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Grasshopper Integrated Pest Management User Handbook

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Foreword

The U.S. Department of Agriculture's (USDA) Animal and Plant Health Inspection Service (APHIS) has the congressional mandate to control grasshoppers on Federal rangeland. Grasshopper infestations blanketed millions of acres of U.S. rangeland in the mid-1980's, and APHIS treated much of this land with insecticides. This demonstrated reliance on chemical control, the cyclical nature of grasshopper outbreaks, and the need to develop environmentally responsible control methods prompted the Department to look for additional control measures. Integrated pest management (IPM) was chosen as the preferred method, and in 1987 Congress provided funds for a large-scale pilot project. IPM demonstration areas in Idaho and North Dakota were chosen as representative of major western ecosystems in which grasshopper outbreaks often occur.

APHIS directed a coalition of Federal agencies that participated in the Grasshopper Integrated Pest Management Project. These agencies included USDA's Agricultural Research Service, Economic Research Service, Forest Service, and Extension Service (now known as the Cooperative State Research, Education, and Extension Service); the U.S. Department of the Interior's Bureau of Land Management, U.S. Fish and Wildlife Service, and National Park Service; and the U.S. Environmental Protection Agency's Office of Pesticide Programs. In addition, State departments of agriculture, land-grant colleges, grazing associations, and private industry joined the effort to develop new strategies for grasshopper IPM.

This Grasshopper Integrated Pest Management User Handbook summarizes the efforts of the many scientists involved in the 8-year project. As an IPM compendium, the Handbook incorporates a variety of disciplines stretching from ecology to entomology and from economics to range management. The looseleaf, three-ring binder allows a reader to lift out a section or chapter of interest. The design also allows the insertion of new information as it becomes available. Various chapters within the Handbook can be used as "standalones" to support technology transfer directly to end users, such as Federal land managers, ranchers, extension agents, and university and State department of agriculture personnel.

This Handbook will serve as an up-to-date resource for implementation of new grasshopper integrated pest management technologies before the next inevitable grasshopper epidemic.

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Acknowledgments

We gratefully acknowledge the help of all scientists and managers who wrote Handbook chapters. The authors constitute only a fraction of the total workforce engaged in the preparation of this Handbook. Sincere appreciation, therefore, is also extended to those unnamed individuals who assisted the authors as technicians, clerical and administrative support personnel, students, and aides. Special recognition is also due for the leaders of each of the seven Handbook sections. Without their leadership and time-management skills, this publication would not have become a reality.

The GHIPM Management Team, who also served as the editorial board for the Handbook, deserves special recognition. Their insight and constructive criticism helped ensure that the information is appropriately written for the intended users. Editorial board members included Bob Brittingham, Nelson Foster, Keith Winks, Gary Cunningham, and Helene Wright, of USDA's Animal and Plant Health Inspection Service (APHIS); and Jerome Onsager, of the Department's Agricultural Research Service.

APHIS' Legislative and Public Affairs unit at headquarters provided editorial oversight (Janet Wintermute), layout oversight (Mary Ann Hines), and printing (Anita McGrady) services.

The Project is indebted to the U.S. Department of the Interior's Bureau of Land Management, Idaho State Office, for providing the space for office staff as well as allowing Project personnel to become a part of their organizational "family."

We believe the Grasshopper Integrated Pest Management User Handbook will be useful to scientists, land managers, and government policymakers for years to come. Late in 1999, as work on the final two sections of the Handbook was wrapping up, USDA's Agricultural Research Service (ARS) announced plans to make a CD-Rom disk containing the entire Handbook plus several other Project-related documents. This all-you-ever-wanted-to-know-about-grasshoppers disk will be available free to the public in 2000. All recipients of the hard copy of this Handbook will receive a letter explaining how to request a copy of the CD-Rom as well. We are especially proud, here at APHIS, that ARS appreciates the value of the investigations done under the GHIPM umbrella and wants to help us transfer this technology in perpetuity in the highly portable and cost-effective CD format.

Gary L. Cunningham and
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Introduction

During the last major outbreak of grasshoppers in the mid-1980's on Western United States' rangelands, Federal and State governments saw the need to develop new and better ways of grasshopper management. From that need, Congress created the Grasshopper Integrated Pest Management Project. (APHIS file photo.)

Introduction

Gary L. Cunningham, Director (1990–94)
Grasshopper Integrated Pest Management Project

Historical Background

In the 1930's, grasshopper infestations covered millions of acres of federally and privately controlled land in 17 Western States. Failed attempts at local control efforts proved that grasshopper outbreaks could be dealt with only on a regional scale. As a result, in 1934 Congress charged the U.S. Department of Agriculture (USDA) with controlling grasshoppers on Federal rangeland. This responsibility is part of the duties of the USDA's Animal and Plant Health Inspection Service (APHIS).

APHIS provided direct supervision and leadership for large-scale grasshopper management programs. Cooperators included other Federal agencies, State departments of agriculture, and private ranchers. The agency's activities complied with National Environmental Policy Act requirements and were authorized by the Incipient and Emergency Control of Pests Act (1937), the Organic Act of the Department of Agriculture (1944), the Cooperation With State Agencies in the Administration and Enforcement of Certain Laws Act (1962), and the Food Security Act (1985). Management strategies are presented in the "1987 Rangeland Grasshopper Cooperative Management Program: Final Environmental Impact Statement."

Cooperative control programs for rangeland grasshoppers are undertaken almost every year in affected parts of the Great Plains and Intermountain West. These programs were most visible in the mid-1980's. In 1985, there were 55 million acres of western rangelands heavily infested with grasshoppers, and APHIS treated 14 million acres with chemicals. Liquid insecticides were aerially applied to blocks of 10,000 acres or more. The chemicals used were chosen for their minimal or negligible impact on the environment. However, treatments of this magnitude did raise concern about the possible effects of insecticides on nontarget organisms, the environment, and the ecosystem.

Grasshopper Integrated Pest Management

In response to the grasshopper epidemic of the mid-1980's, USDA, APHIS, specified in a programmatic environmental impact statement (EIS) that integrated pest management (IPM) be the preferred approach for cooperative programs to control grasshoppers on rangeland. IPM is defined as the coordinated use of pest and environmental information along with available pest control methods (including cultural, biological, genetic, and chemical) to prevent unacceptable levels of pest damage by the most economical means and with the least possible hazard to people, property, and the environment. This approach is often sustainable and complements USDA initiatives in range management, water quality, and food safety. When available, IPM is preferred by Federal and State agencies that manage public lands.

In 1987, APHIS initiated the Grasshopper Integrated Pest Management (GHIPM) Project to develop and demonstrate new IPM technologies and to provide the results to managers of public and private rangelands. The overall purpose of the Project was to develop tools that would help in predicting outbreaks and to develop a combination of preventive tactics that would reduce reliance upon chemical insecticides for control.

The Project's stated objectives were to

- Refine an existing grasshopper phenology (growth and development) model to maximize the efficiency of management activities;
- Demonstrate that early sampling can detect and help classify developing infestations that could be responsive to management with tactics alternative to chemical controls;
- Develop economic thresholds and prescribe treatments to reduce infestations to noneconomic levels with minimal effects on nontarget species;
- Quantify current-season and long-term grasshopper population changes after each different control tactic in order to support a model of population dynamics;
- Develop new biological control methods for grasshopper management, including grasshopper viruses, fungal pathogens, and parasites;

- Provide coordinated research on economics, range management, and ecology as components of a systems approach to grasshopper management; and

- Integrate pertinent data into an expert system that can be used by APHIS and the private sector upon completion of the Project.

Two 1-million-acre demonstration sites were chosen as representative grassland ecosystems. One was in northwestern North Dakota, partially within the USDA, Forest Service's Little Missouri National Grassland, with other large areas managed by the McKenzie County Grazing Association. The second, in south-central Idaho, was managed by the U.S. Department of the Interior, Bureau of Land Management's Shoshone District. Lack of high grasshopper populations in Idaho since 1988 made the Shoshone District site less suitable than the North Dakota site for demonstrating new IPM control technologies.

A major component of the GHIPM Project (1987–94) was a comprehensive research and development program. The Project's technical work group, comprised of representatives from nine Federal agencies, provided oversight for research funding. Under USDA cooperative agreements, more than 50 scientists from Federal and State research institutions were involved in developing new IPM technologies during the life of the Project. Disciplines included agricultural engineering, entomology, plant pathology, ecology, range management, agricultural economics, hydrology, plant physiology, computer science, and wildlife management.

GHIPM User Handbook

This Handbook consolidates extensive information developed over the 8 years of the Project on IPM for grasshoppers. The Handbook is written for anyone who needs practical information, and the intended audience includes Federal and State land managers; USDA, APHIS, Plant Protection and Quarantine (PPQ) officials; State department of agriculture personnel; extension agents working for USDA's Cooperative State Research, Education, and Extension Service at the county level; IPM specialists and researchers; and farmers and ranchers. The Handbook supplements, but does not replace, the APHIS, PPQ, Grasshopper Program Manual (the document that governs PPQ employees in their operational work on grasshopper problems). The Handbook's three-ring binder permits future additions and revisions so it can be kept up to date as a reference tool. The contents are written in a nonscientific format from the perspective of putting new research findings into practical use.

Handbook Format

Seven sections of the GHIPM User Handbook organize and identify the major components developed during the project.

Section I, Biological Control, provides an overview of grasshopper biological control agents and discusses their potential as management tools. Included are protozoa, fungi, bacteria, viruses, insect predators and parasites, mites, nematodes, birds, and wildlife.

Section II, Chemical Control, identifies available chemical insecticides, application methods, and rationale for selecting certain insecticides for grasshopper control. It also discusses new techniques and equipment designed to reduce pesticide load in the environment while retaining cost effectiveness.

Section III, Environmental Monitoring and Evaluation, examines the effects of grasshopper treatments on nontarget organisms and the environment. The section also includes techniques for monitoring and reducing environmental effects of grasshopper treatment programs.

Section IV, Modeling and Population Dynamics, provides an overview of computerized modeling of grasshopper populations and the dynamics associated with grasshopper population development, survival, and buildup. Modeling allows land managers to make more accurate predictions of future outbreaks, based on data bases of past grasshopper populations.

Section V, Rangeland Management, explains how various range-management techniques can affect grasshopper populations and why grazing systems are a factor in grasshopper management. The section includes specific examples for Idaho and for portions of the upper Great Plains.

Section VI, Decision Support Tools, describes and provides "hands-on" management and grasshopper identification tools, including the Hopper decision support software, an expert system that helps in making management decisions. Economic considerations and survey sampling procedures also are discussed. Hopper Helper is a grasshopper identification key included in the Handbook. A much more comprehensive work, Robert Pfadt's "Field Guide to Common Western Grasshoppers," was developed during the GHIPM Project. Section VI includes an example of the guide's contents, which are individual species factsheets. (Recipients of the printed version of this Handbook have also been sent two separate mailouts of Dr. Pfadt's factsheets, which cover some 75 grasshopper species of economic importance.)

Section VII, Future Directions, presents an overview of key trends in and components of grasshopper IPM that need further study and development. These trends and components include new materials for chemical and biological control, environmental studies, the possible effects of grasshopper treatment programs on recreational activities, discussions of grasshopper population and habitat manipulation, and the important role geographic information systems will play in the future.

Within each section, individual chapters have their own bibliographies so readers can seek out more detailed information on the science behind the GHIPM Project's recommendations.

Updating the Handbook

Recipients of the original Handbook will be kept on a mailing list so APHIS can send them new or revised grasshopper materials in the future. New or replacement pages will be punched and labeled, ready for insertion into specified sections in the Handbook.

How To Obtain Copies

Single free copies of the Grasshopper IPM User Handbook may be obtained from USDA, APHIS, PPQ, Operational Support Staff, 4700 River Road, Riverdale, MD 20737. You may request a copy by telephone as well (301–734–8247).

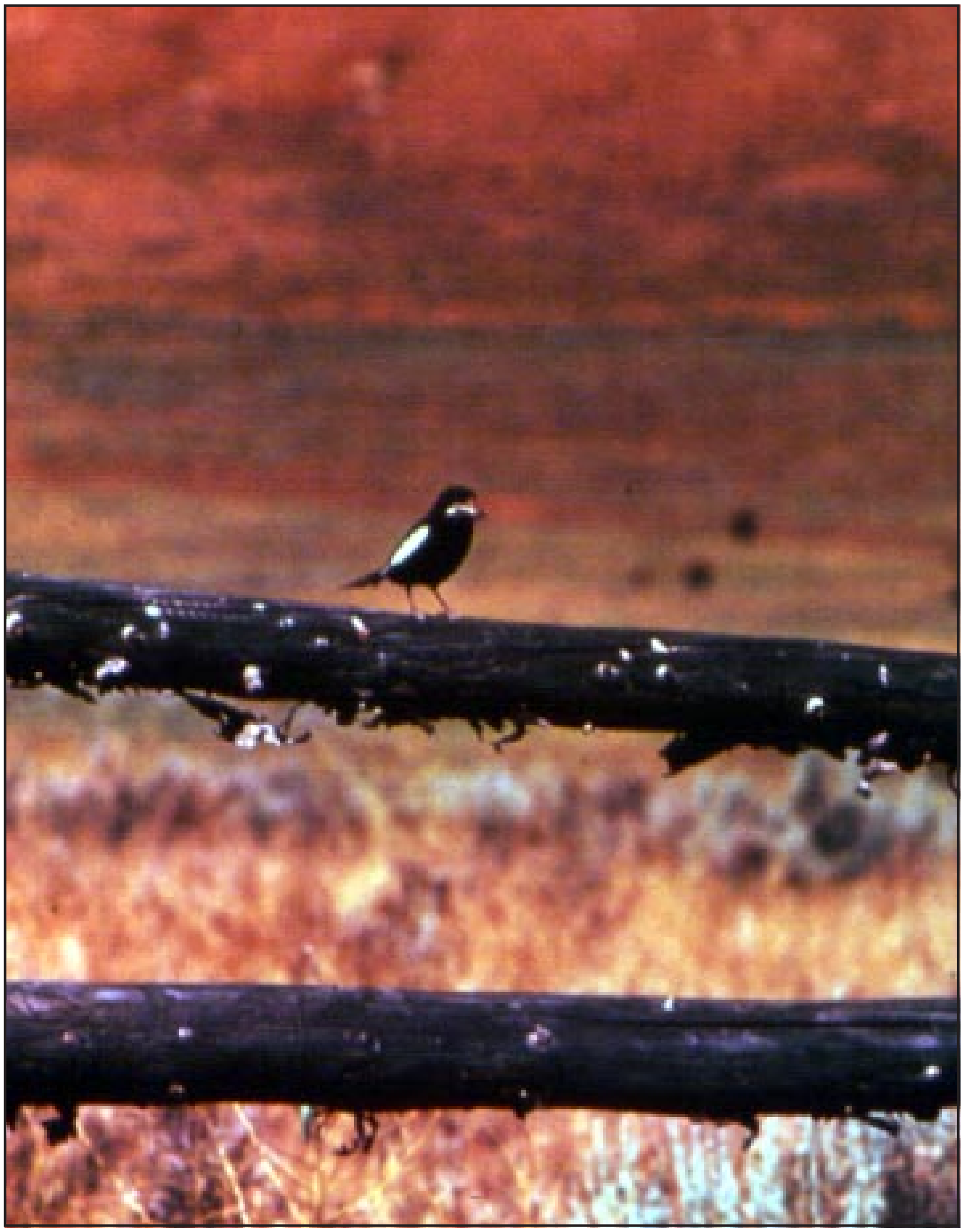
References Cited

U.S. Department of Agriculture, Animal and Plant Health Inspection Service. 1987. Rangeland Grasshopper Cooperative Management Program: final environmental impact statement. Washington, DC: U.S. Department of Agriculture, Animal and Plant Health Inspection Service. 221 p.

Reference Cited—Unpublished

U.S. Department of Agriculture, Animal and Plant Health Inspection Service. 1993. Cooperative Grasshopper Integrated Pest Management Project: annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service. 260 p.

I. Biological Control



Many wildlife species, like this lark bunting, choose grasshoppers as food for their young. Favoring bird populations can help limit grasshoppers in a complementary effort with other control methods. (Photograph by chapter author Lowell C. McEwen, of Colorado State University; used by permission.)

I.1 Biological Control: An Introduction

D. A. Streett

DeBach (1964) defined biological control as "the action of parasites, predators, or pathogens (disease-causing organisms) in maintaining another organism's population density at a lower average than would occur in their absence." A more recent definition proposed by the National Academy of Sciences (1987) for biological control is "the use of natural or modified organisms, genes, or gene products to reduce the effects of undesirable organisms (pests), and to favor desirable organisms such as crops, trees, animals, and beneficial insects and microorganisms."

While many people may share the wider view of biological control that encompasses the methods broadly defined by the National Academy of Sciences, Garcia et al. (1988) make some valid arguments for using DeBach's definition because it emphasizes the concepts of self-sustaining and density-dependent regulation of one species by another. For land managers' purposes, the more traditional definition of biological control proposed by DeBach will be used in this introduction.

Constraints on the use of chemical pesticides may benefit the development of biological control options and their implementation in an integrated pest management (IPM) program. The U.S. Department of Agriculture's (USDA) Animal and Plant Health Inspection Service (APHIS) (1994 unpubl.) defines IPM as "the selection, integration, and implementation of pest management tactics in a systems approach on the basis of anticipated biological, economic, ecological, and sociological indicators." For a more thorough discussion of IPM, refer to the excellent review article by Cate and Hinkle (1993) describing the history and progression of IPM.

Biological control is usually achieved through one or a combination of the following approaches: conservation, augmentation, and classical biological control.

- Conservation is an approach whereby management systems are manipulated to enhance or conserve naturally occurring biological control agents.
- The augmentation approach includes both inoculative and inundative releases of biological control agents. An inoculative release depends upon the biological control agent reproducing, persisting, and spreading on its own accord in the pest population. Inundative releases are more of a short-term control measure with biological control agents causing a more immediate reduction in the pest population but lacking the ability to persist or spread in the environment.
- In the classical approach, exotic (not native) pest species are controlled by the introduction and establishment of exotic biological control agents. Classical biological control has been extremely successful at controlling pests, and current Federal regulations are adequate to monitor and safeguard the importation of biological control agents (Soper 1992).

The approach to classical biological control proposed by Hokkanen and Pimentel (1984, 1989) involves the selection of promising biological control agents from exotic sources for the control of native pest species. Major premises for this approach are a greater likelihood for success using this new association and the ability to control native pests, which represent 60–80 percent of all pest species (Hokkanen and Pimentel 1989).

In the early 1990's, a parasitic wasp and a fungus from Australia were imported into the United States for evaluation as biological control agents against rangeland grasshoppers in the Western United States. Some scientists raised concerns regarding whether the importation of exotic agents would result in some risk to the environment. While concerns about the release of exotic biological control agents are sensible, no major problems are reported from the use of these agents in the United States (Carruthers and Onsager 1993). For a more detailed discussion of this issue, see Lockwood (1993a, b) or Howarth (1991) and Carruthers and Onsager (1993) and/or chapters VII.4 and VII.6 in the Future Directions section of this handbook.

Here in section I, some review chapters on the current status of biological control of grasshoppers discuss the potential of parasites, predators, and pathogens. Various authors in this section describe some research projects funded during the USDA, APHIS, Grasshopper Integrated Pest Management (GHIPM) Project. Topics include identification of fungal pathogens, laboratory assays to assess the effectiveness of *Nosema locustae,* and construction of bird nest boxes. These chapters provide a solid foundation of knowledge on the biological control of grasshoppers. Basic and applied research will continue to be essential in the development and implementation of biological control strategies.

Selected References

DeBach, P. 1964. The scope of biological control. In: DeBach, P. ed. Biological control of insect pests and weeds. New York: Reinhold: 3–20.

Carruthers, R. I.; Onsager, J. A. 1993. Perspective on the use of exotic natural enemies for biological control of pest grasshoppers (Orthoptera: Acrididae). Environmental Entomology 22: 885–903.

Cate, J. R.; Hinkle, M. K. 1993. Insect pest management: the path of a paradigm. Spec. Rep. Alexandria, VA: National Audubon Society. 43 p.

Garcia, R.; Caltagirone, L. E.; Gutierrez, A. P. 1988. Comments on a redefinition of biological control. Biosciences 38: 692–694.

Hokkanen, H.; Pimentel, D. 1984. New approach for selecting biological control agents. Canadian Entomologist 116: 1109–1121.

Hokkanen, H.M.T.; Pimentel, D. 1989. New associations in biological control: theory and practice. Canadian Entomologist 121: 829–840.

Howarth, F. G. 1991. Environmental impacts of classical biological control. Annual Review of Entomology 36: 485–509.

Lockwood, J. A. 1993a. Benefits and costs of controlling rangeland grasshoppers (Orthoptera: Acrididae) with exotic organisms: search for a null hypothesis and regulatory compromise. Environmental Entomology 22: 904–914.

Lockwood, J. A. 1993b. Environmental issues involved in biological control of rangeland grasshoppers (Orthoptera: Acrididae) with exotic agents. Environmental Entomology 22: 503–518.

National Academy of Sciences. 1987. Report of the research briefing panel on biological control in managed ecosystems. Washington, DC: National Academy Press. 206 p.

Soper, R. S. 1992. U.S. Department of Agriculture, Agricultural Research Service national biological control program: policy, and constraints. In: Charudattan, R.; Browning, W. H., eds. Regulations and guidelines: critical issues in biological control. Washington, DC: U.S. Department of Agriculture, Agricultural Research Service: 49–52.

References Cited—Unpublished

U.S. Department of Agriculture, Animal and Plant Health Inspection Service. 1994. Grasshopper program manual. Frederick, MD: U.S. Department of Agriculture, Animal and Plant Health Inspection Service, Plant Protection and Quarantine. 159 p.

I.2 *Nosema locustae*

D. A. Streett

Introduction

Grasshoppers are the most economically important insect pests on rangeland in the Western United States (Hewitt and Onsager 1982). A conservative estimate for the average value of rangeland forage loss to grasshoppers in the West each year is about $393 million (Hewitt and Onsager 1983). Since the late 1960's, controlling major infestations of grasshoppers on rangeland has involved the use of chemical insecticides, primarily malathion and carbaryl. However, increasing awareness of the environmental risk associated with the exclusive use of chemical insecticides led to the establishment of the Grasshopper Integrated Pest Management (GHIPM) Project.

Disease-causing micro-organisms have been investigated as potential biological control agents of grasshoppers for many years. Probably the most well-known case has been the parasite *Nosema locustae,* a pathogen that was selected in the early 1960's for development as a microbial control agent for use in long-term suppression of grasshoppers (Henry 1978, Onsager 1988). *Nosema locustae* is the only registered microbial agent that is commercially available for control of rangeland grasshoppers.

Nosema has been studied more than any other microbial control agent for the suppression of grasshopper populations. Applications of *Nosema* formulated on a wheat bran bait have resulted in numerous successful introductions of the pathogen into field populations. However, while this parasite has proven a potentially effective tool in grasshopper management, several questions have been raised regarding the effectiveness of *Nosema* in the field.

Unpredictability of *Nosema*

Vaughn et al. (I.4) attributed the apparent failures of *Nosema* to low-quality material, equipment failure, poor formulation, inappropriate target species, and unreasonable expectations by users. Onsager (1988) also discussed some of the reasons for this lack of confidence in *Nosema* for controlling grasshopper populations. He noted that the traditional sampling approach used to estimate grasshopper reductions in field trials with chemical insecticides may not be appropriate to assess the effectiveness of *Nosema.* Typically *Nosema* requires much longer to kill a grasshopper than chemicals. Grasshoppers are then able to disperse and conceal differences between treated and control plots.

Reuter et al. (1990) suggested that the standard application rate of *Nosema* (1×10^9 spores/acre) was too low to induce immediate grasshopper population suppression. In a field evaluation, an untreated control plot was compared to plots receiving either the standard rate (1×10^9 spores/acre) or a higher (100×) rate (1×10^{11} spores/acre) of *Nosema.* Density estimates were taken weekly, and bottomless field cages and small rearing cages were used to estimate mortality. The lack of treatment replication, the small plot size, and the close proximity of plots made it impossible to draw firm conclusions about the grasshopper densities or relative rates of suppression after treatment. However, significant mortality was observed at the higher application rate for *Melanoplus sanguinipes* in the small rearing cages 7 weeks after application (Reuter et al. 1990). These preliminary mortality results lend support to Henry's (1981) contention that applying higher dosages of Nosema will not necessarily produce a commensurate gain in density reduction.

A more immediate density reduction has been demonstrated in field studies using wheat bran bait formulations of *Nosema* and carbaryl in which significant short-term response to carbaryl was followed by a later response to *N. locustae* (Onsager et al. 1981). Further studies on the response of grasshoppers to higher application rates of *Nosema* may be warranted.

A review of the literature on the effectiveness of *Nosema* in the field identifies dispersal as a common problem. Movement between plots was cited as affecting results in six of eight studies that evaluated the effects of Nosema in the field (Henry 1971; Henry and Oma 1974, 1981; Henry and Onsager 1982; Henry et al. 1973, 1978). Only Johnson and Henry (1987) suggested that there was little movement of infected individuals into control plots within 31 days of application.

Detection of *Nosema locustae*

In the past, visual examinations with phase contrast microscopy for spores have been required to detect *Nosema* infection in grasshoppers. Generally, *Nosema* spores are detectable about 21 days after application (Henry and Oma 1974). Most protocols recommend microscopic examinations at 28 days following application (Henry 1978). Thus, it has not been possible to assess some of the earlier events in a *Nosema* treatment program.

Dispersal and death that occur prior to the detection of Nosema reduce estimates of its presence in the field. Early detection of *Nosema* infections is therefore necessary to obtain unbiased estimates of initial prevalence. Scientists have developed a sensitive nucleic acid probe for the detection of *Nosema* in grasshoppers. Data indicate that the probe can reliably detect *Nosema* in grasshoppers within 7–10 days after infection. Use of a probe to estimate infection rates should eliminate much of the inherent bias associated with visual examination.

Nosema Transmission

A recent laboratory study by Raina et al. (1995) has reported transovarial transmission of *N. locustae* in *Locusta migratoria migratorioides* with the incidence of infection ranging from 72 percent to 92 percent among progeny up to the F7 generation. *N. locustae* spores also were found in all nymphal instars for the F1 and F2 generations.

The mechanisms and rates of *Nosema* transmission in the field have not been addressed adequately. Spores have been observed in feces (Henry 1969 unpubl.), but the scavenging of *Nosema*-infected cadavers by healthy grasshoppers may represent the greatest potential for transmission to uninfected grasshoppers of the same generation. Scavenging of cadavers is common in many species of grasshoppers (Lavigne and Pfadt 1964, Lockwood 1988). Henry (1969 unpubl.) observed feeding on *Nosema*-infected cadavers in the field. Scavenging may offer a very efficient means for transmission of *Nosema* during the year of treatment and possibly into later generations (O'Neill et al. 1994).

Spores of *Nosema* have been observed in ovaries from and in eggs produced by infected females (Henry 1969 unpubl.). Although Ewen and Mukerji (1980) were unable to find spores in eggs collected from *Nosema*-treated plots, they did observe *Nosema* infection among nymphs raised from field-collected eggs. Henry and Onsager (1982) also reported infection in grasshopper populations during the year after treatment. These observations indicate that transmission to subsequent generations is indeed likely, but the details of *Nosema* transmission in field populations of grasshoppers have never been fully explained.

Effect on Grasshopper Egg Production

Nosema-infected females produce fewer eggs than healthy females (Henry and Oma 1981). Henry (1969, 1971) reported detecting little ovarial or egg debris in infected grasshoppers that were ground up, which suggests that infected females fail to develop reproductively. Ewen and Mukerji (1980) reported substantially lower rates of egg laying after applications of *Nosema* in the field. Henry and Oma (1981) suggested the need to measure the effects of *Nosema* on egg numbers and egg viability. Lockwood and Debrey (1990) also observed some evidence of lower egg production in higher populations (greater than 11.5 grasshoppers/yd^2 or 9.6 grasshoppers/m^2) of grasshoppers treated with *Nosema.*

Conclusions

Until the reasons for the inconsistent response of *Nosema* to grasshoppers are better understood, its effectiveness will probably continue to be disputed (*See* I.4.). The grasshopper species complex, the age of the grasshoppers, and population density can affect the response to a *Nosema* application. Therefore, a more comprehensive approach is needed to adequately assess *Nosema* against grasshoppers. This approach must include a better understanding of the major disease processes of *Nosema.* Vaughn's team (I.4) recommends that *Nosema* be used to suppress rangeland grasshoppers in environmentally sensitive areas where cost and acute insecticide control are not primary concerns and proposes the use of higher rates and/or multiple applications when environmental issues outweigh the economic issues.

References Cited

Ewen, A. B.; Mukerji, M. K. 1980. Evaluation of *Nosema locustae* (Microsporidia) as a control agent of grasshopper populations in Saskatchewan. Journal of Invertebrate Pathology 35: 295–303.

Henry, J. E. 1971. Experimental application of *Nosema locustae* for control of grasshoppers. Journal of Invertebrate Pathology 18: 389–394.

Henry, J. E. 1978. Microbial control of grasshoppers with *Nosema locustae* Canning. In Selected topics of the genus *Nosema.* Miscellaneous Publications of the Entomological Society of America 11: 85–95.

Henry, J. E. 1981. Natural and applied control of insects by protozoa. Annual Review of Entomology 26: 49–73.

Henry, J. E.; Oma. E. A. 1974. Effects of infections by *Nosema locustae* Canning, *Nosema acridophagus* Henry, and *Nosema cuneatum* Henry (Microsporida: Nosematidae) in *Melanoplus bivittatus* (Say) (Orthoptera: Acrididae). Acrida 3: 223–231.

Henry, J. E.; Oma, E. A. 1981. Pest control by *Nosema locustae,* a pathogen of grasshoppers and crickets. In Burges, H. D., ed. Microbial control of pests and plant diseases 1970–1980. New York: Academic Press: 573–586.

Henry, J. E.; Onsager, J. A. 1982. Large-scale field test of control of grasshoppers on rangeland with *Nosema locustae.* Journal of Economic Entomology 75: 31–35.

Henry, J. E.; Tiahrt, K.; Oma, E. A. 1973. Importance of timing, spore concentrations, and levels of spore carrier in applications of *Nosema locustae* (Microsporida: Nosematidae) for control of grasshoppers. Journal of Invertebrate Pathology 21: 263–272.

Henry, J. E.; Oma, E. A.; Onsager, J. A. 1978. Relative effectiveness of ULV spray applications of spores of *Nosema locustae* against grasshoppers. Journal of Economic Entomology 71: 629–632.

Hewitt, G. B.; Onsager, J. A. 1982. Grasshoppers: yesterday, today and forever. Rangelands 4: 207–209.

Hewitt, G. B.; Onsager, J. A. 1983. Control of grasshoppers on rangeland in the United States—A perspective. Journal of Range Management 36: 202–207.

Johnson, D. L.; Henry, J. E. 1987. Low rates of insecticides and *Nosema locustae* (Microsporidia: Nosematidae) on baits applied to roadsides for grasshopper (Orthoptera: Acrididae) control. Journal of Economic Entomology 80: 685–689.

Lavigne, R. J.; Pfadt, R. E. 1964. The role of rangeland grasshoppers as scavengers. Journal of Kansas Entomological Society 37: 1–4.

Lockwood, J. A. 1988. Cannibalism in rangeland grasshoppers (Orthoptera: Acrididae): attraction to cadavers. Journal of the Kansas Entomological Society 61: 379–387.

Lockwood, J. A.; DeBrey, L. D. 1990. Direct and indirect effects of *Nosema locustae* (Canning) (Microsporidia: Nosematidae) on rangeland grasshoppers (Orthoptera: Acrididae). Journal of Economic Entomology 83: 377–383.

O'Neill, K. M.; Streett, D.; O'Neill, R. P. 1994. Scavenging behavior of grasshoppers (Orthoptera: Acrididae): feeding and thermal responses to newly available resources. Environmental Entomology 23: 1260–1268.

Onsager, J. A. 1988. Assessing the effectiveness of *Nosema locustae* for grasshopper control: traditional insecticide-based sampling criteria cannot accurately evaluate efficacy of *Nosema.* Montana AgResearch 5: 12–16.

Onsager, J. A.; Rees, N. E.; Henry, J. E.; Foster, N. 1981. Integration of bait formulation of *Nosema locustae* and carbaryl for control of rangeland grasshoppers. Journal of Economic Entomology 74: 183–187.

Raina, S. K.; Dos, S.; Rai, M. M.; Khurad, A. M. 1995. Transovarial transmission of Nosema locustae (Microsporida: Nosematidae) in the migratory locust *Locusta migratoria migratorioides.* Parasitology Research 81: 38–44.

References Cited—Unpublished

Henry, J. E. 1969. Protozoan and viral pathogens of grasshoppers. Ph.D. dissertation. Bozeman, MT: Montana State University. 153 p.

Reuter, K. C.; Foster, R. N.; Hildreth, M.; Colletto, D.; Cushing, W. J.; Pucelik, M. J.; Kohler, D.; Houston, R.; Scott, A. 1990. Preliminary investigation of the effect of a greatly increased rate of *Nosema locustae* on rangeland grasshopper populations. In: Cooperative Grasshopper Integrated Pest Management Project, 1990 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 165–174.

I.3 Laboratory Bioassays of *Nosema locustae*

Michael B. Hildreth, Chris W. Brey, Billy W. Fuller, and R. Nelson Foster

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Introduction

The use of living insect pathogens as biocontrol agents for insects requires that the virulence (killing power) of these agents must be monitored occasionally, especially just prior to their distribution into the environment. Evaluation of an agent's virulence can be accomplished through the use of laboratory bioassays involving the target insects (raised in the laboratory) and the biocontrol agents that are to be tested.

The first biocontrol agent registered by the U.S. Environmental Protection Agency for grasshopper pests was the protozoan *Nosema locustae*. Grasshoppers acquire *N. locustae* infections by eating its spore stage. *N. locustae* infects the fat bodies of grasshoppers and is only mildly pathogenic to its host. For several years, our lab at South Dakota State University (SDSU) has been bioassaying the viability and virulence of *N. locustae* spores supplied commercially to the U.S. Department of Agriculture, Animal and Plant Health Inspection Service, Plant Protection and Quarantine (USDA, APHIS, PPQ) Grasshopper Integrated Pest Management (GHIPM) Project. Laboratory-reared third-instar *Melanoplus sanguinipes* grasshoppers were used as the target insect for these bioassays, and the grasshoppers were fed the *Nosema* spores on small disks cut from romaine lettuce. The grasshoppers were then kept in the lab for 20 days, and LD_{50} (the calculated dose of pathogen or toxin that kills half of the bioassayed grasshoppers) values were calculated based upon the percentage of grasshoppers that had died by the end of the time period.

When *Nosema* is used to control grasshoppers, spores are typically applied on rangelands with a wheat-bran bait. Lettuce bioassays can be used only to measure the viability of spores prior to the spores' addition to wheat bran. The purpose of this chapter is to describe the protocol used in our laboratory to measure the virulence of *N. locustae* spores stored in water and applied to lettuce disks, and also to describe a bioassay protocol that we've used for measuring the virulence of these spores after their addition to wheat bran. Representative results from these bioassays are reported in this chapter.

Lettuce Bioassay

Methods.—*Nosema locustae* spores used for these studies were provided by various commercial sources. All spores were stored in distilled water at –4 °F (–20 °C) until use. *Melanoplus sanguinipes* grasshoppers used in the studies were a lab-reared Canadian strain that had been maintained at SDSU for several years. These grasshoppers were reared according to the recommendations provided by Henry (1985).

The lettuce bioassay was based upon recommendations supplied by John Henry (personal communication). Spores were counted in a hemocytometer (a special slide used in hospitals to count blood cells) and applied to freshly cut lettuce disks approximately one-third inch (7 mm) in diameter disks using a 10 μL (microliter) pipettor. Six dosages of *N. locustae* spores in 10 μL distilled water (e.g., 0, 1×10^4, $10^{4.5}$, 10^5, $10^{5.5}$, 10^6 spores; $1 \times 10^{4.5}$ is equal to 3.162×10^4 or 31,620) were added to the disks (120 disks per dosage), and allowed to dry for 1 to 4 hours. Each disk was fed individually to a third-instar grasshopper that had been previously starved for 1 day in glass vials at approximately 86 °F (30 °C). To distribute the grasshoppers into vials, the insects needed to be cooled briefly from ambient 86 °F (30 °C) to approximately 39 °F (4 °C). Before adding the appropriate lettuce disks to the vials, the vials were randomly sorted and divided into the appropriate six dosage groups. Once 80 grasshoppers from each group had eaten an entire disk, they were placed in groups of 5 into 16 bioassay tubes (8 inches or 20 cm long, 2.75 inches or 7 cm in diameter) constructed of 0.08-inch (0.02-mm) sheet acetate with screened ends. Generally, grasshoppers ate an entire disk within 2 hours or did not eat it even after 12 hours. The 16 bioassay tubes were divided into 4 replicates of 4 tubes each. In the bioassay tubes, grasshoppers were fed laboratory-reared rye grass daily along with triple sulfa-coated rolled oats (Henry and Oma 1975) and maintained under continuous fluorescent illumination at approximately 86 °F (30 °C).

Each day, we counted the number of dead grasshopper carcasses in the bioassay tubes. Grasshoppers frequently cannibalized other grasshoppers in the tubes, and portions of carcasses often were found. Therefore, we verified the number of living grasshoppers remaining in each tube to not overlook cannibalized individuals. We calculated LD_{50} values by using the software package POLO-PC (LeOra Software, Inc., Cary, NC).

Results.—An example of the typical results obtained from the 22 bioassays conducted in our lab during the past 5 years is shown in figure I.3–1. A few of the uninfected control grasshoppers always died during the 20 days of each bioassay. For all of the 22 bioassays, generally less than 20 percent of the control grasshoppers died before the end of the bioassay. Inoculation of grasshoppers with increasing numbers of *N. locustae* spores consistently increased the mortality rate for grasshoppers infected with 1×10^6, $1 \times 10^{5.5}$, and 1×10^5 spores. Grasshoppers inoculated with 1×10^6 *Nosema* spores typically started to die sooner than the control grasshoppers within the first 8 days postinoculation (p.i.); the largest number of deaths normally occurred between days 10 and 14 p.i. By 20 days p.i., 70 to 100 percent of the grasshoppers infected with 1 million (1×10^6) spores had died among the various bioassays performed. It typically took grasshoppers inoculated with $1 \times 10^{5.5}$ spores longer to die than it did for grasshoppers infected with 1×10^6, and fewer grasshopper had died by the 20-day bioassay period (generally 40 to 90 percent). The mortality rate for grasshoppers dosed with 1×10^5 spores tended to separate gradually from the control mortality, and usually became consistently apparent only after 16 days p.i. Mortality in grasshoppers infected with the two lower concentrations (1×10^4 and $10^{4.5}$) did not consistently differ from those of the controls even at 20 days p.i. The calculated LD_{50} for the bioassay shown in figure I.3–1 was 1.19×10^5 at 20 days p.i.

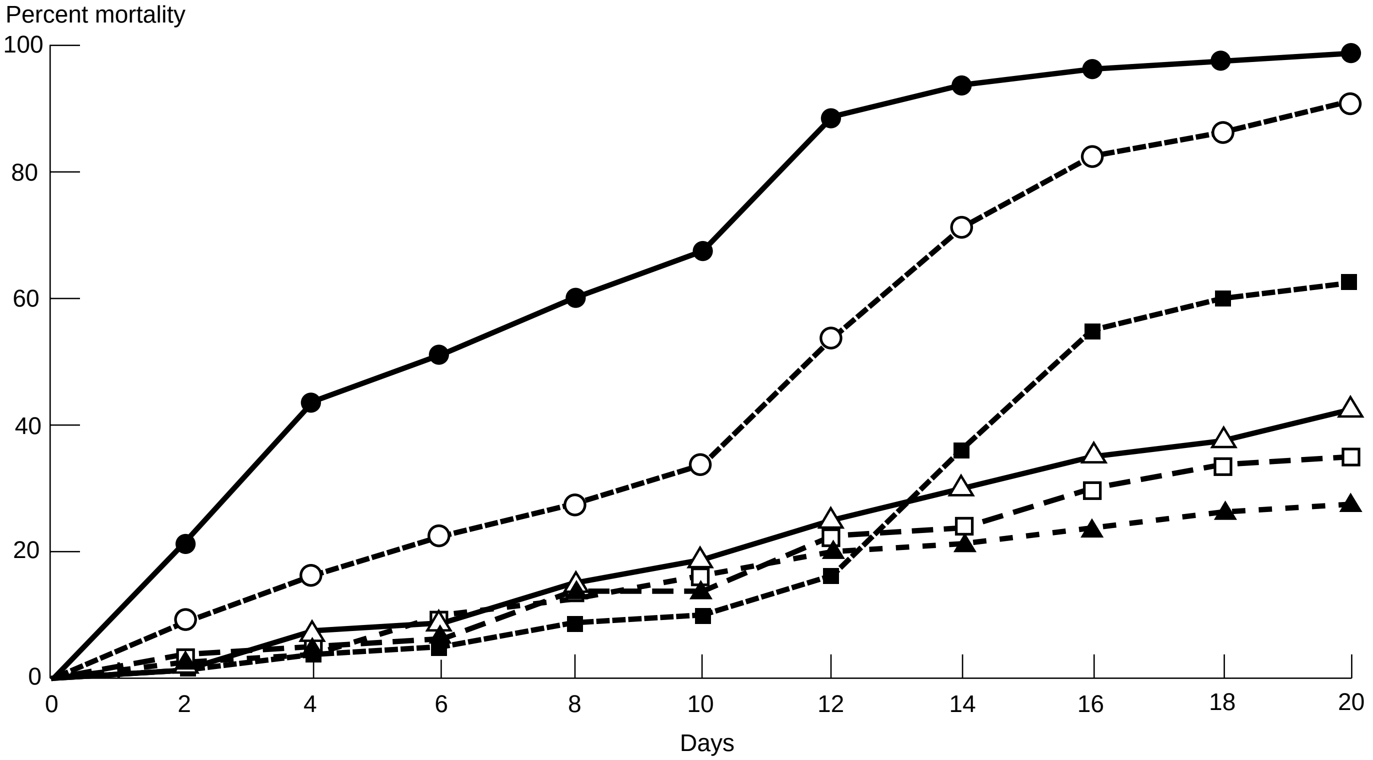

Figure I.3–1—Percent mortality of a 1990 tube bioassay involving third-instar *Melanoplus sanguinipes* grasshoppers treated with varying dosages of *Nosema locustae* spores on lettuce disks (e.g., 0, 1×10^4, $10^{4.5}$, 10^5, $10^{5.5}$, 10^6) and maintained for 20 days postinoculation at approximately 86 °F (30 °C). Solid triangle = 0 spores/grasshopper, open square = 1×10^4 spores/grasshopper, open triangle = $1 \times 10^{4.5}$ spores/grasshopper, solid square = 1×10^5 spores/grasshopper, open circle = $1 \times 10^{5.5}$ spores/grasshopper, and solid circle = 1×10^6 spores/grasshopper.

Bran Bioassay

Methods.—To calculate the theoretical quantity of spores present on average-size flakes of commercially formulated bran, we filtered several grams of the bran through a series of wire sieves with diminishing pore sizes. Most of the flakes were collected on three sieves with pore sizes of 2.36 mm (mesh 8), 2.00 mm (mesh 10), and 1.70 mm (mesh 12). From each of these sieves, 100 flakes were weighed individually. The quantity of spores on each group of flakes was calculated based upon the assumption that each pound of bran contained 1×10^9 spores.

As described in the results section, the theoretical concentration of spores on flakes of commercially formulated bran (an average 1-mg flake should contain 2.2×10^3 spores) was roughly 100 times lower than the concentration of spores easily detected in laboratory bioassays ($1 \times 10^{5.5}$ or 3.16×10^5 spores could easily be detected based upon their effect on grasshopper deaths).

Therefore, in order to bioassay spores on a single bran flake, it was necessary to formulate new bran with spores at a concentration 100 times that of commercially formulated bran (10^{11} spores/lb instead of 10^9 spores/lb). The spores were sprayed onto wheat bran while continually mixing the bran with a small cement mixer. These spores had been recently recovered from grasshoppers and bioassayed on lettuce in our lab (LD_{50} value was 3.29×10^5). In addition to the spores, the spray solution contained 0.2 percent weight to volume (w/v) hydroxymethyl cellulose in distilled water. Hydroxymethyl cellulose is thought to help the spores stick to the bran (Henry et al. 1973). An aerosol sprayer was used to spray the solution on the bran. The treated bran was then allowed to dry and was stored at 39 °F (4 °C).

Attempts were made to bioassay the 100×-treated bran using the same approach used for the lettuce bioassay. One week after formulation of the 100× bran, third-instar grasshoppers were cooled as described above and distributed individually into glass vials. The grasshoppers were starved for 24 hours, randomized, and divided into four groups. Treated bran flakes of different sizes (sieved through mesh 8, 10, or 12) were added to each appropriate vial. Untreated control flakes (sieved only through mesh size 10) were added to the tubes containing control grasshoppers. Once 80 grasshoppers from each group had consumed all bran flakes, they were placed in groups of 5 into 16 bioassay tubes and maintained as described for the lettuce bioassay.

Results from the single-flake bran bioassay study suggested that each grasshopper needed to consume additional bran before any effect could be detected. Therefore, an attempt was made to enable each grasshopper to consume a maximum quantity of treated bran before inclusion in a second bioassay. For that bioassay, 100 grasshoppers were maintained in a large screened rearing cage (30 × 32 × 55 cm) for 48 hours. The only food source during this time was 2.0 g of control or treated bran contained in a standard petri dish. After 24 hours, the uneaten bran was replaced with fresh. Weights were determined from each container of bran and compared to the weights of similar bran maintained similarly just outside the cage. At the end of the bioassay period, the grasshoppers were maintained in bioassay tubes as described for the single-flake bioassay.

Results.—The average weight for each size of Nolo Bait® bran flakes and the estimated number of spores per flake are shown in table I.3–1. The average values ranged from 1.42 mg for larger flakes sieved through mesh 8 to 0.625 mg for flakes sieved through mesh 12. If 1×10^9 spores are added to each pound of bran, then each milligram of flakes should contain 2.20×10^3 spores; therefore, the largest flake weighed in this study (2.2 mg) should contain 4.85×10^3 spores.

Figure I.3–2 illustrates the mortality rates of grasshoppers fed only one flake of 100×-treated bran from each of the various sieves. Because the average flake of bran weighed 1.05 mg, it should contain approximately 2.32×10^5 spores. After 30 days, the mortality rates from the experimental groups of grasshoppers were not significantly greater than that of the controls. In fact, fewer of the grasshoppers receiving the small flakes of experimental bran died than did the control. Unfortunately, however, the mortality rate for the control grasshoppers in this experiment was twice that of previous experiments, and may have obscured any small effects caused by *Nosema.*

Table I.3–1—Average weight in milligrams for each size of bran flakes and estimated spores per flake

Mesh size	Average weight ± standard error of the means	Weight range	Estimated spores per flake
8	1.422 ± 0.0029 mg	0.9–2.2	3.132×10^3
10	1.107 ± 0.0031 mg	0.5–2.2	2.438×10^3
12	0.625 ± 0.0030 mg	0.1–1.1	1.377×10^3

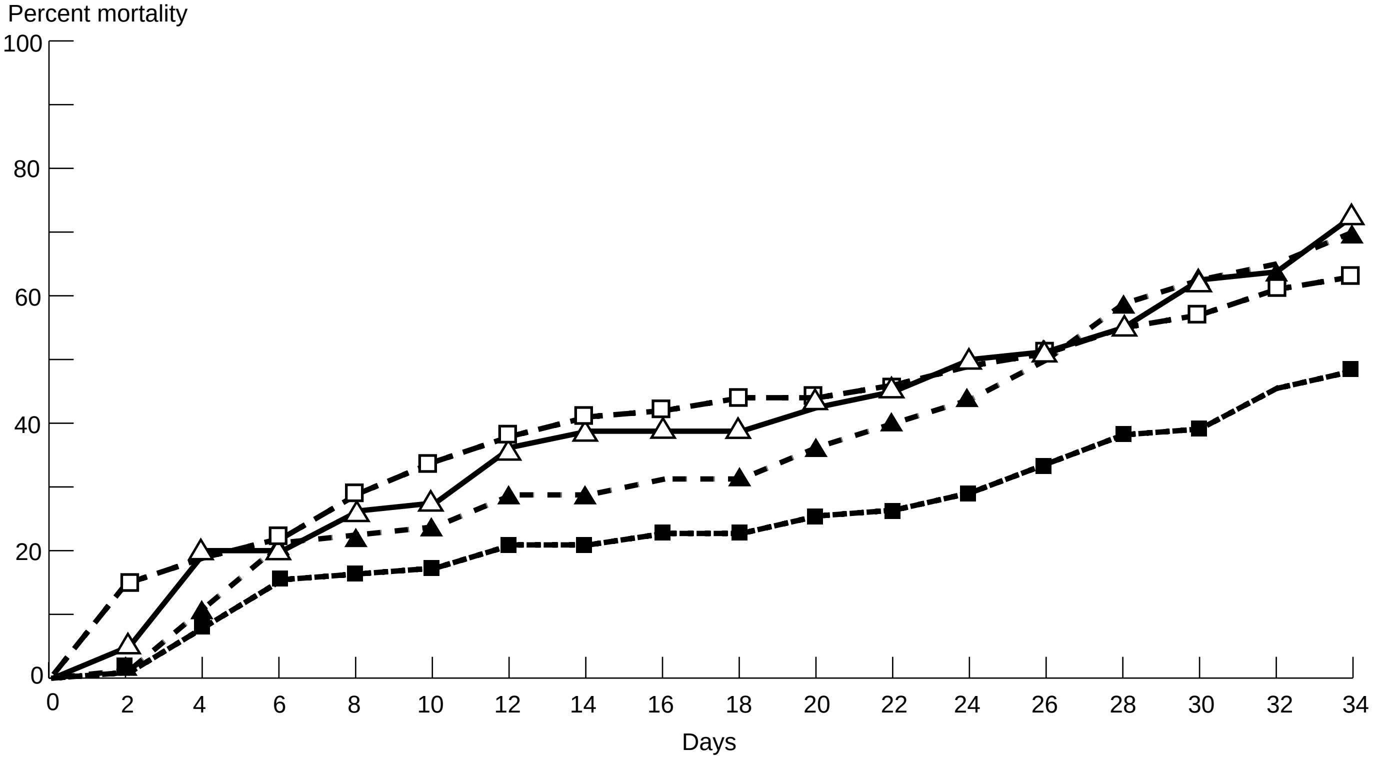

Figure I.3–2—Initial tube bioassay involving *N. locustae*-treated wheat bran flakes given individually to third-instar *M. sanguinipes* maintained for 34 days postinoculation. Solid triangle = grasshoppers given an untreated flake of bran; open triangle = grasshoppers given a treated bran flake that passed through a mesh 7 sieve but not the mesh 8 sieve; open square = flake passed through mesh 8 but not mesh 10; solid square = flake passed through mesh 10 but not mesh 12. Spores had been added to the bran at a concentration of 1×10^{11} spores per pound of bran.

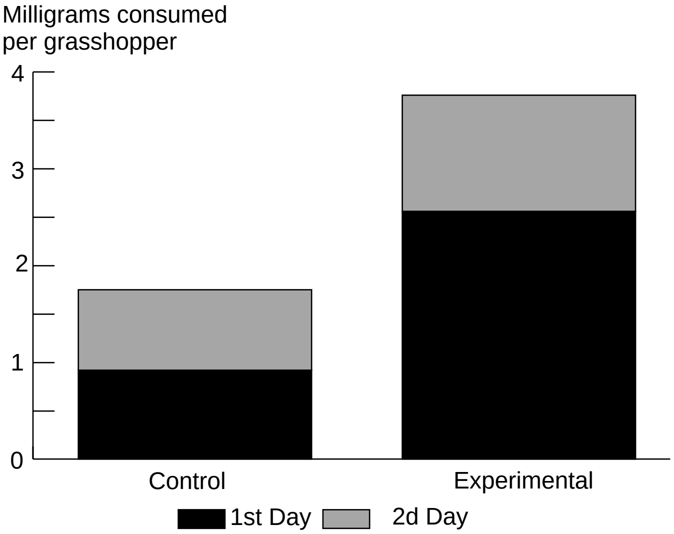

Figure I.3–3—Consumption of control and experimental *(Nosema*-treated) bran by 400 grasshoppers in each group during the first and second day of the inoculation period. Values are expressed in "grams consumed per grasshopper."

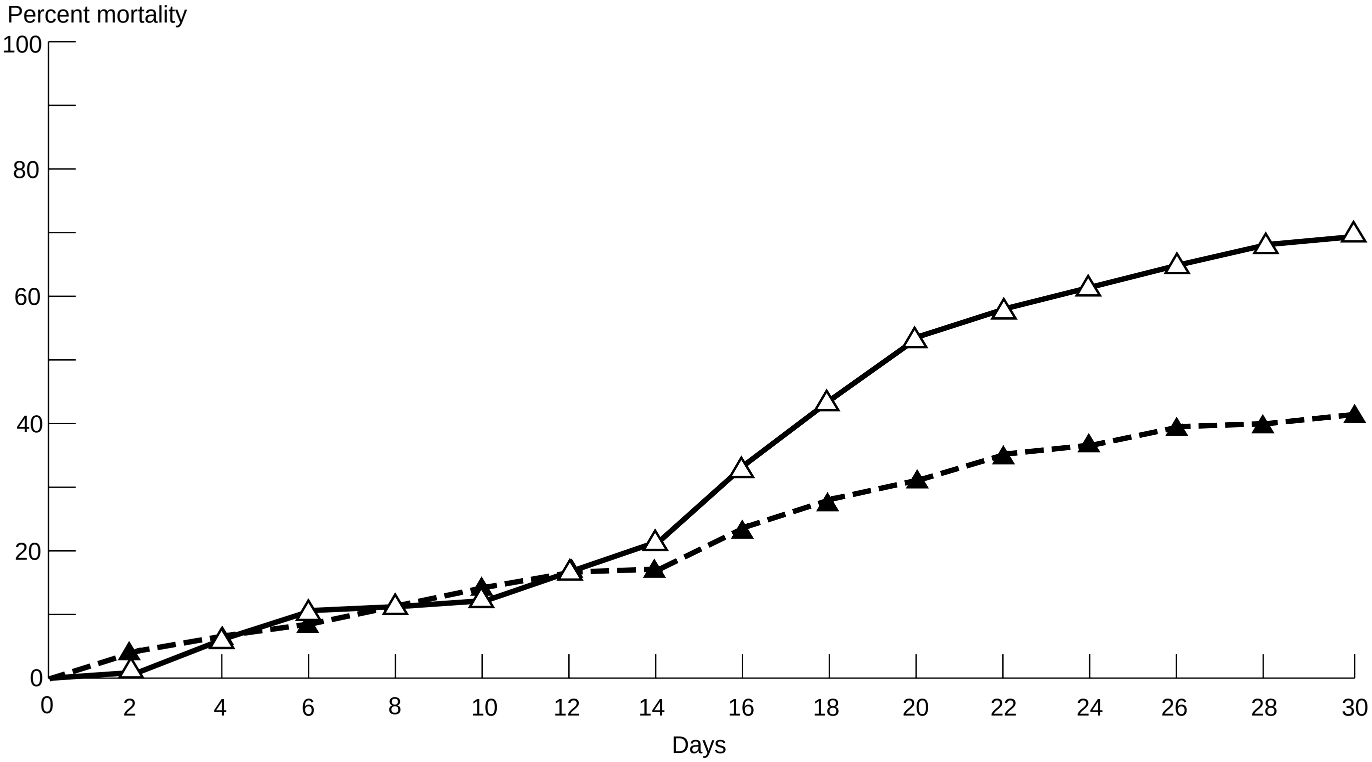

Figure I.3–4—Tube bioassay involving *N. locustae*-treated wheat bran given ad lib (from a petri dish) to third-instar *M. sanguinipes* maintained for 30 days postinoculation. Solid triangle = grasshoppers given untreated flakes of bran; open triangle = grasshoppers given 100×-strength treated bran.

Grasshoppers given as much of the 100×-treated bran as they wanted for 2 days consumed an average of 2.56 mg on the first day and 1.20 mg on the second. Therefore, each experimental grasshopper consumed an average of 3.76 mg of treated bran (roughly 6 small flakes) or 8.27×10^5 spores by the end of the second day. At the end of 2 days, control grasshoppers consumed less than half of the bran consumed by the experimental grasshoppers (fig. I.3–3). Mortality at 30 days p.i. was 75 percent higher for experimental grasshoppers than for those receiving control bran (fig. I.3–4). Mortality rates increased significantly in the experimental grasshoppers after 14 days p.i.

Conclusions

The LD_{50} values determined through the use of lettuce bioassays described in this chapter are generally similar to values reported in other studies. For example, Mussgnug and Henry (1979) calculated the LD_{50} for *N. locustae* in their study of *M. sanguinipes* to be 1.5×10^5 spores based upon a bioassay conducted for 24 days. In lettuce bioassays conducted at SDSU, spore quantities below 1×10^5 did not exhibit mortality rates that were consistently higher than those of the controls. Because the average bran flake from commercially prepared *Nosema*-treated bran theoretically contains only 2.32×10^3 spores, each grasshopper would need to ingest 43 flakes of treated bran to become inoculated with 1.0×10^5 spores. *Melanoplus sanguinipes* grasshoppers that were given only bran flakes during a 2-day period consumed an average of approximately six flakes of bran. In field studies, it is unlikely that many wild grasshoppers ingested more than 40 flakes of *Nosema*-treated bran; therefore, other factors must have influenced the reported effectiveness of *N. locustae* in the field (Henry 1971).

By formulating bran with *N. locustae* spores at a concentration 100 times that which is generally sold commercially (10^{11} spores/lb versus 10^9 spores/lb), it was possible to measure mortality rates caused by the resultant *Nosema* infections. The results generally are consistent with those reported by Reuter et al. 1990 (unpubl.) when the 100× rate—compared to the standard rate and untreated populations only—resulted in significant mortality to one of two field-treated species tested in cages.

References Cited

Henry, J. E. 1971. Experimental application of *Nosema locustae* for control of grasshoppers. Journal of Invertebrate Pathology 18: 389–194.

Henry, J. E. 1985. *Melanoplus* spp. In: Singh, P.; Moore, R. F., eds. Handbook of insect rearing. Amsterdam: Elsevier Science Publishing B.V.: 451–464.

Henry, J. E.; Oma, E. A. 1974. Effects of prolonged storage of spores on field applications of *Nosema locustae* (Microsporida: Nosematidae) against grasshoppers. Journal of Invertebrate Pathology 23: 371–377.

Henry, J. E.; Oma, E. A. 1975. Sulfunamide antibiotic control of *Malameba locustae* (King and Taylor) and its effect on grasshoppers. Acrida 4: 217–226.

Henry, J. E.; Tiahrt, K.; Oma, E. A. 1973. Importance of timing, spore concentrations, and spore carrier levels in application of *Nosema locustae* (Microsporida: Nosematidae) for control of grasshoppers. Journal of Invertebrate Pathology 21: 263–272.

Mussgnug, G. L.; Henry, J. E. 1979. Compatibility of malathion and *Nosema locustae* Canning in *Melanoplus sanguinipes.* Acrida 8: 77–81.

References Cited–Unpublished

Reuter, K C.; Foster, R. N.; Hildreth, M.; Colletto, D.; Cushing, W. J.; Pucelik, M. J.; Kahler, D.; Houston, R.; Scott, A. 1990. Preliminary investigations of the effect of a greatly increased rate of *Nosema locustae* on rangeland grasshopper populations. In: Cooperative Grasshopper Integrated Pest Management Project, 1990 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 169–174.

Bioassays of *Nosema locustae:* An Outline of Procedures

I. Purpose of the outline is to describe two protocols to measure the virulence of *Nosema locustae* spores.

- A. First Protocol: used for spores stored in water
- B. Second Protocol: used for spores already adhered to bran

II. Lettuce Bioassay

- A. Protocol
  1. Obtain 1,000 lab-reared, third-instar *Melanoplus sanguinipes* (Canadian strain) grasshoppers.
  2. Dilute spores to the following concentrations: 0, 1×10^4, $1 \times 10^{4.5}$, 1×10^5, $1 \times 10^{5.5}$, 1×10^6 per 10µL distilled water.
  3. Apply 10µL of the appropriate concentration to 7-mm lettuce disks.
  4. Cool grasshoppers to 39 °F (4 °C), and distribute each grasshopper into a glass vial.
  5. Add disks to vials and wait until the entire disk is consumed.
  6. Distribute grasshoppers into appropriate bioassay tubes.
  7. Maintain grasshoppers for 20 days, daily feeding them lab-reared rye grass and sulfa-coated rolled oats.
  8. Record grasshopper deaths each day.
  9. Calculate the LD_{50} value based upon the total mortality after 20 days p.i.
- B. Results
  1. Largest number of deaths in the grasshoppers infected with 10^6 spores occurred between 10 days and 14 days p.i.
  2. Calculated LD_{50} for the bioassay reported in this study was 1.19×10^5.

III. Bran Bioassay

- A. Protocol
  1. Formulate *Nosema locustae*-treated bran at a concentration of 1×10^{11} spores/lb (100 times higher than the concentration commercially available).
  2. Prepare two large rearing cages each containing 100 lab-reared, third-instar *Melanoplus sanguinipes* (Canadian strain) grasshoppers.
  3. Add 2 g of treated bran (in a petri dish) to one cage and 2 g of control bran to the other cage (add no other food source).
  4. After 24 hours, replace each petri dish with petri dishes containing another 2 g of appropriate bran.
  5. After another 24 hours, distribute grasshoppers into appropriate bioassay tubes, and maintain as described above for 30 days.
  6. Data can be reported only as net percent mortality.
- B. Results
  1. Consumption of control and treated bran can be measured by comparing the weight of the leftover bran inside each cage to the weight of similar bran stored outside the cage.
  2. In our first bran bioassay, on average 3.76 mg of treated bran and 1.90 mg of control bran was consumed by the grasshoppers during the 2-day infection period (theoretically 8.27×10^5 spores consumed per grasshopper).
  3. Experimental grasshoppers exhibited a 75-percent increased level of mortality at 30 days p.i. compared with grasshoppers receiving control bran at rates near 2.5×10^9 per ha on 2 kg (approx. 1×10^9 spores/lb) wheat bran.

I.4 Utility of *Nosema locustae* in the Suppression of Rangeland Grasshoppers

James L. Vaughn, Wayne M. Brooks, John L. Capinera, Terry L. Couch, and Joe V. Maddox

Editorial note: The authors served as an independent review team and prepared this report on *Nosema locustae* in 1991 at the request of the U.S. Department of Agriculture, Animal and Plant Health Inspection Service, Plant Protection and Quarantine's Grasshopper Integrated Pest Management Project. The internal report contains guidelines and recommendations for the use of *Nosema locustae* and is reproduced in the User Handbook because of the importance of the information the report contains. The present version has been edited to be consistent in style and tone with the User Handbook.

Nosema locustae is a microsporidium pathogenic (disease-causing) to a wide range of grasshoppers (more than 90 species are susceptible). It can be easily mass produced and formulated in baits for use as a biological control agent. Although many species of microsporidia are known to act as important naturally occurring biological control agents of insects, very few can be appropriately used as traditional microbial insecticides.

Laboratory studies, simulation models, and some field experiments suggest that *N. locustae* may be successfully utilized for longrange grasshopper control. But there is little or no evidence that *N. locustae* can be used effectively as a microbial insecticide for short-term control of grasshopper populations.

Inducing infections in insect populations is, at best, difficult. Many variables affect the onset and duration of an epizootic (disease outbreak). In the case of grasshoppers, the number and extent of variables are especially troublesome. The number of grasshopper species present, age of grasshoppers, and population density all influence the outcome of field applications. Therefore, the use of *N. locustae* as a grasshopper biological control agent should be considered as part of a long-term suppression effort but not as a microbial insecticide in direct competition with chemical pesticides.

Diseases that affect insects should have great potential for grasshopper control primarily because many grasshopper species readily eat bait into which pathogens can be incorporated. The extensive information generated by *Nosema locustae* studies will be of great help in this area. Domestic and international efforts should be made to identify and isolate other grasshopper pathogens for use as biological control agents.

In preparation for the analysis that is the foundation for this chapter, we were provided with a number of documents, including representative scientific publications, annual reports, and technical reports (see attached list). In addition, we discussed selected questions with Jerome Onsager, Robert Staten, and Jan Meneley.

After consideration of this information, we made the following specific recommendations:

1. *Nosema locustae* should be used to suppress rangeland grasshoppers in environmentally sensitive areas where cost, rapid knockdown, and high levels of control are not primary concerns. In such areas where insecticidal applications are not possible, continued use of *N. locustae* may be warranted. In these areas it may aid in the long-term management of the pest, and its use may allow researchers to address some of the important ecological questions surrounding it. These subjects are discussed in the following section.

2. Higher rates and/or multiple applications should be used where environmental sensitivities outweigh the higher costs involved.

In most of the past field tests with *N. locustae*, the dosage rate of 1×10^9 spores per acre appears to have been predicated more on the economics involved in a grasshopper control program rather than on the actual dose required for effective grasshopper suppression. As estimates of the number of spores per bran flake at this standard rate of application are considerably below LD_{50} (the dose where 50 percent of exposed individuals are killed) rates for *Melanoplus sanguinipes* and *M. bivittatus*, the effectiveness of higher dosage rates needs further evaluation. Laboratory bioassays support the enhanced effectiveness of *Nosema locustae* at higher dosages, although field studies have produced conflicting results.

In tests with up to five times the standard rate, greater reductions in grasshopper densities have not been obtained. However, in tests with 100 times the standard rate and where small field cages were also used to evaluate treatment effectiveness, grasshopper mortality was significantly higher, at least with *M. sanguinipes.* Despite the obvious costs of using higher dosage rates, the potential for enhancing the effectiveness of a readily available and registered biological control agent for use in environmentally sensitive areas may outweigh economic considerations.

In these sensitive areas where higher dosage rates and multiple applications of spores may be used, the methods of evaluation should be improved to include confinement of known numbers of the various grasshopper species in laboratory and field cages. Thus, along with monitoring population densities at appropriate time intervals in field plots, known numbers of treated and untreated grasshoppers should be confined in small field cages on untreated rangeland as well as under laboratory conditions. This evaluation plan will allow more accurate estimates of *N. locustae*'s primary effects on infection and mortality rates, as well the secondary effects on grasshopper food consumption, longevity, fecundity (reproductive capability), and vertical transmission.

3. Use of *Nosema locustae* at presently recommended dosages does not reliably provide an adequate level of suppression. *N. locustae* has been shown to induce measurable reductions in grasshopper longevity, fecundity, and consumption rates under controlled conditions in laboratory and field cages. Also, numerous examples from Canada and the United States indicate that it is possible to obtain significant reductions in grasshopper numbers and damage under field conditions using *Nosema.* However, results are not consistent. Reports of apparent failure also exist and many of the "testimonial-type" data are suspect. Reasons given for the apparent failure of *Nosema locustae* to suppress grasshoppers include

   a. Suboptimal applications of the product: low-quality spores, bad weather, equipment failure, etc.

   b. Poor targeting of the product: grasshopper species of low susceptibility or in the wrong development stage.

   c. Incorrect assessment of the product: inadequate sampling or poor experimental design.

   d. Unreasonable expectations of the product: applicators, evaluators, and land managers expect insecticidal activity from a product that inherently cannot provide rapid or high levels of control.

As long as there are available insecticides that do provide high levels of control (70–95 percent is normal), control by *N. locustae* (30–40 percent under the **best** of conditions) will appear inadequate to ranchers and others concerned with economical, reliable grasshopper suppression. Until the basis for the inconsistencies is better understood, *N. locustae* should be reserved for areas where high levels of control are not essential, or where chemical insecticide usage is not a viable option.

If *N. locustae* is used in ecologically sensitive areas, then research should be conducted to determine the stability characteristics of the formulated bran product. Although data in the literature support the conclusion that the *N. locustae* inoculum is active at the time of formulation, nothing in the literature describes the viability of the *N. locustae* formulations just prior to aerial application.

Pathogens that affect insects are markedly sensitive to elevated temperatures, and significant reduction of activity occurs at temperatures as low as 104 °F (40 °C). If no special handling of the *N. locustae* formulation is routinely done as part of the application program, it is conceivable that the bran formulation could be exposed to temperatures during transit and site storage which could cause a significant, serious biological degradation of the product. It is possible that, in several of the studies, site storage conditions could have had a severe negative effect on the formulation.

Therefore, the committee suggests that a thermal death time-study be developed for the *N. locustae* formulation and storage parameters be defined for the product. These steps will ensure that, if and when future applications are made, shipping specifications and site storage requirements of the formulations can be adjusted to preserve the material's efficacy. With handling protocols in place, the viability of the product can be assured up to the point of application.

In addition, bioassays of samples of the *N. locustae* bran formulation from the aircraft hopper should accompany each application. Information from these assays will aid in determining if the formulation was shipped and stored under the proper conditions as specified by data obtained from the thermal death time-study.

Additional research on application techniques other than bait seem warranted given the dearth of information in the literature. In particular, conventional low-volume and ultralow-volume liquid applications, with various adjuvants (additives) to increase droplet deposition and decrease evaporation, should be investigated.

Nosema locustae References

Availability note: Several of the following citations come from annual reports prepared for the Grasshopper Integrated Pest Management Project but not distributed outside the Animal and Plant Health Inspection Service. Individual photocopies of these materials are available on request from USDA, APHIS, Plant Protection and Quarantine, 4700 River Road, Riverdale, MD 20737.

Brooks, W. M. 1988. Entomogenons protozoa. In: Ignoffo, C. M. ed. CRC handbook of natural pesticides, vol. 5: Microbial insecticides, part A. Boca Raton, FL: CRC Press: 1–149.

Erlandson, M. A.; Ewen, M. K.; Mukerji, M. K.; Gillott, C. 1986. Susceptibility of immature stages of *Melanoplus sanguinipes* to *Nosema cuneatum* Henry and its effect on host fecundity. Canadian Entomologist 118: 29–35.

Ewen, A. B.; Mukerji, M. K. 1980. Evaluation of *Nosema locustae* (Microsporida) as a control agent of grasshopper populations in Saskatchewan. Journal of Invertebrate Pathology 35: 295–303.

Germida, J. J.; Ewen, A. B.; Onofriechuk, E. E. 1987. *Nosema locustae* Canning (Microsporida) spore populations in treated field soils and resident grasshopper populations. Canadian Entomologist 119: 355–360.

Henry, J. E. 1969. Extension of the host range of *Nosema locustae* in Orthoptera. Annals of Entomological Society of America 62: 452–453.

Henry, J. E. 1971. Experimental application of *Nosema locustae* for control of grasshoppers. Journal of Invertebrate Pathology 18: 389–394.

Henry, J. E. 1971. *Nosema cuneatum* sp.n. (Microsporida: Nosematidae) in grasshoppers (Orthoptera: Acrididae). Journal of Invertebrate Pathology 17: 164–171.

Henry, J. E. 1972. Epizootiology of infections by *Nosema locustae* Canning (Microsporida: Nosematidae) in grasshoppers. Acrida 1: 111–120.

Henry, J. E. 1977. Development of microbial agents for the control of Acrididae. Revista de la Sociedad Entomológica de Argentina 36: 125–134.

Henry, J. E. 1981. The value of *Nosema locustae* in control of grasshoppers. U.S. Agency for International Development regional food crop protection project: Biological control of pests. Unpublished paper in Dakar, Senegal. Feb. 9–13.

Henry, J. E. 1982. Production and commercialization of microbials—*Nosema locustae* and other protozoa. In: Invertebrate pathology and microbial control proceedings, IIIrd international colloquium on invertebrate pathology; 6–10 September 1982; Brighton, UK. [Place of publication unknown]: Society of Invertebrate Pathology: 103–106.

Henry, J. E. 1985. Effect of grasshopper species, cage density, light intensity, and method of inoculation on mass production of *Nosema locustae* (Microsporida: Nosematidae). Journal of Economic Entomology 78: 1245–1250.

Henry, J. E.; Oma, E. A. 1974. Effects of infections by *Nosema locustae* Canning, *Nosema acridophagus* Henry, and *Nosema cuneatum* Henry (Microsporida: Nosematidae) in *Melanoplus bivittatus* (Say) (Orthoptera: Acrididae). Acrida 3: 223–231.

Henry, J. E.; Oma, E. A. 1974. Effect of prolonged storage of spores on field applications of *Nosema locustae* (Microsporida: Nosematidae) against grasshoppers. Journal of Invertebrate Pathology 23: 371–377.

Henry, J. E.; Onsager, J. A. 1982. Large-scale test of control of grasshoppers on rangeland with *Nosema locustae.* Journal of Economic Entomology 75: 31–35.

Henry, J. E.; Onsager, J. A. 1982. Experimental control of the Mormon cricket, *Anabrus simplex,* by *Nosema locustae* (Microsporida: Nosematidae), a protozoan parasite of grasshoppers (Orthoptera: Acrididae). Entomophaga 27: 197–201.

Henry, J. E.; Oma, E. A.; Onsager, J. A. 1978. Infection of the corn earworm, *Heliothis zea,* with *Nosema acridophagus* and *Nosema cuneatum* from grasshoppers: relative virulence and production of spores. Journal of Invertebrate Pathology 34: 125–132.

Henry, J. E.; Oma, E. A.; Onsager, J. A. 1978. Relative effectiveness of ULV spray applications of spores of *Nosema locustae* against grasshoppers. Journal of Economic Entomology 71: 629–632.

Henry, J. E.; Tiahrt, K.; Oma, E. A. 1973. Importance of timing, spore concentrations, and levels of spore carrier in applications of *Nosema locustae* (Microsporida: Nosematidae) for control of grasshoppers. Journal of Invertebrate Pathology 21: 263–272.

Hildreth, M. B.; Fuller, B. 1990. Laboratory bioassay to compare virulence of grasshopper pathogens such as *Nosema locustae* from different sources. In: Cooperative Grasshopper Integrated Pest Management Project, 1990 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 204–209.

Hildreth, M. D.; Walgenbach, D. D. 1990. Pathological effects of increased concentrations of *Nosema locustae* on two different grasshopper species with different susceptibilities. In: Cooperative Grasshopper Integrated Pest Management Project, 1990 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 196–201.

Hirsch, D. N.; Foster, N.; Smith, M.; Battaglia, T.; Morgans, M. 1988. Two year evaluation of the 1987 *Nosema locustae* grasshopper control block, North Dakota. In: Cooperative Grasshopper Integrated Pest Management Project, 1988 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 47–51.

Jech, L.; Foster, N.; Sluss, T.; Keim, D.; Miller, J.; Franklin, G. 1990. Application of *Nosema locustae* treated bran bait on grasshopper populations near Delta Junction, Alaska. In: Cooperative Grasshopper Integrated Pest Management Project, 1990 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 55–57.

Johnson, D. L. 1987. Evaluation of grasshopper control with *Nosema locustae* and insecticide on bran carrier. In: Farming for the future. Final Rep. 84-0418. Lethbridge, CN: Agriculture Canada. [Unpublished.] 41 p.

Johnson, D. L.; Henry, J. E. 1987. Low rates of insecticides and *Nosema locustae* (Microsporida: Nosematidae) on baits applied to roadside for grasshopper (Orthoptera: Acrididae) control. Journal of Economic Entomology 80: 685–689.

Johnson, D. L.; Pavlikova, E. 1986. Reduction of consumption by grasshoppers (Orthoptera: Acrididae) infected with *Nosema locustae* (Microsporida: Nosematidae). Journal of Invertebrate Pathology 48: 232–238.

Lockwood, J. A. 1988. Biology and recommendations for use of *Nosema locustae* Canning: a biological control agent of grasshoppers. Bull. B-917. Laramie, WY: University of Wyoming and Wyoming Agricultural Experiment Station.

Lockwood, J. A. 1989. Cannibalism in rangeland grasshoppers (Orthoptera: Acrididae) attraction to cadavers. Journal of the Kansas Entomological Society 61: 379–387.

Lockwood, J. A; DeBrey, L. D. 1990. Direct and indirect effects of *Nosema locustae* (Canning) (Microsporida: Nosematidae) on rangeland grasshoppers (Orthoptera: Acrididae). Journal of Economic Entomology 83: 377–383.

Lockwood, J. A.; Larsen, J. C. 1988. *Nosema locustae:* principles, promises and problems. In: Proceedings, 44th Wyoming weed and pest conference; [date of meeting unknown; Douglas, WY. [Place of publication and publisher unknown]: 18–25.

MacVean, C. M.; Capinera, J. L. 1991. Pathogenicity and transmission potential of *Nosema locustae* and *Vairimorpha* n.sp. (Protozoa: Microsporida) in Mormon crickets (*Anabrus simplex:* Orthoptera: Tettigoniidae). Journal of Invertebrate Pathology 57: 23–36.

Morris, O. N. 1985. Susceptibility of the migratory grasshopper, *Melanoplus sanguinipes* (Orthoptera: Acrididae), to mixtures of *Nosema locustae* (Microsporida: Nosematidae) and chemical insecticides. Canadian Entomologist 117: 131–132.

Mussgnug, G. L.; Henry, J. E. 1979. Compatibility of malathion and *Nosema locustae* Canning in *Melanoplus sanguinipes* (F). Acrida 8: 77–81.

Oma, E. A.; Hewitt, G. B. 1984. Effect of *Nosema locustae* (Microsporida: Nosematidae) on food consumption in the differential grasshopper (Orthoptera: Acrididae). Journal of Economic Entomology 77: 500–501.

Onsager, J. A. 1987. Current tactics for suppression of grasshoppers on rangeland. ARS–50. Bozeman, MT: U.S. Department of Agriculture, Agricultural Research Service: 60–66.

Onsager, J. A. 1987. Integrated management of rangeland grasshoppers. In: Capinera, J. L., ed. Integrated pest management on rangeland: a shortgrass prairie perspective. Boulder, CO: Westview Press: 196–204.

Onsager, J. A. 1988. Assessing effectiveness of *Nosema locustae* for grasshopper control. Montana Ag Research 5: 12–16.

Onsager, J. A.; Henry, J. E. 1977. A method for estimating the density of rangeland grasshoppers (Orthoptera, Acrididae) in experimental plots. Acrida 6: 231–237.

Onsager, J. A.; Henry, J. E. 1988. Efficacy of *Nosema locustae* in control of Mormon cricket nymphs. In: Cooperative Grasshopper Integrated Pest Management Project, 1988 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 308–312.

Onsager, J. A.; Streett, D. A. 1989. Grasshopper pathogen field evaluation: *Nosema.* In: Cooperative Grasshopper Integrated Pest Management Project, 1989 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 214–219.

Onsager, J. A.; Streett, D. A. 1990. Grasshopper pathogen field evaluation: *Nosema*. In: Cooperative Grasshopper Integrated Pest Management Project, 1990 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 192–195.

Onsager, J. A.; Henry, J. E.; Foster, R. N.; Staten, R. T. 1980. Acceptance of wheat bran bait by species of rangeland grasshoppers. Journal of Economic Entomology 73: 548–551.

Onsager, J. A.; Rees, N. E.; Henry, J. E.; Foster, R. N. 1981. Integration of bait formulations of *Nosema locustae* and carbaryl for control of rangeland grasshoppers. Journal of Economic Entomology 74: 183–187.

Pfadt, R. E. 1985. Optimum grasshopper pest management trials in Platte County, Wyoming. Laramie, WY: University of Wyoming. [Unpublished research report.] 9 p.

Pfadt, R. E. 1986. Results of testing bran baits of *Nosema locustae* for extended control of rangeland grasshoppers. Laramie, WY: University of Wyoming and Wyoming Agricultural Experiment Station. 11 p.

Pomerinke, M. A.; French, J. D.; Bomar, C. R.; Lockwood, J. A. 1991. First through third year effects of *Nosema locustae* on grasshoppers of a mixed grass prairie. Metaleptea 13: 14.

Reuter, K. C.; Foster, R. N.; Walgenbach, D. R.; Erickson, S. 1988. Evaluation of *Nosema locustae* bran bait for control of grasshoppers on rangeland near Winnett, Montana, 1988. In: Cooperative Grasshopper Integrated Pest Management Project, 1988 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 23–28.

Reuter, K. C.; Foster, R. N.; Walgenbach, D. R.; Erickson, S. 1988. Immediate and second-year impact of *Nosema locustae* bran bait on populations of grasshoppers on rangeland near Winnett, Montana, 1988. In: Cooperative Grasshopper Integrated Pest Management Project, 1988 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 60–71.

Reuter, K. C.; Foster, R. N.; Hildreth, M.; Colletto, D.; Cushing, W. J.; Pucelik, M. J.; Kahler, D.; Houston, R.; Scott, A. 1990. Preliminary investigation of the effect of a greatly increased rate of *Nosema locustae* on rangeland grasshoppers. In: Cooperative Grasshopper Integrated Pest Management Project, 1990 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 169–174.

Streett, D. A.; Henry, J. E. 1984. Epizootiology of a microsporidium in field populations of *Aulocara elliotti* and *Psoloessa delicatula* (Insecta: Orthoptera). Canadian Entomologist 116: 1439–1440.

Streett, D. A.; Onsager, J. A. 1988. Pathogen research: protozoa and virus. In: Cooperative Grasshopper Integrated Pest Management Project, 1988 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 306–307.

U.S. Department of Agriculture, Animal and Plant Health Inspection Service. 1987. Rangeland Grasshopper Cooperative Management Program: final environmental impact statement. Washington, DC: U.S. Department of Agriculture, Animal and Plant Health Inspection Service. 221 p.

Vinje, E. W. 1991. Personal correspondence to *Nosema* review team, Sept. 3, 1991: 6 p.

I.5 Identification of Fungal Pathogens of Grasshoppers

Michael J. Bidochka and Donald W. Roberts

Introduction

Grasshoppers are host to a wide range of micro-organisms that cause disease. Of these, the fungi provide spectacular appearance of disease symptoms. On a larger scale, fungi can devastate whole populations of grasshoppers. Some of these fungi cannot grow without a grasshopper host (obligate pathogens); other fungi are easily cultured in the laboratory and can infect a wide range of insects including grasshoppers (facultative pathogens). In this chapter, we will examine methods used to discriminate pathogenic fungal infections from bacterial or nonpathogenic fungal growth on a dead insect. We will also discuss the most probable fungal infections found in the field.

Fungi Pathogenic to Grasshoppers

There are two main groups of fungi that have species pathogenic to grasshoppers: the zygomycetes and the deuteromycetes. Some zygomycete species are obligate pathogens of grasshoppers. The deuteromycetes that are pathogenic to grasshoppers are facultative pathogens.

Zygomycetes (Entomophthorales).—The pathogenic Entomophthorales are complex and poorly understood. The only confirmed pathogens that infect grasshoppers belong to the *Entomophaga grylli* complex. There are at least three pathotypes of the *E. grylli* complex. The term pathotype refers to the type of grasshopper that is infected. The three pathotypes also differ with respect to their life cycles, host grasshoppers, and growth requirements (Ramoska et al. 1988). Two of the pathotypes are native to North America, and a third pathotype has been isolated from a grasshopper species in Australia.

Pathotype 1 infects the bandedwinged grasshoppers (Oedipodinae). The grasshopper species most commonly infected are *Camnula pellucida* and *Dissosteira carolina*. Pathotype 2 infects melanopline grasshoppers (Melanoplinae) and the species most commonly infected are *Melanoplus* and *Hesperotettix* spp. Pathotype 3, the Australian isolate, infects bandedwinged and melanopline grasshoppers under laboratory conditions.

Disease Characteristics.—*Entomophaga* spp. are the most common and widespread pathogens of grasshoppers in North America. Disease symptoms in the advanced stage are characteristic and easy to recognize. Shortly before death, infected grasshoppers crawl to the tops of plants, fenceposts, or any other elevated position. There they die with their legs wrapped around the plant stalk and heads pointed upward.

Examining the specimen found in the characteristic “summit disease” is simple. Open the abdomen or poke a hole in it with a sterile toothpick and a place sample of this on a microscope slide with a drop of water. The inside of the grasshopper may contain a variety of fungal bodies, but the most common are large (50 m in diameter), spherical, thick-walled resting spores. If the grasshopper is *Camnula,* the infection is probably pathotype 1; in a melanopline grasshopper, probably pathotype 2.

External sporulation is also used to discriminate between pathotype 1 and 2 infections. Grasshoppers suspect of *E. grylli* infection are placed in a humid environment, such as petri dish containing 1.5 percent agar. Within 24 hours some of the specimens will show sporulation (white rings) on the abdominal segments. Pathotype 1 will show external sporulation (conidia approximately 50 µm in diameter) as well as the internal resting spores. Pathotype 2 will not show external sporulation.

North Dakota Introductions.—Recently, pathotype 3 *(E. praxibuli)* has been introduced into North Dakota from Australia. This fungus infects both oedipodine and melanopline grasshoppers. External growth on a melanopline grasshopper may be indicative of *E. praxibuli* infection. However, we caution against the use of morphology and growth characteristics as tools in differentiating the three *Entomophaga* pathotypes.

We have developed DNA (deoxyribonculeic acid) probes that could be used differentiate the three pathotypes (Bidochka et al., 1995). We have also devised a method by which the resting spores of these fungi can be fractured, and the DNA can be isolated and used as a template for the pathotype-specific probes.

Deuteromycetes.—Worldwide, the most common deuteromycete infections in grasshoppers are *Beauveria bassiana*, *Metarhizium anisopliae*, and *Aspergillus flavus.* In central Africa, *Metarhizium flavoviride* is found more commonly than *M. anisopliae.*

***Disease Characteristics.*—**Grasshoppers that have an external white or green mycelial (filamentlike fungus) growth are also potential suspects of fungal infection. The most common non-*Entomophaga* infections found in the field are *B. bassiana, M. anisopliae* and *A. flavus*. *B. bassiana* infection is characterized by white mycelial growth on parts of the surface of the grasshopper; *M. anisopliae* and *A. flavus* infections are characterized by green surface growth. The conidia of these fungi are much smaller (approximately 5–10 μm in diameter) than the conidia of *Entomophaga grylli*. *M. anisopliae* conidia are rod shaped, but *M. flavoviride* conidia are more rounded or elliptical. *B. bassiana* conidia are globose (round or globelike), and *A. flavus* conidia are spherical. For more detailed descriptions and microphotographs of entomopathogenic fungi, refer to Samson et al. (1988) and Poinar and Thomas (1984).

***Isolating Pathogenic Deuteromycetes.*—**Several selective media for the isolation of *B. bassiana* and *M. anisopliae* have been tested. The best medium for selective isolation of *B. bassiana* and *M. anisopliae* is 30 g of wheat germ in 1 L of water, autoclaved for 10 minutes and filtered through four layers of cheesecloth. To this are added 0.25 g chloramphenicol, 0.75 mg benlate (50 percent benomyl), 0.30 g dodine, 10 mg crystal violet, and 15 g agar (Chase et al. 1986).

The mycelia on the surface of the grasshopper can be picked with a sterile toothpick or sterile wire loop and streaked onto this agar-medium. The petri dishes should be wrapped in aluminum foil because exposure to light delays colony growth. Optimal growth occurs at 79 °F (27 °C) for these fungi. If the fungus grows, then it may be one of the pathogenic deuteromycetes. If the fungus does not grow, it may simply be a nonpathogenic fungus growing on the dead grasshopper.

B. bassiana, *M. anisopliae*, and *M. flavoviride* also can be differentiated based on patterns of DNA fragments generated by random amplification of polymorphic DNA (RAPD) and with molecular probes using the RAPD fragments (Bidochka et al. 1994).

Other fungi that may infect grasshoppers include *Verticillium lecanii*, *Nomuraea rileyi*, and *Paecilomyces* sp.

Assessment of Fungal Disease

To prove that a certain fungal isolate is the causative agent in grasshopper death, lab personnel must follow these steps: (1) The fungus must be isolated from the grasshopper. (2) The fungus must be grown in media. (3) The fungus must cause disease either by injection of conidia into the body cavity or by exposing the insect to fungal conidia. Most entomopathogenic fungi normally infect by passing through the insect exoskeleton. It is preferable that the host insect from which the fungus was isolated be the test insect. This is particularly true for the Entomophthorales. For deuteromycetous fungi, a test insect such as wax moth larvae *(Galleria mellonella)* or silkworm larvae *(Bombyx mori)* may be used. (4) Finally, the fungus must be reisolated from the test insect.

The best diagnostic tools for differentiating *B. bassiana, M. anisopliae, M. flavoviride*, and the *Entomophaga* are molecular probes. The use of these probes is not difficult, and results are generally conclusive. In the near future, the use of such probes will be commonplace in fungal taxonomy.

Suggested References

Bidochka, M. J.; McDonald, M. A.; St. Leger, R. J.; Roberts, D. W. 1994. Differentiation of species and strains of entomopathogenic fungi by random amplification of polymorphic DNA (RAPD). Current Genetics 25: 107–113.

Bidochka, M. J.; Walsh, S.R.A.; Ramos, M. E.; St. Leger, R. J.; Silver, J. C.; Roberts, D. W. 1995. Pathotypes in the *Entomophaga grylli* species complex of grasshopper pathogens differentiated with random amplification of polymorphic DNA and cloned-DNA probes. Applied and Environmental Microbiology 61: 556–560.

Chase, A. R.; Osborne, L. S.; Ferguson, V. M. 1986. Selective isolation of the entomopathogenic fungi *Beauveria bassiana* and *Metarhizium anisopliae* from an artificial potting medium. Florida Entomologist 69: 285–292.

Poinar, Jr., G. O.; Thomas, G. M. 1984. Laboratory guide to insect pathogens and parasites. New York: Plenum Press.

Ramoska, W. A.; Hajek, A. E.; Ramos, M. E.; Soper, R. S. 1988. Infection of grasshoppers (Orthoptera: Acrididae) by members of the *Entomophaga grylli* species complex (Zygomycetes: Entomophthorales). Journal of Invertebrate Pathology 52: 309–313.

Samson, R. A.; Evans, H. C. Evans; Latge, J–P. 1988. Atlas of entomopathogenic fungi. New York: Springer–Verlag.

I.6 Grasshopper Pathogens and Integrated Pest Management

Donald L. Hostetter and Douglas A. Streett

Introduction

Some 97 percent of all animals on Earth are invertebrates, and between 75 and 80 percent of these are insects. One of the most serious gaps in our knowledge of invertebrates in general, and insects specifically, is a thorough understanding of their diseases.

As would be expected, mankind's knowledge of insect parasites and predators preceded that of the disease-causing agents of insects. Although the early interests in insect pathology were primarily concerned with beneficial insects, such as the honeybee and the silkworm, many investigators recognized that harmful insects were subject to disease. Almost from the time of their discovery, insect diseases have been proposed as possible tools for controlling insect pests.

It was not until 1836 that Agostino Bassi, for whom the insect-infecting fungus *Beauveria bassiana* is named, suggested that microorganisms could be used to control destructive insects. Another 43 years would pass before Elie Metchnikoff published his account of a natural infection of the wheat cockchafer *(Anisoplia austriaca)* by the green-muscardine fungus (*Metarhizium anisopliae* [Metchnikoff]) and provided experimental methods for testing the possibility of controlling insects with fungi (Steinhaus 1956).

Micro-organisms with the ability to cause acute and chronic disease in grasshoppers and locusts currently are found among the bacteria, fungi, protozoa, rickettsia, and viruses (Bidochka and Khachatourians 1991).

Bacteria

One of the first attempts to use bacteria as a control agent of insects was against grasshoppers in Mexico (d'Herelle 1911). The bacterium *Coccobacillus acridorum* d'Herelle was isolated from large numbers of dying grasshoppers that had migrated to Mexico from Guatemala. D'Herelle claimed to have begun epidemics among grasshopper populations in Mexico, Colombia, and Argentina, along with some success in Algeria and Tunisia. His results were not reproducible by others and soon viewed with doubt. This bacteria was later determined to be *Aerobacter aerogenes* (Kruse), a member of the coliform group capable of invading warmblooded animals (Steinhaus 1949).

Another bacterium, *Serratia marcescens* Bozio, was isolated from desert locusts (*Schistocerca gregaria* [Forskäl]) raised in a laboratory. *S. marcescens* was cultured, formulated on a bran bait, and used in field tests against the desert locust in Kenya. The results were uncertain (Stevenson 1959). This gram-negative bacterium is found worldwide and is well known as a pathogen of laboratory insects.

The most promising bacteria currently being used for insect control belong to the spore-forming group *Bacillus thuringiensis* Berliner, often referred to as *"Bt."* A diamond-shaped crystalline toxin is produced within the bacteria as they mature and form spores. The toxin is the active ingredient that kills insect larvae. After it is consumed, the toxin is dissolved in the insects' alkaline gut juices and becomes activated. The gut is unable to process food, the larvae stop eating, and the gut ruptures, causing the larvae to die.

Grasshoppers have a built-in barrier against *Bt* because their gut juices are acidic, and the absence of an alkaline environment prevents the toxin from dissolving and becoming activated (Prior and Greathead 1989). Two isolates of *Bt* from the Dulmage Collection originally isolated from grasshoppers were inactive against *M. sanguinipes,* as were 26 other prospective isolates (Streett and Woods 1992 unpubl). Continued examination of the *Bt* group, along with advances in formulation chemistry and genetic manipulation, may produce future successes with these bacteria against grasshoppers.

Fungi

More than 750 species of insect-infecting fungi have been documented (National Academy of Sciences 1979, Roberts and Humber 1984). Although fungi are among the best known and most often reported pathogens associated with grasshoppers and locusts, only a few different fungi have been recorded. The most common are *Beauveria bassiana* (Balsamo) Vuillemin, *Metarhizium anisopliae* (Metchnikoff) Sorokin, and *Entomophaga grylli* (Fresenius) Batko.

Fungi are "contact" pathogens. They do not infect when they are eaten by the insect, as do other pathogens. Fungal infection may occur during the feeding process when conidia contact the mouthparts (Foster et al. 1991 unpubl.). The infection process begins after a spore comes in contact with a suitable host and germinates in the form of a "tube." The tube penetrates the body wall, enters the body cavity, and releases a protoplast that begins asexual reproduction. Rapid growth of the fungus overwhelms the insect host and it dies. After death of the host, the fungus grows back through the body wall and forms vegetative stalks that produce primary spores (conidia) that are forcibly discharged into the atmosphere. These spores are capable of continuing the infection cycle. Toward the end of the season, or if environmental conditions are unfavorable for conidia production, "resting spores" are produced. Resting spores are the environmentally resistant or protective stage that overwinters in the soil litter or in dead grasshoppers.

Beauveria bassiana has been successfully developed and used as a microbial control agent of various insects in the Soviet Union and China (Goettel 1992). Interest in *B. bassiana* as a control agent for rangeland grasshoppers has been renewed with the recent isolation of a strain—virulent to some species of grasshoppers—from a grasshopper in Montana (Johnson et al. 1988 unpubl., Foster et al. 1992 unpubl.).

Extensive laboratory and field testing of this strain has indicated good potential for control of grasshoppers and resulted in the first aerially applied field tests of *B. bassiana* against grasshoppers on rangeland in the United States (Foster et al. 1991–93 unpubl.). Technology for mass production has been developed by Mycotech Corporation (Butte, MT), and a commercial product was registered for use against rangeland grasshoppers by the Environmental Protection Agency in 1995.

B. bassiana is expected to be competitive with current chemical insecticides and could be a very useful microbial control agent in future grasshopper integrated pest management (IPM) programs.

Metarhizium anisopliae is another fungus that has been isolated from grasshoppers and is known to have a worldwide distribution. It also can be mass produced and formulated as a microbial insecticide. One isolate has been used successfully as a control agent against the sugarcane spittlebug in Brazil (Roberts et al. 1991). It has not been tested in the field as a grasshopper control agent but should be considered as a potential tool that merits further tests.

Entomophaga grylli, formerly referred to as a complex of fungi composed of "pathotypes," is now known to consist of at least four species: *E. calopteni* (Bessey) Humber, *E. macleodii*, *E. praxibuli*, and *E. asiatica*. *E. calopteni* is the only species that has been formally described to date (Humber 1989). *E. asiatica*, isolated from one grasshopper in Japan, was screened for activity and placed into the pathogenic insect fungus collection at the U.S. Department of Agriculture's Agricultural Research Service laboratory in Ithaca, NY (Carruthers et al. 1989 unpubl.). All *Entomophaga* spp. isolated from grasshoppers and locusts are infective only for members of this group. This fungus also has a worldwide distribution. *Entomophaga* spp., unlike *B. bassiana* and *M. anisopliae*, cannot be produced in large quantities on or in artificial media at the present time. *Entomophaga* spp. cannot be used as microbial insecticides in large-scale spray applications now.

A classical introduction method uses individually infected grasshoppers, each injected with an amount of the infective stage (protoplasts) of *Entomophaga* sp. that will cause their death within 7 to 10 days. Before dying of the fungus disease, the infected grasshoppers are released into a susceptible population in the field. Distribution of the disease occurs and is dependent upon dispersal of spores from dead, infected grasshoppers to noninfected ones within the population. A series of biological and environmental factors must occur in sequence before such epidemics develop.

One of the native North American fungi, *Entomophaga macleodii* (pathotype I) infects grasshoppers from several genera and produces infective conidia as well as resting spores. The primary host of this fungus is the clear-winged grasshopper *(Camnula pellucida* [Scudder]), which belongs to the bandwinged group of grasshoppers.

The other North American species is *E. calopteni* (pathotype II). It occurs only in a *Melanoplus* species (a member of the spurthroated group) and produces only resting spores upon death of the host.

The Australian fungus, *E. praxibuli,* was isolated from *Praxibulus* sp. grasshoppers in Australia in 1985 during a fungus epidemic. This fungus is similar to *E. macleodii* in producing both infective conidia and resting spores. Laboratory tests and field observations indicate that *E. praxibuli* has a greater host range than *E. macleodii* and is infective for at least 14 species of grasshoppers from the three major subfamilies: the spurthroated, slantfaced, and bandwinged grasshoppers.

Following a review of the known literature and a series of laboratory evaluations, the Australian isolate *E. praxibuli* was selected as a candidate for a classical biological control program for grasshopper populations in western North Dakota (Carruthers et al. 1989–91 unpubl.).

Protozoa

The microsporidia comprise the most important group of the protozoan pathogens of insects with over 250 species currently documented (Maddox 1987). The most probable route of infection occurs when insects' food is contaminated with spores. Upon ingestion into the midgut of a host, the spores forcibly extrude a hollow filament that penetrates or is placed near the epithelial cells lining the gut. The infective sporoplasm travels through the tube and into the cell, where asexual reproduction of spores begins. Spores can be released prior to death of the infected host through regurgitation or in feces.

Microsporidia also can be passed on to the next generation of host insects on the surface of eggs, or within eggs laid by infected females. Some microsporidia may also be mechanically transmitted by the feeding or ovipositing activities of insect parasites of the infected host. Microsporidial infections can range from acute, leading to death in several days, to chronic, with little evidence of infection and prolonged life stages. Microsporidia can be serious pathogens in laboratory colonies of insects.

Within the family Microsporida, the genera *Nosema* and *Vairimorpha* have proven to contain the most promising candidates for grasshopper and locust control. *Nosema locustae* (Canning) was first isolated from infected migratory locusts in a laboratory colony in Great Britain (Canning 1953). It has received the most attention as a biological control agent for grasshoppers. *Nosema* was thoroughly investigated in a series of laboratory and field evaluations, registered, and developed as the first commercial microbial product for grasshopper control (Henry 1978 and 1982, Henry and Oma 1981). Applications were difficult to evaluate and did not meet expectations. *N. locustae* was widely acclaimed but unfortunately is not extensively used in grasshopper control programs. For grasshopper control in environmentally sensitive areas, *N. locustae* is still worthy of consideration. In many cases, in sensitive areas, no action is chosen over *N. locustae* for economic reasons and because results with *Nosema* have been irregular *(See* I.4.).

Nosema acridophagus Henry and *N. cuneatum* Henry are two other grasshopper-isolated species of microsporidia that have potential as microbial control agents (Henry 1967, Henry and Oma 1974). Both have demonstrated variable virulence and have been adapted to production in surrogate hosts (certain species of caterpillars). These agents may have a place in future IPM programs (Streett 1987).

A *Vairimorpha* sp. was isolated from Mormon crickets *(Anabrus simplex* Haldeman) in Utah and Colorado during an epidemic in 1989. The crickets are very susceptible to this *Vairimorpha* and it may be considered as a control agent for Mormon crickets. Field observations indicate that infection causes increased mortality among crickets while decreasing development of nymphs and adult reproduction (Henry and Onsager 1989 unpubl.).

Viruses

The only viruses isolated from grasshoppers and cricket species to date are members of the entomopoxvirus and crystalline array virus groups. The entomopoxviruses are the best known of the viruses reported from grasshoppers and crickets. The entomopoxviruses isolated from *M. sanguinipes* have received the closest examination and evaluation (Henry and Jutila 1966). Fewer than 10 entomopoxviruses have been isolated from grasshoppers (Streett et al. 1986). Two other poxviruses, one from

Arphia conspersa Scudder and one from the African grasshopper *Oedaleus senegalensis* (Krauss), are potential microbial control agents (Streett 1987). These viruses were originally viewed with caution because of their resemblance to vertebrate orthopoxviruses (Bidochka and Khachatourians 1991). Examination of this group has revealed no biochemical similarity or infectivity of vertebrates, however (Arif 1984, Streett and McGuire 1990).

The crystalline array viruses do closely resemble the picornaviruses of vertebrates and are not currently considered to be exploitable as a microbial agent for grasshoppers (Greathead 1992).

Nuclear polyhedrosis viruses (NPV's), probably the most common of insect viruses, have not been isolated from grasshoppers or crickets. One report has documented transmission (by feeding) of an NPV from *Spodoptera littoralis* (a caterpillar) to both *Schistocerca gregaria* and *Locusta migratoria*, resulting in a phenomenon known as "dark cheeks" (Bensimon et al. 1987).

Summary

Grasshoppers and locusts, like all other animals, are subject to pathogenic micro-organisms. Representatives from all of the major groups of known pathogens have been isolated from grasshoppers and crickets. The fungi *Entomophaga* spp. and *Beauveria* spp. are the most frequently reported and observed pathogens. Spectacular mortality due to *Entomophaga* sp. is often observed within grasshopper populations throughout the world. Fungi, at the current time and state of technology, probably have the greatest potential as microbial control agents.

Bacterial pathogens do not exhibit much promise as tools for grasshopper control now. Technological advances in molecular biology may lead to development of strains of *Bacillus thuringiensis* that will be active against grasshoppers. Efforts to isolate bacteria, particularly sporeformers, from grasshoppers and crickets on a worldwide scale should be supported.

Protozoans, particularly *Nosema* spp. and *Vairimorpha* spp., are also promising candidates for reducing grasshopper populations in environmentally sensitive areas. Although *Nosema locustae*, the first registered and commercially produced microbial control agent for grasshopper suppression, has not met expectations, it still remains a viable alternative to chemical control in long-term management programs.

Continued research with grasshopper and cricket viruses undoubtedly will result in new isolates that may be considered as management tools. Viruses have the potential to be "tailored" to fit specialized control requirements in localized areas and may become a tool of choice—with substantial research and development—for long-term population reduction in grasshoppers in the future. Insect pathogens will play a larger role in future grasshopper management strategies as requirements for control are redefined and evolve in the decades ahead.

References Cited

Arif, B. M. 1984. The entomopoxviruses. Advances in Virus Research 29: 195–213.

Bensimon, A. S.; Zinger, E.; Gerasi, A.; Hauschner, I.; Sela, I. 1987. "Dark cheeks," a lethal disease of locusts provoked by a lepidopterous baculovirus. Journal of Invertebrate Pathology 50: 254–260.

Bidochka, M. J.; Khachatourians, G. G. 1991. Microbial and protozoan pathogens of grasshoppers and locusts as potential biocontrol agents. Biocontrol Science and Technology 1: 243–259.

Canning, E. U. 1953. A new microsporidian, *Nosema locustae n.* sp., from the fat body of the African migratory locust, *Locusta migratoria migratorioides* Reiche and Fairmaire. Parasitology 43: 287–290.

d'Herelle, F. 1911. Sur une épizootie de natur bactérienne sévissant sur les sauterelles au Mexique. Compt. rend. acad. sci. Paris: 152: 1413–1415.

Goettel, M. S. 1992. Fungal agents for biocontrol. In: Lomer, C. J.; Prior, C., eds. Biological control of locusts and grasshoppers. Wallingford, UK: C.A.B. International: 122–132.

Greathead, D. J. 1992. Natural enemies of tropical locusts and grasshoppers: their impact and potential as biological control agents. In: Lomer, C. J.; Prior, C., eds. Biological control of locusts and grasshoppers. Wallingford, UK: C.A.B. International: 105–121.

Henry, J. E. 1967. *Nosema acridophagus* sp. *n.*, a microsporidian isolated from grasshoppers. Journal of Invertebrate Pathology 9: 331–341.

Henry, J. E. 1978. Microbial control of grasshoppers with *Nosema locustae* Canning. Miscellaneous Publications Entomological Society America 11: 85–95.

Henry, J. E. 1982. Production and commercialization of microbials: *Nosema locustae* and other protozoa. In: Invertebrate pathology and microbial control, Proceedings 3rd. international colloquium on invertebrate pathology; [dates of meeting unknown]; Brighton, UK. [Place of publication and publisher unknown]: 103–106.

Henry, J. E.; Jutila, J. W. 1966. The isolation of a polyhedrosis virus from a grasshopper. Journal of Invertebrate Pathology 8: 417–418.

Henry, J. E.; Oma, E. A. 1974. Effects of infections by *Nosema locustae* Canning, *Nosema acridophagus* Henry, and *Nosema cuneatum* Henry (Microsporida: Nosematidae) in *Melanoplus bivittatus* (Say) (Orthoptera: Acrididae). Acrida 3: 223–231.

Henry, J. E.; Oma, E. A. 1981. Pest control by *Nosema locustae*, a pathogen of grasshoppers and crickets. In: Burges, H. D., ed. Microbial control of pests and plant diseases 1970–1980. New York: Academic Press: 573–586.

Humber, R. A. 1989. Synopsis of a revised classification for the Entomophthorales: (Zygomycotina). Mycotaxon 34: 441–460.

Maddox, J. V. 1987. Protozoan diseases. In: Fuxa, J. R.; Tanada, Y., eds. Epizootiology of insect diseases. New York: John Wiley & Sons: 417–452.

National Academy of Sciences. 1979. Microbial processes: promising technologies for developing countries. Washington, DC: National Academy of Sciences.

Prior, C.; Greathead, D. J. 1989. Biological control of locusts: the potential for the exploitation of pathogens. Plant Protec. Bull. 37. Rome: United Nations Food and Agriculture Organization: 37–48.

Roberts, D. W.; Humber, R. A. 1984. Entomopathogenic fungi. In: Roberts, D. W.; Aist, J. R., eds. Infection process of fungi, a conference report of the Rockefeller Foundation. New York: Rockefeller Foundation. 201 p.

Roberts, D. W.; Fuxa, J. R.; Gaugler, R.; Goettel, M.; Jaques, R.; Maddox, J. 1991. Use of pathogens in insect control. In: Pimentel, D., ed. CRC handbook of pest management in agriculture. Boca Raton, FL: CRC Press: 243–278.

Steinhaus, E. A. 1949. Principles of insect pathology. New York: McGraw–Hill. 757 p.

Steinhaus, E. A. 1956. Microbial control—the emergence of an idea, a brief history of insect pathology through the nineteenth century. Hilgardia 26: 107–160.

Stevenson, J. P. 1959. Epizootology of a disease of the desert locust, *Schistocerca gregaria* (Forskäl), caused by nonchromogenic strains of *Serratia marcescens* Bizio. Journal of Insect Pathology 1: 232–244.

Streett, D. A. 1987. Future prospects for microbial control of grasshoppers. In: Capinera, J. L., ed. Integrated pest management on rangeland: a shortgrass prairie perspective. Boulder, CO: Westview Press: 205–218.

Streett, D. A.; McGuire, M. R. 1990. Pathogenic diseases of grasshoppers. In: Chapman, R. F.; Joern, A., eds. Biology of grasshoppers. New York: John Wiley & Sons: 484–516.

Streett, D. A.; Oma, E. A.; Henry, J. E. 1986. Characterization of the DNA from Orthopteran entomopoxviruses. In: Samson, R.; Vlak, J.; Peters, D., eds. Fundamental and applied aspects of invertebrate pathology. Wageningen, The Netherlands: Grafisch Bedrijf Ponson and Looijen: 408.

References Cited—Unpublished

Availability note: Copies of the annual reports from the Grasshopper Integrated Pest Management Project are available from USDA, APHIS, PPQ, 4700 River Road, Riverdale, MD 20737.

Carruthers, R. I.; Humber, R. A. 1989. *Entomophaga grylli* as a biological control agent for grasshoppers. In: Cooperative Grasshopper Integrated Pest Management Project, 1988 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 330–335.

Carruthers, R. I.; Humber, R. A.; Ramos, M. E. 1989–91. Development of *Entomophaga grylli* as a biological control agent for grasshoppers. In: Cooperative Grasshopper Integrated Pest Management Project, 1988–90 annual reports. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service.

Foster, R. N.; Reuter, K. C.; Bradley, C. A.; Wood, P. P. 1991. Preliminary investigations on the effect of *Beauveria bassiana* on several species of rangeland grasshoppers. In: Cooperative Grasshopper Integrated Pest Management Project, 1991 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 203–208.

Foster, R. N.; Reuter, K. C.; Bradley, C.; Britton, J.; Black, L.; Drake, S.; Leisner, L.; Tompkins, M.; Fuller, B.; Hildreth, M.; Kahler, E.; Radsick, B. 1992. Development of the fungus *Beauveria bassiana* as a bio-insecticide for grasshoppers on rangeland. In: Cooperative Grasshopper Integrated Pest Management Project, 1992 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 207–213.

Foster, R. N.; Reuter, K. C.; Tompkins, M. T.; Bradley, C.; Britton, J.; Underwood, N.; Black, L. R.; Vigil, E.; Daye, G.; Radsick, B.; Fuller, B.; Brinkman, M.; Kahler, E. 1993. Development of the fungus *Beauveria bassiana* as a bio-insecticide for grasshoppers on rangeland. In: Cooperative Grasshopper Integrated Pest Management Project, 1993 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 233–238.

Henry, J. E.; Onsager, J. A. 1989. Efficacy of *Nosema locustae* and *Vairimorpha* sp. against young nymphs of the Mormon cricket, *Anabrus simplex.* In: Cooperative Grasshopper Integrated Pest Management Project, 1989 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 241–48.

Johnson, D. L.; Pavlik, E.; Bradley, C. 1988. Activity of *Beauveria bassiana* sprayed onto wheat against grasshoppers. Pesticide Res. Rep. Ottawa, ON: Agriculture Canada, Expert Committee on Pesticide Use in Agriculture.

Streett, D. A.; Woods. S. A. 1992. Grasshopper pathogen evaluation. In: Cooperative Grasshopper Integrated Pest Management Project, 1992 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 179–185.

I.7 Insect Predators and Parasites of Grasshopper Eggs

Richard J. Dysart

Introduction

The following remarks are intended to provide a brief overview of the life cycle and habits of the various insects that attack grasshopper eggs. Individually, these natural enemies may not seem significant, but collectively they determine how many grasshopper eggs will hatch and reach the nymphal stage. Also, it is important that land managers recognize the damage done to these beneficial insects when pesticides are sprayed during grasshopper control campaigns. Currently, when rangeland grasshoppers are treated with pesticides, the chemical of choice is usually malathion because it is effective and inexpensive and relatively nontoxic to mammals and birds. However, malathion is not selective, killing virtually all of the exposed insects, including the beneficials.

Grasshopper Egg Laying

Grasshopper eggs are normally deposited in clusters, called egg-pods, placed just below the surface of the soil. The egg-pod is covered by a fairly durable coating of soil particles mixed with a glutinous substance excreted by the female as she lays her eggs in the soil. The female thrusts her abdomen into the soil to a depth of an inch or two (5–10 cm) and starts laying her eggs. When the cavity formed by her abdomen is filled with eggs, she commonly blocks the hole above the eggs with a glandular secretion forming a “froth plug.”

The egg-pod may contain from 2 to more than 100 eggs, depending on the species of grasshopper. The eggs are quite tough and very resistant to cold. They are able to survive the most severe winters if the ground is not disturbed. Also, there is usually enough moisture in the surrounding soil to keep the eggs from drying out even in drought conditions. After the eggs have been deposited in a suitable spot, the female grasshopper provides no maternal or defensive care and merely abandons them.

Natural Enemies of the Egg Stage

The eggs of some species hatch in a few weeks and thus escape destruction by many natural enemies. Most of the grasshoppers in the Western United States lay their eggs in summer and fall and they remain in the ground during the winter in a state of suspended development called diapause, and they do not hatch until the following spring. These eggs are unprotected and exposed to their enemies for some 9 months of the year.

In spite of the fact that grasshopper eggs are available to natural enemies for such long periods, there are surprisingly few insect enemies of the egg stage. It must be noted that locating grasshopper egg-pods in the soil is usually a lengthy and difficult task. Because finding pods in soil and vegetation is so unpredictable, it is easy to miss egg-pods, and especially the larval stages of predators. Thus it is difficult to obtain accurate density counts per unit of area.

The insects that feed on grasshopper eggs can be divided into two groups, predators and parasites, based upon the insects’ method of feeding.

Egg Predators

Predators attack the egg-pod as a whole, feeding externally on the grasshopper eggs. Predators are capable of moving from one egg or egg-pod to another as they complete their development. Most insect predators of grasshopper eggs are generalists. They pose a threat to grasshopper egg populations, but in an undirected way. Some of these predators are no more than scavengers. They locate egg-pods somewhat at random, taking advantage of targets of opportunity. The following groups of grasshopper egg predators are discussed in their approximate order of importance in the Northern Plains.

Coleoptera: Meloidae.—In North America, the larvae of blister beetles (meloids) are an important group of predators of grasshopper eggs. However, in Australia, Africa, and other parts of the world, blister beetles are of little or no importance. The adult stages are called blister beetles because their body fluids can cause blistering of the human skin. Although the larvae of this group of beetles are predaceous, the adults feed exclusively on vegetation, and certain species can become numerous enough on crops such as alfalfa to require treatment with pesticides. In this family, the beneficial aspect of the larva frequently is offset by the destructive habit of the adult.

Rees (1973) lists 26 species of meloids whose larvae are known to attack grasshopper eggs in North America. In early summer, the female blister beetle lays a group of 100–200 eggs in an earthen chamber. When the young larva hatches from the egg, it is quite mobile and begins to search through the soil for a grasshopper egg-pod. Once a pod is located, the meloid larva transforms into a fat white grub and usually eats all of the eggs within the egg-pod. In fact, if the larva still has not completed its development, it will seek out another egg-pod on which to feed. Some species require 2 years to complete their life cycle.

Diptera: Bombyliidae.—The larvae of certain bombyliid flies are also important predators of grasshopper eggs. As many as 13 genera have the habit of consuming acridid (grasshopper) eggs. The adults are called bee flies because certain species have furry bodies resembling a bumble bee. Also they hover in midair and dart swiftly from place to place, moving like bees. When the flies are at rest, the wings are held away from the body. Eggs are deposited in soil cracks and crevices in the vicinity of ovipositing grasshoppers.

After a brief incubation period, the eggs hatch and the larvae wander through the soil in a random search for food. Encounters with grasshopper egg-pods appear to be more or less accidental. A bee-fly larva can completely consume the contents of a pod, but at times only a few eggs are eaten in each of several pods. In this way, many egg-pods can be damaged, allowing the entry of other scavengers. The bee-fly larval stage can last for several years. The number of egg-pods destroyed per individual often exceeds three (Rees 1973). When the bee-fly larva is fully developed, it leaves the egg-pod and pupates near the surface of the soil.

Coleoptera: Carabidae.—Both the adult and larval stages of this family are predaceous on other insects, but members of the family are known as generalists in their choice of hosts. The adults are commonly called ground beetles. The larvae of carabid beetles are predaceous on grasshopper eggs, and in some local situations, they seem to be of importance (Greathead 1963).

Miscellaneous Groups.—On occasion, the larvae of certain members of the following families of beetles and flies have been noted as soil-inhabiting predators of acridid egg-pods, but none seem to be dependent on grasshopper eggs for their survival. These include three Coleoptera families (Cleridae, Tenebrionidae, and Trogidae) and three Diptera families (Asilidae, Calliphoridae, and Chloropidae) (Greathead 1992). Note: during the Grasshopper Integrated Pest Management Project study, larvae of two new chloropid flies were found to be predators of grasshopper eggs in the Northern Plains (Dysart 1991, Sabrosky 1991).

Egg Parasites

Parasites feed internally and complete their development within a single egg. In general, parasites of the eggs of insects usually are tiny hymenopterous wasps that come from one of several different families. However, the eggs of grasshoppers are attacked by wasps of the family Scelionidae only.

Hymenoptera: Scelionidae.—Members of this group are the only true parasites of grasshopper eggs. The North American species of Scelionidae that develop as parasites in the eggs of grasshoppers belong to two genera: the genus *Scelio*, which contains about 19 species, and the genus *Synoditella*, represented by 2 species (Muesebeck 1972). *Scelio* species occur throughout the world wherever grasshoppers are found. Only a single wasp develops within a grasshopper egg. *Scelio* adults live only a very short time, usually no more than 3 weeks under the best conditions. The sex ratio varies among species, but there are usually more females than males by a considerable margin.

The factors involved in host selection are not entirely clear, but it seems certain that the adult female is attracted by some chemical in the egg-pod froth. After locating a suitable egg-pod, the female wasp chews a passageway through the froth until she encounters the grasshopper eggs. Then the wasp backs out, reenters the passageway tail first, and, using her long ovipositor, lays eggs in as many host eggs as she can reach. After the *Scelio* larva hatches, it feeds internally on the contents of the host egg.

When mature, the larva pupates within the host egg shell, and the adult wasp emerges during the summer months. In the Northern Plains, *Scelio* species are thought to have only one generation per year. The most abundant and most widespread of the North American species is *Scelio opacus*. Host records from the literature and my own studies (Dysart 1995) show that it has been reared from eggs of nine different grasshopper species.

Discussion

Many articles in the literature describe the habits and life history of grasshopper parasites and predators, but few good ecological studies describe the impact of these natural enemies on grasshopper populations.

In his general review of predators and parasites of North American grasshoppers, Rees (1973) speculated that grasshopper egg predators probably have more effect on grasshopper populations than do predators of nymphs and adults. Based on a 10-year study in North Dakota and Montana, Parker (1952) estimated that predators destroyed 20 percent of the eggs laid by grasshoppers. Parker and Wakeland (1957) cite results from a studies made at 16 sites in 7 States. Average annual destruction of egg-pods by predators was about 18 percent (9 percent by blister beetles, 6 percent by bee flies, and 3 percent by ground beetles).

Prior and Greathead (1989) estimated that, in Africa, scelionid egg parasites *(Scelio* spp.) were the predominant cause of egg mortality in solitary locust populations. However, scelionids were rather ineffective mortality factors in the egg beds of gregarious species, such as the desert locust. In Australia, parasitism by *Scelio* species at certain sites has been found in up to 90 percent of the egg-pods. In my study areas in Montana and North Dakota, *Scelio* parasitism never reached such high levels. I found that a complex of four species of *Scelio* parasitized about 11 percent of the egg-pods (Dysart 1995). Parasitism figures from the literature indicate that a range of 5 to 15 percent of pods are attacked by *Scelio* spp. in the Northern United States and the Prairie Provinces of Canada.

References Cited

Dysart, R. J. 1991. Biological notes on two chloropid flies (Diptera: Chloropidae), predaceous on grasshopper eggs (Orthoptera: Acrididae). Journal of the Kansas Entomological Society 64: 225–230.

Dysart, R. J. 1995. New host records of North American *Scelio* (Hymenoptera: Scelionidae), parasites on grasshopper eggs (Orthoptera: Acrididae). Journal of the Kansas Entomological Society 68: 74–79.

Greathead, D. J. 1963. A review of the insect enemies of Acridoidea (Orthoptera). Transactions of the Royal Entomological Society of London 114: 437–517.

Greathead, D. J. 1992. Natural enemies of tropical locusts and grasshoppers: their impact and potential as biological control agents. In: Lomer, C. J.; Prior, C., eds. Biological control of locusts and grasshoppers. Wallingford, UK: C.A.B. International: 105–121.

Muesebeck, C.F.W. 1972. Nearctic species of Scelionidae (Hymenoptera: Proctotrupoidea) that parasitize the eggs of grasshoppers. Smithsonian Contribution to Zoology 122: 1–33.

Parker, J. R. 1952. Grasshoppers. In: Insects, the yearbook of agriculture 1952. Washington, DC: U.S. Department of Agriculture: 595–603.

Parker, J. R.; Wakeland, C. 1957. Grasshopper egg pods destroyed by larvae of bee flies, blister beetles, and ground beetles. Tech. Bull. 1165. Washington, DC: U.S. Department of Agriculture, Agricultural Research Service: 1–29.

Prior, C.; Greathead, D. J. 1989. Biological control of locusts: the potential for the exploitation of pathogens. United Nations Food and Agriculture Organization Plant Protection Bulletin 37(1): 37–48.

Rees, N. E. 1973. Arthropod and nematode parasites, parasitoids, and predators of Acrididae in America north of Mexico. Tech. Bull. 1460. Washington, DC: U.S. Department of Agriculture, Agricultural Research Service. 288 p.

Sabrosky, C. W. 1991. A new genus and species of Chloropidae (Diptera) predaceous on grasshopper eggs. Journal of the Kansas Entomological Society 64: 221–224.

I.8 Natural Enemies Attacking Grasshopper Nymphs and Adults

D. L. Hostetter

Introduction

There are 548 recognized species of North American grasshoppers, with about 400 of these occurring on the 650 million acres of rangeland in the 17 Western States (Pfadt 1988). Around two dozen of these are considered potential agricultural pests. Several species may be considered beneficial because of their preference for weeds, and the remainder are either harmless, cause only minor damage, or are beneficial as food sources for wildlife. Along with mammals, grasshoppers are the most significant grazers in the world's temperate grasslands, where people produce most of their food.

This large and diverse group of extremely successful insects occupies many habitats worldwide. Grasshoppers are a food source for equally large and diverse groups of parasites and predators—insects, spiders, and other animals collectively referred to as "natural enemies." These complex animal groups maintain a continual pressure, although variable in degree, on grasshoppers throughout their range. Natural enemies significantly affect grasshopper populations and present the first line of defense before outbreak events. Natural enemies should be factored into regulatory strategies with efforts to conserve them when resorting to chemical control operations.

Decisionmakers need to consider the impact on nontarget and beneficial insects of chemical insecticides and application rates used to control rangeland grasshoppers. The effect of economically and politically expedient chemical control programs should be monitored constantly through the "window of natural enemies" in the process of regulation.

This chapter presents a brief review of life histories, occurrence, and distribution and briefly details facts relating to some of the more important arthropod natural enemies of grasshoppers in western North America.

Grasshopper Parasites and Predators

Order Diptera (flies).—

Family Anthomyiidae.—Members of the family Anthomyiidae are medium in size, about a quarter-inch (6 mm) long and closely resemble the common housefly. Adults are often characterized by slender, dark-colored bodies and rarely possess any metallic coloration or noticeable "bristles" on their body (Cole 1969). Some species are of economic importance, but very few are true parasites. This family occurs worldwide with more than 550 species known in North America, and many are quite common (Borror and DeLong 1971). One species is reported to parasitize grasshoppers, and two species have been reported as egg predators of grasshoppers, although the validity of these reports has been challenged (Rees 1973).

Acridomyia canadensis Snyder is the only species in North America known to parasitize grasshoppers. It has been classified as an "important" parasite in Canada and mentioned as "occurring" in Montana and Idaho (Rees 1973). This fly is known to parasitize at least 16 species of grasshoppers within all 3 grasshopper subfamilies of the family Acrididae, the slantfaced, the spurthroated, and the bandwinged grasshoppers. *Melanoplus bivittatus* and *M. packardii* are reported as this parasite's preferred host species.

Details of the life history of this species are summarized by Rees (1973). Pupae overwinter in the soil, and adults emerge during June, July, and early August. *A. canadensis* typically has one generation per year; however, some adults do not emerge until the second year. Mating occurs upon emergence. After a short but unspecified gestation period, the female flies begin stalking hosts. Upon selection of a suitable host, the female uses its rasping mouthparts to penetrate the host's body and then feeds upon the body fluids. After feeding, the female inserts her ovipositor into the feeding wound and lays eggs in the body cavity. The ovipositor is barbed, which prevents the host from escaping once penetration has occurred. Up to 70 eggs are deposited in each host, and they hatch within 48 hours. Larvae (20–70 per host) develop simultaneously and complete three instars in about 16 to 20 days. Mature larvae then emerge from the host, enter the soil, and pupate. Death of the host usually precedes emergence of the larvae.

Two species of this family have been reported as predators of grasshopper eggs: *Hylemya angustifrons* (Meigen) and *H. platura* (Meigen) = *Delia platura (Meigen*). *H. angustifrons* was reported as a predator only of *M. spretus* eggs. *D. platura,* the seedcorn maggot, may have been incorrectly associated with grasshopper eggs and confused with *Acridomyia canadensis,* which it closely resembles (Rees 1973). These assertions of predation recorded in the early literature should be viewed with caution in light of current systematic knowledge of this group.

Family Calliphoridae.—This is a common group occurring worldwide and well known as blow flies. Adults, about the size of the common housefly, are recognized by their abdomen. Adults of different species have abdomens of different colors–usually a variation of metallic blue or green. Most members of this group are scavengers that live in carrion and excrement. They are similar to flesh flies, family Sarcophagidae, which are important parasites of grasshoppers in North America.

Calliphorids and sarcophagids can be separated visually by structural differences of the antenna and thorax. It is uncertain whether differentiating *Calliphora vicina* Robineau–Desvoidy, the only species ever associated with grasshoppers in North America and collected by Riley (1877), from the now extinct Rocky Mountain locust, *Melanoplus spretus* (Walsh), was an error. Cole (1969) reported that Packard and Thomas, two other entomologists of that era, also recorded *C. vicina* as parasitic on *M. spretus* and indicated that reproduction was by paedogenesis (reproduction in the larval stage), suggesting to Cole that erroneous observations had been made.

Family Asilidae.—Members of the Asilidae are known as robber flies. These raptors of the insect world are strong fliers noted for their voracious appetites and predatory behavior toward flying insects (Rees and Onsager 1985). There are 856 species of Asilidae in North America; 26 are reported as predators of grasshoppers (fig. I.8–1). Six species exhibit a definite preference for grasshoppers (Rees 1973).

Asilids display a variety of identifying structural characters. Those that prey on grasshoppers are large with elongated, tapering bodies and long legs. Bright colors are rare in this group. Most species exhibit gray to silvery coloration, and nearly all are bearded and bristly (Cole 1969). Life cycles range from 1 to 3 years. The adults are very territorial and cannibalistic.

Figure I.8–1—Unidentified robber fly (Family Asilidae) feeding on a fifth-instar *Camnulla pellucida,* Twin Buttes, Owyhee County, ID. (This photograph and all others in this chapter were taken by D. L. Hostetter of the U.S. Department of Agriculture's Agricultural Research Service.)

A 6-year study in Wyoming by Lavigne and Pfadt (1966) documented that three species, *Stenopogon coyote* Bromely, *S. neglectus* Bromley, and *S. picticornis* Loew, feed primarily on rangeland grasshoppers. These authors concluded that these species, along with 9 others associated with grasshoppers in Wyoming, can reduce grasshopper populations by 11 to 15 percent.

Family Sarcophagidae.—Most sarcophagids or flesh flies are scavengers as larvae, but some are parasites of insects. This family is distributed almost worldwide with more than 2,000 described species, most of which occur in tropical to warm temperate areas (Shewell 1987). There are about 21 to 23 species that are parasites of grasshoppers in North America. Sarcophagidae are without exception ovoviviparous, meaning that their eggs hatch within the uterus and the female deposits a live larva on the host (Shewell 1987).

The five most prominent North American species are *Acridophaga aculeata* (Aldrich), *Kellymyia kellyi* (Aldrich) = *Blaesoxipha kellyi* (Aldrich), *Opsophyto opifera* (Coquillett) = *Blaesoxipha opifera* (Coquillett), *Protodexia hunteri* (Hough) = *Blaesoxipha hunteri*, and *Protodexia reversa* (Aldrich) = *Blaesoxipha reversa* (Aldrich).

"Larviposition" by *A. aculeata* and *K. kellyi* occurs during flight of the fly with the selected grasshopper. This airborne interception often knocks the targeted grasshopper to the ground. The flies attack during natural flight or when the host has been flushed by livestock or otherwise disturbed. *Opsophyto opifera, P. hunteri,* and *P. reversa* all stalk grasshoppers on the ground. When within striking range, the female flips a larva from the tip of her abdomen onto the grasshopper. The larva quickly penetrates the host's body through an intersegmental space and begins feeding on the body fluids and tissue (fig. I.8–2). One species, *Servaisia falciformis* (Aldrich) = *Protodexia* = *Sarcophaga falciformis* (Aldrich), possesses a sharp ovipositor that is used to insert a larva into the large muscle of the hind leg of the grasshopper. The larva begins to feed in the leg and eventually migrates into the body cavity, where it continues feeding until mature (Middlekauff 1959).

Sarcophagid larvae complete three instars (growth stages) in 6 to 9 days within the host before reaching maturity. The mature larva exits through a hole in the grasshopper body wall and pupates in the soil. One to three generations are possible, depending on the species, number of suitable hosts available, and environmental conditions. These flies target last-stage nymphs and adults and are generally considered the most effective group of grasshopper parasites (Rees 1973).

Figure I.8–2—Mature third-instar *Sarcophagidae* sp. larva prior to emergence from an adult *Melanoplus sanguinipes* (magnification 1.6×).

Reports documenting the incidence of parasitization in grasshoppers in the Northern United States and Western Canada vary from less than 1 percent to 50 percent (Middlekauff 1959, Lavigne and Pfadt 1966, Rees 1973). A detailed compilation of the North American species of Sarcophagidae associated with grasshoppers, preferred hosts, geographic distribution, and life histories and habits is presented in Rees (1973).

Family Tachinidae.—Tachinid flies also occur worldwide and represent the second largest family in the order Diptera with nearly 1,300 North American species (Borror and DeLong 1971). The larvae are primarily parasites of caterpillars in the order Lepidoptera. Most tachinids deposit their eggs directly on the surface of the host. Upon hatching, the larva burrows into the host and feeds internally on body fluids and tissue. The larva completes three instars feeding within the caterpillar. The host dies prior to emergence of the larva, which then pupates and overwinters in the soil. Six species have been reported from grasshoppers, but only the following three are considered important parasites in the United States and Canada (Smith 1958, Rees 1973).

Acemyia tibialis is the principal tachinid parasite of grasshoppers and has been reported from *Melanoplus bivattatus* and *M. sanguinipes*. Canadian reports indicate parasitism ranges between 16 and 65 percent (Rees 1973). *Ceracia dentata* (Coquillett) and *Hemithrixion oestriforme* Brauer and Bergenstamm have been reported from grasshoppers collected in the United States and Canada with parasitism rates ranging between 1 and 5 percent (Rees 1973).

Family Nemestrinidae.—Members of this cosmopolitan family are commonly known as tangle-veined flies. They are medium-sized, stout-bodied, fast fliers that can hover persistently. There are only six North American species. Two, *Neorhynchocephalus sackenii* (Will.) and *Trichopsidea* (= *Parasymmictus*) *clausa* (Osten Sacken) (Smith 1958) are parasites of grasshoppers. *N. sackenii* is the smaller of the two species and is readily distinguished from *T. clausa* by having an elongated proboscis.

Nemestrinids have only one generation a year. They overwinter in the soil as mature larvae, pupate in the spring, and emerge as adults from late May through mid-July (Smith 1958). Females deposit as many as 4,700 eggs in crevices or holes in dead weeds, fenceposts, and other similar structures at elevations ranging from 3 to 40 feet. Eggs hatch in 8 to 10 days, producing a small (0.5-mm) cream-colored larva. Larvae are thought to be distributed by the wind, a scenario enhanced by the fact that females prefer to lay their eggs on elevated sites (Prescott 1955). Contact with a host is thought to be a random event facilitated by the ability of larvae to survive up to 14 days in the free-living state.

When a suitable grasshopper host (fourth- and fifth-stage nymphs or adults) is contacted, penetration of the body wall occurs within 30 minutes. Once inside the host, the nemestrinid constructs an elongated, spiral respiratory tube attached internally to the body wall of the host (fig. I.8–3). The small end of the tube opens at the surface of the body wall and is the source of air for the larva. The larger end of the tube forms a respiratory sleeve, which fits snugly over the breathing spiracles on the rear of the larva. The larva feeds on the host's fat and reproductive tissue and completes four instars before emerging from the host (fig. I.8–4). The larva emerges just prior to death of the grasshopper and burrows into the soil, where it overwinters as a larva (Prescott 1955).

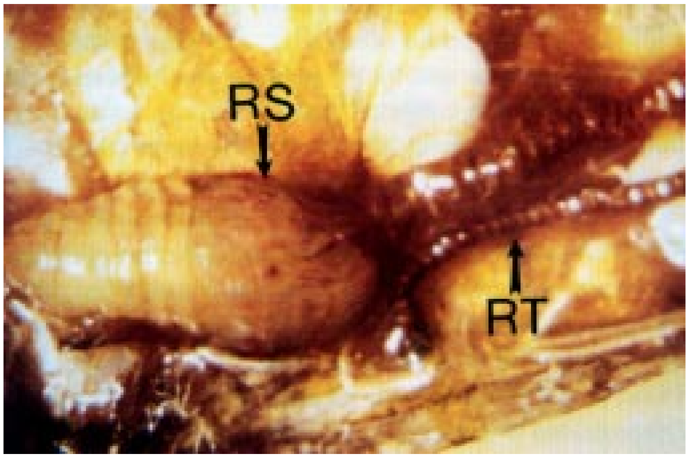

Figure I.8–3—Second-instar *Neorhynchocephalus sackenii* (Family Nemestrinidae) in adult *Oedaleonotus enigma.* Note the respiratory sleeve (RS) and respiratory tube (RT) attached to body wall (magnification 1.6×).

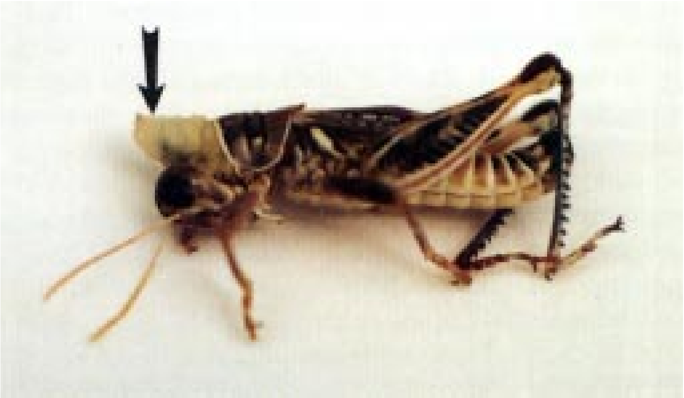

Figure I.8–4—Mature *Neorhynchocephalus sackenii* larva emerging between head and pronotum of mature brachypterous *Oedaleonotus enigma*.

Nemestrinids favor rangeland and "idle acres" habitats and those grasshoppers with similar habitat preferences. The flies are seldom found in cultivated areas or cropland (Prescott 1960). *Ageneotettix deorum* (Scudder), *Camnulla pellucida* (Scudder), *Metator pardalinus* (Saussere), and *Aeropedellus clavatus* (Thomas) are preferred hosts of *N. sackenii* with parasitization rates between 30 and 95 percent (Prescott 1955). *Oedaleonotus enigma* (Scudder) is a preferred host of *N. sackenii* in south-central Idaho (Hostetter et al. 1991 unpubl.).

Order Hymenoptera.—

***Family Formicidae.*—**This family consists of the ants, a large and very successful group found worldwide in almost every habitat. Ants can be formidable predators of hatchling grasshoppers if they are found in an ant colony's territory. Ants are localized, general predators and have little effect on grasshopper populations. Four species have been observed as predators of rangeland grasshoppers: *Formica rufa obscuripes* Forel, *F. obtusopilosa* Emery, *Myrmica sabuletti americana* Weber, and *Solenopsis molesta validiuscula* Emery (Lavigne and Pfadt 1966).

Family Sphecidae.—This is a large family of solitary wasps consisting of eight subfamilies, most of which nest in wood, construct mud cells, or burrow in the soil. Twenty-nine species are recorded as parasitizing grasshoppers in Canada and the United States (Rees 1973). Female "digger wasps" prefer grasshoppers as provisions for their nests and are sometimes mentioned as efficient grasshopper predators (Lavigne and Pfadt 1966). A typical species of this group, *Prionyx parkeri* Bohart and Menke, requires about 1 hour to capture, cache, and lay an egg on an adult grasshopper. Upon hatching, the wasp larva begins to consume the live grasshopper, which remains paralyzed. These wasps are generally rare in most grasshopper habitats, but there is a report in Idaho (Newton 1956) of three *Tachysphex* spp. reducing a population of *Oedaleonotus enigma* (Scudder) by 84 percent.

Order Arachnida.—

Family Araneidae.—The spiders are probably the least studied of the grasshopper predators. Nine species of spiders have been reported as predators of grasshoppers, but the list is known to be incomplete and undoubtedly is much longer (Lavigne and Pfadt 1966, Rees 1973). The wolf spider, *Schizocosa minnesotensis* Gertsch, and a jumping spider, *Pellenes* sp., are two species of nonweb-builders that are often quite abundant on rangeland and are reported (Lavigne and Pfadt 1966) as predatory on various rangeland grasshopper species. The black widow spider, *Latrodectus mactans* (F.), is also a common rangeland predator of grasshoppers in Wyoming and Idaho (Lavigne and Pfadt 1966, and my own personal observations).

The feeding habits and preferences of spiders in the rangeland ecosystem are largely unknown and difficult to measure. Most species are generalists and opportunistic feeders on grasshopper nymphs and adults.

Family Trombidiidae.—This is the most important of three known families of mites that have been reported as parasites of grasshoppers and locusts. Red mites have been universally observed attached to the wings of their host (Uvarov 1928). The most thorough biological studies of the commonly observed North American species *Eutrombidium locustarum (= trigonum)* Walsh were by L. O. Howard (1918) and H. C. Severin (1944).

Adult mites appear early in the spring and begin searching for grasshopper egg-pods. Mites remain in the pods feeding on individual eggs until the mites become sexually mature. Mating takes place in the egg-pod, but eggs are laid in cells (300–700 per cell) in the soil. Larvae emerge after 28 to 30 days and actively seek a suitable host. Larvae usually attach at the base of the wings on adults. Feeding continues until the larva is engorged; it then drops off the host, burrows into the soil, and transforms into a "nymph," an eight-legged immature mite that closely resembles the adult. Nymphs leave the grasshopper toward the end of the summer, when fresh egg pods become available for additional feeding. After more feeding on eggs, the nymph transforms into the adult and overwinters in the soil.

The value of these mites as regulators of the grasshopper is not significant, but they are minor factors in the grasshopper life cycle. Mites belonging to the genus *Gonothrombium* were collected from grasshoppers in Wyoming in 1963 (Lavigne and Pfadt 1966). The investigators reported that 21 of 35 species of grasshoppers (adults) collected during studies in Wyoming between 1959 and 1962 were infested with mites. Conversely, only 8 of 454 grasshopper nymphs collected during the same period were infested with mites. Numbers of mites per individual grasshopper ranged from 1 to 41 with 2 the most common number. No attempt was made to determine detrimental effects, but Lavigne and Pfadt concluded that the mites had little if any effect on the grasshopper hosts.

Order Nematoda.—

Family Mermithidae.—Three species of nematodes belonging to this family are parasitic on grasshoppers: *Agamermis decaudata* Cobb, Stiener, and Christie, *Agamospirura melanopli* Christie, and *Mermis subnigrescens* Cobb (Rees 1973). A *Hexamermis* sp. has also been recovered from the greenstriped grasshopper, *Chortophaga viridifasciata* (DeGeer), in Missouri (Blickenstaff and Sharifullah 1962, Puttler and Thewke 1971).

Nematodes are long-lived animals with from 2 to 4 years required for each generation (Rees 1973). Spring rains and moist soil force the pregnant females from the soil to the vegetation. Eggs are deposited on foliage, where they remain alive for most of the summer. Grasshopper nymphs eat vegetation contaminated with these eggs. The infective nematode larva is released from the egg during the digestive process. The larva eventually penetrates through the host's gut wall into the body cavity, where it remains for 4 to 10 weeks. The mature larva exits the host (usually killing it) late in the summer and overwinters in the soil. The final molt, resulting in the adult, occurs in the spring.

Known North American distribution is limited to the upper Midwest, Northeast, and small, restricted areas in the Western United States. Moisture in the microhabitat, probably in the form of free water, is required for successful development of nematodes. When the required conditions occur, the incidence of infestation in localized areas can exceed 60 percent (Rees 1973).

Family Gordiacea.—Members of this class of roundworms (Nematomorpha) are known as horsehair worms or Gordian worms. They closely resemble nematodes in general body features (Hegner and Engemann 1968). Adults are free living and aquatic. Larvae are parasitic in crustaceans, grasshoppers, crickets, and beetles. Females lay thousands of eggs in long, gelatinous strings in water.

Upon hatching, larvae seek an immature form of aquatic insect as the primary host. Later the larvae become terrestrial and seek a secondary host (usually a cricket, grasshopper, or beetle), where they feed and continue to develop. The mature larva exits the host (causing death) and returns to an aquatic habitat (ponds, animal watering troughs, intermittent pools, streams, or similar area), where it reaches sexual maturity. Roundworms are opportunists that attack many hosts and are considered incidental parasites of grasshoppers and Mormon crickets (Rees 1973).

References Cited

Blickenstaff, C. C.; Sharifullah, N. 1962. Infestation of overwintering nymphs of *Chortophaga viridifasciata* by mermithids. Journal of Economic Entomology 55: 268.

Borror, D. J., Delong, D. M. 1971. An introduction to the study of insects, 3d ed. New York: Holt, Rinehart and Winston. 811 p.

Cole, F. R. 1969. The flies of western North America. Berkeley, CA: University of California Press. 693 p.

Hegner, R. W.; Engemann, J. G. 1968. Invertebrate Zoology, 2d ed. New York: MacMillan Inc. 619 p.

Howard, L. O. 1918. A preliminary report on the Trombidiidae of Minnesota. Rep. 17. [Place of publication unknown]: State Entomologist of Minnesota: 111–144.

Lavigne, R. J.; Pfadt, R. E. 1966. Parasites and predators of Wyoming rangeland grasshoppers. Monogr. 3. Laramie, WY: University of Wyoming and Wyoming Agricultural Experiment Station. 31 p.

Middlekauff, W. W. 1959. Some biological observations on *Sarcophaga falciformis,* a parasite of grasshoppers (Diptera: Sarcophagidae). Annals of the Entomological Society of America 52: 724–728.

Newton, R. C. 1956. Digger wasps, *Tachysphex* spp., as predators of a range grasshopper in Idaho. Journal of Economic Entomology 49: 615–619.

Pfadt, R. E. 1988. Field guide to common western grasshoppers. Bull. 912. Laramie, WY: University of Wyoming and Wyoming Agricultural Experiment Station. 25 p.

Prescott, H. W. 1955. *Neorhynchocephalus sackenii* and *Trichopsidea clausa,* nemestrinid parasites of grasshoppers. Annals of the Entomological Society of America 48: 392–402.

Prescott, H. W. 1960. Suppression of grasshoppers by nemestrinid parasites (Diptera). Annals of the Entomological Society of America 53: 513–521.

Puttler, B.; Thewke, S. E. 1971. Notes on *Hexamermis* spp. (Nematoda: Mermithidae) occurring in the vicinity of Columbia, Missouri. Annals of the Entomological Society of America 64: 1177–1178.

Rees, N. E. 1973. Arthropod and nematode parasites, parasitoids, and predators of Acrididae in America north of Mexico. Tech. Bull. 1460. Washington, DC: U.S. Department of Agriculture. 288 p.

Rees, N. E.; Onsager, J. A. 1985. Parasitism and survival among rangeland grasshoppers in response to suppression of robber fly (Diptera: Asilidae) predators. Environmental Entomology 14: 20–23.

Riley, C. V. 1877. The locust plague in the United States. Chicago: [publisher unknown]. 236 p.

Shewell, G. E. 1987. Sarcophagidae. In: McAlpine, J. F., ed. Manual of nearctic Diptera, vol. 2. Monogr. 28. Lethbridge, CN: Agriculture Canada, Research Branch, Biosystematics Research Center: 1159–1186.

Severin, W. C. 1944. The grasshopper mite *Eutrombidium crigonum* (Hermenn) an important enemy of grasshoppers. Tech. Bull. 3. Brookings, SD: South Dakota Agriculture Experiment Station: 1–36.

Smith, R. W. 1958. Parasites of nymphal and adult grasshoppers (Orthoptera: Acrididae) in western Canada. Canadian Journal of Zoology 36: 217–262.

Uvarov, B. P. 1928. Locusts and grasshoppers. A handbook for their study and control. London: Imperial Bureau of Entomology: 102–143.

References Cited—Unpublished

Hostetter, D. L.; Breeding, S. L.; Broemeling, D. K.; Zugnoni, S. L. 1991. The impact of insect parasites and predators on grasshopper populations in Idaho. In: Cooperative Grasshopper Integrated Pest Management Project, 1989 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 223–236.

I.9 Mites and Nematode Parasites of Grasshoppers

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Very little is known about the nonfungal, nonbacterial, and nonprotozoan pathogens (macroparasites) of grasshoppers. Two major groups of macroparasites for grasshoppers are mites (Acarina) and roundworms (Nematoda). In some instances, the different species of these natural enemies of grasshoppers have not even been identified, let alone studied for their impacts upon grasshopper populations. Therefore, macroparasites are a largely unexploited set of biocontrol agents that might be used to manage grasshopper populations.

Mites

Mites provide an excellent example of the potential opportunity for pest managers to exploit macroparasites in grasshopper control, as well as exemplifying the general lack of understanding about the ecology of parasites that prevents pest managers from using them.

At least two mite species are known to parasitize grasshoppers. The most common is the red mite *(Eutrombidium locustarum)* found on the wings of grasshoppers; another red mite is found on the legs and antennae of grasshoppers and has not yet been formally named. These mites have complex life cycles, going through at least three stages of development (larvae, nymph, and adult), and the complete life cycle requires from 2 months to a year (Rees 1973). Larvae of both mite species attach to the external surface (are ectoparasites) of grasshoppers and suck their blood (hemolymph). In addition, at least the wing mite as a nymph and adult also preys upon grasshopper eggs.

Little is known about the egg predation by mites because this occurs in the soil. However, based upon the mites' consumption needs (Rees 1973), their predatory depression of grasshopper egg survival could be substantial. Each mite nymph requires more than two grasshopper eggs to become an adult. Adult males require three eggs to be able to reproduce and adult females require seven to eight eggs to reproduce. Furthermore, each female mite deposits up to 4,000 eggs (Rees 1973), providing mite populations the potential to increase rapidly and substantially as grasshopper population numbers increase.

When studied in the laboratory, the ectoparasitic effects of larval mites were thought to be of no consequence to grasshopper survival or reproduction (Huggans and Blickenstaff 1966). This conclusion is not unexpected because the grasshoppers had greater quantities of high-quality food than they could consume and were maintained at near optimal temperatures and humidities. Unlike the laboratory studies, our field investigations indicate that larval mites can reduce grasshopper survival and reproduction dramatically.

In western Montana, we have studied the survival and reproduction of *Melanoplus sanguinipes* in cages that were placed over field vegetation and that maintained field temperature and moisture conditions. We have found that the grasshopper densities attained in the cages were comparable to field densities and were food limited (Belovsky and Slade 1994). In another set of experiments conducted in the same fashion, we stocked cages with grasshoppers that either had no wing mites on them, or had one or more wing mites on them.

When we compared the survival of grasshoppers with and without mites in the cages, we found that mites reduced the survival of grasshopper nymphs and adults by an average of 29 percent, and female reproductive output was reduced by an average of 47 percent (fig. I.9–1). Rather than an inconsequential effect, the ectoparasitism by wing mites reduced the grasshopper population's overall egg production by 62 percent.

The effect of ectoparasites in reducing the grasshopper population's egg production becomes stronger when grasshoppers experience greater intraspecific competition for food (higher densities). For example, cages initially stocked with 4 adults exhibited only a 45-percent reduction in total egg production, while cages initially stocked with 10 adults exhibited a much greater reduction, 69 percent. Therefore, the loss of hemolymph to wing mites must be considered in the context of environmental conditions, and the judgment that mite ectoparasitism is unimportant from laboratory studies is of little value. Similar results for the leg mite and the grasshopper *Ageneotettix deorum* were observed with total egg production being reduced by 41 percent (fig. I.9–1).

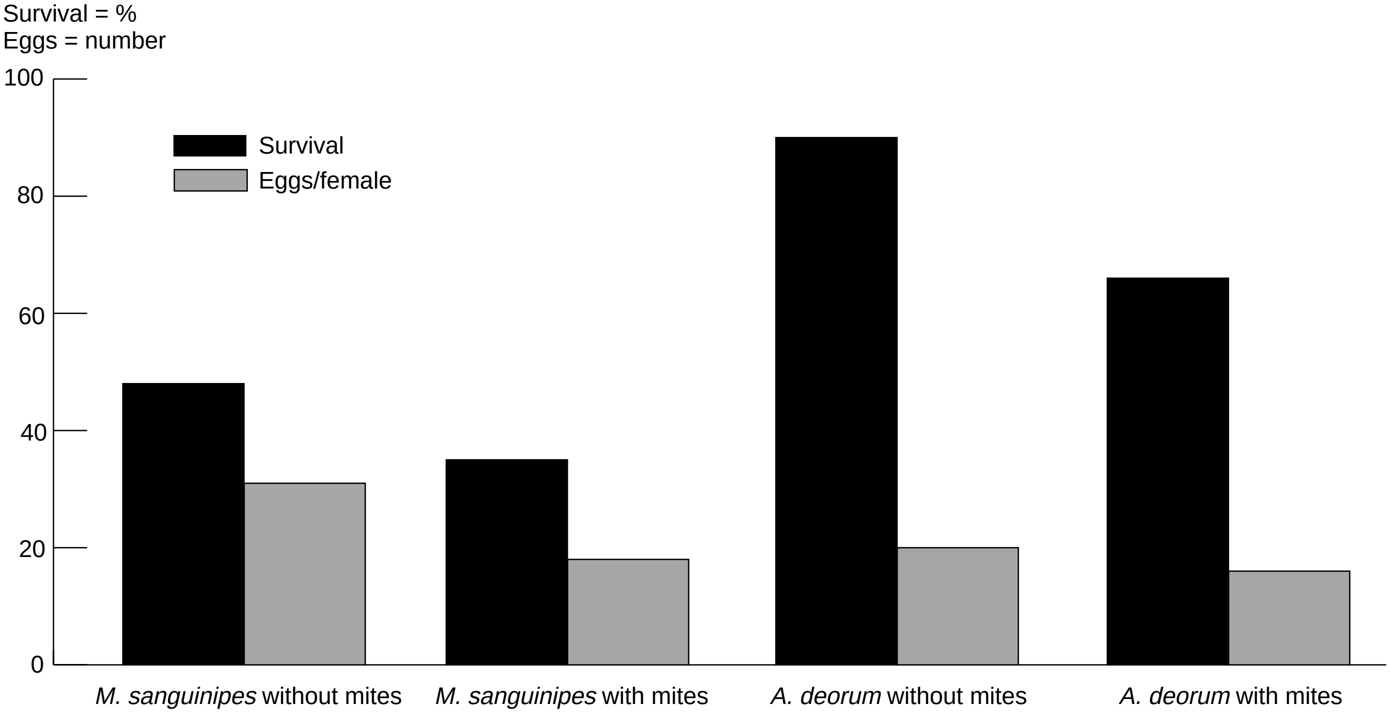


Figure I.9–1—Comparison of the survival and reproduction for two grasshopper species with and without mite infections. Results are statistically significant, and the values represent the means of at least 10 caged populations for each treatment.

The importance of egg predation by nymphal and adult mites and ectoparasitism by larval mites in controlling grasshopper numbers depends upon the abundance of mites. Predation and ectoparasitism effects will be of little importance if there are not large enough numbers of mites relative to grasshopper numbers.

In our field experiments, the grasshoppers that were infected had an average of 3.5 mites. Samples from grasshopper populations in different habitats in western Montana showed that from 0 to 75 percent of the grasshoppers were infected (average = 20.5 percent) at a site, and the individuals that were infected had an average of 2.5 mites. Extending our experimental results on ectoparasitism to field grasshopper populations indicates that larval mites may reduce overall egg production on average by 9 percent, with the effect varying from 0 to 33 percent in different populations.

The predicted natural reductions in total egg production by mites are not adequate in many instances to serve as a viable control method. However, the impact of ectoparasitism by mites could potentially help control grasshopper numbers if the percentage of grasshoppers infected can be increased.

We compared the percentage of grasshoppers infected by mites at different sites in western Montana with environmental characteristics (average daily air temperature, average solar radiation, average soil surface temperature, average soil temperature at less than an inch to almost 2 inches (2–5 cm), average relative humidity, percent cover by vegetation, soil moisture, and the rate of water passing through the soil). We found that infection increased with the rate of water passing through the soil, indicating that mite abundance may be limited by the soil's drainage (the poorer the drainage the fewer the mites). Because the egg, nymphal, and adult stages of the mites live in the soil, we suspect that survival of these stages, rather than survival of the ectoparasitic larval stage, is reduced in soils with poor drainage.

Consequently, to take advantage of the mites' efficiency in controlling grasshopper egg production, a pest manager would need to counteract the local environmental conditions that lead to poor drainage. This type of habitat management may be difficult. Pest managers may be able to raise mites in large numbers and release them into the environment to overcome the poor survival of mite eggs, nymphs, and/or adults in the soil. Raising large numbers of mites in the laboratory is difficult because of the mites' complex life cycle and varied needs for survival and reproduction.

Nematodes

Nematodes are parasites that live within the grasshopper's body (endoparasites), and they are even less well understood than mites. Two species, *Mermis nigrescens* and *Agamermis decaudata*, are important parasites of grasshoppers. These species are even more difficult to identify taxonomically than the mites. These roundworms have a 2- to 3-year life cycle. The larval stages live in the hemolymph of grasshoppers and are considered parasites because they obtain nourishment by absorbing nutrients from the hemolymph. Nematodes are considered parasites rather than parasitoids because parasitoids would consume the grasshopper's body and nematodes do not.

Grasshoppers become infected with *Mermis nigrescens* when they ingest the nematode's eggs, which have been deposited on vegetation. Grasshoppers become infected with *Agamermis decaudata* when the newly hatched larvae penetrate a grasshopper's body (Streett and McGuire 1990). The infection generally lasts for 1 to 3 months and usually results in the death of the grasshopper when the adult nematode(s) exits from the grasshopper's body. The remainder of the nematode's life is largely spent in the soil except when adult females emerge for egg deposition.

In western Montana, we have found, by dissecting large numbers of *M. sanguinipes* in different years and habitats, that nematodes infected less than 10 percent of the grasshoppers at most sites in most years. The highest infestation level we observed at one site in a single year was more than 90 percent. We also found that nematode-infected female grasshoppers still produced eggs, but egg production was reduced by 85 percent.

Nematodes have the potential to be used as a biological control agent if pest managers could enhance nematode numbers by improving survival in the soil or by supplementing their numbers by releases. However, nematode ecology is even more poorly understood than that of mites, and in nature, nematode numbers are usually even lower than mite numbers.

Future Prospects

Employing mites and nematodes actively as biological control agents will require a better understanding of these parasites' natural histories and their ecological impacts on grasshoppers. Also, nobody knows if these parasites can be raised economically in the laboratory. Scientists may be able to take advantage of these natural grasshopper enemies through habitat manipulation that increases their populations or by adding to their natural populations. Mites and nematodes are native enemies of our grasshoppers and may potentially provide an environmentally "friendly" control strategy that can be sustainable for longer periods of time with less attention by pest managers.

References Cited

Belovsky, G. E.; Slade, J. B. 1994. Dynamics of some Montana grasshopper populations: relationships among weather, food abundance and intraspecific competition. Oecologia 101: 383–396.

Huggans, J. L.; Blickenstaff, C. C. 1966. Parasites and predators of grasshoppers in Missouri. Res. Bull. 903. Columbia, MO: University of Missouri and Missouri Agricultural Experiment Station. 40 p.

Rees, N. E. 1973. Arthropod and nematode parasites, parasitoids, and predators of Acrididae in America North of Mexico. Tech. Bull. 1460. Washington, DC: U.S. Department of Agriculture, Agricultural Research Service. 288 p.

Streett, D. A.; McGuire, M. R. 1990. Pathogenic diseases of grasshoppers. In: Chapman, R. F.; Joern, A., eds. Biology of grasshoppers. New York: John Wiley and Sons: 483–516.

I.10 Birds and Wildlife as Grasshopper Predators

L. C. McEwen, B. E. Petersen, and C. M. Althouse

In the early years of this country's agriculture, birds were considered the first line of defense against insect damage. The first laws to protect birds were proposed in 1877 (U.S. Entomological Commission 1878). The act establishing the U.S. Department of Agriculture (USDA) in 1862 made reference to "the introduction and protection of insectivorous birds" (McAtee 1953). A Section of Economic Ornithology and Mammalogy was formed in USDA's Division of Entomology in 1885, and it was expanded into a Division of Food Habits Research in 1921. Much of the wildlife food-habits work was summarized in a book by Martin et al. (1951) in which the authors reported almost universal predation on grasshoppers by insectivorous and omnivorous birds, mammals, and reptiles.

It is interesting that most of the early studies in economic ornithology were not done by ornithologists (people studying birds) but rather by entomologists (those studying insects). For example, S. A. Forbes, an entomologist, founded the field of economic ornithology more than 100 years ago and defined many of the principles of integrated pest management (IPM) as we know them today (Metcalf 1980). The results of examination of more than 40,000 bird stomachs were reviewed by W. L. McAtee (1953). More than 200 species of birds were found to prey on grasshoppers (fig. I.10–1).

Figure I.10–1—More than 200 species of birds are known to feed on grasshoppers. Even seed-eating species, such as this Savannah sparrow, depend on insects for high-protein food during the breeding season. (Photograph by C. K. Miller, of the Nature Conservancy, Clear Lake, SD; used by permission.)

Some of the larger species, such as kestrels (sparrow hawks) (fig. I.10–2), gulls, and meadowlarks, could capture in excess of 100 grasshoppers per day. Swainson's hawks are known to gather in flocks of several hundred to feed on grasshoppers when they become abundant (Wakeland 1958). More recently Johnson et al. (1987) observed large flocks of these hawks capturing about 100 grasshoppers per bird per day in Idaho.

It is not surprising that grasshoppers are so important as food for wildlife because they (1) have high energy value and contain 50–70 percent crude protein (Ueckert et al. 1972, DeFoliart 1975), (2) are widely distributed and available in most western habitats, and (3) are large enough to easily exceed the energy cost of capture by foraging birds and wildlife. Grasshoppers are especially important for successful raising of young by the majority

Figure I.10–2—Research on kestrels has shown their high rate of predation on grasshoppers and their compatibility with grasshopper IPM control materials, such as dimilin, *Beauveria bassiana,* and Sevin 4-Oil ®. (Photograph by B. E. Petersen; used by permission.

of bird species (McEwen 1987) and for many mammals as well. Nestlings and chicks must go through a period of rapid development and growth to survive and perpetuate their species. Even many species that, as adults, eat mostly seeds and plant materials are completely insectivorous in early life (fig. I.10–3). Grasshoppers are highly preferred for feeding young of many kinds of songbirds, upland shore birds, game birds (quail, grouse, pheasants, and turkeys), and even certain hawks and owls (McAtee 1935, 1953).

Grasshoppers are beneficial to a healthy, vigorous, grassland ecosystem when they are at low to moderate (noneconomic) densities. This family of insects preceded today's rangeland plant species and vertebrate animal life by millions of years (Carpenter 1953). Grasshoppers developed in the rangeland ecosystem during a long period of coevolution with other flora and fauna. Grasshoppers' ecologic role (Van Hook 1971) of providing food for wildlife, stimulating plant growth, creating plant litter for the soil, and cycling elements and nutrients was developed as a functional part of the whole ecosystem. Land managers should view grasshoppers as pests only when the insects increase to densities that are clearly damaging to the rangeland plant cover and ecosystem.

Figure I.10–3—Lark bunting with grasshopper captured to feed nestlings. Grasshoppers are a preferred food for young wildlife because of the high protein content and nutritional value. (Photograph by Lowell C. McEwen; used by permission. The shot was taken on the GHIPM Project's grasshopper spray area in Bighorn County, Wyoming.)

Although there is much evidence that birds and wildlife prey on grasshoppers, little research has been done to learn whether wildlife predators actually reduce grasshopper populations or prevent outbreaks. A few recent experiments determined the reduction in grasshopper densities attributed to birds on rangeland. Results show that bird predation commonly reduces grasshopper densities on rangeland by 30–50 percent (Joern 1986, Fowler et al. 1991, Bock et al. 1992). But predation is not so effective in some habitats (Belovsky et al. 1990). Studies of bird predation on other insect pest species also have found that birds significantly reduce pest numbers (McFarlane 1976, Takekawa et al. 1982, Crawford and Jennings 1989, Marquis and Whelan 1994).

Capture of grasshoppers for food by mammals has not received much attention as a suppressing force on grasshopper populations. Small mammals, such as shrews, ground squirrels, deer mice, and grasshopper mice, and larger species, including skunks, foxes, and young coyotes, all eat grasshoppers when available (Martin et al. 1951). Many reptiles and amphibians do the same (fig. I.10–4).

Most investigators agree that predation is more important before, rather than after, insect pests reach the outbreak stage. Bird and mammal predation on grasshoppers is considered a stabilizing force on grasshopper populations. Wildlife predation acts as a preventive factor to grasshopper outbreaks, rather than a means of quick reduction after a buildup to high pest densities. However, instances have been recorded (Wakeland 1958) of flocks of birds saving valuable forage from destruction by grasshopper outbreaks. Perhaps the best known example is the arrival of gulls to save crops in Utah from Mormon crickets (Forbush 1907).

The recognition of the value of birds in combating insect pests has led to efforts not only to protect insectivorous species but also to increase their numbers by providing nest boxes and improving habitat. Nest boxes have been successfully used for hundreds of years on a large scale in Europe to attract birds that control forest insect pests (Takekawa et al. 1982). In the United States, forest management effects on bird populations and relationships to insect outbreaks were reviewed by Thomas (1979) and Crawford and Jennings (1989). A study of insectivorous

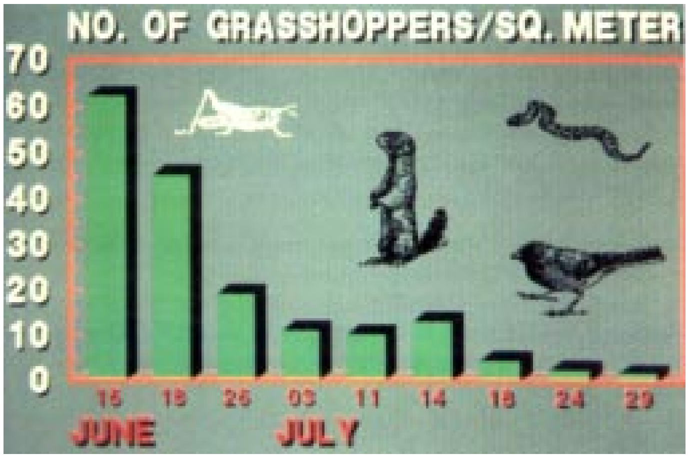


Figure I.10–4—Over the summer, relatively low grasshopper densities on a rangeland area can be diminished further by natural biological factors, including bird, mammal, and reptile predation. But wildlife predation is effective only when grasshopper densities are low (noneconomic) and should be viewed as a preventive force against grasshopper outbreaks, not as a total control measure after outbreaks have occurred.

birds feeding on the insect pests of white oak *(Quercus alba)* concluded that bird predation reduced insect numbers by 50 percent and resulted in one-third greater growth of the oaks (Marquis and Whelan 1994).

Control of locusts that had been a chronic problem on 8,200 acres of grassland in China was achieved by using birds. This was done by creating nesting habitat, planting small shrubs, and digging water seeps to increase the number of insectivorous birds (Anonymous 1988, Yu 1988). Control was successful over many years. Predation on grasshoppers by birds was found in food-habit studies of rangeland birds foraging at edges of Montana wheat fields (McEwen et al. 1986).

Bird densities on the semiarid western rangelands of the United States are generally lower than in other ecosystems that receive higher precipitation. However, numbers of highly insectivorous birds, such as meadowlarks and grasshopper sparrows, can be increased by improving range condition and increasing perennial grass and forb cover. The wildlife associated with healthy stands of native grasses, forbs, and shrubs can contribute greatly to prevention of grasshopper outbreaks (McEwen 1982, McEwen 1987). Figure I.10–1 shows an example of grasshopper suppression by wildlife.

An investigation of bird numbers and range grasshopper densities on the North Dakota Grasshopper Integrated Pest Management Project Demonstration Area indicated a significant negative relationship (George and McEwen 1992). This relationship was a strong indication of possible effects of avian predation on grasshopper densities.

Although bird population densities vary on rangeland, most studies show a normal population range of 1 to 3 birds/acre in the late spring to summer breeding season. Models of predation (McEwen 1987) by birds at these densities show a grasshopper reduction potential of at least 50 percent. In a recent review of the role of birds in controlling insect pests, Kirk et al. (1996) developed a model that indicates even greater potential for regulation of grasshoppers–based on bird numbers, capture rates, and energetics.

Wildlife populations are an important biological control factor in natural suppression of rangeland grasshoppers. Management practices that improve range condition and habitat for insectivorous and omnivorous wildlife can dampen or prevent extreme grasshopper population fluctuations and help reduce damage to vegetation.

References Cited

Anonymous. 1988. Recruiting locust-eating birds on the prairies of Tianshan Mt. area in China. IPM Practitioner 10: 18.

Belovsky, G. E.; Slade, J. B.; Stockhoff, B. A. 1990. Susceptibility to predation for different grasshoppers: an experimental study. Ecology 71: 624–634.

Bock, C. E.; Bock, J. H.; Grant, M. C. 1992. Effects of bird predation on grasshopper densities in an Arizona grassland. Ecology 73: 1706–1717.

Carpenter, F. M. 1953. The geographical history and evolution of insects. American Scientist 41: 256–270.

Crawford, H. S.; Jennings, D. T. 1989. Predation by birds on spruce budworm *Choristoneura fumiferana:* functional, numerical, and total responses. Ecology 70: 152–163.

DeFoliart, G. R. 1975. Insects as a source of protein. Bulletin of the Entomological Society of America 21: 161–163.

Forbush, E. H. 1907. Useful birds and their protection. 2d ed. Boston: Massachusetts Board of Agriculture. 437 p.

Fowler, A. C.; Knight, R. L.; George, T. L.; McEwen, L. C. 1991. Effects of avian predation on grasshopper populations in North Dakota grasslands. Ecology 72: 1775–1781.

George, T. L.; McEwen, L. C. 1992. Relationships between bird density, vegetation characteristics, and grasshopper density in mixed grass prairie of western North Dakota. In: McCullough, D. R.; Barrett, R. H., eds. Wildlife 2001: populations. London and New York: Elsevier Publishers: 465–475.

Joern, A. 1986. Experimental study of avian predation on coexisting grasshopper populations (Orthoptera: Acrididae) in a sandhills grassland. Oikos 46: 243–249.

Johnson, C. G.; Nickerson, L. A.; Bechard, M. J. 1987. Grasshopper consumption and summer flocks of nonbreeding Swainson's hawks. The Condor 89: 676–678.

Kirk, D. A.; Evenden, M. L.; Mineau, P. 1996. Past and current attempts to evaluate the role of birds as predators of insect pests in temperate agriculture. Current Ornithology 13: 175–269.

Marquis, R. J.; Whelan, C. J. 1994. Insectivorous birds increase growth of white oak through consumption of leaf-chewing insects. Ecology 75: 2007–2014.

Martin, A. C.; Zim, H. S.; Nelson, A. L. 1951. American wildlife and plants. New York: McGraw–Hill. 500 p.

McAtee, W. L. 1935. Food habits of common hawks. Circ. 370. Washington, DC: U.S. Department of Agriculture. 36 p.

McAtee, W. L. 1953. Economic ornithology. In: Fifty years' progress in American ornithology. Lancaster, PA: American Ornithologists Union: 111–129.

McEwen, L. C. 1982. Review of grasshopper pesticides vs. rangeland wildlife and habitat. In: Peek, J. M.; Dalke, P. D., eds. Proceedings of the wildlife–livestock relationships symposium; 20–24 April 1981; Coeur d'Alene, ID. Moscow, ID: University of Idaho: 362–382.

McEwen, L. C. 1987. Function of insectivorous birds in a shortgrass IPM system. In: Capinera, J. L., ed. Integrated pest management on rangeland: a shortgrass prairie perspective. Boulder, CO, and London: Westview Press: 324–333.

McEwen, L. C.; DeWeese, L. R.; Schladweiler, P. 1986. Bird predation on cutworms (Lepidoptera: Noctuidae) in wheat fields and chlorpyrifos effects on brain cholinesterase activity. Environmental Entomology 15: 147–151.

McFarlane, R. W. 1976. Birds as agents of biological control. The Biologist 58: 123–140.

Metcalf, R. L. 1980. Changing role of insecticides in crop protection. Annual Review of Entomology 15: 147–151.

Takekawa, J. Y.; Garton, E. O.; Langelier, L. A. 1982. Biological control of forest insect outbreaks: the use of avian predators. Transactions of North American Wildlife and Natural Resources Conference, March 1982, Portland, OR. Washington, DC: Wildlife Management Institute: 47: 393–402.

Thomas, J. W., ed. 1979. Wildlife habitat in managed forests: the Blue Mountains of Oregon and Washington. Agric. Handb. No. 553. Washington, DC: U.S. Department of Agriculture. 511 p.

Ueckert, D. N.; Yang, S. P.; Albin, R. C. 1972. Biological value of rangeland grasshoppers as a protein concentrate. Journal of Economic Entomology 65: 1286–1288.

U.S. Entomological Commission. 1878. First annual report of the United States Entomological Commission for 1877. Washington, DC: U.S. Government Printing Office.

Van Hook, R. I. 1971. Energy and nutrient dynamics of spider and orthopteran populations in a grassland ecosystem. Ecological Monographs 41: 1–26.

Wakeland, C. 1958. The high plains grasshopper. Tech. Bull. No. 1167. Washington, DC: U.S. Department of Agriculture, Agricultural Research Service. 168 p.

Yu, J. H. 1988. Locust-eating birds and their recruitment in prairies of Tianshan Mt. Chinese Journal of Biological Control 4(2): 6870.

I.11 Enhancing Biological Control of Grasshoppers by Construction and Placement of Bird Nest Boxes

B. E. Petersen, L. C. McEwen, and C. M. Althouse

Wildlife can play a significant role in the regulation of grasshopper population dynamics (see chapter I.10). Placement of nest boxes for American kestrels (also known as sparrow hawks), bluebirds, and other insect-eating bird species can provide a strong and stabilizing factor to help control grasshopper populations and prevent outbreaks (fig. I.11–1). Kestrels take large numbers of grasshoppers and in some areas are called "grasshopper hawks." Many other bird species that nest in cavities and nest boxes also feed on grasshoppers during the breeding season and feed them to their young. These insects are a very important source of protein and other nutrients for young birds during growth and development.

Because the birds listed in table I.11–1 are limited by the number of natural cavities available, their abundance can be increased significantly by the construction and placement of nest boxes. Each cavity-nesting bird species prefers nest boxes of a particular size but frequently will nest in larger boxes when smaller ones are not available. Placement of nest boxes on a large scale by land managers might reduce the need for, and cost of, chemical spraying and could be important in a grasshopper integrated pest management system.

Kestrels and bluebirds (eastern, western, and mountain bluebirds) are among the most common species attracted to properly placed nest boxes. Plans and directions for construction are shown in figures I.11–2 and I.11–3. Because most cavity-nesting species are territorial, placement of boxes should not be too close together so that birds avoid using them. Defended territories vary with the species of birds, food availability, and their other needs.

American kestrels have the largest territories compared to other cavity-nesting species. In open country, where the boxes are within direct line of sight, the distance between them should be at least 2,460 ft (750 m). When trees intervene, such as along a meandering river or irregular woodland edges, the boxes can be as close as 656 ft (200 m). Entrance holes should face south to southeast, away from prevailing winds and storms. Preferred height of boxes should be a minimum of 10 ft (3 m) but lower posts (7–9 ft) (2–2.7 m) also may be used. Boxes can be wired at top and bottom to posts, poles, or smaller trees or nailed through the holes to large-diameter trees. Add 1 inch of wood chips or dried grass for nest material as kestrels do not bring in their own nesting material. Boxes should be cleaned out and fresh chips or grass added before each nesting season.

The three species of bluebirds defend smaller areas surrounding their nests than do kestrels; therefore, greater numbers of nest boxes can be provided per unit area. In open country, where bluebird boxes are within direct line

Figure I.11–1—Nest boxes placed on poles or trees bordering open rangeland readily attract birds, such as kestrels, that require cavities for nesting. Cavity-nesting birds are higly insectivorous and contribute to biological control of grasshopper populations. A young kestrel can be seen peeking out of the entrance hole. (Photograph by B. E. Petersen; used by permission.)

of sight, the distance between can be as short as 300 ft (92 m). Entrance holes also should face south to southeast, away from prevailing winds and storms. Boxes can be wired at top and bottom to posts, poles, or smaller trees or nailed through the holes to large-diameter trees at a height of about 5 ft (1.5 m) for ease in checking. No nesting material need be added to boxes because bluebirds bring in their own nesting material; but boxes should be cleaned out each year after the nesting season by removing debris and old material.

Information on construction and optimum placement of the various kinds of nest boxes can be obtained from State wildlife agencies or conservation organizations, such as the Bluebird Recovery Program, Box 566, Minneapolis, MN 55458; the North American Bluebird Society, Box 6295, Silver Spring, MD 20906–0295; or a local chapter of the Audubon Society.

Table I.11–1—Dimensions (in inches) of nest boxes for several avian species

Species	Diameter of entrance	Entrance height above bottom	Depth of cavity	Bottom of cavity
American kestrel	3.0	12 – 14	14 – 18	8 × 8
Downy woodpecker	1.25	6 – 8	8 – 10	4 × 4
Northern flicker	2.5	14 – 16	16 – 18	7 × 7
Red-headed woodpecker	2.0	9 – 12	12 – 15	6 × 6
House wren	1.0	5 – 6	6 – 8	4 × 4
Bluebird	1.5	7 – 8	8 – 10	5 × 5
Tree swallow	1.5	4 – 5	6	5 × 5
Chickadee	1.25	6 – 8	8 – 10	4 × 4

Note: Entrance should face south to southeast. Height of box is variable: larger birds prefer greater heights (about 10 feet or more), and smaller birds use lower boxes (about 5 feet or more above the ground).

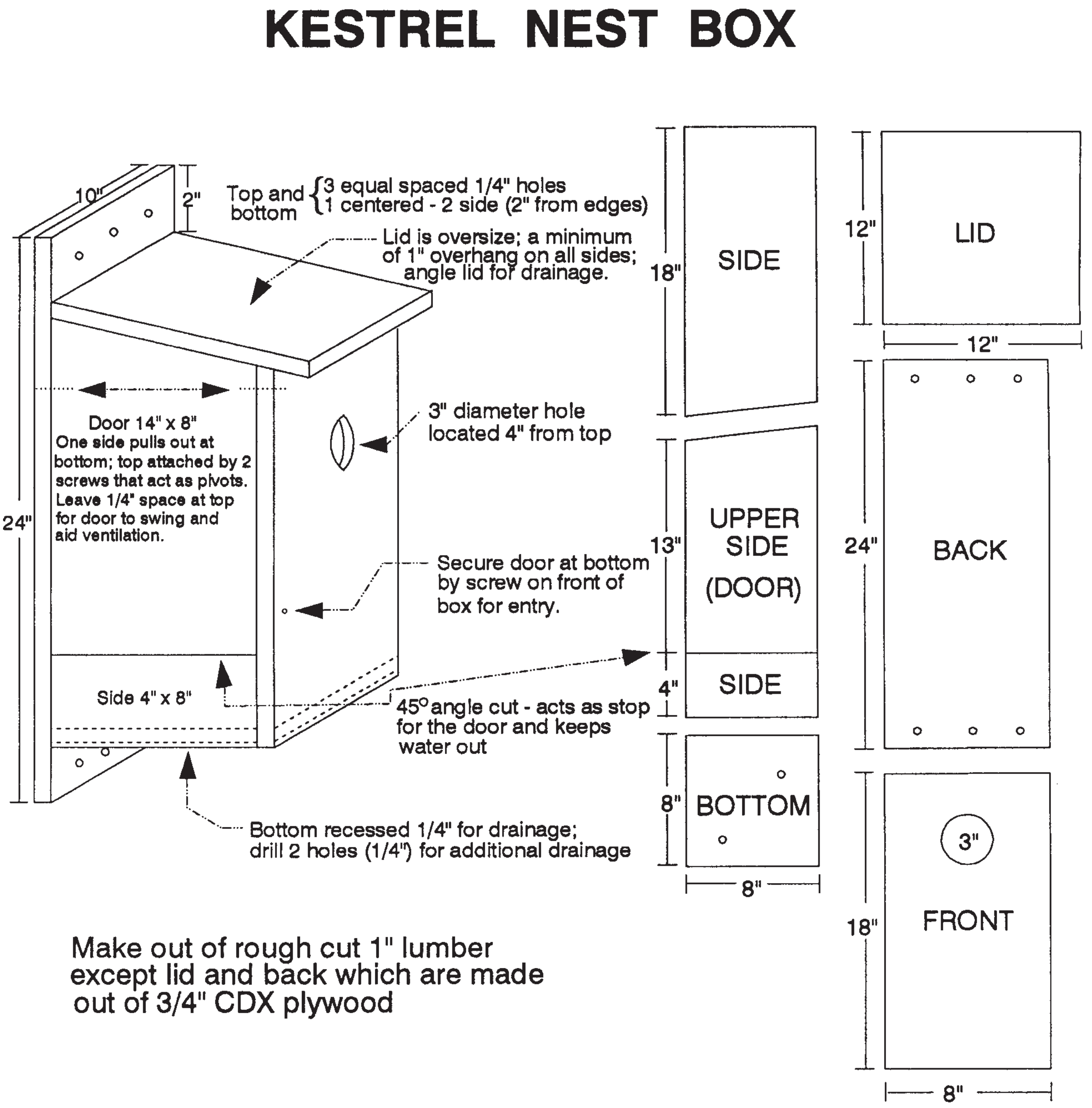
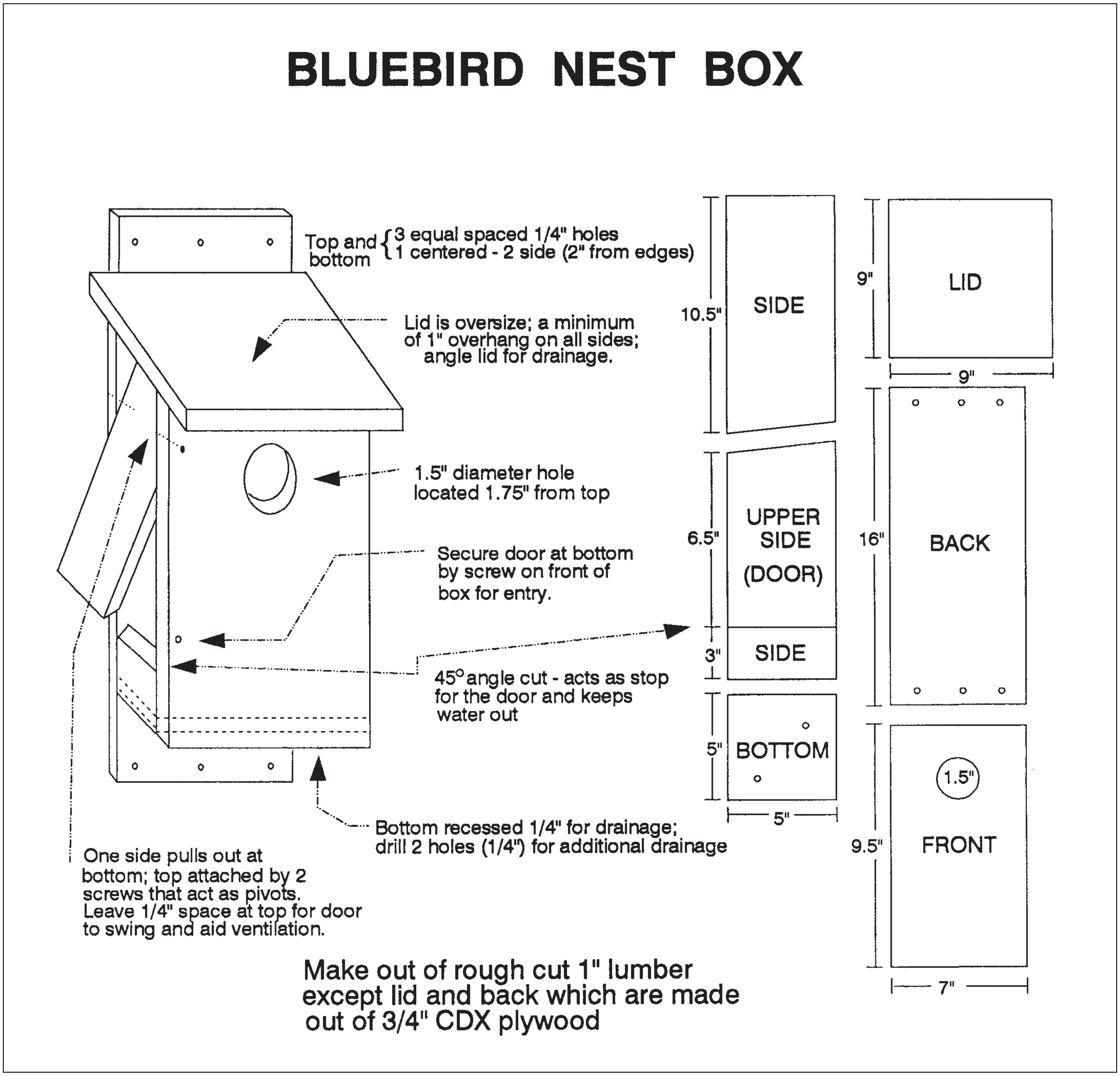


Figure I.11–2—American kestrel nesting box construction plan with dimensions and description of door mechanism. Entrance should face south to southeast, away from prevailing winds and storms. Boxes can be attached to trees, poles, or posts. Optimum height of boxes is a minimum of 10 ft (3 m), but lower attachments can sometimes be successfully used if taller ones are not available.



BLUEBIRD NEST BOX

Top and bottom { 3 equal spaced 1/4" holes 1 centered - 2 side (2" from edges)

Lid is oversize; a minimum of 1" overhang on all sides; angle lid for drainage.

1.5" diameter hole located 1.75" from top

Secure door at bottom by screw on front of box for entry.

45° angle cut - acts as stop for the door and keeps water out

Bottom recessed 1/4" for drainage; drill 2 holes (1/4") for additional drainage

One side pulls out at bottom; top attached by 2 screws that act as pivots. Leave 1/4" space at top for door to swing and aid ventilation.

Make out of rough cut 1" lumber except lid and back which are made out of 3/4" CDX plywood

Figure I.11–3—Bluebird nesting box construction plan with dimensions and description of door mechanism. Entrance should face south to southeast, away from prevailing winds and storms. Boxes can be attached to fenceposts, small trees, or poles at preferred heights of 5 ft (1.5 m) or higher.

I.12 The Biological Control Potential of Parasites, Predators, and Fungal Pathogens

D. L. Hostetter and R. J. Dysart

Introduction

Grasshoppers, like all other animals, are subject to a large number of parasites, predators, and pathogens, including fungi, protozoa, and viruses (Henry et al. 1985, Prior and Greathead 1989, Streett and McGuire 1990). Parasites, predators, and pathogens can be used as "classical" biological control agents. Classical biological control is defined as "the importation and release of an organism outside its natural range for the purpose of controlling a pest species" (Howarth 1991). Another approach, "augmentative" biological control, uses native or exotic organisms that are released periodically to enhance mortality in a targeted pest population. Insect pathogens generally fall in this category because many can be mass-multiplied and applied as biological pesticides (Prior and Greathead 1989).

Insect Parasites and Predators

Classical Introduction Approach.—According to a review article by Prior and Greathead (1989), the classical biological control of a pest grasshopper using an insect parasite or predator as the beneficial agent has been attempted on nine occasions: there were two cases using bombyliids or bee flies, three cases using sarcophagid flies, two cases using meloid beetles, and two cases using scelionid wasps. Only two of these nine attempts resulted in the establishment of the introduced beneficial, a meloid beetle in Corsica and a scelionid wasp in Hawaii. However, the only project that has been claimed as a success was the introduction of a *Scelio* sp. from Malaysia, released against the rice grasshopper in Hawaii.

As suggested by Greathead (1992) and by Siddiqui et al. (1986), the possibilities for classical work certainly have not been exhausted, particularly with any scelionid egg parasites having an acceptable degree of host specificity. A controversy surrounding the request by Richard J. Dysart for permission to release a species of *Scelio* from Australia against pest grasshoppers in the United States seemed to pivot around the issue of host specificity. In spite of the constraints involved in the classical biological control approach, there are even more problems to consider in the augmentative approach.

Augmentative Approach.—Using insect parasites or predators as substitutes for chemical insecticides is not considered feasible for the control of grasshoppers. In his recent review of biological control options for tropical locusts and grasshoppers, Greathead (1992) expressed the same sentiments. In order for this approach to be workable, the natural enemy to be used must have a number of attributes:

- An acceptable level of host specificity, assuring some degree of safety to nontarget organisms,
- The ability to be easily reared in a laboratory situation and be produced in large quantities, and
- Costs of production and delivery to the target areas low enough so that the cost of using the biocontrol organism is competitive with the cost of using chemicals.

Concerns about host specificity would eliminate several groups of natural enemies, for example, the meloid and carabid beetles, whose larvae wander through the soil in search of a wide range of hosts. Similarly, certain beneficial groups can be eliminated from consideration because they are not amenable to handling in captivity, for example, the egg predators (Bombyliidae, Meloidae) and the nemestrinid parasites (Greathead 1992).

Although certain scelionid egg parasites can be reared easily in the laboratory, the rearing process is dependent on a constant supply of grasshopper eggs of a certain age. Considering the immense areas that would require release of parasites, plus the logistics of rearing and delivery, it is certain that the costs of using *Scelio* sp. parasites in an augmentative approach would be unacceptable.

Classical Introduction Approach to the Use of Fungi

One of the first documented reports of attempting to use *Entomophaga* (= *Empusa) grylli* Fresenius (Batko) as a classical biological agent occurred in South Africa in 1896 (Howard 1902). A man named Arnold Cooper, of Richmond, Natal (South Africa), noticed grasshoppers dying apparently from a fungous disease. He took specimens to the Bacteriological Institute at Grahamstown, where a fungus capable of infecting healthy grasshoppers was isolated. Subcultures of the isolate were made, and vials containing them were distributed to planters in areas where grasshoppers were abundant. Planters such as H. H. Wells chronicled the situation in 1899: "I dipped captured adult grasshoppers into fluid containing the fungus then released them into the swarm over a period of two to three days...to my profound astonishment I found grasshoppers hanging in clusters all over my farm...millions of them." Many other equally favorable reports were received by the Bacteriological Institute, and distribution of the culture tubes continued.

Questions concerning the precise "nature" of the fungus were raised in 1899 and 1900. Specimens sent to the Royal Botanic Gardens, Kew, England, were identified as a *Mucor* sp. The same determination had been made simultaneously in Victoria, Australia, from similar specimens received from Natal. Circumstantial evidence suggests that perhaps two different fungi were in fact distributed. *Mucor* sp., which is easily cultivated and was readily identified by the authorities of the day, could have been contaminated with resting spores of *Entomophaga* sp. This scenario would support the reports of "clusters of diseased grasshoppers" by planters such as H. H. Wells and early photographs showing dead grasshoppers hanging from the tops of foliage. That phenomenon provides strong evidence of infection by *Entomophaga* sp. It is also apparent that "mixtures of fungal cultures" originating in South Africa were freely distributed to Australia and North America during the period 1899–1901 (Howard 1902).

Documents indicate that fungus cultures were obtained from South Africa by Dr. L. O. Howard in 1900 for subculture and release against grasshoppers in Colorado. A total of 223 "probable releases" were made in 24 States plus the Philippine Islands and Cuba during the period 1901–02 (Howard 1902). Howard further states that "No effort was made to determine the exact nature of the fungus contained in the culture tubes received from South Africa in the spring of 1900, but subsequent events indicate plainly that the Bacteriological Institute at Grahamstown is sending out more than one kind of fungus."

Professor L. Bruner (1901) also reported on a fungus, *Sporotrichum* sp. (= *Beauveria* sp.) he discovered infecting locusts in Argentina in 1897–98. He noted that "it is quite similar to the fungus which is used in destroying chinch-bugs in some portions of the United States." Bruner also stated "that [although] considerable time has been spent in experimenting with this South American fungus upon our North American grasshoppers, thus far the results have all been negative since not a single insect has died from the disease."

These early attempts to use entomopathogenic fungi as "classical" biological control agents set the precedent for introduction and distribution of exotic pathogens in North America. It is apparent that numerous releases of unknown species from a wide variety of locations were made with little concern for environmental consequences beyond reduction of the pest species of the day.

For more than 100 years, the literature on grasshopper fungi has documented the evolution of a wide range of biological facts and observations. Habitat and climatic requirements are most often alluded to as dampening factors for the expression of fungus disease. The initial association between cool, wet, spring weather and an ensuing fungus epizootic plus other observations led to the current data base.

Many entomologists have reported the importance of microhabitats and macrohabitats for the development and expression of fungus epizootic among grasshopper populations. Reports indicate that fungus-infected grasshoppers are often restricted to roadside ditches; perimeters of cropland; low-lying, moist swales and intermittent waterways in pastures and hayfields; and various other noncultivated habitats (Hostetter et al. 1992 unpubl., Packham et al. 1993, McDaniel 1987).

A review of the accumulated information suggests that perhaps entomopathogenic fungi can be exploited in a "classical" sense through novel manipulations and applications already existing in North American agroecosystems.

The theoretical basis for the use of pathogens in biological control has been thoroughly discussed by many authors; most notably by Anderson (1980, 1982) and Hochberg (1989).

A mathematical model derived by Hochberg (1989) shows that host populations may be regulated to low and relatively constant densities if sufficient numbers of pathogens are translocated from reservoirs to habitats where transmission can occur. The model accounts for host–pathogen interactions based on heterogeneity; pathogen populations are not uniform. Transmissibility and lifespan of the pathogen differ among individuals or life stages in the environment. Pathogens are considered as two distinct subpopulations; one as transmissible and short lived, and one as nontransmissible and long lived (e.g., *Entomophaga macleodii* and *E. grylli* pathotype 3, conidia and resting spores).

Infective entities of the pathogen can cause infection only when they are translocated (abiotically or biotically) from the reservoir to the susceptible host. Hochberg suggests that, to increase the efficacy of indigenous pathogens of insects, the focus should be on the identification and manipulation of pathogen reservoirs between nontransmissible and transmissible subpopulations.

The model suggests that for the introduction of exotic pathogens as classical biological control agents, the conditions for the likelihood of success are (1) long lifespan of pathogen stages residing in reservoirs and (2) the propensity of these stages to be translocated to the habitat of the host for transmission.

Two practical applications of this model would be the use of existing Conservation Reserve Program (CRP) land and Federal and State highway rights-of-way as reservoirs or "refugia" for hosts, pathogens, parasites, and predators (Parker 1971).

The CRP program, which was devised in accordance with Title XII of the Food Security Act of 1985 (P.L. 99–198), provides for farmers to enter voluntarily into multiyear (10-year minimum) contracts with USDA to take specified highly erodible cropland out of annual production and put it into some other permanent vegetation. CRP acreage has been identified, quantified, and mapped for each county in each State by personnel of USDA's Agricultural Stabilization and Conservation Service. Blocks of land most often occur in multiples of 40 acres and will be available as a stabilized system (for a minimum of 10 years).

It may be feasible to isolate grasshopper populations on CRP acreage with timely applications of chemical agents or mechanical barriers followed by inoculation/suppression with biological agents utilized in concert with naturally occurring parasites. Geographical imaging systems (GIS) are in place and could be used to delineate graphically and link strategic release areas based on ecological requirements of natural enemies across vast acreages. Host–pathogen reservoirs could be maintained and manipulated by augmentative releases of pathogens, parasites, and predators.

Manipulation of the habitat could be effected in a variety of ways: (1) clearcutting or stripcutting of foliage, which forces susceptible stages of the target species to concentrate in an area favorable to pathogens and arthropod natural enemies; (2) regulation of irrigation practices to create optimum habitat (cover crops) within the reservoir; (3) timely use of disruptive techniques (cultivation, spring-tooth harrow, mowers) to facilitate movement of pathogens from the soil (reservoir) to the host habitat (transmission–infection arena).

The current soil conservation program under the aegis of P. L. 99–198 will probably be succeeded by another "idle acres" program that may provide an exceptional opportunity for demonstrating the principles of IPM.

Federal and State highway rights-of-way could be manipulated to become "beltway reservoirs" for beneficial organisms across entire States. Millions of dollars are spent each year throughout the rangeland States for highway beautification and maintenance programs (e.g., landscaping, mowing, spraying). Monies diverted into development and conservation of habitat may be a wise investment toward long-term stability in the agrosystem. Perhaps a highly visible program of conservation and manipulation of "reservoirs of natural enemies" along the Nation's roadways would pique public interest and support.

Augmentative Approach.—Presently, entomopathogenic fungi have the greatest probability of exploitation as microbial control agents for managing grasshopper populations. The wide range of orthopteran hosts and environments from which fungi have been isolated has revived interest in this group over the last decade.

Worldwide, at least 10 genera of fungi are known to be entomopathogens of grasshoppers and locusts (Prior and Greathead 1989). Use in the initial phase will be "augmentative": "insecticidal" formulations and applications will used to augment natural enemies in the target area (Foster et al. 1991–94 unpubl.).

The most promising candidates are found among the *Beauveria* spp., *Metarhizium* spp., and *Entomophaga* spp. *Beauveria* spp. and *Metarhizium* spp. have host-specific strains and are purported to be nonhazardous to nontarget organisms (Prior and Greathead 1989). Conidia, or spores (the infective entity), are easily produced on commercially available solid substrates or in fermentation processes and can be formulated and applied similarly to other contact chemical pesticides (Foster et al. 1991–94a and b unpubl.).

Because they are lipophilic, the conidia of *Beauveria* spp. and *Metarhizium* spp. can be formulated with oil carriers and applied via ultralow-volume techniques. Oil droplets have the advantage in that droplets of smaller volume (mean diameter) can be generated at the nozzle (time of release), and the oil prevents evaporation during travel to impact on the target (grasshopper cuticle). Oil formulations have the advantage of spreading over the also lipophilic insect cuticle, thereby carrying conidia to intersegmental membranes and joints. Delivery to those areas increases the probability of penetration and infection of the insect (Prior and Greathead 1989).

Vegetable, soybean, or corn oils produced within or near insecticide-application areas could provide sustainable, nontoxic, environmentally safe formulation bases. The use of vegetable oils could decrease reliance on petroleum-based carriers.

The augmentative application of *Entomophaga grylli*, pathotype 1 (= *E. calopteni* [Bessey] Humber), was attempted in South Dakota (McDaniel 1987). McDaniel noticed the presence of *E. grylli* while conducting grasshopper surveys in 1979–80. Among other observations, he noted that the majority of grasshoppers dying from the fungus were found in areas not subject to cultivation (e.g., field borders, roadside ditches, alfalfa fields) and from the edges of corn and soybean fields.

McDaniel reported that he "triggered two fungus outbreaks in the spring of 1982 in plots in Hughes county near Blunt, SD and at a location 21 miles west on the Bad River road in Stanley county." The triggering was accomplished by collecting 4,468 plant sections, each of which had a fungus-killed grasshopper attached; taking them to an area known to be free of the fungus disease; and taping them to the tops of tall grasses and alfalfa plants.

Fungus-killed grasshoppers were observed 15 days after inoculation and a 53-percent reduction of the population occurred within 45 days. McDaniel also reported that the fungus continued to kill grasshoppers at these plots through 1986 with no additional inoculum of spores.

McDaniel developed a method of extracting resting spores from cadavers for inoculation of field plots. He extracted 2 gal of pure spores from 38 gal of hand-picked, dead, fungus-killed grasshoppers. He was able to effect disease in release plots using infected grasshoppers or by applying with a grass-seed spreader ground-up bodies of *Melanoplus differentialis* (Thomas), *M. bivittatus* (Say), and *M. sangunipes* (F.) that had been treated with fungal spores.

McDaniel (1987) attributed the unsuccessful inoculations done with pure resting spores to the fact that they had been stored for several months at room temperature between collection in late fall and application in early spring.

Entomophaga spp.—particularly the Australian isolate, *Entomophaga grylli* pathotype 3—may be best utilized as "classical biological control agents." Members of this complex cannot be produced easily on axenic substrates or in large enough quantities to be used as insecticidal treatments. Current ideology views this as a limitation of the present state of technology; however, perhaps not all entomopathogenic fungi or other microbial agents are best used as insecticides.

The best utilization of entomopathogens will evolve over time along with increased understanding of the ecology and the systems that regulate it. The many avenues of availability are just beginning to be explored. Exploitation will require long-term commitment, innovative approaches, and the willingness to tailor management practices within the principles of ecology.

Selected References

Anderson, R. M. 1980. Depression of host population abundance by direct life cycle macroparasites. Journal of Theoretical Biology 82: 283.

Anderson, R. M. 1982. Theoretical basis for the use of pathogens as biological control agents of pest species. Parasitology 84: 3.

Bruner, L. 1901. Locusts or grasshoppers. Bull 70, vol. XIII. Lincoln, NE: University of Nebraska and Nebraska Agricultural Experiment Station: 43–54.

d'Herelle, F. 1911. Sur une épizootie de natur bactérienne sévissant sur les sauterelles au Mexique. Compt. Rend. Acad. Sci. 152: 1413–15.

Greathead, D. J. 1992. Natural enemies of tropical locusts and grasshoppers: their impact and potential as biological control agents. In: Lomer, C. J.; Prior, C., eds. Biological control of locusts and grasshoppers. Wallingford, UK: C.A.B. International: 105–121.

Henry, J. E.; Wilson, M. C.; Oma, E. A.; Fowler, J. L. 1985. Pathogenic micro-organisms isolated from West African grasshoppers (Orthoptera: Acrididae). Tropical Pesticide Management 31: 192–195.

Hochberg, M. E. 1989. The potential role of pathogens in biological control. Nature 337: 262–265.

Howard, L. O. 1902. Experimental work with fungous diseases of grasshoppers. In: The yearbook of agriculture: 1901. Washington, DC: U.S. Department of Agriculture: 459–470.

Howarth, F. G. 1991. Environmental impacts of classical biological control. Annual Review of Entomology 36: 485–509.

Huffaker, C. B.; Messenger, P. S.; DeBach, P. 1971. The natural enemy component in natural control and the theory of biological control. In: Huffaker, C. B., ed. Biological control. New York: Plenum Press.

McDaniel, B. 1987. Grasshoppers of South Dakota. Tech. Bull. 89. Brookings, SD: South Dakota State University and South Dakota Agricultural Experiment Station. 163 p.

Packham, S. O.; Kis, L. P., Brusven, M. A. 1993. Relationship between spring-fed swales and adjacent xeric grasslands on the incidence of *Entomophaga calopteni* (Entomophthorales: Entomophthoraceae) among grasshoppers on southwest Idaho rangeland. Environmental Entomology 22: 1156–1160.

Parker, F. D. 1971. Management of pest populations by manipulating densities of both host and parasites through periodic releases. In: Huffaker, C. B., ed. Biological control. New York: Plenum Press.

Prior, C.; Greathead, D. J. 1989. Biological control of locusts: the potential for the exploitation of pathogens. FAO Plant Protection Bulletin 37: 37–48.

Siddiqui, R. K.; Irshad, M.; Mohyuddin, A. I. 1986. Digest: *Scelio* spp. as biocontrol agents of acridids. Biocontrol News and Information 7(2): 69–76.

Streett, D. A.; McGuire, M. R. 1990. Pathogenic diseases of grasshoppers. In: Chapman, R. F.; Joern, A., eds. Biology of grasshoppers. New York: J. Wiley & Sons, Interscience. Chapter 15.

Steinhaus, E. A. 1956. Microbial control—the emergence of an idea, a brief history of insect pathology through the nineteenth century. Hilgardia 26: 107–160.

References Cited—Unpublished

Foster, R. N.; Reuter, K. C.; Bradley, C. A.; Wood, P. P. 1991. Preliminary investigations on the effect of *Beauveria bassiana* on several species of rangeland grasshoppers. In: Cooperative Grasshopper Integrated Pest Management Project, 1991 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 203–210.

Foster, R. N.; Reuter, K. C.; Bradley, C.; Britton, J.; Black, L.; Drake, S.; Leisner, L.; Tompkins, M.; Fuller, B.; Hildreth, M.; Kahler, E.; Radsick, B. 1992. Development of the fungus *Beauveria bassiana* as a bio-insecticide for grasshoppers on rangeland. In: Cooperative Grasshopper Integrated Pest Management Project, 1992 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 207–215.

Foster, R. N.; Reuter, K. C.; Tompkins, M. T.; Bradley, C.; Britton, J.; Underwood, N.; Black, L. R.; Vigil, E.; Daye, G.; Radsick, B.; Fuller, B.; Brinkman, M.; Kahler, E. 1993. Development of the fungus *Beauveria bassiana* as a bio-insecticide for grasshoppers on rangeland. In: Cooperative Grasshopper Integrated Pest Management Project, 1993 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 233–238.

Foster, R. N.; Reuter, K. C.; Black, L.; Britton, J.; Bradley, C.; Delgado, F.; Post, B.; James, C.; Radsick, B. 1994a. Field development of the fungus *Beauveria bassiana* as a bio-insecticide for grasshoppers on rangeland. In: Cooperative Grasshopper Integrated Pest Management Project, 1994 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 201–209.

Foster, R. N.; Reuter, K. C.; Black, L. R.; Bradley, C.; Britton, J.; Post, W. L.; Kahler, E. 1994b. Laboratory bioassays of *Melanoplus sanguinipes* grasshoppers treated with various production batches of *Beauveria bassiana.* In: Cooperative Grasshopper Integrated Pest Management Project, 1994 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 211–214.

Hostetter, D. L.; Ramos, M. E.; Breeding, S. L.; Brey, C. W.; Johnson, A.; Shimek, C.; Unterseher, J. 1992. Development and implementation of classical biological control of rangeland grasshoppers with *Entomophaga praxibuli,* an Australian isolate of the *Entomophaga grylli* complex. In: Cooperative Grasshopper Integrated Pest Management Project, 1992 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 187–201.

Section II. Chemical Control

For blocks of rangeland in the thousands of acres, aerial application of insecticides provides one of the most cost-effective methods of grasshopper management. Research has shown that aerial application of bait in the form of treated wheat bran can also be cost effective, especially in environmentally sensitive areas. (APHIS photo by Mike Sampson.)

II.1 Introduction to Chemical Control

R. Nelson Foster

NOTE: Acephate is no longer approved by EPA for rangeland grasshopper control.

Since the beginning of recorded history, outbreaks of grasshoppers have plagued humanity, coming in direct competition with people for life-sustaining food. Humans were initially helpless against grasshopper outbreaks. Natural control through grasshopper predators, parasites, diseases, and unfavorable weather conditions offered the only relief that could be expected.

Colonial America recorded grasshopper outbreaks in the mid-1700's. From 1718 to 1767, the founders of California missions faced near famine from grasshopper plagues (Schlebecker 1953). During 1874 to 1877, the outbreak of the Rocky Mountain locust (grasshopper) became widespread and severe. The U.S. Congress established the U.S. Entomological Commission to deal with grasshopper problems (Parker 1952). The first effective chemical control of U.S. grasshopper populations took place in 1885 with the use of bran and arsenic-based bait.

From then until the middle 1900's, poison baits that grasshoppers would eat were the most commonly used type of chemical control for combating these pests. Baits laced with arsenic were popular until 1943, when sodium fluosilicate became the active ingredient of choice.

Through increased research, baits were improved, and by 1950 the chlorinated hydrocarbons chlordane, toxaphene, and aldrin replaced sodium fluosilicate. Aerially applied sprays containing the newer chemicals saw use in the late 1940's and were so effective that bait treatments essentially disappeared in the 1950's (Parker 1952). Improved baits are now enjoying a renewed interest, primarily because of environmental concerns and improved application technology. By the mid to late 1960's, malathion spray applied at ultralow volume became the most common chemical for controlling grasshoppers on rangeland. In the early 1970's, the Sevin 4-Oil® formulation of carbaryl became available. By the early 1980's, acephate was added to the group of chemicals recommended for controlling grasshoppers.

There are several other chemicals highly toxic to grasshoppers, but they are not registered for use on rangeland, where treatments occasionally contact domestic livestock and wildlife. For grasshopper control programs that the U.S. Department of Agriculture (USDA), Animal and Plant Health Inspection Service (APHIS) oversees, only chemicals with minor impact on the environment and nontarget organisms are used. These chemicals give acceptable performance on grasshoppers. Currently, malathion, carbaryl, and acephate remain the three recommended chemicals for use in large-scale, aerially applied control programs against grasshopper outbreaks.

Because grasshopper outbreaks often are so extensive that individual land managers and owners alone cannot control them, Congress charged USDA in 1934 to help protect rangeland and cropland from the destructive populations of grasshoppers (U.S. Department of Agriculture 1979). In the 1980's, for example, the Federal Government sprayed millions of acres of public and private western rangeland for grasshopper control. Control programs on a smaller scale take place almost every year in some States. Congress authorized USDA involvement in large-scale, coordinated efforts against damaging outbreaks of grasshoppers by the Incipient and Emergency Control of Pests Act, 1927; the Organic Act of the Department of Agriculture, 1944; the Cooperation with State Agencies in the Administration and Enforcement of Certain Federal Laws Act, 1962; and the Food Security Act, 1985.

Currently, two major programs administrated by USDA exist for managing grasshoppers on or near rangeland areas. They are the Rangeland Grasshopper Cooperative Management Program and the Cropland Protection Program. USDA is also involved when grasshoppers reach certain levels on Conservation Reserve Program lands.

The work to develop alternatives to chemicals for suppression and control of grasshopper outbreaks is ongoing. However, advances are slow, and currently the proven options are few at best. The small number of effective tools and strategies for managing grasshoppers dictates continued reliance on chemical control as a major option within grasshopper management. When outbreaks reach crisis proportions, chemical control of some form may be the only remaining option.

A primary goal of integrated grasshopper management is to prevent the buildup of populations to damaging levels. However, some periodic outbreaks will inevitably occur, and some will require immediate intervention in the form of fast-acting chemical control. The traditional use of chemicals has been to control grasshoppers to the greatest possible extent. However, recent improvements in equipment and application methods and the development of a system for analyzing the economics of alternate strategies are expanding the role of chemicals. These developments may lead to strategies with objectives other than maximum control and ultimately will allow the use of a lower dosage of chemicals previously believed to produce unacceptable results.

The following section will explore some major techniques and issues related to current chemical control tools and tactics and will also discuss and propose some future tactics. The chapters in this Chemical Control section of the Grasshopper Integrated Pest Management User Handbook serve as a state-of-the-art source of information about the role chemical control has in integrated rangeland grasshopper management technology.

Suggested References

Parker, J. R. 1952. Grasshoppers. In: Insects: the Yearbook of Agriculture, 1952. Washington, DC: U.S. Department of Agriculture: 595–604.

Schlebecker, J. T. 1953. Grasshoppers in American agricultural history. Agricultural History 27: 85–93.

U.S. Department of Agriculture, Animal and Plant Health Inspection Service. 1979. Final environmental impact statement, USDA-APHIS-ADM-79-I-F. Rangeland Grasshopper Cooperative Management Program. Washington, DC: U.S. Department of Agriculture, Animal and Plant Health Inspection Service. 177 p.

II.2 Evaluation of Rangeland Grasshopper Controls: A General Protocol for Efficacy Studies of Insecticides Applied From the Air

R. Nelson Foster and K. C. Reuter

NOTE: Acephate is no longer approved by EPA for rangeland grasshopper control.

Introduction

Many chemical compounds are registered for use against grasshoppers, but only a few are used in the large-scale cooperative private–State–Federal rangeland grasshopper management programs directed by the U.S. Department of Agriculture's Animal and Plant Health Inspection Service (USDA/APHIS). APHIS chooses and approves compounds based on (1) effective performance against grasshoppers on rangeland, and (2) minimal or negligible impact on the environment and nontarget species. On rangeland, APHIS normally uses these compounds at the lowest active ingredient (AI) level listed on the label.

To be approved for use by APHIS, chemical insecticides must be evaluated for effectiveness, or efficacy. Efficacy testing determines the levels of performance for a specific compound formulation at different doses of active ingredient and in different application volumes of diluent (a diluting liquid or solid) per unit of surface area. Candidate treatments may be newly developed compounds, new formulations of currently used compounds, or registered compounds proposed for rangeland use for the first time. Based on 15 years of development, the following describes the protocol (procedure) used to evaluate candidate treatments for use on rangeland grasshoppers in APHIS-managed programs.

Geographic Location

The first step in an efficacy test is selecting a location for the study. The test is only as good as the location where it is conducted. The location should be typical of areas commonly treated in cooperative large-scale management programs. Also, the location should have a typical population mix of rangeland grasshoppers or a majority of species commonly considered as potentially damaging to rangeland. Average population levels should be at least 10–25 grasshoppers/yd^2. Lower populations may limit the level and type of statistical analysis performed on the data.

Test locations commonly are selected from areas experiencing a significant outbreak of grasshoppers and near where control programs are planned. These locations have two major advantages. First, such locations allow researchers to experience firsthand some of the local problems that may exist in controlling grasshoppers. Second, the proximity to a major control program activity allows a control program manager a firsthand view of the potential tool.

While there are distinct advantages in locating research and program activities near each other, doing so may cause problems. First, the large-scale program and the researcher may be competing for the same infested land. The program manager is interested in improving the control plot by simplifying boundaries or protecting its integrity from migration of grasshoppers from untreated plots in the research design. The researcher looks for desirable population and topographic features typical of a program. For the private party, a cost share will be required if the land is included in the control program, but charges are generally not assessed for land used in research. Close communication with the program manager is the only solution to these potential conflicts.

Sometimes, the test area may be located adjacent to the program area. In such cases, researchers must take extra precautions to ensure that no contamination from the control block will compromise the integrity of the test area. In many cases, it is easier to choose a test area separated from a nearby control block. With appropriate approval, both public and private lands can be used. Permission to use public lands usually requires additional procedures compared to private lands. Because of the brief period of time between locating a test area and beginning the test (occasionally as few as 3–4 days), researchers most often choose private land with approval of landowners, lessees, or others who may be involved. Tests on rangeland usually require the use of trail bikes and the temporary positioning of other equipment. Researchers discuss use of these items with and get approval from the landowner as one of the first steps in site selection.

Once general permission for use of the land is obtained, a preliminary survey on the parcel of land proposed for the test is conducted. The preliminary survey generally consists of conducting population estimates every $^1/_4$ mi and a cursory examination of terrain and vegetation types. This survey ensures adequate uniformity in the general vegetation types and grasshopper population levels for the study proposed. The absence of livestock during the study period is not a requirement but simplifies counting and eliminates the need to build temporary fences for protection of any required specialized equipment.

Close proximity of the test area to a landing strip or airport is extremely important. Many experiments require several changes in equipment and formulations, and since only 1–2 hours of application time may be available each day, ferrying distances should be kept to a minimum. Lodging close to the test area also is worth consideration. Daily travel will be needed during setup and application and usually for 2–4 weeks after the final application.

Types and Sizes of Experiments

Several general types and sizes of experiments take place when APHIS evaluates a candidate treatment for potential program use. The evaluation usually begins with replicated (repeated) small rangeland plots and eventually progresses to larger blocks. Each type of experiment is important in producing a complete evaluation and recommendation that both industry and the user communities will accept. Later, for treatments used in cooperative programs, APHIS evaluates each program to document the performance of the compound and the success of the program in which it was used.

Small-Scale Replicated Plot Studies.—After a compound has shown a potential for producing mortality to the target pest either in the laboratory or on small (less than 10 acres) field plots, the evaluation process graduates to replicated field plots of substantial size. At this stage in the development of a treatment, testing for the first time incorporates the aerial application aspect. APHIS typically designs the experiments to determine the (1) lowest effective dose of active ingredient, (2) minimum volume of application, and (3) optimal type of diluent (water, oil, or solid bait carrier). These experiments also serve to determine if proposed formulations are compatible with existing commercial aerial application equipment. Experiments also may be designed to determine the optimal nozzle type and size to be used with the final formulation.

Plots are typically square and 40 acres in size ($^1/_4$ mi by $^1/_4$ mi). This size allows for a buffer zone on all sides of the centrally located evaluation site. The buffer area protects the evaluation site from grasshoppers that have been exposed to different treatments and may migrate from adjacent plots. Additionally, buffer areas ensure that any drift contamination near the edges of plots will not jeopardize the integrity of the evaluation site. In studies of aerially applied insecticide on rangeland, smaller plots are simply inadequate for evaluating treatment impact on grasshopper populations. Plots larger than 40 acres may be used. Larger plots increase the protection of the evaluation area but rapidly use up valuable rangeland test acreage. In small-scale studies, using four replications of each aerially applied treatment is typical and is considered minimal.

An example of a typical small-scale study follows. Grasshopper mortalities resulting from three dosages of a candidate formulation at a fixed volume of application are compared with each other. Mortalities are also compared to those produced by a treatment currently used for controlling grasshoppers, called a standard. Mortalities resulting from all four treatments are compared with mortalities in untreated plots. These untreated plots will show the mortality rate that naturally occurs during the experiment. In this experiment, there are five different kinds of plots called “treatments” with each replicated four times. The entire experiment takes 20 plots and uses 800 acres. The untreated control plots are the most important in the experiment. All other treatments are judged against the controls. Control plots are part of the experimental design and must be included in the process of assigning treatments to specific plots. Other actual examples can be seen in Foster et al. (1983 unpubl.) and Jech et al. (1993).

Because densities of grasshoppers may vary considerably over the study area, it is important to ensure that any one treatment is not assigned exclusively to high or low grasshopper population levels. In small-scale experiments, the population-level values of the plots are typically arranged in descending order of density. In the case of the above example, each of the five treatments are randomly assigned to plots within the top five densities, five treatments to the next five densities, and so on until the desired number of replications have been performed. This ensures that all treatments are tested against similar population densities. Typically, one or more treatments of those tested in small replicated plot studies will be suitable for large-scale testing. (See table II.2–1 and fig. II.2–1.)

Large-Scale Simulated Program Studies.—After successful small-scale testing, the next step is to evaluate the candidate formulations under simulated program conditions. Doing this ensures that the level of performance seen in tightly controlled small-scale experiments can be expected when much larger acreages are treated. These tests challenge the formulation (1) under environmental and meteorological conditions expected during a program, and (2) for compatibility with commercial spraying equipment for extended periods of time. Successful performance in these studies may result in recommendations for program use.

In these experiments, application flights of at least 1 mi in length are desirable. Plot size typically ranges from 640 acres (a section) to 1,000 acres. With a plot of this size and a single aircraft such as an Ag Truck, researchers can use much or all of acceptable early morning application time in a single plot. The changing meteorological conditions that occur over this time period allow for assessment over the varying conditions that occur during a typical control program application day. Aircraft altitude (application height) in these studies will be similar to those APHIS uses during programs.

A typical large-scale study may consist of one or two different formulations of a candidate compound, a standard treatment, and an untreated control plot, each on a minimum of 640 acres. Because of the size of acreage involved in these tests, true statistical replication, in the general vicinity, is usually impossible. However, it is common to conduct the same test in other areas or States. Typically, the candidate and standard treatments, as well as the untreated control, are randomly assigned to one of several (in this case, three) adjoining plots. Before treatment, these plots are assessed to make sure they are suitable for the experiment. Unfortunately, in many cases, enough grasshopper-infested acreage is not available. In such cases, the untreated check sites are established outside of the treated plots and at a distance to ensure that there is no contamination from treatment.

A large-scale experiment usually relies on 9–10 evaluation sites per treatment plot. Without prior knowledge of plant communities, soil characteristics, or species composition of grasshoppers, the researchers determine the location of each evaluation site using topographic and county maps. These sites generally are distributed evenly over the entire plot (see fig. II.2–2). With this technique, each type of habitat is represented proportionately in the evaluation of each plot. An actual example can be found in Foster et al. (1993 unpubl.).

Efficacy Evaluation of Control Programs

Evaluation of performance continues even after treatments have been recommended for cooperative programs. APHIS evaluates each program to determine the performance of the treatment and to document the level of success of the program in which it was used.
Cooperative programs may vary greatly in size, from 10,000 acres to 100,000-plus acres, and may rely on several aircraft flying in formation for application. Evaluation of a program treatment is similar to that which occurs for program-simulated experiments. Evaluation sites are evenly distributed within the treatment area, while allowing for access by roads or trails. Sites are selected at 1 per 1,000 acres for the first 100,000 acres, and 1 per each 10,000 acres above 100,000 acres. Where programs are less than 10,000 acres, we recommend using a minimum of 10 treatment evaluation sites. We identify the evaluation sites before application. Evaluation of those sites is in addition to the more cursory visual mortality checks, commonly conducted on all cooperative control programs.

APHIS also establishes an equal number of untreated check sites that can be used for comparison in the evaluation. The untreated sites are mandatory. However, because a program goal is usually to treat all land infested with grasshoppers that cause damage at economic levels in a given area, untreated control sites within the treated block are not possible. Consequently, untreated control sites are situated outside, but near to, the boundary of the program block and surround the entire perimeter of the area tested.

Plot and Evaluation Site Setup

In both small- and large-scale simulated program studies, corner boundaries of all plots have flexible poles to which streamers of flagging tape are attached. We use two colors, usually orange and white, to increase visibility. Corners also are marked with a wooden stake labeled to identify the plots.

We mark evaluation sites with flexible poles and wooden stakes. In replicated small-plot studies, only a single color of tape is attached to the site markers to prevent confusion with corners. At each evaluation site, we use 0.1-m^2 aluminum rings (Onsager and Henry 1977) to delimit 40 areas for counting grasshoppers. Starting at the wooden stake, we arrange the rings about 5 yd apart in a large circle about 64 yd in diameter. Placement of individual rings is simply a random drop at the end of each 5-yd interval.

The circle arrangement provides for a curved transect of 200 yd which allows the sample counter to finish at the initial stake. Compared to techniques where counting areas are concentrated and uniform habitat is desired, this arrangement of sample rings allows for sampling a more diversified habitat. The circular arrangement also ensures that counting at all sites will be affected by wind and sun angles from all directions. Ring spacing of 5 yd between rings ensures that there is no disturbance to the next area to be counted during an ongoing count. In some programs, we may base pesticide effectiveness on estimates of grasshoppers in 18 visualized 1-ft^2 areas at evaluation sites rather than counts from rings. While not as accurate as counting from rings, the resulting data generally yield good estimates of the level of control achieved by the treatment.

Application

Calibration of the aircraft delivery system (spreader for baits and spraying systems for liquids) is the most important aspect of application. The accuracy of application in experiments and programs depends on repeatable precision obtained through the use of proven calibration procedures. Details of some of these procedures are in the chapters on “Calibration of Aerially Applied Sprays” (II.8) and “Equipment Modification, Swath Width Determination, and Calibration for Aerial Application of Bran Bait with Single-Engine Fixed-Wing Aircraft” (II.18) in this section of the User Handbook.

In small-scale replicated plot experiments, we consider the order of treatments. Similarly based formulations are grouped together in the sequence of application to minimize equipment cleanup and changeover time between treatments. We arrange the dosages tested in increasing or decreasing order depending on the complexity of mixing required for test formulations.

Conventional replication in an experiment requires all treatments to be applied once before repeating. Then all treatments are applied a second time before a third treatment is applied, and so forth. The arguments against this type of sequencing are numerous and usually win out to preserve time and money and to maintain a uniform grasshopper age structure against which the treatments are applied. Typically, we apply each treatment to all of its assigned plots before changing over equipment for the next formulation in the sequence of application.

Table II.2–1—Pretreatment grasshopper densities per square meter, arranged in descending order with randomly assigned treatments for each density group

Grasshopper density per m^2	Plot number	Assigned treatment
41	17	Treatment 2
41	16	Treatment 1
36	13	Treatment 3
36	1	Untreated
29	11	Standard
29	3	Treatment 1
25	18	Treatment 2
23	12	Treatment 3
22	6	Untreated
19	20	Standard
18	19	Treatment 1
18	2	Standard
14	7	Untreated
13	15	Treatment 3
13	4	Treatment 2
11	10	Untreated
9	5	Standard
9	9	Treatment 3
9	14	Treatment 2
6	8	Treatment 1

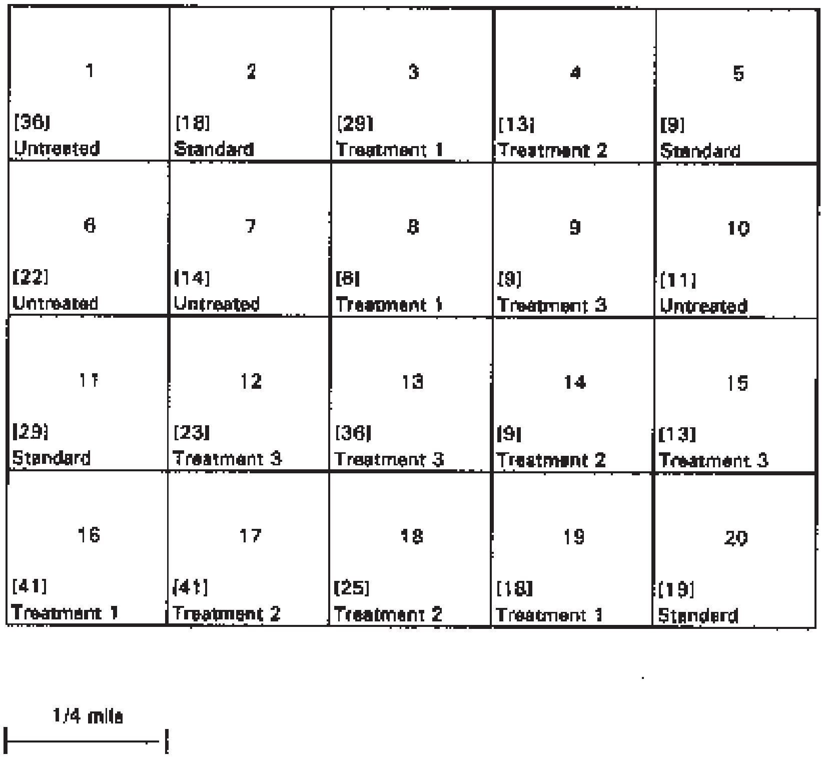


Figure II.2–1—Plot map showing pretreatment mean density of grasshoppers per square meter, in parentheses, and assigned treatments from table II.2–1.

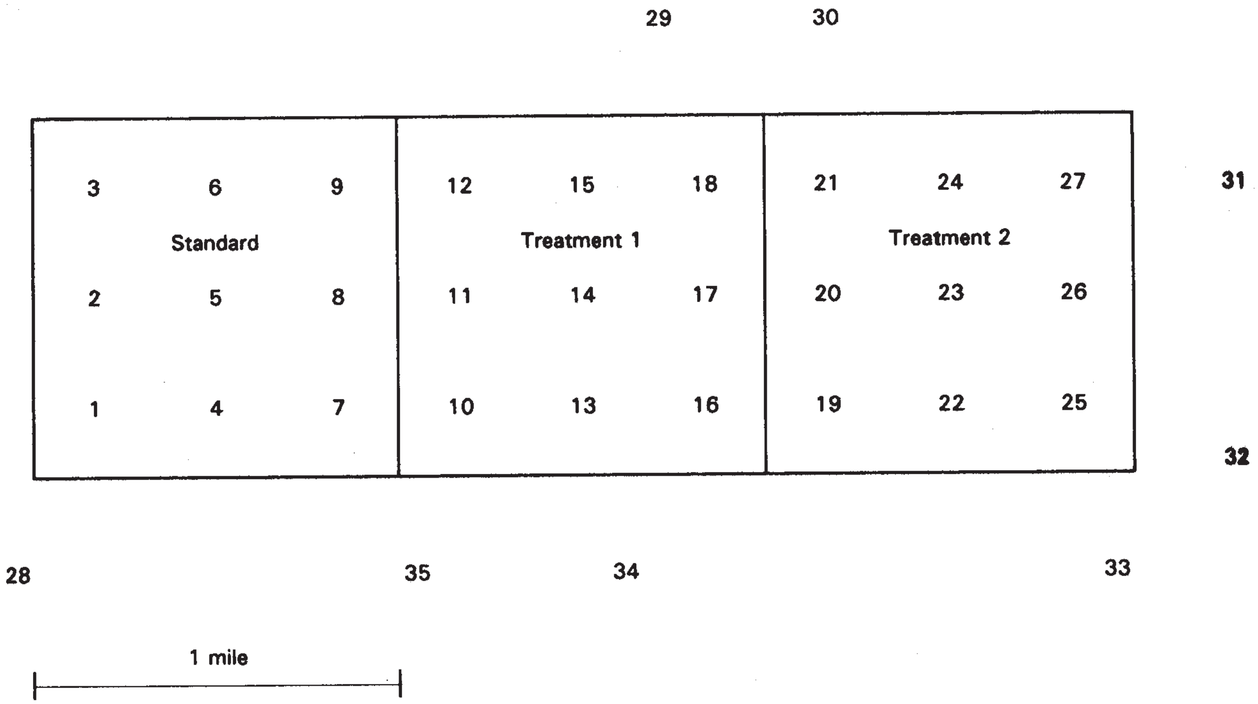


Figure II.2–2—Map showing 640-acre (1-section) plots showing evaluation sites numbered within the plots and numbered untreated evaluation sites located around the perimeter of the treated plots.

Deciding when to start and stop application is not only a decision made daily, but one made on each pass or run of an applying aircraft. Decisionmaking requires consideration of windspeed, ground and air temperatures, amount of moisture on vegetation, and the possibility of precipitation.

In some States, laws define some of the guidelines under which applications are made. Generally the smaller the plot size, the more restricted the guidelines for application become. Typically, with 40-acre replicated plots, application is stopped when winds exceed 3–4 miles per hour or ground temperatures exceed air temperatures. Monitoring spray-sensitive cards in adjacent plots or designated no-spray areas during application is important to determine unacceptable pesticide drift.

Aircraft Guidance

Guidance of aircraft during application varies from sophisticated electronic guidance systems used in many programs to simple but effective flag-waving provided by ground personnel in small plots. However, all guidance depends on the specific swath width assigned to a particular type of aircraft and equipment and the material being applied. Ground crews can determine the location of each swath by using measuring tapes or calibrated wheels or by accurately pacing a known distance equal to the desired swath width. Also, ground crews can make and mark these measurements ahead of time or as application is occurring.

The width of a swath is determined through extensive testing prior to small-plot or program application. Swath widths of 75 ft for most water-based formulations and 100 ft for most oil-based formulations are typical for small-plot work with a Cessna Ag Truck aircraft, for example. Swath width assignments for other types of aircraft are found in the USDA-APHIS-Aerial Application Prospectus. APHIS generally conducts applications at a height equal to 1 $^1/_2$ times the wingspan of the aircraft.

Recordkeeping

Recordkeeping is essential in assessing any treatment in both test work and program use. At the airport, it is important to maintain a record of the final calibration for comparison with the actual acreage covered and material used for each flight. In the field, it is important to measure and record numerous parameters: (1) beginning and ending time of actual application, (2) windspeeds during application, (3) ground and air temperatures during application, and (4) passes that the aircraft makes when applying material. In experimental work, these parameters should be measured and recorded at the beginning and ending of treatment for each plot treated. In programs where multiple aircraft are used, the number and location of partially or completely inoperable spray tips on each aircraft should immediately be reported to the official in charge. In test work, seeing such occurrences requires landing the aircraft to correct the problem.

Evaluation Site Data

The basic types of data collected are grasshopper species composition and density. The conditions, including weather, present during data collection are recorded. Depending on the specific study, we may collect other types of data for association with population estimates, such as vegetation composition and quality or spray droplet size and frequency.

We estimate the grasshopper population by counting the number of grasshoppers found in 40 0.1-m^2 rings at each site. We count and record each ring separately. In our evaluations, the order of counting is always the same, counterclockwise from the site stake. A more detailed description and discussion of procedures for counting grasshoppers is in the chapter on survey in the Decision Support Tools section of the User Handbook.

A typical square mile of infested rangeland will contain 15 to 40 different grasshopper species, some of which may not be causing damage. Estimating the relative abundance of each species is important in order to determine the need for control and the effectiveness of treatments on target species. Base estimates on samples taken from the population with a sweep net. Such sampling is done by taking equal numbers of low–slow (ground level) and high–fast (canopy level) sweeps uniformly along the margin of the circle of rings. Low–slow sweeps ensure the capture of early instar and slow-moving species, while high–fast sweeps ensure the capture of older instars and more-active species. Try to get a collection of at least 100 grasshoppers at each site. Do this by conducting 100–200 low–slow and high–fast sweeps each. Determine the density of the individual species by multiplying the frequency of occurrence, from the sweep sample, by the total density of grasshoppers at the site (counts from rings). Except in some program evaluations, take sweep samples whenever a grasshopper count is conducted.

Make pretreatment counts to determine the population levels against which posttreatment levels are compared. In small replicated plot studies, use the initial pretreatment count to assign treatments appropriately. These studies require additional pretreatment counts conducted closer to the date of treatment for comparison with posttreatment counts. If at all possible, take pretreatment counts 0–48 prior to treatment.

Counts from untreated and treated sites taken on the same day will allow for converting reduction calculations (posttreatment count divided by pretreatment count) to a percentage control value (Conin and Kuitert 1952). This formula is discussed in the chapter "Bait Acceptance by Different Grasshopper Species and Instars." Using the untreated control-plot data in this fashion allows for adjustment for any natural mortality that occurs and will provide a value of the actual mortality that can be attributed to the treatment. Just as important, if not more so, this procedure will provide an adjusted value that accommodates the day-to-day meteorological changes (such as wind, temperatures, and precipitation) that affect the actual grasshopper counts.

The interval between treatment and the posttreatment count depends on the purpose of the evaluation and the treatment(s) used. With solid baits or fast-acting, short residual sprays, posttreatment intervals of 2, 4, and 7 days are typical. For slower acting or longer residual treatments, weekly intervals at 1, 2, 3, and 4 weeks posttreatment are typical. If two or more treatments that work at different speeds are to be compared, collect the data at similar posttreatment intervals for all treatments. In such cases, an end-of-study or season comparison is helpful in addition to evaluation at specific intervals.

Conclusion

The above protocol is not a detailed standard operating procedure but is intended to serve as a general guideline for several types of treatment evaluations on rangeland grasshoppers. The kinds of data and methods of collection discussed here will allow researchers and program evaluators to use numerous kinds and strategies of analysis.

References Cited

Connin, R. V.; Kuitert, L. C. 1952. Control of the American grasshopper with organic insecticides in Florida. Journal of Economic Entomology 45: 684–687.

Foster, R. N.; Reuter, K. C.; Gourd, J. M.; Enis, P. J. 1983. Field experiments on the toxicity of acephate for control of grasshoppers (Orthoptera: Acrididae) on rangeland. Canadian Entomologist 15: 1163–1168.

Jech, L. E.; Foster, R. N.; Colletto, D.; Walgenbach, D. D.; Roland, T. J.; Rodriguez, G. D.; Bohls, R.; Houston, R. D.; Meeks, W. K.; Queener, R. L.; Jackson, C. L.; Dines, J. L.; Puclik, M. J.; Scott, A. K. 1993. Field evaluation of diflubenzuron and carbaryl bran baits against grasshopper (Orthoptera: Acrididae) populations in South Dakota. Journal of Economic Entomology 86: 557–565.

Onsager, J. A.; Henry, J. E. 1977. A method for estimating the density of rangeland grasshoppers (Orthoptera, Acrididae) in experimental plots. Acrida 6: 231–237.

References Cited—Unpublished

Foster, R. N.; Reuter, K. C.; Black, L. R.; Daye, G.; Vigil, E. L.; Walz, A.; Radsick, B. 1993. Field evaluation of aerial application of two formulations of diflubenzuron for rangeland grasshopper control under simulated control program operational conditions. In: Cooperative Grasshopper Integrated Pest Management Project, 1993 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 93–100.

II.3 Sprays *versus* Baits

R. Nelson Foster and Jerome A. Onsager

NOTE: Acephate is no longer approved by EPA for rangeland grasshopper control.

Chemicals can be applied in two different forms, liquid sprays or solid-based baits, to suppress or control populations of grasshoppers on rangeland. Both forms have distinct advantages and disadvantages, depending on the situation in which they are used. The diverse habitat, topographical features, meteorological conditions, economic concerns, and environmental constraints associated with grasshoppers on rangeland play an important role in choosing the best form of treatment. This chapter briefly discusses the advantages and disadvantages of both liquid and bait formulations and the eight major factors to be considered in the selection of a type of treatment.

Advantages and Disadvantages

Cost of Aerial Application.—Generally, contract costs are substantially lower for applying sprays than baits. These differences are primarily a result of the wider swaths used in spray application. Bait application costs also may be higher because an acre equivalent of bait typically occupies more space than a liquid. Some types of aircraft and bait-dispensing equipment produce about the same swath width with both sprays and baits. However, most of the few systems that have been evaluated to date produce a narrower swath with baits.

Of the spreader–aircraft combinations evaluated to date, the Bull Thrush (Thrush 1,200 hp) and a Transland 22007 spreader produced the bait swath most similar to the swaths from liquid sprays. The Bull Thrush has a spray swath of 150 ft for oil mixtures and 100 ft for water mixtures and produces a 100-ft swath with bran bait using the 22007 spreader. In contrast, the Turbine Thrush with the same swath widths for oil and water mixtures produced only a 45-ft swath with bran bait and a Transland 20250 spreader. Bait application can become more cost effective if new spreaders, which produce wider swaths, are used and/or if application objectives are changed to omit the old requirement of complete coverage of the treatment area.

Amount of Active Ingredient Required.—Baits typically require significantly less toxicant than sprays. For example, when carbaryl is used in a spray, it is typically applied at 0.375–0.5 lb of active ingredient (AI) per acre. When it is used in a bait, it is typically applied at 0.04 lb (by ground) to 0.03 lb (by air) of AI per acre. The lower amount of active ingredient is attractive from the standpoint of both cost and possible impact on the environment.

Level of Control.—On a typical assemblage of grasshopper species (the total population), sprays applied properly always produce a higher average level of mortality than baits. All species of grasshoppers do not feed equally on currently registered baits, and some species seem to avoid almost any contact with bait on the ground. For species susceptibility to bait, see the chapter “Bait Acceptance by Different Grasshopper Species and Instars” (II.12). Sprays typically produce higher levels of mortality on all species of grasshoppers, through both direct contact with the grasshopper itself and by the grasshopper’s feeding on contaminated vegetation (ingestion).

Grasshopper Density and Species Composition.—Sprays produce similar levels of mortality regardless of the grasshopper density. Baits cause highest mortality against low densities of grasshoppers where the dominant species readily consume bait. When very high densities of susceptible grasshoppers (greater than 30–40/yd^2) are treated with bait, there simply are not enough bait particles for all the grasshoppers. According to theoretical models, 1.5 lb of 2 percent carbaryl bait per acre can kill about 65 grasshoppers/yd^2 under perfect conditions. In actual practice, however, it is not likely that this dosage will kill more than 20 to 30 grasshoppers/yd^2. Increasing the amount of bait will increase the level of control slightly but usually not enough to be justified economically.

Nontarget Arthropods.—Sprays kill by both contact and ingestion; baits kill by ingestion. Sprays may affect to some degree both canopy-dwelling and ground-dwelling arthropods, such as insects and spiders. In particular, sprays have the potential to affect those arthropods that feed or rest on the vegetation that has been sprayed. Because baits fall through the vegetation to the ground and work by ingestion only, they may affect only some of the ground-dwelling arthropods that feed on the bait. Both treatments could produce some secondary poisoning of arthropods that scavenge upon affected grasshoppers.

Calibration of Equipment.—It is a misconception that calibration of bait-applying equipment is more difficult than calibration of spray equipment for liquid chemical insecticides. This common misconception is based on lack of experience with bait equipment and its calibration techniques and procedures. Insecticide applicators typically have much more experience with the equipment used to disperse sprays. The Aircraft and Equipment Operations unit of the U.S. Department of Agriculture's Animal and Plant Health Inspection Service lists, to date, 28 different types of fixed-wing aircraft that have been studied and approved for sprays. In contrast, only three different types have been approved for application of baits. With experience, applicators should encounter no substantial difference in the difficulty of equipment calibration for sprays or bait. (A procedure for calibrating bait equipment is found in this section's chapter on "Equipment Modification, Swath Width Determination, and Calibration for Aerial Application of Bran Bait With Single-Engine Fixed-Wing Aircraft" [II.18].)

Aerial Drift and Length of Application Day.—Sprays are much more susceptible than baits to wind-assisted drift and can be carried much greater distances. Drift is a function of wind and temperature at the time of application and the weight of the liquid or solid particle being dispensed. A rise in temperature increases the evaporation and reduces the droplet size in sprays. These changes result in increased buoyancy and drift. For further discussion on the effect of wind and temperature on sprays, see the chapter "Factors Affecting Application and Chemical Deposition" (II.7).

Changes in temperature do not affect the drift of bait. Bait can be very confidently directed to the area of treatment. It is not unusual to discontinue spray application when either wind or temperature conditions might result in unacceptable drift. Winds generally must reach levels that threaten the safety of flight operations before application of baits is discontinued.

Established buffers around bodies of water reflect the dangers of drift and the reduced risk when baits are used. In large-scale cooperative programs, baits can be used within 200 ft of water; sprays require a 500-ft buffer. Spray application usually happens early in the morning, shortly after sunrise, when meteorological conditions are acceptable. These conditions may last for only 1–3 hours. Application of bait can take place at any time during daylight hours, when safe operation of the equipment may be ensured.

Ease of Application.—In spray operations, the applicator must spot clogged nozzles. Applicators can prevent most clogging problems by ensuring that the spraying system is absolutely clean before the material to be sprayed is loaded. Baits require more attention during application. The pilot must manage the physical process of opening the hopper gate of the aircraft consistently. In addition, the pilot must constantly watch for signs of uneven flow of bait during application.

Baits must be carefully inspected for lumps before they are loaded into the aircraft. These lumps will cause partial or complete blockage at the aircraft gate opening and result in nonuniform flow during application. Bait requires more space than sprays. An acre's worth of bait (2 percent carbaryl at 1.5 lb/acre) occupies space equal to about 90 fluid oz, requiring about 3–11 times as much space as an acre's worth of spray material (acephate 32 oz/acre, carbaryl 20 oz, and malathion 8 oz).

How To Decide What To Do

In discussing the eight major considerations that could affect the choice of spray versus bait treatments, no priorities are offered here because no simple rules apply. There are situations where any one of the eight considerations may be the most important determinant of a decision to use either bait or liquid sprays. The complexity of the decision process was one of the reasons why the Grasshopper Integrated Pest Management Project developed Hopper, a computer-based decision support system (see "Decision Support Tools" section of this handbook).

The preferred procedure for deciding on bait versus liquid spray treatment is to gather as much information as possible on the eight considerations under discussion and key that information into Hopper. If specific data on certain questions are lacking, Hopper will generate "default" or representative values that will be reasonably close over a variety of rangeland sites. However, it is likely that accurate site-specific data will yield better recommendations than default values. Hopper will also accept specific data in the form of a range of values, with upper, middle, and lower levels being used to compare decisions under worst-case, best-case, and most likely scenarios. Finally, a manager is free to accept or reject the assessments of Hopper because there may be considerations that only the manager can evaluate for relative importance. However, Hopper's advice can help a manager maximize the chances of making a good decision.

II.4 A Review of Chemical Sprays in Cooperative Rangeland Control Programs

NOTE: Acephate is no longer approved by EPA for rangeland grasshopper control.

R. Nelson Foster and Jerome Onsager

The chemical sprays used against rangeland grasshoppers today and the current cooperative rangeland grasshopper management program are both results of an evolving solution to an age-old problem. That problem is one of how best to control or suppress damaging populations of grasshoppers over widespread areas. The following chapter will review the history and evolution of chemical sprays in rangeland grasshopper control to the present day.

History

In the United States, the history of grasshopper control is interwoven with that of the Mormon cricket. Control was conducted primarily to protect crops, but rangeland also was treated to save forage and prevent insect migration to nearby cropland. During the first half of the 20th century, control relied almost exclusively on poison baits. Although sprays such as paris green and sodium arsenate were used, these compounds fell from favor because the poisoned vegetation endangered livestock (Parker 1952). Both State and Federal assistance were provided for organizing and financing control efforts, particularly during outbreak years.

In the late 1940's and early 1950's, several major developments occurred that significantly changed the way grasshoppers were controlled.

1. Perhaps the most important was the development of the chlorinated hydrocarbon insecticides. They were extremely effective in small amounts against grasshoppers. They could easily be formulated into baits, acted quickly, and had a longer residual effect than previously used baits. Because of these qualities, chlordane and toxaphene in 1949 and aldrin in 1951 quickly replaced previous baits (Parker 1952).

2. Large-scale (thousands of acres) aerial application of bait became more commonplace. Compared to older wet baits, the new compounds could be formulated dry, which made distribution easier. In Montana and Wyoming during 1949–50, aerial application of chlordane and toxaphene baits were the major tools used against grasshoppers (Parker 1952).

3. Sprays of these compounds were also developed at the same time. In addition to being extremely effective, they were much cheaper than baits. Sprays of chlordane, toxaphene, and aldrin first were used in grasshopper control programs in 1947, 1948, and 1950, respectively (Parker 1952).

4. Organized, large-scale programs to control rangeland grasshoppers were started. In 1949, a cooperative program provided for the aerial treatment of toxaphene and chlordane baits to 40,000 acres in Wyoming. Within 2 years, the cooperative program had switched to aldrin spray (Pfadt and Hardy 1987).

5. In 1952, several State departments of agriculture and the U.S. Department of Agriculture (USDA) formed an agreement through a memorandum of understanding that the cooperative grasshopper control programs would be reserved for rangeland. Because of the low cost of the chlorinated hydrocarbons, treatment for crop protection could be borne by the private sector. In the past, government involvement in the form of direct financial aid had been available for treatment to both crop and rangeland. The federally sponsored cooperative grasshopper control program was now focused only on rangeland, both private and public (Dick S. Jackson, personal communication).

The acceptance of these new chlorinated hydrocarbon compounds was short lived. Almost as quickly as they appeared for control of rangeland grasshoppers, their use was discontinued. One of the initially attractive features of the chlorinated hydrocarbons, that of longevity, began to be recognized as a problem. The compounds began to accumulate in the food chain and thus posed a threat to not only the pests they were designed for but to nontarget organisms also. In 1962, Dieldrin, which had been used in cooperative rangeland grasshopper spray programs in 1960–62, was discontinued for use, along with other chlorinated hydrocarbons (Dick S. Jackson, personal communication).

In 1962, carbaryl in the form of the Sevin® 80 S spray formulation became available for use in the cooperative rangeland grasshopper programs. It was used on about 4,000 to 36,000 acres of rangeland annually from 1962 through 1967 (Foster et al. 1983). However, during this time, control was not as high or as consistent as that previously expected of the chlorinated hydrocarbons, and compatibility problems between the spray and aerial spraying systems were commonplace.

In the early 1960's, ultralow-volume (ULV) application—defined as less than 0.5 gal/acre (Maas 1971)—was refined for grasshopper control in the United States. By 1964, Malathion ULV® Concentrate had become the most frequently applied chemical spray for controlling grasshoppers on cooperative rangeland programs.

By 1972, the formulation of carbaryl had been greatly improved and the Sevin 4-Oil® formulation replaced the 80 S formulation as a recommended treatment in the rangeland grasshopper programs.

From 1979 through 1982, research led to the development of formulations of acephate sprays for use against grasshoppers. Acephate in the form of the Orthene® 75 S formulation was adopted as an option for controlling grasshoppers in the cooperative programs in 1982. However, it has been rarely used in the control programs to date. Compared to carbaryl and malathion, the mixing required for acephate made it less desirable.

Through the 1980's, malathion was the most frequently used spray for large-scale cooperative programs. Additional improvements in the formulation of carbaryl have increased its use so that today it is used almost as frequently as malathion in large-scale programs against grasshoppers in the United States.

The three chemical sprays currently approved by USDA's Animal and Plant Health Inspection Service (APHIS) for use on large-scale rangeland grasshopper control programs are acephate, malathion, and carbaryl.

Malathion

Malathion is the common name for the 0,0-dimethyl phosphorodithioate ester of diethyl mercaptosuccinate. It is a broad-spectrum organic phosphate insecticide–acaricide developed by American Cyanamid in 1950. Malathion is registered for control of a wide variety of insects on beef cattle, sheep, goats, swine, grain, fruit and vegetable crops, forests, rangeland, pastures, agricultural premises, poultry ranges, stored grains, and in homes and gardens.

The toxicity of chemicals is measured in relative terms by determining the amount of active ingredient (AI) (in weight) that will kill 50 percent of a test group of laboratory animals. This concept is referred to as the "acute oral LD_{50} (lethal dose)." The LD_{50} of malathion technical material on white albino rats is 1,375 mg per kg of the rats' body weight. This figure marks malathion as moderately toxic to mammals. Malathion exhibits slight to moderate toxicity to birds and moderate to high toxicity to some fish species and other aquatic organisms. It is highly toxic to most insects, including bees and all species of grasshoppers.

While several formulations of the pesticide are available, only the formulations of Cythion® ULV, Fyfanon® ULV, and Malathion ULV Concentrate have been used USDA/APHIS-managed cooperative programs.

For controlling grasshoppers on rangeland, malathion is typically sprayed at 8 fluid oz/acre. The per-acre dose of active ingredient at the application rate ranges from 0.58 lb to 0.61 lb, depending on the concentration of malathion in the particular formulation used.

Malathion provides control through both direct contact and ingestion, although when these types of mortalities are separated in experiments, ingestion results in a greater percentage of mortality (Pfadt et al. 1970).

Malathion is relatively nonpersistent in soil, water, plants, and animals. Residual activity (control) against grasshoppers can be seen for 2 to 5 days after treatment. Malathion is quick acting, usually producing high levels of control during the first and second days following application. When treatment occurs during good conditions for application, control can range from 92 to 96 percent.

Malathion should be used during warm and dry conditions. The air temperature for the expected daytime high should be higher than 80 °F, and rain should not be predicted for the day of treatment. With lower temperatures, the grasshoppers may feed less and be less likely to move into direct contact with spray droplets. Rain soon after an application can reduce mortality dramatically. Foster et al. (1981) discovered rain-related mortality rates as low as 33 percent.

An area of several thousand acres typically contains grasshoppers of as many as 40 different species. Because of the short residual activity of malathion, it is generally selected for use later in the season when the majority of the grasshopper species in an area to be treated have hatched. As a result, the earlier hatching species often have reached adulthood when the applications occur. In these cases, the overall average age of the population could typically be fourth instar to adult.

Waiting to treat a population until it is mostly made up of adults is not a problem unless the grasshoppers have started to mate and lay eggs. But once grasshoppers have reached the adult stage, by definition, forage loss in the area of treatment has taken place.

On small areas, such as “hot-spots,” where only a few species may be predicted to occur or in a large area where only early season species are expected to be the problem, an earlier treatment of malathion targeted to third instars could be preferable. In outbreak years, when economic infestations of large acreages in numerous places within a State occur, timing all treatments ideally becomes difficult. In large outbreak years, malathion may be used later in the season because earlier treatments were logistically impossible. Malathion is most often used late in the season for quick control of older grasshoppers when conditions are hot and dry.

Carbaryl

Carbaryl is the common name for 1-naphthyl N-methylcarbamate. It is a broad-spectrum carbamate insecticide developed by Union Carbide in 1956. Carbaryl is registered for control of a wide variety of insects on fruit and vegetable crops, forests, rangelands, pastures, agricultural premises, poultry houses, horses, dogs, cats, and ornamental and lawn plants, and indoors. Carbaryl demonstrates low to moderate toxicity to mammals (acute oral LD_{50} of technical material on white albino rats, 500 mg/kg), low toxicity to birds, and moderate toxicity to fish, but extreme toxicity to aquatic invertebrates. It is extremely toxic to many insects, including bees and all species of grasshoppers.

The Sevin 4-Oil and Sevin 4-Oil ULV formulations of carbaryl have been used by the USDA/APHIS-managed cooperative programs. For controlling grasshoppers on rangeland, it is typically sprayed at 15 to 20 fluid oz/acre at 0.375 lb AI to 0.5 lb AI. Control is provided through both contact and ingestion, although when the types of mortalities are separated in experiments, ingestion provides the majority of the mortality (Lloyd et al. 1974).

Carbaryl is relatively nonpersistent in the environment. Its residual activity against grasshoppers lasts for 14 to 21 days. Carbaryl is slower acting than malathion or acephate. Depending on conditions, mortality during the first 2 days after treatment may range from 30 to 80 percent. Under good application conditions, mortality may reach 90 percent. However, mortalities ranging from 95 to 99 percent have been recorded in experiments with excellent application conditions.

Carbaryl can be used over a broader range of general climatic conditions than malathion or acephate. Although carbaryl performs well at temperatures in the 60–80 °F range, it kills slower at lower temperatures. This trait may not be as bad as it seems. Under cooler conditions, both grasshopper development and the rate of forage destruction decrease. The Sevin 4-Oil formulation is relatively resistant to removal by rainfall after the spray has dried on the vegetation.

In two major experiments where Sevin 4-Oil was applied to wet vegetation, mortalities eventually exceeded 90 percent. Subtle changes have been made in the formulation of Sevin 4-Oil during the last few years, leading up to today’s Sevin 4-Oil ULV formulation. Along with improved handling characteristics, a trend toward slightly higher mortalities has accompanied these improvements.

Because of the residual activity of the Sevin 4-Oil ULV formulation, it can generally be selected for use both early and late in the season (third instar to adults). However, care must be taken not to use it against grasshoppers that are within a few days of laying eggs because the insects may lay eggs before dying.

Use of carbaryl spray against small hot-spots may not be advantageous if quick migration from the treated area is expected. However, if additional acres adjacent to the hot-spots are treated, use of carbaryl could be acceptable, especially if additional hatch is predicted.

As circumstances dictate, the 0.5-AI dose may be used for older instars and mature grasshoppers. The 0.375-AI dose may be used where younger stages of grasshoppers are present and early treatment can be accomplished or when lower or economically marginal densities of grasshoppers exist.

Where dense vegetation or difficult topography requires greater coverage, a volume of 20 fluid oz/acre should be used. A total volume-per-acre treatment as low as 15 oz/acre may be used when vegetation is sparse. The decision can be made only on a case-by-case basis and by the local personnel involved. The Sevin–ULV spray formulation is typically used under cool conditions in years when rain in the treatment area is not unusual.

Acephate

Acephate is the common name for 0,S-dimethyl acetylphosphoramidothioate, a broad-spectrum organic phosphate insecticide developed by Chevron Chemical Co. in 1972. Acephate controls a wide variety of insects on several grain and vegetable crops, forests, rangeland, pastures, grass, trees, shrubs, cotton, and ornamentals.

Acephate demonstrates low to moderate toxicity to most terrestrial and aquatic animals, including mammals (acute oral LD_{50} of technical material on white albino rats, 866 mg/kg). It is highly toxic to many insects, including bees and all species of grasshoppers.

While several formulations of the pesticide are available, only Orthene® 75S and Orthene Specialty Concentrate® will be addressed here. For controlling grasshoppers on rangeland, acephate is typically sprayed at an application dose of 0.094 lb of AI in 32 oz of water, plus an antidrift additive such as Orthatrol or Nalcotrol (at 9 fl oz per 100 gal of mix) and unsulfured molasses (at 3 percent of the total volume). The addition of unsulfured molasses to the formulation results in slightly quicker action. It is unclear whether this is a result of attractance, additional protection from photo degradation, increased anti-evaporation qualities, or a combination of these actions. Control is provided through both contact and ingestion. When the types of mortalities are separated in experiments, ingestion results in greater mortality (Foster et al. 1984).

In soil, acephate is readily degraded through biological activity: its half life is about 11 days in soils with moisture levels and organic content comparable to those in the West and Midwest. Residual activity against grasshoppers is intermediate, between that of malathion and carbaryl. Some activity can be seen for up to 10 days, but most mortality occurs by the fourth day after treatment. When treatment occurs during good conditions for application, mortality can range from 92 to 94 percent.

With acephate, maximum mortality is reached slower than with malathion but quicker than with carbaryl. Acephate can be used during warm and dry conditions. The air temperature for the expected daytime high should be higher than 75 °F, and rain should not be predicted for the day of treatment. Because of the longer residual activity compared to malathion, acephate can be used in some cases where the lack of residual activity would be a concern for malathion. Conditions for acephate's use more closely parallel those for malathion than for carbaryl. Acephate can be used on small hot-spots where some migration is expected and on third-instar to adult grasshoppers, provided that most females are not ready to lay eggs.

More is known about the efficacy of lower doses of acephate against grasshoppers than that of low-dose malathion or carbaryl. In some cases, such knowledge may allow greater flexibility in selecting lower dosages to fulfill economic considerations.

Duration of Control

When landowners or managers consider directly investing money to control grasshoppers on rangeland, one of the major questions is how long control will last following treatment. The question would not apply if large-scale outbreaks lasted for only 1 year, but they often last several years. The main question of control duration may be further divided into four basic questions:

1. What are the chances that grasshopper populations will remain as high or go higher next year?

2. If control measures are not applied and grasshoppers remain high, how long are they likely to stay high?

3. If control is used during an outbreak, how long are the benefits likely to continue?

4. What are some things that can jeopardize the length of control expected?

The answers to these questions vary with where you live and where your acreage is in the outbreak cycle. In the past, ranchers with rangeland prone to grasshopper infestations had to base decisions on intuition and experience. Now, particularly with the development of the Grasshopper Integrated Pest Management (GHIPM) Project, quantifiable data are available to provide a more precise decisionmaking process.

Kemp (1987) and Lockwood and Kemp (1987) and Lockwood et al. (1988) have published information on questions 1 and 2 for some counties of Montana and Wyoming. Their data are important. They found that the likelihood of grasshopper populations staying high or increasing from 1 year to the next is only about 56 percent in Garfield County, MT, but 96 percent in Johnson County, WY. In the absence of control, high populations are likely to stay high for 2.25 years in Gallatin County, MT, but up to 23 years in Sheridan County, WY.

Blickenstaff et al. (1974) and Pfadt and Hardy (1987) provided important clues to “best case scenario” answers to the question of control duration. In a study of the time interval between treatment and required retreatment of 1,200,000 acres of Wyoming rangeland, Blickenstaff’s team reported an average retreatment rate of 3.8 percent per year. In other words, about 96 percent of the treated area probably enjoyed benefits for only 1 year, 92 percent for 2 years, and 81 percent likely received some benefits for at least 5 years. Similarly, Pfadt and Hardy (1987) reported at least partial protection of treated range for 3 to 6 years after treatment.

The above reports establish beyond doubt that the concept of multiple-year benefits is valid in some large cooperative programs conducted by State and Federal personnel. Such benefits are not guaranteed. Blickenstaff et al. (1974) reported six mechanisms that can negate, in total or part, the potential for future benefits:

1. Reinvasion by flight. This occurrence is a distinct possibility for highly mobile species like *Melanoplus sanguinipes,* which is a major component of infestation in some areas, like Arizona (Nerney 1960) or eastern Montana (Kemp 1992). However, in other areas, such as Platte and Goshen counties in Wyoming, *M. sanguinipes* comprised less than 5 percent of infestations that were suppressed for 3 to 6 years by treatments (Pfadt 1977).

2. Natural declines in untreated populations. The probability of this event is 100 percent minus the chances that infestation will stay the same or go up.

3. Occurrence of 2-year life cycles at high altitudes.

4. Extended hatching periods (note that this would be aggravated by poor timing of treatment or improper selection of a short-lived chemical when persistence is required).

5. Ability of survivors to increase rapidly (note that this would be aggravated by low levels of control).

6. Failure to treat infested areas in their entirety (note that APHIS prefers to treat entire infestations and has special provisions to allow such treatment).

In any one particular case, protection beyond the year of treatment depends on where in the outbreak cycle (buildup or decline) the program is conducted. If control tactics are not initiated until the populations are on the decrease, then protection is limited to the year of treatment because the population would be of no concern the next year (smaller or negligible population because of the continuing decrease). However, many large-scale treatments occur during the early or middle years of an outbreak. In these cases, multiple years of protection are expected and usually realized.

Conclusions

Traditionally, the use of chemical sprays against grasshoppers on rangeland has been that of a corrective tool. Sprays were used against grasshoppers in outbreak crisis situations as a last resort where the objective was to control the greatest number of grasshoppers.

With the development of the integrated pest management approach and the emerging technologies resulting from the GHIPM Project, chemical sprays are positioned for an expanded role in controlling grasshoppers. This new role will be preventive as well as corrective. Grasshopper treatments should be considered while populations are building. The historical mindset was one where managers waited for the pests to reach outbreak numbers before anything was done. In the future, the use of chemical sprays will be integrated with other strategies, such as managed livestock grazing and treatment of hot-spots for reducing damaging and outbreak-threatening populations of grasshoppers.

While enjoying an expanded role, the traditional use of sprays in emergencies probably never will be eliminated. Chemical sprays are but one weapon in the fight against grasshoppers, and pesticides will remain as an excellent insurance against damaging populations that require immediate attention in the form of fast-acting chemical control.

Selected References

Blickenstaff, C. C.; Skoog, F. E.; Daum, R. J. 1974. Long-term control of grasshoppers. Journal of Economic Entomology 67: 268–274.

Foster, R. N.; Henderson, J. A.; Huddleston, E. W.; Bullard, R. G. 1981. Effect of Triton X-190 and water on malathion and carbaryl related rangeland grasshopper mortalities, 1978. Insecticide and Acaricide Tests 6: 136.

Foster, R. N.; Reuter, K. C.; Kinzer, J. G.; Fischer, J. M. 1984. Manner of pickup of acephate by *Melanoplus occidentalis* (Orthoptera: Acrididae) from aerially sprayed rangeland. Canadian Entomologist 116: 1 149–1151.

Foster, R. N.; Reuter, K. C.; Gould, J. M.; Enis, P. J. 1983. Field experiments on the toxicity of acephate for control of grasshoppers (Orthoptera: Acrididae) on rangeland. Canadian Entomologist 115: 1163–1168.

Kemp, W. P. 1987. Probability of outbreak for rangeland grasshoppers (Orthoptera: Acrididae) in Montana: application of Markovian principles. Journal of Economic Entomology 80: 1100–1105.

Kemp, W. P. 1992. Temporal variation in rangeland grasshopper (Orthoptera: Acrididae) communities in the steppe region of Montana, USA. Canadian Entomologist 124: 437-450.

Lockwood, J. A.; Kemp, W. P. 1987. Probabilities of rangeland grasshopper outbreaks in Wyoming counties. Bull. B-896: Laramie, WY: University of Wyoming and Wyoming Agricultural Experiment Station. 17 p.

Lockwood, J. A.; Kemp, W. P.; Onsager, J. A. 1988. Long-term, large-scale effects of insecticidal control on rangeland grasshopper populations (Orthoptera: Acrididae). Journal of Economic Entomology 81: 1258–1264.

Lloyd, J. E.; Pfadt, R. E.; Ali, M. 1974. Manner of pickup of ULV carbaryl by *Aulocara elliotti* from aerially sprayed rangeland. Journal of Economic Entomology 67: 596–597.

Maas, W. 1971. ULV application and formulation techniques. Amsterdam, Netherlands: N. V. Phillips-Duphar, Crop Protection Division. 164 p.

Nerney, N. J. 1960. Grasshopper damage on short-grass rangeland at the San Carlos Apache Indian Reservation, Arizona. Journal of Economic Entomology 53: 640–646.

Parker, J. R. 1952. Grasshoppers. In: Insects: the Yearbook of Agriculture, 1952. Washington, DC: U.S. Department of Agriculture: 595–604.

Pfadt, R. E. 1977. Some aspects of the ecology of grasshopper populations inhabiting the short grass plains. Tech. Bull. 310: St. Paul, MN: Minnesota Agricultural Experiment Station: 73–79.

Pfadt, R. E.; Hardy, D. M. 1987. A historic look at rangeland grasshoppers and the value of grasshopper control programs. In: Capinera, J. L., ed. Integrated pest management on rangeland: a shortgrass prairie perspective. Boulder, CO, and London: Westview Press 183–195.

Pfadt, R. E.; Lloyd, J. E.; Ali, M.; Sharafi, G. 1970. Manner of pickup of ULV malathion by grasshoppers from aerially sprayed rangeland. Journal of Economic Entomology 63: 1210–1214.

Thomson, W. T. 1992. Agricultural chemicals: Book I Insecticides. Fresno, CA: Thomson Publications. 301 p.

U.S. Department of Agriculture, Agricultural Research Service. 1959. Residues in fatty tissue, brain and milk of cattle from insecticide applied for grasshopper control on rangeland. Journal of Economic Entomology 52: 1206–1210.

U.S. Department of Agriculture, Animal and Plant Health Inspection Service, Plant Protection and Quarantine. 1987. Rangeland Grasshopper Cooperative Management Program. Final Environmental Impact Statement—1987. Washington, DC: U.S. Department of Agriculture, Animal and Plant Health Inspection Service.

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II.5 Success With Reduced Rates of Carbaryl, Malathion, and Acephate Sprays

NOTE: Acephate is no longer approved by EPA for rangeland grasshopper control.

K. Christian Reuter and R. Nelson Foster

Carbaryl, malathion, and acephate have become the chemical insecticide control alternatives in the U.S. Department of Agriculture, Animal and Plant Health Inspection Service's (APHIS) grasshopper cooperative-management programs. Extensive field and laboratory testing of these chemicals over the years have shown that they are very effective in controlling grasshoppers (Skoog et al. 1965; Onsager 1978; Foster et al. 1981 a and b; 1983, 1984, 1985, 1986). Generally, with proper timing of application and acceptable climatic conditions, these treatments will kill at least 90 percent of grasshoppers in the treatment area.

All three chemicals exhibit relatively low toxicity to mammals and have been approved by the Environmental Protection Agency for rangeland grasshopper control. The third factor accounting for the popularity of these three chemicals is their ready availability from suppliers. Often during outbreak situations, and on short notice, there are demands for large quantities of an insecticide to be used anywhere in the Western United States.

Lowering the application rates of these chemicals would be desirable because of reduced costs of the product as well as lessened impact on nontarget organisms. Until viable nonchemical control tools are available for large-scale programs, however, managers of rangeland must take advantage of existing control tools and strive to make them more efficient.

Carbaryl

Current labeling recommends per-acre application rates of carbaryl at 0.375 to 1.0 lb (12–32 fluid oz) active ingredient (AI) in at least 15 oz of spray volume for rangeland grasshopper control. APHIS cooperative programs are restricted to rates of 0.375 to 0.5 lb AI per acre. Sevin 4-Oil® (Rhone-Poulenc) is generally the formulation of choice for rangeland programs at a standard rate of 0.5 lb AI per acre in 20 oz total volume.

In a recent study, Reuter et al. (1993) showed that a 25-percent-reduced rate of an oil formulation of carbaryl was statistically as effective as the standard rate of carbaryl on rangeland grasshoppers. At 1 week after treatment, this reduced formulation had lowered the grasshopper population by 95 percent. At 3 weeks after treatment, mortality remained at 95 percent. In another study (Onsager 1978), a water-diluted formulation of carbaryl at a 50-percent-reduced rate (0.25 lb AI per acre) compared favorably with the standard rate, yielding mortalities of 76 percent at 7 days and 91 percent at 21 days after treatment. There are no data available on the effects of these reduced rates on nontarget organisms, but it is naturally assumed that there would be a reduced impact. Continued control in these studies 1 to 3 weeks after treatment indicate some persistence of the chemical even at a reduced rate. Persistence would be advantageous in controlling additional hatch or migration, especially in early season control efforts.

Malathion

Current labeling recommends per-acre application rates of malathion at 0.58 to 0.87 lb AI (8–12 fluid oz) for rangeland grasshopper control. Criteria in APHIS' cooperative programs restrict treatments to 0.58 lb AI per acre or 8 fluid oz/acre. Several ultralow-volume (ULV) formulations are available and range from 91 to 95 percent active ingredient. In the past, Cythion® ULV was generally the brand name formulation of choice for rangeland programs. At this time, Fyfanon® ULV is the brand name formulation available for programs.

In a study by Foster et al. (1989), results showed that 25- and 50-percent reductions of malathion with an inflight encapsulation material (a polymeric medium) were statistically as effective as the standard rate of malathion on rangeland. At 25 percent less active ingredient, the treatment reduced the grasshopper population 95 percent at 7 days and 92 percent at 21 days. At 50 percent less active ingredient, the treatment reduced the population 92 percent at 7 days and 85 percent at 21 days. Increased persistence of the active ingredient, even at reduced levels, could be economically and environmentally attractive. In a crop protection study by Herbaugh et al. (unpublished data), results with a strip treatment of 4 oz of malathion per acre on rangeland grasshoppers adjacent to cropland showed 74-percent mortality at 2 days after treatment.

Acephate

Current labeling recommends per-acre spray application rates of acephate at 0.094 to 0.125 lb AI in a minimum of 0.5 gal of carrier. APHIS cooperative programs use the minimum of 0.094 lb AI, originally delivered in 1 qt of carrier. Orthene® 75S is the brand-name formulation of choice for rangeland programs and is formulated with Nalcotrol® (an antidrift additive) at 9 fluid oz Nalcotrol per 100 gal of mix plus unsulfured molasses at 3 percent of total volume.

Foster et al. (1979) demonstrated that results from acephate applied at rates 33 and 67 percent below the standard rate were statistically comparable to the standard 12 to 13 days after treatment (78 percent and 60 percent mortality, respectively), although the reduced rates did not produce mortality as consistently among replications as the standard rate. Orthene is generally thought to persist in the field from 7 to 10 days after application. Persistence of Orthene is somewhat less than that of Sevin-4 Oil but greater than that of Cythion, which lasts only for a few days.

Discussion

Large-scale grasshopper outbreaks generally demand immediate attention and significant reductions in a short time. These demands can be met with carbaryl, malathion, or acephate sprays as each can greatly reduce grasshopper populations in a week or less, and each is readily available from suppliers. The same cannot be said for carbaryl bran bait, *Nosema locustae* (a biological control organism), bran bait, or other alternatives in the developmental stages. Carbaryl bran bait is readily available but not particularly effective against high densities of diverse grasshopper assemblages. *Nosema locustae* has never consistently proven effective for grasshopper control, and production capabilities would be a limiting factor for large-scale programs.

Success with reduced rates of these established chemical sprays is both environmentally and economically attractive. Further reductions in treatment rates are certainly attainable with the advent of improved formulations and additives in conjunction with sound applied research. Although reduced rates may yield lower control, the availability of Hopper software (Grasshopper Decision Support System) makes it possible to evaluate each treatment option in accordance with various management scenarios. Lower control percentages may ultimately prove to be acceptable in terms of economic benefits and costs.

References Cited

Foster, R. N.; Henderson, J. A.; Huddleston, E. W.; Bullard, R. G. 1981a. Effect of Triton X-190 and water on malathion and carbaryl related rangeland grasshopper mortalities, 1978. Insecticide and Acaricide Tests 6: 136.

Foster, R. N.; Onsager, J. A.; Reuter, K. C.; Roland, T. 1985. Field evaluation of an aqueous formulation of carbaryl for rangeland grasshopper control, 1983. Insecticide and Acaricide Tests 10: 237.

Foster, R. N.; Reuter, K. C.; Gourd, J. M.; Enis, P. J.; Wooldridge, A. W. 1983. Field experiments on the toxicity of acephate for control of grasshoppers (Orthoptera: Acrididae) on rangeland. Canadian Entomologist 115: 1163–1168.

Foster, R. N.; Reuter, K. C.; Henderson, J. A.; Wooldridge, A. W. 1981b. Orthene with Nalcotrol for control of grasshoppers on rangeland 1979. Insecticide and Acaricide Tests 6: 137.

Foster, R. N.; Reuter, K. C.; Smith, M. E.; Swain, J. L.; Dyer, R. 1984. Comparison of acephate formulations for control of adult grasshoppers on rangeland, 1983. Insecticide and Acaricide Tests 9: 288.

Foster, R. N.; Reuter, K. C.; Walgenbach, D. D.; Bohls, R. A.; Swain, J. L.; Roland, T. J. 1989. Field comparison for rangeland grasshopper control, 1986. Insecticide and Acaricide Tests 14: 265–266.

Foster, R. N.; Staten, R. T.; Reuter, K. C.; Henderson, J. A. 1979. Low rates of Orthene for control of grasshoppers on rangeland, 1979. Insecticide and Acaricide Tests 6: 137.

Onsager, J. A. 1978. Efficacy of carbaryl applied to different life stages of rangeland grasshoppers. Journal of Economic Entomology 71: 269-273.

Reuter, K. C.; Foster, R. N.; Jech, L. E.; Walgenbach, D. D.; Walgenbach, D. R.; Roland, T. J. 1993. Field evaluation of a reduced rate of carbaryl spray on rangeland grasshoppers (Orthoptera: Acrididae). Journal of the Kansas Entomology Society 66(2): 231–236.

Skoog, F. E.; Cowen, F. T.; Messenger, K. 1965. Ultra-low-volume aerial spraying of dieldrin and malathion for rangeland grasshopper control. Journal of Economic Entomology 58: 559–565.

References Cited—Unpublished

Foster, R. N.; Reuter, K. C.; Staten, R. T.; Swain, J. L.; Roland, T. J.; Walgenbach, D. D.; Bohls, R. A.; Villaveces, A. 1989. Field evaluation of controlled released malathion against grasshopper populations (Orthoptera: Acrididae) in South Dakota, 1986. In: Pink Bollworm and Range Pests Station, 1989 annual report, Phoenix, AZ: U.S. Department of Agriculture Animal and Plant Health Inspection Service.

Herbaugh, L. L.; Foster, R. N.; Reuter, K. C.; Roland, T. J.; Bennett, D. 1986. Evaluation of selected liquid sprays and baits to control migration of grasshoppers from rangeland to adjacent cropland (unpublished report).

II.6 Using Hopper To Adapt Treatments and Costs to Needs and Resources

John Larsen and R. Nelson Foster

NOTE: Acephate is no longer approved by EPA for rangeland grasshopper control.

Total treatment cost may be the most critical factor in determining whether grasshopper control on rangeland is feasible, especially because profits from grazing lands are usually much lower than profits from croplands on a per-acre basis. The simplest ways to reduce treatment costs are to use less insecticide or to treat less land. Both solutions require the land manager to accept reduced grasshopper control compared to the level of mortality achieved through traditional control methods. However, reduced grasshopper mortality as a result of less vigorous treatment may be practical when the treatment produces a favorable benefit–cost ratio, adequate forage production, and an acceptable reduction in the number of grasshopper eggs produced by the survivors of the treatment.

Hopper is a recently developed computer-based decision support tool that allows users to conduct sophisticated, precise, and repeatable economic analyses of proposed treatment actions. In the treatment decisionmaking process, Hopper can help users choose from among a greater number of options by analyzing a range of reduced treatments.

There are two techniques for reducing total treatment expenses—interval swath spacing and direct dosage reduction. These techniques can be used separately or jointly in adapting grasshopper control treatments to individual financial resources and circumstances. When these techniques are used, the traditional goal of controlling the maximum number of grasshoppers no longer applies.

Interval Swath Spacing

This technique leaves, by design, an untreated strip of infested land (interval) of predetermined width between treated swaths. The technique has a high potential for reducing costs. Both the cost of the insecticide and the cost of application are reduced because less acreage is treated.

The potential savings of this technique become apparent when its costs are compared to costs of traditional control techniques on a fixed size of rangeland. For example, if the pesticide used costs $2/acre and application of the pesticide costs $2/acre, on a 10,000-acre block of rangeland with traditional control techniques, the total treatment costs would be $40,000 (table II.6–1).

Using interval swath spacing on the same 10,000-acre block and leaving 20 percent of the block (2,000 acres) untreated in narrow intervals between the treated swaths reduces treatment costs to $32,000 (table II.6–1).

Table II.6–1—Costs to treat a 10,000-acre block of rangeland when minimum grasshopper control is the goal and when interval swath spacing and direct dosage-reduction techniques are employed. Costs in this table are for example purposes only.

	Pesticide costs	Application costs[1]	Total treatment cost
	$/acre	*$/acre*	
Traditional technique			
All 10,000 acres treated with conventional pesticide dosage	$2 ($20,000 +	$2 $20,000) =	$40,000
Interval swath technique			
20% of the 10,000 acres left untreated; conventional pesticide dosage used	$2 ($16,000 +	$2 $16,000) =	$32,000
Reduced dosage technique			
All 10,000 acres treated with a 25% reduction in pesticide applied	$1.50 ($15,000 +	$2 $20,000) =	$35,000
Combined technique			
20% of the 10,000 acres left untreated; 25% less pesticide applied to the 8,000 treated acres	$1.50 ($12,000 +	$2 $16,000) =	$28,000

[1] Figures in this column include $0.30/acre for costs associated with typical aerial spray applications (travel, pay, vehicles, flagging, etc.).

Direct Dosage Reduction

This technique simply uses less pesticide per treated acre. For example, on the same 10,000-acre block of rangeland, the pesticide cost of $2/acre for the traditional program results in a total pesticide cost of $20,000. With a direct dosage reduction of 25 percent, the total pesticide cost is $15,000 (75 percent × $2/acre × 10,000 acres). With both traditional and direct-dosage-reduction techniques, the application costs are identical—$20,000. Total treatment costs are $40,000 for a traditional program and $35,000 for a direct-dosage-reduction program.

Combining Techniques

Both of the techniques discussed above demonstrate substantial savings compared to a traditional program. But, by using both techniques jointly, further treatment cost savings can be realized. For example, on the same 10,000 acres, let's assume that both a 25-percent reduction in direct dosage is used and that 20 percent of the block is left untreated in narrow intervals between treated swaths. For example, a pesticide that is traditionally used at 8 fluid oz/acre is used at 6 fluid oz/acre (a 25-percent reduction). Table II.6–1 illustrates these additional savings of treatment costs when compared to traditional treatment.

This example of using interval swath spacing and reduced pesticide together results in a total cost of $28,000 for the treatment. Additionally, there is a 40-percent reduction in pesticide applied on the 10,000-acre block. (For example, in a traditional program, 10,000 acres × 8 fluid oz/acre = 80,000 total fluid oz and combined techniques 8,000 acres × 6 fluid oz/acre = 48,000 total fluid oz.)

Cost reductions on this scale could be highly significant in deciding whether or not pesticide treatment is economically feasible in a given situation. By keeping costs low, land owners and managers can make grasshopper control more affordable on rangelands.

Comparison of Typical Traditional and Combined-Techniques Programs

The following list illustrates a typical cooperative grasshopper management program for the early 1990's when maximum control of grasshoppers is the goal and malathion is the insecticide chosen.

10,000 acres	
Pesticide cost	$1/acre
Application costs	$1/acre
Associated costs (travel, pay, vehicles, flagging, etc.)	$0.30/acre
Total treatment cost	**$2.30/acre**
($23,000 for a 10,000-acre block)	

In an example of a combined program of interval swath spacing and direct dosage reduction, a 20-percent interval swath is used (20 percent of the block is left untreated in narrow intervals between treated swaths). In addition, the per-acre amount of pesticide applied is reduced by 25 percent. This example reduces the overall cost per acre within the 10,000-acre block by 30 percent and the pesticide applied by 40 percent (table II.6–1).

Managers could implement this example by directing the pilot of a spray aircraft who normally flies a 100-ft swath to space the swaths at 120 ft with the 100-ft calibration. This gives a 20-ft untreated interval between treated swaths. A 25-percent reduction in pesticide applied per acre could be achieved by lowering the dosage rate from 8 to 6 fluid oz/acre.

The following two examples compare data from two different Hopper test runs. Example A is for current grasshopper treatments used on the U.S. Department of Agriculture, Animal and Plant Health Inspection Service, Plant Protection and Quarantine-administered cooperative grasshopper management program. Example B is for the same scenario but with a 20-percent interval-swath-spaced treatment and a 25-percent reduction in pesticide applied per acre treated (combined interval swath spacing and direct dosage reduction).

The Hopper test run data show yield in pounds per acre, total cost of treatment, return (dollar value saved by treatment), benefit–cost ratio (B/C) (returns divided by cost), and grasshopper eggs per square yard. You can calculate the net return by subtracting cost from return. In most cases, net returns will also be important to your decision. Keep in mind that these are only example test runs. Each real-world situation is different. You will need to do several test runs on Hopper to get an idea of the appropriateness of reduced treatments for any given situation. Notice that the mortality values entered are different among these examples. This difference is important as the expected mortality value you enter when using Hopper has a large impact on the analysis. As a rule of thumb, if you use interval swathing, the expected level of mortality in the intervals left untreated is conservatively set at zero.

Example A

The following is a list of parameter definitions and values as currently seen on the Hopper 4.0 screen on a computer:

```
Weather at time of treatment                         hot and dry
Survey Date                                             06/22/93
Treatment Date                                          06/30/93
Environmentally sensitive (no chemicals)          Isolated Areas
Managed Bees in the area                                      No
Protect beneficial insects                                    No
Average stage at survey                                     3.06
Average stage at treatment                                  3.67
Percent early season target species                        40.00
Closed canopy                                                 No
Egg hatch completed                            greater than 90%
Grasshoppers density is greater than 22/yd²                  Yes
Weed biocontrol insectaries present                           No
```

The following is a list of economic definitions and values you would find on one of the Hopper screens:

Forage and Grasshopper Models
Sheridan Historical Levels of Trt

```
GRASS FEEDING HOPPERS (#/yd²)                                 15
MIXED FORAGE FEEDING HOPPERS (#/yd²)                          20
PEAK EDIBLE FORAGE PRODUCTION                                550
FORAGE PROD. MULTIPLIER                                     1.00
% Warm Season Grass                                           40
% Cool Season Grass                                           40
% Forbs                                                       20
Normal Soil Moisture (% by Wt.)                               23
Inches of Rain to fill dry soil to field capacity              5
Soil Water Holding Capacity (% by Wt)                         25
Days for saturated soil to dry to 10% Water                   65
```

```
                                TREATMENT COSTS

             Treatment                Cost         Mortality %
             Acephate                $2.30              91
             Carbaryl Bait           $4.50              73
             Carbaryl Spray          $3.50              92
             Malathion               $2.30              90
             Nosema Bait             $4.75              —

Survey date: 06/22/93 Stage: 3.1, Treatment date: 06/30/93 Stage: 3.7.  Yield
Without Treatment:  449 #/acre. Acres to be treated: 16044.  Eggs per sq yd
without treatment: 29.8

Treatment       Yield      Cost       Return          B/C Ratio            Eggs
               (lbs/a)      ($)        ($)       Current  + 2 Years      per yd²
Acephate         533      36900      44848        1.22       3.27          1.8
Carbaryl Bait    514      72196      35310        0.49       1.32          8.2
Carbaryl Spray   524      56153      40196        0.72       1.93          2.8
Malathion        534      36900      45072        1.22       3.29          1.8
Nosema Bait      480      76207      16895        0.22       0.60         13.3
```

Example B

The following is a list of parameter definitions and values as currently seen on the Hopper 4.0 screen on a computer:

```
Weather at time of treatment                              hot and dry
Survey Date                                                  06/22/93
Treatment Date                                               06/30/93
Environmentally sensitive (no chemicals)               Isolated Areas
Managed Bees in the area                                           No
Protect beneficial insects                                         No
Average stage at survey                                          3.06
Average stage at treatment                                       3.67
Percent early season target species                             40.00
Closed canopy                                                      No
Egg hatch completed                                 greater than 90%
Grasshopper density is greater than 22/yd²                        Yes
Weed biocontrol insectaries present                                No
```

The following is a list of economic definitions and values you would find on one of the Hopper screens.

Forage and Grasshopper Models
Sheridan Historical Levels of Trt

```
GRASS FEEDING HOPPERS (#/yd²)                                  15
MIXED FORAGE FEEDING HOPPERS (#/yd²)                           20
PEAK EDIBLE FORAGE PRODUCTION                                 550
FORAGE PROD. MULTIPLIER                                      1.00
% Warm Season Grass                                            40
% Cool Season Grass                                            40
% Forbs                                                        20
Normal Soil Moisture (% by Wt.)                                23
inches of Rain to fill dry soil to field capacity               5
Soil Water Holding Capacity (% by Wt)                          25
Days for saturated soil to dry to 10% Water                    65
```

```
                        TREATMENT COSTS

     Treatment                 Cost         Mortality %
     Acephate                 $1.61             73
     Carbaryl Bait            $4.50             73
     Carbaryl Spray           $2.45             75
     Malathion                $1.61             72
     Nosema Bait              $4.75             —
```

```
Survey date: 06/22/93 Stage: 3.1, Treatment date: 06/30/93 Stage: 3.7.  Yield
Without Treatment:  449 #/acre. Acres to be treated: 16044.  Eggs per sq yd
without treatment: 29.8
```

```
Treatment       Yield      Cost       Return         B/C Ratio           Eggs
               (lbs/a)     ($)         ($)       Current  + 2 Years     per yd²
Acephate         517      25830       36696        1.42       3.82         6.3
Carbaryl Bait    514      72196       35310        0.49       1.32         8.2
Carbaryl Spray   496      39307       25122        0.64       1.72        10.5
Malathion        516      25830       35938        1.39       3.74         7.0
Nosema Bait      480      76207       16895        0.22       0.60        13.3
```

Decisions and Conservation Practices

Another practical aspect of these reduced treatment strategies may be the conservation of nontarget organisms. In pest management, conservation techniques are practices that conserve nontarget organisms. Conservation techniques, such as treatments with reduced active ingredient and interval swath spacing, may significantly reduce the pesticide exposure of nontarget insects.

Natural enemies of grasshoppers, such as parasites and predators, may be affected to a lesser degree when conservation practices are employed. Interval swath spacing could be employed within treated areas to create refuges that may provide significant protection for naturally occurring and released biological control agents. These conservation practices may provide useful grasshopper integrated pest management options in areas where the presence of biological control agents is important to pesticide use decisions. These practices may become more important in the future as biological control of rangeland weeds is implemented on a wider scale in rangeland areas where grasshopper management is also a problem.

You should consider reduced treatment options when some level of reduced grasshopper control can be accepted and for conservation and/or economic purposes. To enter useful data into Hopper, users need to have a good understanding of how these reduced treatment techniques affect both treatment cost and expected mortality. Reduced treatment options provide an opportunity to adapt treatment programs to resources and site-specific circumstances. The models in Hopper produce much of the information needed in such decisionmaking.

Considerations

While reducing the amount of pesticide used to control grasshopper pests is extremely attractive, use caution when deciding to leave a significant portion of the pest population. In geographic locations where grasshoppers seldom or never cause problems 2 or more years in a row, or during times when the overall trends for the general area indicate grasshopper populations to be in decline, such a strategy could be used with minimal risk. In these cases, grasshoppers remaining after reduced treatments pose little chance of propagating a problem for the next season, and single-year economic analysis can be used to support significantly reducing pesticide use.

In locations where grasshopper populations historically cause damage over several years, or in years when general grasshopper populations show no indication of quickly declining on their own, the potential risk associated with a reduced-pesticide strategy should be carefully considered. The risk is one of leaving enough grasshoppers to propagate populations of damaging levels that could require treatment the next year. The argument for leaving some grasshoppers may be supported by a favorable benefit–cost analysis for the season of treatment.

If the remaining grasshoppers result in populations that require treatment the next year, the strategy may be seriously questioned. But even if populations the next season reach damaging levels, the benefit–cost ratio could still be favorable in the succeeding year if treatment was again required. However, even though benefit–cost analysis for 2 years in a row may have proven economical, treating the same acreage 2 years in a row, even at reduced pesticide level, would probably be much more expensive than treating one time with a standard rate of pesticide for maximum control in the initial year.

The strategies of interval swath spacing and reduced doses of pesticide offer exciting possibilities and afford numerous advantages if employed under the right conditions. The trick is deciding where and when risking the need for a second-year (next-year) treatment is too high. Attention to the history of the area and knowledge of current grasshopper population trends will help in making this decision.

II.7 Factors Affecting Application and Chemical Deposition

Robert Sanderson and Ellis Huddleston

Control of spray deposition is vital if pesticides are to be delivered safely and effectively to the intended target. Numerous studies have shown that drift (off-target movement of material) and deposition of pesticides are affected by application equipment, release height, windspeed, air turbulence, air temperature, humidity, and formulation characteristics. It is important for pest managers and applicators to understand the factors that influence the movement of spray droplets on their journey to the target. Drift can become a critical factor when environmentally sensitive areas are in or near spray operations.

Droplet Size

Droplet size is recognized as the major factor in the transport to and the collection of spray by the target. Agricultural sprays contain droplets of varying sizes, but the selection of proper equipment, spray delivery pressure, and nozzle selection play important roles in maintaining a reasonably uniform droplet size. In agricultural sprays, droplets are usually measured in micrometers (µm)—units that are often referred to as microns. Large droplets are influenced primarily by gravity and tend to fall within the target area, whereas small droplets, falling more slowly, are susceptible to wind or turbulence effects and can be moved off target.

A 200-µm droplet would require only 5.4 seconds to fall a distance of 3 m while a 20-µm droplet would take 230 seconds. With only a 1.5-m/second wind, the 20-µm droplet could drift 338 m while the 200-µm droplet would drift only a few meters. Droplets below 100–150µm are generally considered to be the primary driftable portion of the spray. The following table describes droplet characteristics.

Although drift potential may be reduced by increasing the size of droplets, spray coverage on target surfaces may not be as effective at a given volume application rate if most of the liquid volume is contained in very large droplets. Good spray coverage on the target is necessary for efficient insect or weed control. The number of droplets per unit area is a function of droplet size. The relationship between droplet volume and diameter (d) is expressed by the equation

$$\text{Volume} = \Pi d^3/6.$$

Doubling a droplet's diameter will increase its volume by a factor of eight. Therefore a 400-µm droplet has a volume eight times that of a 200-µm droplet. Alternatively, eight 200-µm droplets contain the same volume of spray as a single 400-µm droplet. This formula is an important consideration when determining or assessing deposits on target surfaces.

If thorough coverage is required for pest control, small droplets will be more effective than large droplets, but small ones will be more susceptible to off-target movement by the wind. The droplet size selected for a particular application is often a compromise between coverage with smaller droplets and reduced drift with larger droplets.

Nozzles

Application equipment is very important in determining the droplet sizes contained in the spray. Most agricultural nozzles produce a spray containing a range of droplet sizes, referred to as the droplet size spectrum. The droplet size spectrum is often described by the volume median diameter (vmd or $D_{v0.5}$), which is the droplet size at which one-half of the total spray is in larger droplets and one-half is in droplets smaller than the vmd. A parameter often used to express the range of droplet sizes in the spray is the relative span and is given by the expression $(D_{v0.9}–D_{v0.1})/D_{v0.5}$. Large relative span values indicate wide range of droplet sizes. Typical relative span values for agricultural sprays are in the range 0.8–1.2.

The main types of nozzles used in agriculture are hydraulic, which uses pressure to atomize; gaseous, which uses shear between two fluids; and rotary, which uses centrifugal force. When they are used at practical field application rates, all nozzles produce a range of droplet sizes. Under certain conditions, rotary atomizers can produce a reasonably narrow droplet size spectrum, giving rise to the term "controlled droplet application."

The hydraulic or pressure nozzle is the type most often used in aerial and ground application of pesticides. Droplets are produced by forcing liquid through a small opening, or orifice, under pressure. The size and type of the nozzle tip determine the flow rate and to some extent the droplet size produced. The fan tip produces a flat fan of spray; the disc-core nozzle produces a hollow cone pattern.

In general, a larger nozzle orifice will produce a spray with a larger mean droplet size. Increasing the operating pressure for a given nozzle will increase the flow rate, decrease the mean droplet size, and generally increase the proportion of small droplets. Nozzles on aircraft tend to produce sprays with smaller mean droplet size at similar pressures because of additional shear forces due to the high-speed movement of the aircraft through the air. Increased flying speed or directing the orientation of nozzles forward into the airstream will produce sprays with a smaller droplet size.

As nozzles are used, abrasion and erosion will increase the orifice size and alter the flow rate and droplet size. Nozzles should be checked frequently for calibration and discarded if the flow rate has increased by more than 10 percent.

Examples of rotary atomizers are the Micronair and the Beecomist. The droplet size produced by rotary atomizers is dependent on rotational speed. Higher rotational speeds produce smaller droplets. Rotary nozzles can produce sprays with a smaller mean droplet size than those pressure nozzles can.

Evaporation

Droplets can become smaller as they move toward the target due to evaporation of the spray material. Evaporation, especially in the low-humidity conditions of the Southwest, results in rapid reduction in the size of water droplets. The evaporation rate increases as temperature rises or humidity decreases. At a temperature of 86 °F and relative humidity of 50 percent, a 50-µm droplet of water will completely evaporate in 4 seconds while only falling 15 cm. Spray deposition within the target area can drastically decrease as the temperature increases during the day, an important factor to take into account during a spray operation. Table II.7–2 describes evaporation characteristics.

Evaporation rate is affected by formulation properties as well as air temperature and relative humidity. An oil droplet is less volatile than a water droplet and would not decrease in size so rapidly. Suppliers of a number of spray additives claim their products reduce evaporation. In most cases, these claims lack scientific validation, but the addition of a nonvolatile substance may provide some drift control by preventing the droplet from evaporating to extinction. For example, a 400-µm droplet with 12.5-percent nonvolatile composition would stabilize at 200 µm because of the nonvolatile fraction.

Effects of Formulation Properties

Properties of the pesticide formulation or mixture can influence droplet size. Formulations with low viscosity (thickness) or surface tension generally produce sprays with slightly smaller mean droplet size because less energy is required to break up and atomize the material. Formulations that contain emulsifiers usually have low surface tension and tend to produce sprays with smaller mean droplet size. Also, many of the solvents used in pesticide formulations are highly volatile. Their incorporation into the spray mix can accelerate the decrease in droplet size due to evaporation, and using these volatile additives may increase the drift potential of certain formulations.

Numerous adjuvants (additives) are available for mixing with pesticide sprays as “spray modifiers.” For example, spray thickeners are often added to pesticide sprays in an attempt to reduce the proportion of small, driftable droplets. These adjuvants generally increase the viscosity of the spray mixture, resulting in the production of large droplets; however, studies have shown that adjuvants can also increase the number of very fine droplets. The diverse functions, chemistry, concentrations, and interactions of thickeners, surfactants, and surface active agents make it difficult to predict the effect of these products on droplet size and spray deposition.

Dispersal of Spray

Weather plays an important role in spray dispersal and deposition. Wind displaces spray material, and the distance spray material moves depends on droplet size, the strength of the wind, and the spray release height. Strong winds and higher spray release heights will cause droplets to move a greater distance. Strong winds can cause even large droplets to move off target and become a hazard. Spray operations should be shut down if windspeeds increase excessively. As an example, the U.S. Department of Agriculture's Animal and Plant Health Inspection Service normally stops spraying with ultra-low-volume pesticides when the windspeed reaches 10 miles per hour. Other conditions and State laws may dictate even lower windspeeds.

There is always some downwind displacement of spray droplets, even in light winds. If spray applications are made by moving into the wind, this displacement will move spray back behind the sprayer. If applications are made in a crosswind, the spray will be moved slightly downwind from the sprayer. This occurrence is known as swath displacement and should be taken into account when switching on and off the sprayer. With crosswind swath displacement, multiple spray passes are needed to obtain the desired deposition.

Table II.7–1—Selected characteristics of various size spray droplets of water

Droplet diameter	Terminal velocity	Fall time from 3 m	Drift distance (3-m fall with 5-km/h wind)	Drops/cm^2 from 10 a/ha application
(µm)	*(M/sec)*	*(Sec)*	*(M)*	*(No./cm^2)*
10	0.003	1,020	1,372	190,990
50	0.075	40	54	1,530
100	0.279	11	15	192
200	0.721	5.4	5	24
500	2.139	1.6	2	1.5

Table II.7–2—Evaporation characteristics for water droplets under two environmental conditions

Droplet size	Time to extinction	Fall distance	Time to extinction	Fall distance
(µm)	*(Sec)*	*(M)*	*(Sec)*	*(M)*
50	14	0.5	4	0.15
100	57	8.5	16	24
200	227	136.5	65	39

Air Temperature

In strong winds, frictional turbulence produces mechanical stirring of the air and promotes strong mixing in the atmosphere that tends to lessen the effects caused by any localized temperature differences. In lighter winds, especially where there is intense radiation, temperature can vary significantly with height. Temperature variations are caused by solar radiation and heat exchange between air, soil, and vegetation. The change in temperature with height is called the vertical temperature gradient. The temperature gradient has an important effect on atmospheric stability because it can increase or decrease air mixing. Under normal atmospheric conditions, the air is warmer at ground level and gets cooler with an increase in height due to the decrease in air pressure with height. Under these conditions, the temperature decrease is approximately 1.8 °F for every 100-m height increase. This factor is known as the adiabatic lapse rate.

If the temperature decreases more rapidly, there is a superadiabatic lapse rate, characterized by strong convection currents and turbulence. Under these conditions, the air layer is said to be unstable. High levels of spray drift can occur when a large number of small droplets are caught in the convection currents and fall out of the target zone.

If the temperature change is less than the adiabatic lapse rate, the air layer is considered stable. Under certain conditions, temperature can increase with height. This condition, known as inversion, is extremely stable. Inversions can occur only over a limited height range because there must be an overall drop in temperature with increase in height. Inversions usually occur when the wind is zero or very slight and may develop by the “sinking” of cold, dense air pushed in by weather fronts, or by radiational cooling of the surface, especially on clear nights. Off-target spray drift can occur under these condition because the inhibited mixing permits the formation of a mass or cloud of small droplets that can move great distances with little dispersal.

II.8 Calibration of Aerially Applied Sprays

Billy Tanner and T. J. Roland

Calibration is the process of measuring and adjusting the amount of pesticide your equipment will apply to the target area. Pesticide applicators need to be sure they are using the correct amount of pesticide: Too little can result in inadequate control; too much can result in injury to people, plants, or animals, illegal residues, excess runoff or movement from the target, and lawsuits and fines.

Calibration was a frightening word to most early aerial applicators. Their procedures were to mix, load, and fly. Pilots continually adjusted boom pressure and swath width as they went along to make the pesticide come out right for the acreage. Some areas were overdosed; others were underdosed or completely missed. Advancing technology, education, demands by ranchers and farmers, pesticide laws, and label requirements are forcing the modern-day aerial applicator to be calibration conscious.

An aircraft with a properly calibrated dispersal system reduces the workload of the pilot. He or she has enough to watch from the cockpit without constantly monitoring the amount of chemical remaining in the hopper and adjusting boom pressure to make chemical and acreage come out right.

The manufacturers of various nozzles, atomizers, and spray tips provide calibration formulas and/or procedures to calibrate their equipment properly. The formula used by the Plant Protection and Quarantine unit of the U.S. Department of Agriculture's Animal and Plant Health Inspection Service to calibrate aerial liquid systems is simple and accurate.

Before calibration procedures begin, learn the airspeed, swath width, application rate per acre, spray tip size (output per minute per nozzle), and the flow factor for the chemical being used. With these known factors, you can use the following calibration formulas:

- (Miles per hour × swath width in feet) ÷ 495 (a constant) = **acres per minute**

- (Acres per minute × rate per acre in ounces) ÷ 128 (oz in 1 gal) = **gallons per minute**

- Gallons per minute ÷ nozzle output = **number of nozzles to install using water**

- Number of nozzles for water × chemical flow factor = **number of nozzles to install on the aircraft for the chemical being used.**

A Practical Example of Aerial Spray Calibration

Cessna Ag Truck
Airspeed = 120 miles per hour (mi/h)
Swath width = 100 ft
Pesticide = malathion
Application rate = 8 oz/acre
Nozzle tip size = 8002 flat fan
Nozzle output = 0.2 gal/minute using water at 40 pounds per square inch (lb/in^2)
Correction flow factor for malathion = 1.1

Step 1. Calculate the acres per minute that the aircraft will cover.

(120 mi/hour × 100 ft) ÷ 495 = **24.24 acres/minute**

Step 2. Calculate the number of gallons per minute that the aircraft will put out at the desired rate per acre.

(24.24 acres/minute × 8 oz/acre) ÷ 128 (oz in 1 gal) = **1.52 gal/minute**

Step 3. Calculate the number of nozzles required to apply water at 8 oz/acre and pressure set at 40 lb/in^2.

1.52 gal/minute ÷ 0.2 (output per minute per nozzle) = **7.58 nozzles for water**

Step 4. Calculate the number of nozzles to install correcting for viscosity (flow factor—see table II.8–1 at the end of this chapter) of the chemical being used.

7.58 (nozzles) × 1.1 (flow factor) = **8.3 nozzles**

Step 5. Round to the nearest whole number.

8.3 rounded down to **8 nozzles to install on the aircraft.**

Step 6. Conduct a calibration run either static (run the system on the ground and collect discharge from each nozzle into containers to determine the actual output per minute) or fill the spray tank to a known reference mark and fly the aircraft for 1 min. Refill the tank to the known reference mark and determine the amount used. If the output was light or heavy, make small adjustments to the pounds-per-square-inch setting to achieve the correct output per minute. The final calibration check should be accomplished during actual application with a small load. The following information and flow factor table will help calibration for most sprays and aircraft.

Useful Information and Calculations

128 oz/gal ÷ rate per acre (ounces) = acres/gal
128 oz ÷ 8 oz = 16 acres/gal
128 oz ÷ 12 oz = 10.67 acres/gal
128 oz ÷ 16 oz = 8 acres/gal
128 oz ÷ 20 oz = 6.4 acres/gal
128 oz ÷ 32 oz = 4 acres/gal
128 oz ÷ 40 oz = 3.2 acres/gal
128 oz ÷ 96 oz = 1.33 acres/gal

- Total program acres ÷ acres per gallon = total gallons required

- Airspeed (mi/hour) × swath width in feet ÷ 495 (a constant) = acres per minute

- Acres per minute ÷ acres per gallon = gallons per minute

- Gallons per load ÷ gallons per minute = dispersal time per load

- Gallons dispersed ÷ acres covered × 128 = rate per acre in ounces

- Swath width in feet ÷ 8.25 = acres per mile

- Acres per mile ÷ acres per gallon = gallons per mile

- Gallons per mile × swath length in miles = gallons per swath

- Aircraft load in gallons ÷ gallons per swath = number of swaths per load

To convert knots to miles and miles to knots, multiply
Knots × 1.15 (a constant) = mi/hour
Example: 160 knots × 1.15 = 184 mi/hour
mi/hour × 0.868976 (a constant) = knots
Example: 135 mi/hour × 0.868976 = 117 knots

- 1 mi^2 = 640 acres
- 1 acre = 43,560 ft^2 = 0.405 hectare (ha)
- 1 ha = 2.471 acres
- 1 gal/acre = 9.35 L/ha
- 1 gal = 128 fluid oz = 8 pints = 4 quarts
- 1 gal = 3.785 L = 3,785 Ml
- 1 mi = 5,280 ft = 1,610 m = 1.61 km

Table II.8–1—Flow factor table for spraying solutions other than water

Weight of solution	Specific gravity	Conversion factors
(lb/gal)		
7.0	0.84	0.92
8.0	.96	.98
8.34	1.00	1.00
9.0	1.08	1.04
10.0	1.20	1.09
10.65—28% Nitrogen	1.28	1.12
11.0	1.32	1.14
12.0	1.44	1.20
14.0	1.68	1.29

II.9 Ground Equipment for “Hot-Spot” Treatments With Chemical Sprays

Ellis Huddleston, Robert Sanderson, and James Ross

Aerial application of ultra-low-volume (ULV) malathion at 8 oz/acre has proven to be a very successful method of controlling grasshoppers in the United States and other parts of the world. Using aircraft is the most efficient way to treat large infestations.

In the integrated pest management (IPM) mode, program managers often strive to reduce grasshopper numbers on small areas to lessen the chances of spread of the infestation or to protect valuable forage and crops. In much of the Western United States, aircraft simply are not available or are far too expensive to treat small infestations (up to 1,000 acres). Ground application or no control are the only options. Conventional row-crop sprayers with booms are not sturdy enough for treating rangeland and are not adapted to volumes in the ULV range for malathion.

In an IPM program to control range caterpillar in New Mexico, (a wind-assisted dispersal system for “hot-spot” treatment with ground equipment was successfully developed. This approach is used on thousands of acres each year. New Mexico State University has adapted this approach to rangeland grasshopper control and also found it to be very successful for black grassbug control in New Mexico.

Equipment

We conducted experiments in western New Mexico in late May–early June 1986, on predominantly blue gramma grass rangeland. The principal grasshoppers were *Aulocara elliotti* (bigheaded grasshopper) and *Melanoplus sanguinipes* (migratory grasshopper), and most were adults at the time of spraying. The experiments included a completely random design with a minimum of five replicates per treatment. Square 40-acre plots were treated using a swath spacing of 100 ft.

A mist blower (Model MM55-S, Automatic Equipment Mfg. Co., Pender, NE) was mounted in a trailer pulled behind a half-ton pickup truck. A motorized backpack mist blower (Solo Port 423, Solo Inc., Newport News, VA) was mounted in the back of the truck. The truck was driven at 10 miles per hour (mi/hour) perpendicular to the prevailing wind with both sprayers calibrated to deliver 8 oz/acre of ULV malathion. Grasshopper density was checked 1 day prior to treatment and 1 day after treatment. We counted densities in 40 0.1-m^2 rings in a circle 165 ft in diameter in the center of each plot. Mortality was estimated from pre- and posttreatment counts.

Control

The MM55-S mist blower provided excellent control when used in windspeeds of 4 to 20 mi/hour. For six replications of the test, the average grasshopper mortality was 93 percent with a range of 87 to 100 percent. Two additional replicates evaluated adverse conditions in which effectiveness was greatly reduced (64 percent compared with 93 percent) when this piece of equipment was used with 100-ft swaths in light and variable winds. The Solo 423 was found to provide 95-percent control (range 91 to 100 percent) when used at windspeeds in excess of 5 mi/hour. The results of a single trial were similar to those for the MM55-S mist blower in light and variable winds.

Using the Equipment in the Field

Results showed that both the MM55-S and the Solo 423 mist blowers delivered ULV malathion at the same volume per acre as aircraft and provided control at least equal to that of malathion delivered from aircraft. Both pieces of equipment were equally effective, and both require a steady, fairly strong wind to be effective.

ULV malathion is available in 5-gal containers at a 1994 cost of about $24/gal (Helena Chemical Co., Terra Int.). At 8 oz/acre, the chemical cost is $1.50/acre. Because no mixing is required, unused material can be stored in the original container and should have a shelf life of at least 2 years if stored properly.

Using a 100-ft swath and 10 mi/hour vehicle speed, mist sprayers can cover 2 acres/min. Counting lost time turning, coverage of 80–100 acres/hour is possible. The MM55-S has a cab-mounted remote control that changes the spray from right to left, so whenever the driver turns, he or she can direct the spray downwind. A device to attach the Solo 423 to the tailgate and ropes and pulleys to change the direction of the spray should be easy to build. One rancher in New Mexico has a mist blower that is similar to the MM55-S but does not have a remote control to switch the spray directions. He simply drives forward on one swath and backs up on the next.

Calibration of a sprayer is simply making sure that the sprayer is delivering the correct amount of spray per acre. For the example used here (100-ft swath and 10 mi/h), the sprayer will cover 2 acres/min.

Here's how that figure was calculated:
10 mi/hour = 52,800 ft/hour ÷ 60 = 880 ft/minute × 100-ft swath = 88,000 ft^2/minute.
88,000 ÷ 43,560 ft^2 in an acre = 2.02 acres/minute.
2 acres/minute × 8 oz/acre = 16 oz/minute = 1 pt/minute.

Solo does sell a ULV attachment for the Solo Port 423. Instead, a metering orifice or flow regulator can be inserted in the plastic line between the tank and the nozzle. These orifices and accessories are available from suppliers of agricultural sprayer parts. The larger mist blowers use a pump and pressure regulator, which may be adequate. If not, use a metering orifice.

ULV malathion flows enough like water that water can be used for the initial calibration. For the Solo, pour 3 gal of water in the tank and make sure the supply hose is full. Run the sprayer for 2 minutes and measure the amount of water left, including that in the supply tube. This calibration normally will use 1 qt. You may need a larger or smaller orifice to get the desired rate. For the mist blowers with pumps, you can use a similar procedure or you can catch the output from the nozzle without the fan blowing. Changing the pressure and/or the metering orifice will change the flow rate. During spraying operations, applicators should check the flow rate of the ULV malathion and make required adjustments.

Mist blowers are an effective way to control grasshoppers on rangeland with ground equipment. We prefer the relatively inexpensive motorized backpack mist blower because of cost and versatility. Users can adapt the blower to all-terrain vehicles, and a mist blower is handy for spraying trees and small gardens.

II.10 Treating Localized Hot-Spots of Rangeland Grasshoppers: A Preventative Strategy With Promise

Jeffrey A. Lockwood, Michael J. Brewer, and Scott P. Schell

The Problem

In most years, and in most locations, most grasshopper species are innocuous or even beneficial to grassland ecosystems, but large-scale outbreaks can inflict serious economic damage to western rangelands. Figure II.10–1 illustrates the duration of grasshopper outbreaks in Wyoming. Some areas show grasshopper activity for up to 20 of the last 50 years. Although the grasshopper population on a broad scale collapsed across the Western United States in 1988–89 and has remained low through 1994, historical records suggest that the population is likely to resurge in this decade (fig. II.10–2).

Current economic conditions and mounting environmental concerns strongly suggest that the massive grasshopper treatment programs of the past 40 years will not be repeated. Therefore, economically viable, environmentally sound alternatives need to be found in the immediate future.

A Solution?

Scientists' understanding of North American rangeland grasshopper outbreaks is in its infancy. According to Alan Berryman's outbreak theory (1987), insect outbreaks take one of two forms, and the form of an outbreak is critical to pest management.

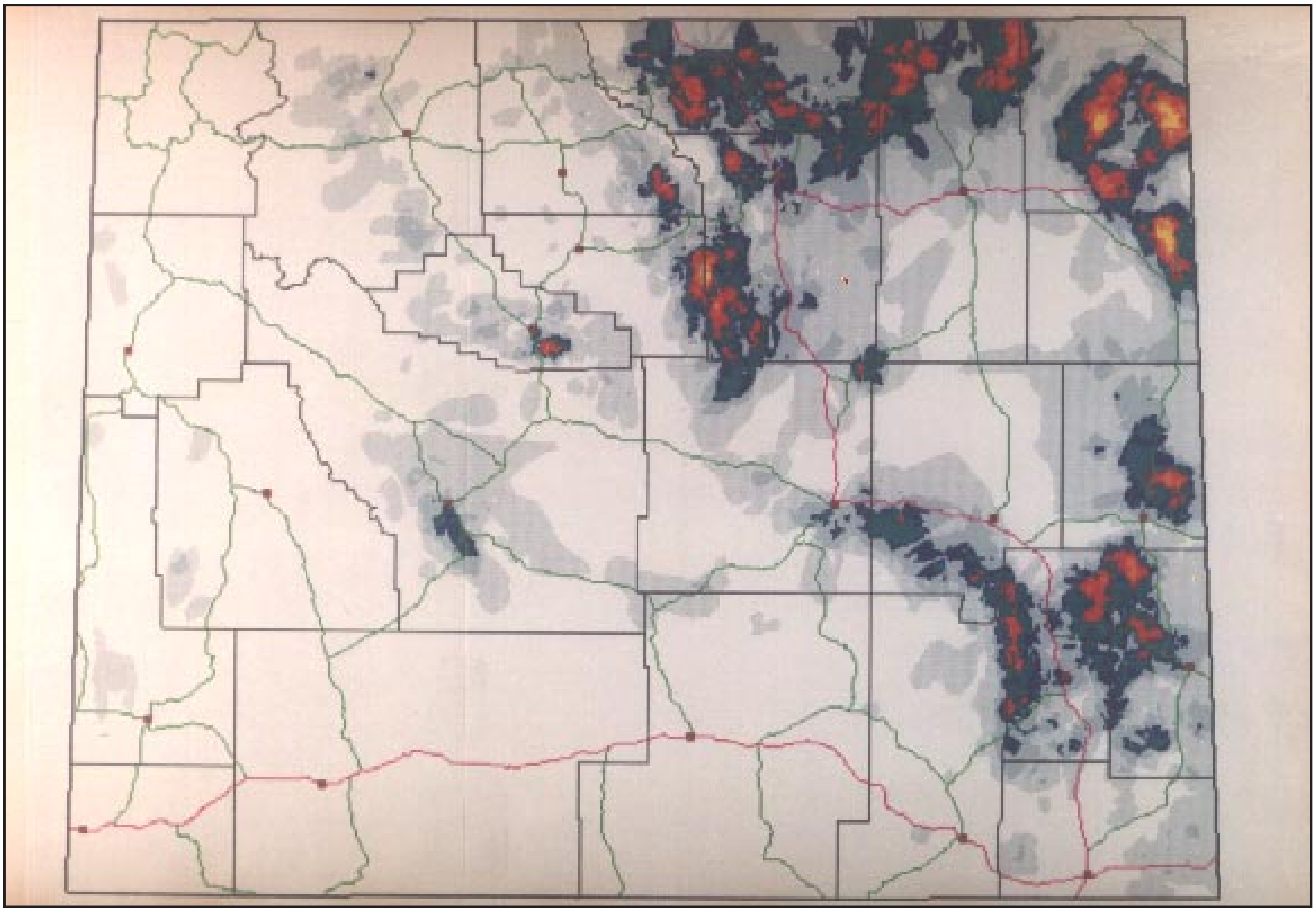

Figure II.10–1—Spatial distribution of rangeland grasshopper outbreaks in Wyoming from 1944 to 1993 (white = no infestations, light gray = 1–2 yr infested, gray = 3–4 yr infested, black = 5–6 yr infested, bluish green = 7–8 yr infested, blue = 9–10 yr infested, red = 11–12 yr infested, orange = 13–14 yr infested, and yellow = 15–20 yr infested). Interstate highways are magenta, and main State roads are yellow-green. County borders are in black, and county seats are brown squares.

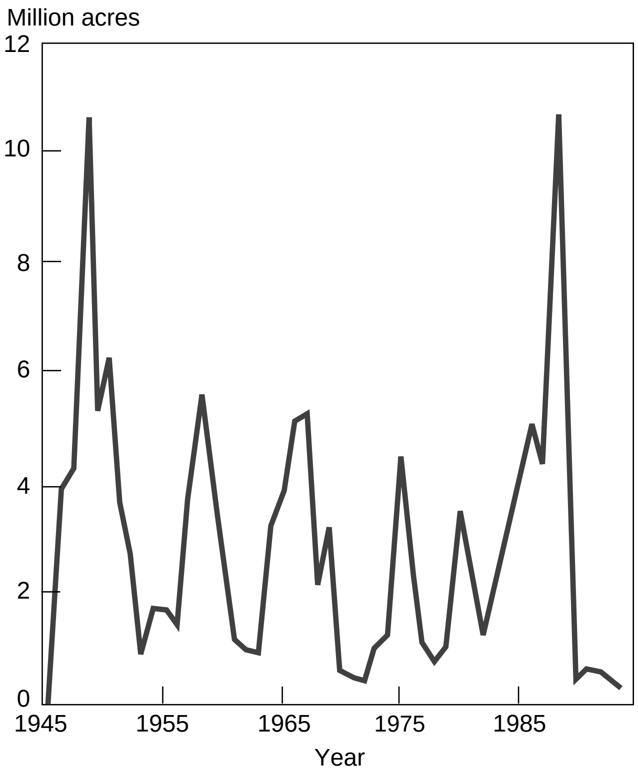

Figure II.10–2—History of rangeland grasshopper outbreaks in Wyoming. Note the erratic pattern of infestation (>8 grasshoppers/yd^2), including the massive outbreak in 1987 and the remarkably low area of infestation since 1989.

The first is the eruptive outbreak, characterized as starting from a "hot-spot" that expands through a self-perpetuating process to encompass increasingly large areas. This type of outbreak occurs with the mountain pine beetle and the gypsy moth. With eruptive dynamics, large-scale outbreaks can be prevented if the hot-spots are controlled. This strategy is analogous to suppressing small fires caused by lightning strikes to prevent large-scale forest fires. The treatment of hot-spots from which outbreaks arise has been an effective tool in the management of several pests of natural and agricultural resources, including African locusts. Indeed, it appears that the extinction of the Rocky Mountain locust was the consequence of agricultural practices having effectively (albeit unwittingly) destroyed through cultivation of soils the highly localized eruptive foci of this species in the 1800's.

The second form of outbreak dynamics is termed "gradient." Gradient outbreaks occur when pest populations fluctuate over broad areas in response to external conditions, without growth from a local hot-spot. This type of outbreak is seen in forest insects, such as many cone and seed insects, some defoliators, and "nonaggressive" bark beetles. If gradient dynamics lie at the heart of grasshopper outbreaks, then little can be done with respect to prevention. By analogy, local, tactical actions will not prevent droughts.

Over the last several years, the hot-spot treatment strategy has been studied in Wyoming through the collaborative efforts of the University of Wyoming and the U.S. Department of Agriculture (USDA), Animal and Plant Health Inspection Service (APHIS), Grasshopper Integrated Pest Management Project (Lockwood and Schell, in press). In the context of traditional APHIS operations, Lockwood and Schell defined a hot-spot as an area of less than 10,000 acres of rangeland infested with at least 8 grasshoppers/yd^2. Although the results of this experiment are not yet definitive, the investigators believe that continuing, long-term studies of grasshopper population dynamics will eventually clarify the process of outbreak formation. At present, there is sufficient information to provide some preliminary insights and recommendations.

Current Knowledge

Evidence for Eruptive Dynamics.—There are four lines of evidence that support the process of an eruptive outbreak dynamic. First, the existence of highly localized infestations is a necessary precursor to an eruptive outbreak. The discovery of numerous hot-spots (table II.10–1, fig. II.10–3), from which larger areas could become colonized, suggests the potential for eruptive dynamics. Although they are a necessary condition for eruptive dynamics, the existence of these hot-spots cannot be considered sufficient evidence of this outbreak form.

Next, the observation that two of the nine hot-spots for which there are data over at least 2 yr sustained or expanded with time demonstrates that these infestations can give rise to larger outbreaks (table II.10–1).

Although only one hot-spot developed into an outbreak, it should be noted that eruptive dynamics do not require that all or most of the hot-spots give rise to large-scale outbreaks. By analogy, very few lightning strikes result in major forest fires.

Third, no continued outbreak was found in the areas around hot-spots treated with insecticides (table II.10–1). If outbreaks were gradient, then treating a localized site should simply result in a “hole” in a larger region of high densities.

Finally, it appears that at least one grasshopper species (the bigheaded grasshopper, *Aulocara elliotti)* has high rates of reproduction at both very low densities and moderately high densities. This “bimodal” reproductive feature is necessary for the self-perpetuating dynamics of an eruptive outbreak.

Evidence for Gradient Dynamics.—The possibility of gradient outbreaks is supported by four lines of evidence. First, two large-scale outbreaks (greater than 15,000 acres) were found that were apparently not preceded by a hot-spot (table II.10–1). One might argue that these areas were simply very large hot-spots, but there was no evidence of continued expansion (there were no topographic or other features limiting expansion in all directions), as would be expected from eruptive dynamics.

Next, seven out of nine documented hot-spots for which at least 2 yr of data exists disappeared the season after their discovery, even without treatment (table II.10–1). This finding suggests that expansion of hot-spots into eruptive outbreaks is not common. But as with forest fires, sometimes it only takes one lightning strike to cause major destruction.

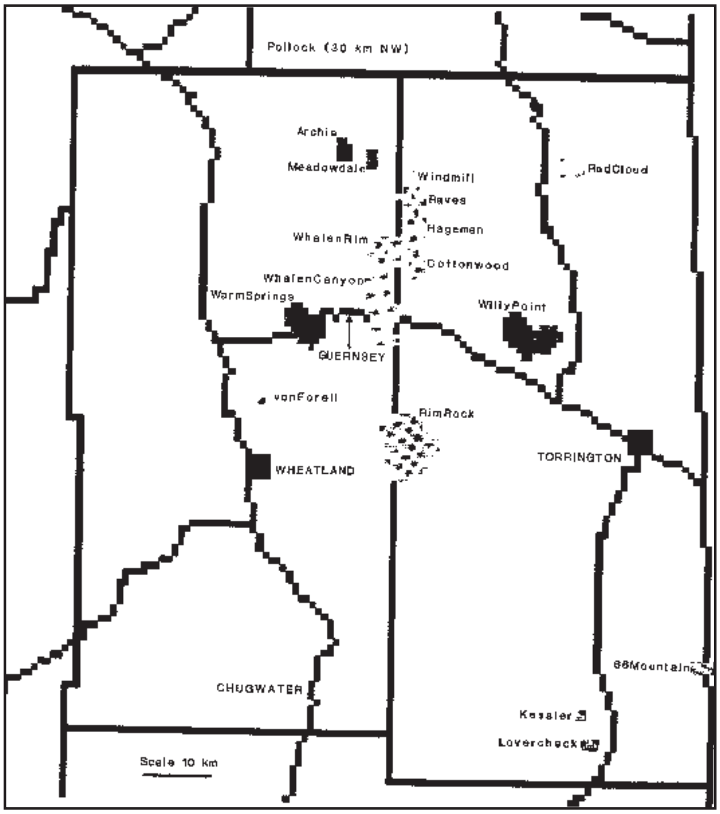

Figure II.10–3—Locations of hot-spots in Platte and Goshen counties in southeastern Wyoming (light shading = 1990, moderate shading = 1991, black shading = 1992). Hot-spots and outbreaks reduced to <10,000 acres are labelled with upper- and lower-case letters; weather stations are labelled in upper-case letters.

Table II.10–1—Dynamics of control (untreated) and treated grasshopper hot-spots and outbreaks in southeastern Wyoming

			Area			
Site	Category	Status	1990	1991	1992	1993
			Acres			
Rave	Hot-spot	Untreated	500	0	0	0
vonForell	Hot-spot	Untreated	500	0	0	0
Red Cloud	Hot-spot	Untreated	1,900	0	0	0
Whalen Canyon	Hot-spot	Untreated	7,920	10,340	1,460	0
Hageman	Hot-spot	Treated	2,140	0	0	0
Pollock	Hot-spot	Treated	2,400	0	0	0
Willy Point	Outbreak	Untreated	38,880	34,080	9,430	4,960
Kessler	Hot-spot	Untreated	0	[1]170	0	0
66 mountain	Hot-spot	Untreated	0	[1]790	0	0
Lovercheck	Hot-spot	Untreated	0	[1]240	0	0
Cottonwood	Hot-spot	Untreated	0	790	0	0
Windmill	Hot-spot	Untreated	0	1,340	1,370	0
Whalen Rim	Hot-spot	Treated	0	1,150	0	0
Rim Rock	Outbreak	Untreated	0	17,760	9,310	[2]0
Archie	Hot-spot	Untreated	0	0	460	0
Warmsprings	Hot-spot	Untreated	0	0	5,380	3,840
Meadowdale	Hot-spot	Treated	0	0	1,030	0
Table Mt.	Outbreak	Untreated	0	0	18,530	2,400
Kincaid Draw	Hot-spot	Untreated	0	0	0	640

[1] Hot-spot collapsed during heavy spring rains in 1991.
[2] Hot-spot collapsed during heavy summer rains in 1993.

Third, the species composition of a hot-spot can change dramatically between years—a discovery that suggests that dominant species may be tracking available resources. For example, a species that prefers needle grasses, *Amphitornus coloradus*, comprised only 2 percent of the hot-spot communities in a dry year (when needle grasses were sparse) but comprised 16 percent in a wet year (when needle grasses were abundant). This resource-tracking phenomenon is consistent with gradient outbreak dynamics.

Finally, most hot-spots have unique soil and topographic properties, compared to adjacent lands. Hot-spots generally occur in foothills with relatively poor soils. Thus, it appears that external factors (rather than a self-perpetuating process) give rise to these localized infestations.

A Hybrid Case?

The evidence regarding the processes that give rise to large-scale outbreaks supports both gradient and eruptive dynamics. This continuing ambiguity calls into question the viability of the current outbreak theory. Unfortunately, the matter becomes more complex as a function of spatial scale.

The scale of resolution used in our study was derived from the management needs of USDA; cooperative programs with APHIS are standardly triggered once a grasshopper outbreak exceeds 10,000 acres. Perhaps the populations examined at finer or coarser resolutions are regulated by different processes and exhibit unique dynamics. Additionally, the rate of change in the density, area, and species composition of an infestation may be related to its size; small infestations may include fewer species and change more rapidly than large outbreaks.

Indeed, such differences in the rates of change may be seen within the size range of hot-spots. For example, small hot-spots may be more susceptible to suppression by mobile predators (a 25-acre infestation of *Camnula pellucida* was eliminated by the immigration and feeding of starlings over a 2-wk period). We found that no hot-spot less than 1,200 acres persisted for more than a single year, and the only hot-spot to increase in size began at 8,000 acres.

As scientists continue to investigate the outbreak dynamics of rangeland grasshoppers, it may be important to consider the possibility that the population dynamics of these insects cannot be effectively classified using the existing theory. This theory was developed based primarily on forest pests, and there are potentially important ecological differences between forest and rangeland pest outbreaks. For example, forest pest outbreaks often involve a single insect species feeding on a single tree species, while rangeland grasshopper outbreaks often involve 10 or more species feeding on dozens of plant species. Given the complexity of rangeland grasshopper communities, it is possible that some species have eruptive potential while others exhibit gradient dynamics.

Management Practices

Although there is uncertainty about the outbreak dynamics of rangeland grasshoppers, some management strategies can be inferred from existing data. Available evidence provides some insights regarding survey strategies, treatment tactics, and programmatic obstacles with respect to a hot-spot management program. However, it should be kept in mind that these inferences are derived from work conducted in southeastern Wyoming from 1990 to 1993, and grasshopper population dynamics may be different in other times and regions.

Hot-Spot Detection

We believe that four approaches may be useful in improving the efficiency of searching for localized hot-spots. First, hot-spots are most likely to occur in areas of historically chronic infestations (figs. II.10–3 and –4). Historical maps of grasshopper outbreaks may provide vital clues as to the areas in which survey efforts should be concentrated. Unfortunately, there does not appear to be a single, consistent outbreak species on which to focus attention. The species composition of hot-spots varies dramatically between sites and years. Slantfaced grasshoppers are the most common species in hot-spots of southeastern Wyoming (especially *Ageneotettix deorum, Amphitornus coloradus, Aulocara elliotti,* and *Cordillacris* spp.). However, we also have found hot-spots dominated by spurthroated and bandwinged species *(Melanoplus sanguinipes* and *Trachyrhachys kiowa,* respectively).

Next, several features of ecosystems and habitats are associated with hot-spots. Hot-spots generally occur in foothills, the areas of transition between mountains and plains. Areas with 8 to 10 in of annual precipitation also appear to be most likely to support hot-spots. At a finer scale, hot-spots are clearly associated with poorer soils.

Within a region, soils with relatively low nitrate, phosphate, and potassium should be considered prime candidates for hot-spots. Low salt levels and high clay content may also be associated with grasshopper hot-spots. There do not appear to be substantial differences in the plant communities inside and outside of hot-spots.

Third, hot-spots apparently develop, persist, and occasionally expand during periods of normal to dry weather and collapse with the onset of wet conditions. These phenomena suggest more intense surveys in years with dry conditions.

Finally, landowners and managers need training to survey for grasshoppers. The exclusive use of federally funded scouts for the intensive surveys required to locate hot-spots over large expanses of land is cost prohibitive. With materials in this handbook, land users can take an active role in pest management, thereby allowing site-specific strategies to be effective. Along with training, systems need to be developed for the coordinated communication of potential hot-spots to APHIS and local pest-management authorities.

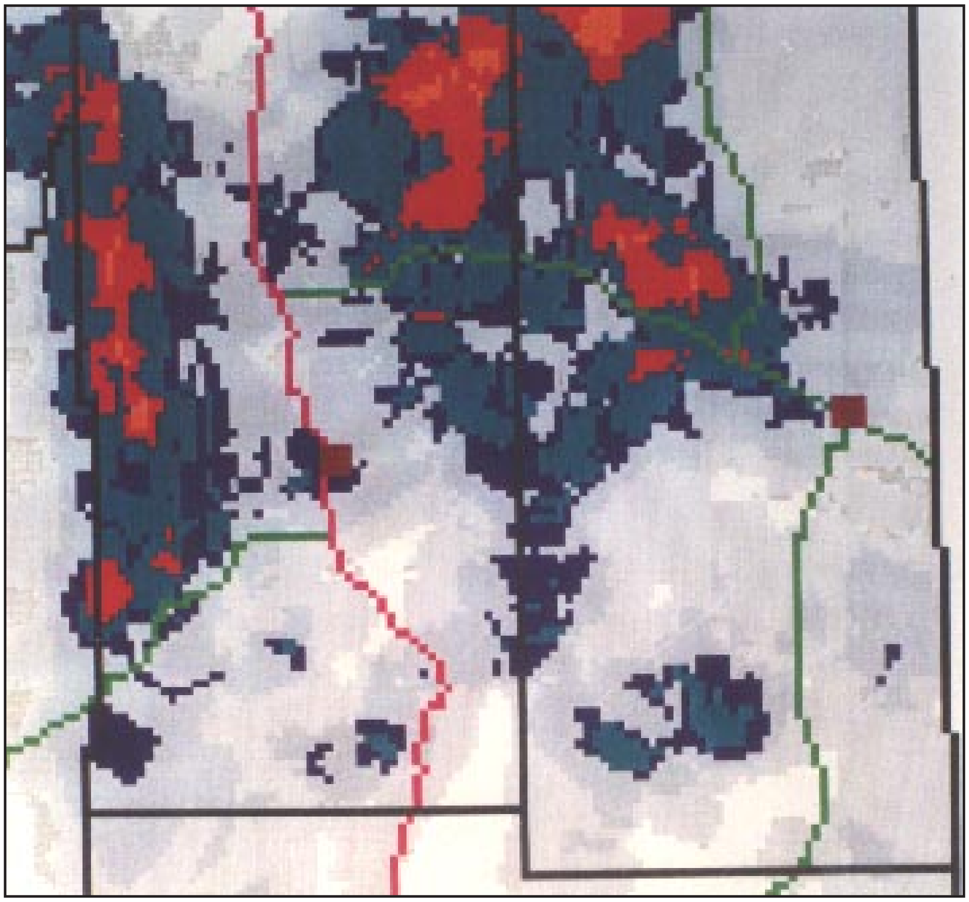

Figure II.10–4—Expanded view of southeastern Wyoming from 1960 through 1993 (Platte and Goshen counties; see figure II.10–1 for spatial reference; white = no infestations, light shading = 1–2 yr infested, dark shading = 3–4 yr infested, purple = 5 yr infested, green = 6–7 yr infested, red = 8–9 yr infested, orange = 10–11 yr infested, and yellow = 12–15 yr infested).

Treatment Strategies

With regard to the tactics of treating hot-spots for the purpose of preventing larger scale infestations, three elements bear consideration. First, it appears that most hot-spots collapse without treatment. In particular, hot-spots of less than 1,000 acres have not been found to persist or expand with time. So these areas should probably not be treated, although it may be prudent to monitor them.

Second, the annual expansion of persistent hot-spots is relatively limited, with a documented maximum of 30 percent, although the rate of expansion could be greater prior to a large-scale outbreak. Given the documented rates and likelihoods of expansion, it would appear that no hot-spot should be treated in the year of discovery. Only if the infestation persists into the subsequent year should treatment be considered.

Finally, the benefits of small-scale insecticide treatments with respect to the preservation of beneficial arthropods may potentially offset the relatively higher costs per acre of hot-spot treatments. With regard to beneficial insects, treating small areas reduces the number of beneficial insects killed by insecticides and increases the recolonization rate. These beneficial organisms may be responsible for the sustained suppression of a hot-spot after treatment. Given that the inadvertent, large-scale suppression of beneficial arthropods through the use of broad-spectrum liquid insecticides has been found to aggravate grasshopper outbreak dynamics in Wyoming (Lockwood et al. 1988), the benefits of small-scale treatments are potentially substantial.

Obstacles to Implementation

The implementation of a hot-spot program is confounded by four obvious obstacles: the Federal cost-share program, the requisite sampling intensity, the "principle of the commons," and the current state of knowledge. Fortunately, all of these problems have potential solutions.

First, the Federal cost-share program discourages preventive practices and local survey efforts and encourages large-scale treatments by triggering APHIS involvement when outbreaks exceed 10,000 acres. For the treatment of hot-spots to become an accepted grasshopper management strategy, the cost-share formula must reward participants in small-scale programs. In its most simple form, such a cost-share formula could be inversely proportional to the number of acres infested, so that the Federal cost-share would increase as the number of infested acres decreases:

$$\text{Federal cost-share proportion} = \frac{1}{\text{thousand infested acres}}$$

For example, a treatment of 10,000 acres would result in a 10-percent Federal cost-share (1/10 = 0.10 = 10 percent), while a treatment of 2,000 acres would result in a 50-percent Federal subsidy (1/2 = 0.50 = 50 percent).

Second, the intensity of survey necessary to discover the relatively small areas of infestation that constitute hot-spots effectively precludes such a program being conducted solely by USDA/APHIS. Adequately surveying Platte and Goshen counties in Wyoming required the equivalent of six full-time field scouts in May and June of each survey year. This dedication of personnel is not viable for even the high-risk rangelands, let alone for the entire West. Ranchers and land managers must become active participants in a coordinated survey effort for a hot-spot program to be a viable management strategy. Again, a cost-share formula that rewards local participation or at least does not discourage such activity would be beneficial.

Third, the principle of the commons (derived from European grazing practices) suggests that people generally act to maximize their individual gains when given access to a common or collective resource. In terms of a hot-spot program, there is a potential conflict between individual and collective interests.

Because hot-spots are not uniformly distributed and treating a hot-spot potentially protects and benefits adjacent lands from future damage, this strategy tends to individualize the costs and collectivize the benefits. One solution to this problem is to collectivize the costs, perhaps through the formation or utilization of grazing and pest-management districts in order to support the higher short-term costs of survey and treatment in a hot-spot program.

Fourth, not enough long-term data have been gathered to provide a definitive answer to the viability of the hot-spot strategy. Current field data are not adequate to determine the population ecology of most rangeland grasshopper species, and existing information can be used to support aspects of both eruptive and gradient dynamics.

Summary

The Western United States has been in an interoutbreak period since 1987, so the processes leading to the extreme infestations (such as 50,000 acres) associated with the major outbreak periods have yet to be observed. With continued tracking of rangeland grasshopper dynamics, investigators may be able to determine the feasibility of a preventive approach to grasshopper outbreaks. For now, local experiments with this strategy should be encouraged as a means of confirming the usefulness of hot-spot programs across different rangeland systems.

Selected References

Barbosa, P.; Schultz, J. C., eds. 1987. Insect outbreaks. New York: Academic Press. 578 p.

Berryman, A. A. 1987. The theory and classification of outbreaks. In: Barbosa, P.; Schultz, J. C., eds. Insect outbreaks. New York: Academic Press: 3–30.

Hewitt, G. B.; Onsager, J. A. 1983. Control of grasshoppers on rangeland in the United States—a perspective. Journal of Range Management 36: 202–207.

Joern, A.; Gaines, S. B. 1990. Population dynamics and regulation in grasshoppers. In: Chapman, R. F.; Joern. A., eds. Biology of grasshoppers. New York: Wiley: 415–482.

Lockwood, J. A.; Kemp, W. P.; Onsager, J. A. 1988. Long-term, large-scale effects of insecticidal control on rangeland grasshopper populations. Journal of Economic Entomology 81: 1258–1264.

Lockwood, J. A.; Schell, S. P. In press. Outbreak dynamics of rangeland grasshoppers: Eruptive, gradient, both, or neither? In: Proceedings, 6th meeting of the Orthopterists' Society; Hilo, HI.

II.11 Baits for Controlling Rangeland Grasshoppers: An Overview

R. Nelson Foster

NOTE: Acephate is no longer approved by EPA for rangeland grasshopper control.

The first use of baits for grasshopper control began in the late 1800's. In 1878, the U.S. Entomological Commission reported bait experiments with mixtures of paris green and flour. In 1885, a bran bait containing arsenic, sugar, and water was used against grasshoppers in the San Joaquin Valley of California (Coquillet 1886). Over the next several decades, there was extensive testing to improve baits.

The work to improve baits concentrated on testing substances for attractiveness to grasshoppers and substitutes or diluents (diluting agents) for bran. Some of these substances were molasses (beet and cane), salt, calcium chloride, citrus fruits, lemon and vanilla extracts, geraniol nitobenzine, amyl acetate, propyl acetate, butyl acetate, apples, apple flavoring, anise, corn oil, fusel oil, saccharin, sugar, vinegar, stale beer, sawdust, shorts (grain byproducts), whey, soap, and even horse manure (Shotwell 1942). Some of the substrates studied to replace bran were sawdust, cottonseed hulls, rolled wheat, ground wheat screenings, citrus meal, chopped and ground alfalfa, ground flax fiber, ground peanut shells, bagasse, pear and apple pomace, peat moss, ground beet pulp, ground corncobs, chopped cornstalks, cornmeal, soybean meal, pea bran, oat hulls, and low-grade wheat flour (Parker 1952).

Over the years, different toxic substances were studied for effectiveness against grasshoppers. These toxins included paris green, white arsenic, dry and liquid sodium arsenate, barium fluosilicate, and sodium fluosilicate (Shotwell 1942). However, until 1942, when sodium fluosilicate became the preferred toxic agent, arsenic was most often used (Parker 1952). The chlorinated hydrocarbon insecticides introduced in the 1940's soon replaced the previously used toxic agents. Because sprays of these insecticides were so effective, widespread use of baits discontinued by 1950.

New insecticides that were equally effective, but environmentally safer, later replaced the chlorinated hydrocarbons. The development of acceptable spray agents and spray technology, even though extremely efficient, did not eliminate the use of bran bait completely. Bait commonly was used against Mormon cricket (a longhorn grasshopper) in the 1970's and continues today.

Although liquid sprays are very effective and economically superior, baits offer several environmental advantages, and work has continued to improve them. Ewen (1990) reviewed some of the more recent reported results with baits. His review included studies on the organophosphates (dimethoate, pyridaphenthion, fenitrothion, and malathion), the carbamates (propoxur, carbofuran, carbaryl, and cloethocarb); and the synthetic pyrethroids (fenvalerate and cypermethrin). In addition to these chemicals, chlorpyrifos and acephate, both organic phosphates, and diflubenzuron, an insect growth regulator, have also recently been studied in bait formulations. Studies of these toxicants in baits are noted in the references at the end of this chapter.

Of the toxicants recently studied, dimethoate, fenitrothion, carbofuran, cloethocarb, chlorpyrifos, diflubenzuron, and carbaryl are very effective in bait formulations against susceptible species of grasshoppers. However, most of these toxicants are not currently registered for use in baits against grasshoppers. Carbaryl is currently registered for use in the United States against grasshoppers and is commonly used on rangeland when bait treatments are indicated. It has been extensively used as a preventive "hot-spot" treatment in the Grasshopper Integrated Pest Management Project's North Dakota demonstration area. Dimethoate is registered for use in Canada in baits against grasshoppers.

Even though extensive research has been conducted with baits, two general areas of concern still detract from their widespread use against grasshoppers. Grasshopper populations on rangeland are seldom composed of only species that readily consume baits, and control of bait-consuming species is usually less with baits than with sprays. The cost of applying baits, particularly by air, usually exceeds the cost of applying sprays. Also, because applicators have less experience with baits, they perceive more difficulty in calibrating equipment for baits than for sprays.

On the other hand, baits have some considerable environmental advantages. The increased interest in protecting the environment and reducing the effects on nontarget species make baits more attractive than in the past. Compared to sprays, baits require less active ingredient to achieve reduction in grasshopper populations and are much more specific toward grasshoppers and affect significantly fewer nontarget organisms than sprays. Baits are also easier to direct toward the target area than sprays. Also, the increased knowledge that allows for use of treatments that do not provide almost total control of pest species adds to the attractiveness of baits. Other chapters in this section describe the recent developments, methods, and potential strategies for the use of bait formulations for controlling grasshoppers.

Suggested References

Capinera, J. L.; Hibbard, B. E. 1987. Bait formulations of chemical and microbial insecticides for suppression of crop-feeding grasshoppers. Journal of Agricultural Entomology. 4: 337–344.

Coquillet, D. W. 1886. Report on the locust of the San Joaquin Valley, California. Washington, DC: U.S. Commission on Agriculture Annual Report: 1885: 289–303.

Ewen, A. B. 1990. A synthesis paper on the use of insecticides formulated as baits for grasshopper control in Canada and the USA. Boletin de Sanidad Vegetal Plagas (Fuera de Serie) 20. 83–89.

Ewen, A. B.; Mukerji, M. K. 1987. Field evaluation of carbofuran bait against grasshopper (Orthoptera: Acrididae) populations in Saskatchewan. Canadian Entomologist 119: 537–540.

Ewen, A. B.; Mukerji, M. K. 1980. Evaluation of *Nosema locustae* (Microsporida) as a control agent of grasshopper populations in Saskatchewan. Journal of Invertebrate Pathology 35: 295–303.

Jech, L. E.; Foster, R. N.; Colletto, D.; Walgenbach, D. D.; Roland, T. J.; Rodriguez, G. D.; Bohls, R.; Houston, R. D.; Meeks, W. K.; Queener, R. L.; Jackson, C. L.; Dines, J. L.; Puclik, M. J.; Scott, A. K. 1993. Field evaluation of diflubenzuron and carbaryl bran baits against grasshopper (Orthoptera: Acrididae) populations in South Dakota. Journal of Economic Entomology 86: 557–565.

Johnson, D. L.; Henry, J. E. 1987. Low rates of insecticides and *Nosema locustae* (Microsporidia: Nosematidae) on baits applied to roadsides for grasshopper (Orthoptera: Acrididae) control. Journal of Economic Entomology 80: 685–689.

Mukerji, M. K.; Ewen, A. B. 1984. Field evaluations of cypremethrin and carbaryl as sprays and baits for grasshopper (Orthoptera: Acrididae) control in Saskatchewan. Canadian Entomologist 116: 5–9.

Mukerji, M. K.; Ewen, A. B.; Craig, C. H.; Ford, R. J. 1981. Evaluation of insecticide-treated bran baits for grasshopper control in Saskatchewan (Orthoptera: Acrididae). Canadian Entomologist 113: 705–710.

Onsager, J. A.; Henry, J. E.; Foster, R. N.; Staten, R. T. 1980a. Acceptance of wheat bran by species of rangeland grasshoppers. Journal of Economic Entomology 73: 548–551.

Onsager, J. A.; Henry, J. E.; Foster, R. N. 1980b. A model for predicting efficacy of carbaryl bait for control of rangeland grasshoppers. Journal of Economic Entomology 73: 726–729.

Parker, J. R. 1952. Grasshoppers, In: Yearbook of Agriculture. Washington, DC: U.S. Department of Agriculture: 595–604.

Quinn, M. A.; Kepner, R. L.; Walgenbach, D. D.; Bohls, R. A.; Pooler, P. D.; Foster, R. N.; Reuter, K. C.; Swain, J. L. 1989. Immediate and second-year effects of insecticide spray and bait treatments on populations of rangeland grasshoppers. Canadian Entomologist 121: 589–602.

Shotwell, R. L. 1942. Evaluation of baits and bait ingredients used in grasshopper control. Tech. Bull. 796. Washington, DC: U.S. Department of Agriculture: 51.

Suggested References—Unpublished

Colletto, D.; Jech, L.; Foster, R. N.; Houston, R.; Puclik, M.; Scott, A. 1990. Efficacy of three different toxicants on three carriers in combinations for grasshopper control on improved rangeland near Medora, N.D., 1990. In: Cooperative Grasshopper Integrated Pest Management Project, 1990 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 164–168.

Colletto, D.; Jech, L.; Foster, R. N.; Roland, T. J.; Jackson, C.; Patterson, J.; Meeks, W. 1989. Preliminary insecticidal value field trials on rangeland grasshoppers near Edgemont, South Dakota, 1989. In: Cooperative Grasshopper Integrated Pest Management Project, 1989 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 190–193.

Jech, L. E.; Foster, R. N.; Colletto, D.; Drake, S.; Houston, R.; Black, L. 1991. Field trials of defatted corn and flaky wheat bran treated with either carbaryl or acephate for control of rangeland grasshoppers near Wheatland, Wyoming, 1991. In: Cooperative Grasshopper Integrated Pest Management Project, 1991 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 87–89.

Jech, L. E.; Foster, R. N.; Colletto, D.; Walgenbach, D. D.; Bohls, R.; Rodriguez, G.; Burgess, J.; Meeks, W.; Queener, R. 1988. Field trials of chlorpyrifos and carbaryl bran baits for control of grasshopper on rangeland near Buffalo, South Dakota, 1988. In: Cooperative Grasshopper Integrated Pest Management Project, 1989 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 194–206.

II.12 Bait Acceptance by Different Grasshopper Species and Instars

Jerome A. Onsager, R. Nelson Foster, and Larry Jech

The Grasshopper Integrated Pest Management (GHIPM) Project provided unique resources and opportunities that allowed investigators to gather a large amount of data on the responses of rangeland grasshoppers to carbaryl bait. A total of 39 different species were recorded in 24 different control experiments at 14 different sites in the western parts of North Dakota and South Dakota. All species were not present in sufficient numbers to provide useful information, but the data base allowed GHIPM-funded investigators to study many questions that could not have been examined without it.

Data Collection

The monitoring procedure was to establish from 4 to 10 monitoring sites, each consisting of 40 0.1-m^2 rings spaced about 5 m apart in circles, both in plots that were scheduled for treatment and in adjacent plots that remained untreated. Density counts and sweep-net collections were made as close as possible (usually 24 hours) before scheduled treatments, and again as close as possible to 48 hours after treatment. The information from all sample sites per plot for each sampling date was then combined for further study.

Each sweep sample was examined to determine the species and stage of development for every grasshopper in the sample. Each total density count was then converted to density per instar per species by multiplying observed total density times the appropriate proportions of composition within the sweep samples. The procedure is identical to that described in chapter II.2, “Evaluation of Rangeland Grasshopper Controls,” except that density was estimated for each instar of a species as well as for all individuals of a species.

Computer tabulations of different species recorded in different experiments revealed a potential for 253 independent determinations of species-specific response to carbaryl bait. Pretreatment and posttreatment data for each species in each experiment were then examined to assess which of the possible determinations would be meaningful. A total of 101 potential data sets were declared useless, leaving 152 legitimate determinations.

Reasons for rejecting some data sets included initial presence in such low density that subsequent reduction would not be measurable (in most cases, at least five specimens in pretreatment samples were required), absence of specimens at untreated sample sites (which prohibited estimation of mortality in the absence of treatment), and higher estimated mortality in untreated plots than in treated plots (a common artifact of sampling error among low-density samples).

The 152 data sets accepted as legitimate provided opportunities to study a variety of questions about response to carbaryl bait. The simplest assessment concerned the average percent control among all individuals of a species. This average percent control was calculated with a variation of the formula by Connin and Kuitert (1952):

Percent control = $100(1 - (\mathrm{Ta} \times \mathrm{Ub} \div \mathrm{Tb} \div \mathrm{Ua}))$, where
Tb is density in treated plots before treatment,
Ta is density in treated plots after treatment,
Ub is density in untreated plots before treatment, and
Ua is density in untreated plots after treatment.

The formula does not yield “simple” or “raw” control data—that is, the percentage of the total infestation that “disappeared” in treated plots. Rather, it yields “adjusted” control data: the percentage of the total infestation that most likely was killed by carbaryl bait.

The formula is useful for two major reasons. First, grasshopper infestations suffer some mortality each day due to natural causes, so the formula “removes” that natural mortality from consideration. The formula essentially uses data from untreated sites to estimate what the posttreatment counts at treated sites would have been in the absence of treatment. Percent control then represents the difference (if any) between expected and observed posttreatment density in treated plots. Second, without the formula, the percent control that is estimated will be grossly different, depending on how much time elapses between pretreatment and posttreatment counts. These problems can be illustrated with an example.

Let us assume that an infestation of 30 grasshoppers/yd^2 comprises 6 *Aeropedellus clavatus,* 15 *Melanoplus sanguinipes,* and 9 *Amphitornus coloradus.* We decide to treat half and leave half, and we sample both halves on the day before treatment (day –1), and on days 2, 3, 4, and 5 after treatment. Table II.12–1 shows typical density data.

Table II.12–1—A representative example of typical grasshopper density data in untreated plots versus plots that were treated (on day zero) with carbaryl bait

Time	*A. clavatus*		*M. sanguinipes*		*A. coloradus*		All species	
(days after treatment)	Untreated plot	Treated plot	Untreated plot	Treated plot	Untreated plot	Treated plot	Untreated plot	Treated plot
–1	6	6	15	15	9	9	30	30
+2	3.68	2.95	13.69	3.42	7.71	7.56	25.08	13.93
+3	3.13	2.51	13.28	3.32	7.33	7.18	23.74	13.01
+4	2.66	2.13	12.88	3.22	6.96	6.82	22.5	12.17
+5	2.26	1.81	12.49	3.12	6.61	6.48	21.36	11.41

Looking only at the raw density for “All species” in only the treated plot, a reader might believe that this bait treatment achieved about 54- to 62-percent average control of the infestation. The fallacy is that if a similar strategy is applied to data from untreated plots, a reader could estimate 16- to 29-percent control where nothing was done. Use of the formula yields more conservative and more realistic estimates of about 44- to 46-percent adjusted control of “All species.”

Raw estimates for individual species can also be very misleading. For example, *A. clavatus* usually is the first species that hatches in the spring. By the time of typical bait treatments to control later-hatching major pest species, *A. clavatus* often is present as very old adults that suffer very high daily mortalities likely associated with the process of aging. Raw estimates indicate 51- to 70-percent population reduction, but adjusted estimates reveal only 20-percent control due to the bait, meaning the raw estimates placed control at 2.5 to 3.5 times higher than it actually was.

Notice in the example that discrepancies between raw and adjusted mortalities for *A. coloradus* are even greater than they were for *A. clavatus.* This is because adjusted response to treatment (2-percent control) was less than the daily loss due to natural mortality (5 percent per day). In such a case, raw estimates yield greatly distorted results. As one might then expect, raw estimates are closest to adjusted estimates in cases like the *M. sanguinipes* example, where natural mortality was relatively low (3 percent per day) and adjusted control was relatively high (75 percent). Nevertheless, it should be noted that all raw estimates for *M. sanguinipes* still were too high, and the degree of error increased as the amount of time between pretreatment and posttreatment samples was increased. Similar errors are guaranteed to occur in real life (in field experiments or commercial control projects) if natural mortality is ignored.

Relative Susceptibility of Different Species

The results of GHIPM experiments were combined with a number of previous studies by the authors and others (see Swain [1986] and Quinn et al. [1989]) to produce table II.12–2. It divides grasshoppers into three broad classes of susceptibility. The “sensitive” class contains species that readily seek out and eat wheat bran bait and therefore usually suffer a high degree (average = 56–87 percent) of adjusted (true) mortality. The “vulnerable” class contains species that usually either suffer only a moderate degree (30–55 percent) of adjusted mortality or else exhibit such great variation among different tests that one cannot safely depend on more than moderate results. The “nonsusceptible” class (less than 30-percent adjusted mortality) contains species that eat little or no bait and therefore usually are not markedly affected by bait.

Most of the experiments that contributed to table II.12–2 were applied when the majority of target pest grasshopper species were in third, fourth, or fifth instars. A few very early species like *A. clavatus* and *M. confusus* typically were treated as adults or fifth instars, while some relatively late species like *P. nebrascensis* and *P. quadrimaculatum* were occasionally treated as first or

Table II.12–2—Classification of grasshopper species according to susceptibility to carbaryl wheat bran bait

Class and expected levels of control	Species
Sensitive (>55-% control) Control is expected to average about 70%. Worst-case and best-case scenarios will be about 55% and 85%, respectively.	*Ageneotettix deorum* *Anabrus simplex* *Aulocara elliotti* *Camnula pellucida* *Hadrotettix trifasciatus* **Melanoplus bivittatus* *Melanoplus confusus* *Melanoplus dawsoni* *Melanoplus foedus* **Melanoplus infantilis* **Melanoplus occidentalis* **Melanoplus packardii* *Melanoplus sanguinipes* *Spharagemon equale* *Stenobothrus brunneus* **Mermiria bivittata*
Vulnerable (30- to 55-% control) Control is expected to average about 42%. Worst-case and best-case scenarios will be about 12% and 72%, respectively.	**Aulocara femoratum* *Eritettix simplex* *Melanoplus femurrubrum* *Oedaloenotus enigma* *Opeia obscura* *Phoetaliotes nebrascensis* *Psoloessa delicatula*
Nonsusceptible (<30-% control) Control is expected to average about 15%. Worst-case and best-case scenarios will be about 0% and 30%, respectively.	*Aeropedellus clavatus* *Amphitornus coloradus* *Cordillacris crenulata* *Cordallacris occipitalis* *Hesperotettix viridis* *Metator pardalinus* **Phlibostroma quadrimaculatum* *Trachyrhachys kiowa*

*These species are not likely to suffer best-case scenario levels of control.

second instars where they were incidental rather than primary target species.

Relative Susceptibility of Different Developmental Stages

Some of the GHIPM experiments provided data that allowed the comparison of the relative susceptibility of different instars of a species to bait. In general, the requirements for a meaningful test were the presence of at least four or more different stages in reasonable numbers (usually at least five individuals per instar in pretreatment sweep samples) in two or more different experiments. In those cases, the authors calculated adjusted percent control for each instar and used analyses of covariance, with instar as the covariant, to test susceptibility by instar. When covariance was significant (when percent control was affected by instar), the slope of the relationship indicated whether larger or smaller instars were most susceptible.

A total of eight species were tested, six of which were considered in table II.12–2 to be sensitive. Younger instars of three species, *A. deorum, M. packardii,* and *M. sanguinipes,* were found to be significantly more susceptible to bait than older instars. Susceptibility was not affected by instar in the cases of *A. elliotti, C. pellucida, M. infantilis, P. nebrascensis,* or *T. kiowa.*

Relative Susceptibility of Different-Aged Populations

Some of the GHIPM experiments provided data that allowed the researchers to examine the effect of age on susceptibility of populations to bait. Age was expressed as average instar, which is calculated as the sum of each instar number multiplied by the number of grasshoppers in the instar (adults are considered instar 6 for this procedure) divided by the total number of grasshoppers present. The requirements for a meaningful test were significant adjusted control observed in three or more experiments (incidences of zero control were excluded from these calculations). The relationship between average instar and percent adjusted mortality was examined by linear regression techniques.

A total of 17 species was tested, 10 of which were considered in table II.12–2 to be sensitive or vulnerable. For three of those species, *A. elliotti, A. deorum,* and *M. sanguinipes,* percent adjusted control increased significantly with average instar.

Summary and Recommendations

Grasshopper species vary considerably in their inclination to feed on wheat bran and in their susceptibility to carbaryl-treated bait. In addition, levels of control that follow bait treatments are considerably lower and much less predictable than control achieved with liquid sprays. The GHIPM Project greatly increased the knowledge base for both acknowledged pest grasshopper species (the primary target species) and for incidental (nontarget) species. Project researchers now feel that they can offer some general guidelines, based on species susceptibility (table II.12–2), for the appropriate use of carbaryl bait.

Individuals should not attempt to control nonsusceptible pest species with bait. If such species comprise a significant proportion of an infestation, a conservative manager should simply assume that bait will give no control of that proportion. Vulnerable species may or may not be markedly controlled by baits, but what regulates that degree of success remains unknown, and at this time those results cannot be predicted. Past situations have documented dramatic reductions in vulnerable species from the use of bait, as well as cases of almost total failure. In the future, managers should not use bait against vulnerable species without seriously weighing the consequences of failure. Control of the sensitive species with bait is generally reliable.

Questions about optimum timing for bait treatments remain somewhat perplexing, but it fortunately appears that timing is not of extreme importance, perhaps because of compensatory factors. Some tests support early treatments in that, at least for some species, younger instars were more susceptible than older instars. This is logical because smaller grasshoppers are killed by smaller doses of toxicant. Another advantage of early bait treatment is that natural control agents have more time to act upon surviving grasshoppers.

Other tests, however, support late treatments in that total percent control was greater for older populations than for younger populations. While these results may seem contrary, they also can be considered strong evidence that something like changes in behavioral traits (perhaps searching capabilities) or habitat characteristics (perhaps cover, litter, or bare ground) make baits more accessible as the season progresses. If such compensating factors exist, the mechanisms cannot be accurately described at the present time. Fortunately, however, for most species (14 of 17 tested), adjusted percent control was not markedly affected by population age. It therefore appears that timing of bait treatments is not of extreme importance as long as it occurs when most of the primary target grasshoppers are in third, fourth, or fifth instars.

References Cited

Connin, R. V.; Kuitert, L. C. 1952. Control of the American grasshopper with organic insecticides in Florida. Journal of Economic Entomology 45: 684–687.

Quinn, M. A.; Kepner, R. L.; Walgenbach, D. D.; Bohls, R. A.; Pooler, P. D. 1989. Immediate and 2nd-year effects of insecticide spray and bait treatments on populations of rangeland grasshoppers. Canadian Entomologist 121: 589–602.

Swain, J. L. 1986. Effect of chemical grasshopper controls on nontarget arthropods of rangeland in Chaves County, New Mexico. Master’s thesis. Las Cruces, NM: New Mexico State University. 102 p.

II.13 What, When, and Where Do Grasshoppers Eat?

Larry Jech

Some species of grasshoppers do not readily take baits. As a result, the effectiveness of grasshopper control through bait applications can be limited. Various researchers have attempted to increase bait effectiveness. These studies have focused primarily on comparing toxicants, varying applications timing, and varying the amount of toxicant on the bait applied. Carefully designed and executed experiments with alternate insecticides and time-of-day application did not lead to increases in grasshopper mortality among the species that did not feed on bait in other experiments. The Grasshopper Integrated Pest Management (GHIPM) Project conducted observation studies to improve baits through better understanding of grasshopper feeding behavior.

Findings of Direct Observations

During the summers of 1990 and 1991, GHIPM Project experiments involved direct observation of grasshoppers feeding on host plants in rangelands. The study focused on species that readily take bait and species that do not. The study sites were typical prairies in western South Dakota and North Dakota. The grasshopper densities were representative of those targeted for bait control programs (greater than 10 but less than 25 grasshoppers/m^2). Observation involved watching individual grasshoppers from daybreak to dusk and recording their behavior every 15 seconds.

Most of the behavior observed had very little to do with feeding. Grasshoppers basked in the sun, moved about their habitat, and exhibited avoidance behavior. Most observations were of third-instar (young grasshoppers) to adults.

The study included four common species that are not easily controlled by bait applications at the standard rate of 1.5 lb/acre containing 2 percent carbaryl. These species were *Amphitornus coloradus* (Thomas), *Cordillacris occipitalis* (Thomas), *Trachyrhachys kiowa* Thomas, and *Phlibostroma quadrimaculatum* (Thomas). Also, the study compared these four species' behavior with that of two species that are easily controlled with baits—*Aulocara elliotti* (Thomas) and *Ageneotettix deorum* (Scudder).

Usually grasshoppers spent the early morning basking. After the air temperature reached 81 °F, the grasshoppers began to feed. Grasshoppers allowed time for their crops to empty between feeding sessions and repeated feeding and resting cycles regularly. The insects generally groomed their antennae and eyes before feeding, but grooming apparently was not a prerequisite to feeding.

Feeding continued throughout the day if temperatures remained below 90 °F. When temperatures rose above 95 °F, the grasshoppers perched on stems or took shelter under vegetation to avoid excessive heat. While the temperature remained elevated, the grasshoppers did not actively feed; active feeding resumed when the temperature fell. In other experiments designed to determine the optimal time of bait application (including experiments during the GHIPM Project), temperatures remained below 90 °F so that timing of application was not a significant factor for most of the grasshopper population.

Very little feeding took place when winds exceeded 15 miles per hour (mi/hour) or during cool, cloudy days. The insects would remain quiet until weather conditions improved. Grasshoppers also stopped feeding when rain was imminent. After showers or rains passed and the ground warmed, grasshoppers returned to feeding.

Although grasshoppers spent one-seventh of their time moving, the movement appeared to be random. Most of the time, grasshoppers were on the soil surface and climbed the plants only to feed. The exception was *Amphitornus coloradus.* This species would enter a clump of grass and position itself so its body was nearly vertical. The upright position, combined with its cryptic body markings, gave the grasshopper maximum protection from predators. For this species, feeding behavior seemed to be balanced carefully between the need to feed and to remain hidden.

Grasshoppers were very discriminating in their food choices. They would sample a blade of grass before feeding on it and occasionally move back to a portion of the blade or another blade passed over previously. *T. kiowa,* one that does accept bran bait, often would feed on a plant, move a short distance, and then return to the same plant and resume feeding. The activity showed the grasshopper was capable of relocating a suitable host plant. Grasshoppers fed on the tips of leaf blades or would clip the tip of a blade and then feed on the tip while grasping it with their forelegs. When the latter feeding habit occurred, the grasshoppers usually ate all of the clipped portion. The other common feeding pattern was to bite a portion out of a leaf margin, leaving it notched.

Aulocara elliotti and *Ageneotettix deorum*, the two species that readily eat bran bait, often picked up bits of plant litter from the soil surface and tasted and consumed those food items in addition to feeding actively on live tissue. These two species also clipped the leaf tips but dropped the clippings to the ground and later fed on the sun-dried clippings. The four species that do not accept bran bait seldom fed on materials found on the soil surface and preferred live tissue.

Additional tests showed species that feed on live tissue and do not take baits would accept baits glued to host plants. *Cordillacris occipitalis* and *Aulocara elliotti* were caged on a host plant that is acceptable to both species. Bait particles were glued to the host at the leaf tip, midleaf, and at the leaf base. Grasshoppers were allowed to browse for 8 hours. Grasshoppers caged on untreated leaves had no mortality, while both species caged on treated leaves showed equal mortality.

Conclusions

Grasshoppers in this study spent only a small portion of their time feeding. They fed in sessions interspersed with rest or movement (see table II.13–1.) Grasshopper species that were easily controlled with baits fed on plant litter and detrital material on the ground and were therefore predisposed to feed on bran baits. Grasshoppers that did not take baits fed on living host plants.

One approach to enhancing bait effectiveness would be to treat the bait with a sticking agent as the bait is applied. Some of the treated bait would then be encountered by grasshoppers feeding on live host plants. Bait falling on the soil surface will remain available to ground-feeding species.

Attracting grasshoppers that feed on live tissue to bait and positioning bait in the known feeding locations are some areas for the next stage of research.

Table II.13–1—Summary of feeding behaviors for six species of grasshoppers

	Percent of time engaged in:			Total
Species	Basking	Moving	Feeding	hours
Ageneotettix deorum	81.8	13.9	4.4	14.9
Aulocara elliotti	69.5	17.2	13.2	25.5
Amphitornus coloradus	77.4	8.4	14.2	57.8
Cordillacris occipitalis	81.0	9.1	9.8	18.8
Phlibostroma quadrimaculatum	76.8	18.4	5.8	14.4
Trachyrhachys kiowa	36.8	31.4	31.7	14.9

II.14 Effect of Multiple Concentrations and Rates of Carbaryl–Bran Bait

Mark A. Quinn, R. Nelson Foster, and K. C. Reuter

Introduction

Insecticidal baits are a viable alternative to conventional insecticidal sprays for controlling grasshoppers (Quinn et al. 1989). Baits are particularly effective when the grasshopper community is composed largely of bran "acceptors," or those species that readily consume bran baits (see chapter II.12 on bait acceptance). Most of the recent studies involving bran baits have used concentrations in the range of 2–5 percent toxicant at rates near 1.5 lb/acre. The efficacy of higher concentrations and rates has not been studied extensively. As part of the Grasshopper Integrated Pest Management Project, a study was conducted in northwestern South Dakota to determine the effects of multiple concentrations and rates of carbaryl bran bait on grasshoppers on mixed-grass rangeland.

Multiple Concentrations and Rates of Bran Bait—A Case Study

Fifty-one 40-acre plots were treated with aerial applications of carbaryl bran bait in the following concentrations and rates: 2 percent carbaryl at 0.5, 1, 2, 5, and 10 lb/acre; 5 percent carbaryl at 0.5, 1, and 2 lb/acre; and 10 percent carbaryl at 0.5, 1, and 2 lb/acre. An additional nine plots were used as controls. The baits were applied with a Cessna Ag Truck operating at an altitude of 40–60 ft at 115 miles per hour (mi/hour) and equipped with a standard Transland 20244 spreader. Swath widths were 45 ft. Treatments were applied over a 17-day period from June 27 to July 13, 1987. Approximately 56 percent of grasshoppers were in the nymphal stage at the time of treatments (table II.14–1).

Densities of grasshoppers were estimated in the center of each plot by counting grasshoppers in 40 0.1-m^2 rings (Onsager and Henry 1977) placed approximately 16 ft apart in a 210-ft-diameter circle. Relative abundance of each grasshopper species and instar was determined by collecting grasshoppers near the circle of rings with a sweep net. Densities of individual species were estimated by multiplying their relative abundance by total grasshopper density. Grasshopper populations were monitored before treatment and 2, 4, and 7 days after treatments. Populations were monitored approximately daily from June 26 to July 20 in the control plots.

Changes in densities of total grasshoppers, bran-accepting species, and bran-rejecting species in the control plots were compared with changes in plots treated with the insecticidal baits to determine overall treatment effects. Major bran-accepting species included *Melanoplus sanguinipes*, other *Melanoplus* species, *Ageneotettix deorum, Phoetaliotes nebrascensis*, and *Aulocara elliotti*. Bran-rejecting species included *Aeropedellus clavatus, Amphitornus coloradus, Trachyrhachys kiowa*, and *Opeia obscura*. Although *O. obscura* may be vulnerable to insecticidal baits (see the bait acceptance chapter in this section), we included it in the bran-rejector category because it was not affected by the insecticidal bait in our particular study.

Mean pretreatment densities of total grasshoppers ranged from 13.1 to 22 grasshoppers/yd^2 in the treatment plots. *M. sanguinipes, A. deorum*, and *A. clavatus* constituted 32, 15, and 14 percent of all grasshopper species, respectively, during the pretreatment period (table II.14–1). Bran acceptors constituted 72 percent of all species.

All insecticidal bait treatments, except the 2 percent carbaryl at 0.5 lb/acre, caused significant reductions in total grasshopper density compared with controls (table II.14–2). The greatest mean mortalities, ranging from 72 to 86 percent, occurred in plots treated with 2 percent carbaryl bran bait at 5 and 10 lb/acre, 5 percent carbaryl bran bait at 1 lb/acre, and 10 percent carbaryl at 2 lb/acre. The more standard treatments of 2 percent carbaryl at 1 and 2 lb/acre gave intermediate results, causing average mortalities of 52 and 64 percent, respectively. Applications of bran bait at 0.5 lb/acre were least effective, killing less than 50 percent of all grasshoppers.

All treatments caused significant mortality of bran-accepting species of grasshoppers compared with controls (table II.14–2). The greatest mortality occurred in plots treated with 2 percent carbaryl at 10 lb/acre (97 percent), 5 percent carbaryl at 2 lb/acre (90 percent), 2 percent carbaryl at 5 lb/acre (90 percent), and 5 percent carbaryl at 1 lb/acre (88 percent). The commonly used treatments of 2 percent carbaryl at 1 or 2 lb/acre caused 72 and 89 percent mortalities, respectively, of bran-accepting grasshopper species. Applications of 2 and 5 percent carbaryl at 0.5 lb/acre caused 45–54 percent reductions in the bran acceptors. Densities did not change in control plots.

Table II.14–1—Relative abundance of grasshopper species and instars and number of plots occupied on the pretreatment sampling dates, June 26–July 7, 1987, Harding County, SD

Species	No. of plots occupied	Percentage of grass- hoppers[1]	Percentage of individuals in each instar					
			I	II	III	IV	V	Adult
Melanoplus sanguinipes (F.)	55	32.31	0.0	0.9	13.6	17.0	32.1	36.4
Ageneotettix deorum (Scudder)	55	14.35	0.0	0.8	4.6	15.2	57.6	21.8
Aeropedellus clavatus (Thomas)	51	13.95	0.0	0.0	0.0	0.0	0.0	100.0
Melanoplus dawsonii (Scudder)	40	5.31	0.4	4.7	23.8	27.7	20.6	22.8
Melanoplus confusus Scudder	47	4.57	0.0	0.0	0.0	0.0	0.0	100.0
Amphitornus coloradus (Thomas)	50	4.55	0.0	0.0	2.7	9.5	43.1	44.6
Melanoplus infantilis Scudder	44	3.76	0.2	2.8	15.9	15.3	31.7	34.0
Trachyrhachys kiowa Thomas	48	2.50	0.0	0.6	10.5	16.8	35.0	37.1
Melanoplus spp.	38	2.32	25.0	74.6	0.0	0.0	0.4	0.0
Orphulella speciosa (Scudder)	31	2.13	0.4	2.0	11.8	34.1	29.5	22.2
Phoetaliotes nebrascensis (Thomas)	39	2.10	4.5	40.1	36.7	14.8	3.9	0.0
Aulocara elliotti Thomas	38	1.92	0.0	0.0	0.5	0.8	20.0	78.7
Melanoplus packardii (Scudder)	46	1.47	0.6	4.9	16.0	38.8	32.8	8.7
Melanoplus femurrubrum (DeGeer)	17	1.36	6.1	20.8	42.8	15.6	11.0	3.7
Melanoplus bivittatus (Say)	34	1.30	0.0	1.5	5.7	12.8	15.1	64.8
Opeia obscura (Thomas)	39	1.19	0.0	2.4	15.3	39.3	36.1	6.9
Others (26 species)	—	4.91	6.8	10.6	13.6	14.3	23.0	31.7
All species	55	100.00	1.1	4.3	10.0	13.7	27.3	43.6

[1] Based on a total of 12,063 grasshoppers collected.

Table II.14–2—Pretreatment densities and reductions in grasshopper densities 7 days after treatments with different rates and concentrations of carbaryl bran bait, Harding County, SD

Grasshopper variable	Treatment[1]	Pretreatment density[2] ($\bar{x}$ ± SEM) standard error of the means	Percent reduction[3] ($\bar{x}$ ± SEM)
Total grasshoppers	Control	13.7 ± 1.91a	8.1 ± 12.66a
	2% — 0.5	12.5 ± 2.43a	31.3 ± 10.69abcd
	2% — 1.0	13.8 ± 0.61a	51.7 ± 17.01be
	2% — 2.0	17.4 ± 2.81a	63.9 ± 2.17efgh
	2% — 5.0	17.4 ± 4.21a	75.3 ± 8.20gk
	2% — 10.0	20.1 ± 6.57a	85.9 ± 7.91k
	5% — 0.5	16.4 ± 1.40a	37.4 ± 15.58bcdf
	5% — 1.0	18.6 ± 5.53a	77.9 ± 7.54hk
	5% — 2.0	18.0 ± 4.92a	56.0 ± 8.05ceg
	10% — 0.5	12.0 ± 2.83a	49.9 ± 5.98bcdf
	10% — 1.0	13.9 ± 2.39a	58.7 ± 3.81deg
	10% — 2.0	17.3 ± 2.33a	72.3 ± 4.71ek
Bran acceptors	Control	9.4 ± 1.43a	5.7 ± 12.77a
	2% — 0.5	8.7 ± 1.42a	45.2 ± 19.72b
	2% — 1.0	8.4 ± 0.60a	60.2 ± 17.80bcd
	2% — 2.0	11.3 ± 1.40a	77.5 ± 4.85cdef
	2% — 5.0	13.6 ± 4.53a	89.6 ± 5.79fg
	2% — 10.0	17.0 ± 5.23a	97.4 ± 2.34g
	5% — 0.5	12.6 ± 1.30a	53.5 ± 11.42bc
	5% — 1.0	15.1 ± 4.45a	87.9 ± 6.98efg
	5% — 2.0	10.2 ± 2.44a	89.8 ± 1.25efg
	10% — 0.5	8.2 ± 2.22a	72.1 ± 8.67cde
	10% — 1.0	10.1 ± 1.12a	69.5 ± 4.07bcd
	10% — 2.0	13.8 ± 1.74a	80.8 ± 8.47def
Bran rejectors	Control	3.4 ± 0.63a	0.1 ± 16.62ab
	2% — 0.5	3.1 ± 0.86a	–34.6 ± 24.01b
	2% — 1.0	3.2 ± 0.54a	34.6 ± 29.41ac
	2% — 2.0	5.7 ± 1.90a	–27.0 ± 44.99ab
	2% — 5.0	2.5 ± 0.28a	37.8 ± 8.79ab
	2% — 10.0	4.6 ± 1.13a	59.6 ± 28.41c
	5% — 0.5	3.7 ± 0.44a	8.5 ± 27.83ab
	5% — 1.0	3.4 ± 1.12a	33.4 ± 13.77ab
	5% — 2.0	7.7 ± 2.47a	9.1 ± 20.79ab
	10% — 0.5	3.2 ± 0.45a	15.4 ± 20.98ab
	10% — 1.0	3.3 ± 1.33a	12.2 ± 43.52ab
	10% — 2.0	3.3 ± 1.00a	–112.0 ± 117.23b

[1] Percent of carbaryl applied—application rate in lb/acre.

[2] No./yd^2.

[3] A negative percent reduction indicates an increase in grasshoppers.

Note: Means within columns followed by the same letter are not significantly different at the 0.1 level (Fisher's protected SD).

In general, bran-rejecting species were not affected by the treatments (table II.14–2). However, the greatest reduction in bran rejectors (60 percent) occurred in plots treated with 2 percent carbaryl at 10 lb/acre. Because changes in densities in these plots were highly variable, it could not be determined if this reduction was caused by mortality or natural variation in grasshopper populations.

Conclusions

The following conclusions can be drawn from the study. First, the quantity of carbaryl bran bait applied to rangeland affects grasshopper mortality. Baits applied at 0.5 lb/acre are relatively ineffective. The highest rates (5 and 10 lb/acre) were very effective in controlling grasshoppers. These results do not suggest, however, that more bait is always better (see chapter II.15 on multiple applications of bran bait). For example, 78 percent mortality was achieved in plots treated with 5 percent carbaryl at 1 lb/acre. Also, the small increase in mortality caused by higher rates may not be economically justifiable.

Second, the concentration of carbaryl seemed less important than the rate of application. For example, 2 and 10 percent carbaryl applied at a rate of 2 lb/acre caused similar grasshopper mortalities.

Finally, high mortality of grasshoppers was achieved because the grasshopper community was composed mainly of the bran-accepting *Melanoplus* species. Insecticidal baits are less effective when there is a higher proportion of bran-rejecting species (Quinn et al. 1989, Jech et al. 1993).

References Cited

Jech, L. E.; Foster, R. N.; Colletto, D.; Walgenbach, D. D.; Roland, T. J.; Rodriguez, G. D.; Bohls, R.; Houston, R. D.; Meeks, W. K.; Queener, R. L.; Jackson, C. L.; Dines, J. L.; Puclik, M. J.; Scott, A. K. 1993. Field evaluation of diflubenzuron and carbaryl bran baits against grasshopper (Orthoptera: Acrididae) populations in South Dakota. Journal of Economic Entomology 86: 557–565.

Onsager, J. A.; Henry, J. E. 1977. A method for estimating the density of rangeland grasshoppers (Orthoptera: Acrididae) in experimental plots. Acrida 6: 231–237.

Quinn, M. A.; Kepner, R. L.; Walgenbach, D. D.; Bohls, R. A.; Pooler, P. D.; Foster, R. N.; Reuter, K. C.; Swain, J. L. 1989. Immediate and 2nd-year effects of insecticidal spray and bait treatments on populations of rangeland grasshoppers. Canadian Entomologist 121: 589–602.

II.15 Comparison of Single and Multiple Applications of Bran Bait

Mark A. Quinn, R. Nelson Foster, and K. C. Reuter

Introduction

Insecticidal baits generally kill 30 to 70 percent of all rangeland grasshoppers (Quinn et al. 1989, Ewen 1990, Jech et al. 1993). Several factors influence the overall effectiveness of insecticidal baits. These include (1) the species composition of grasshoppers in the treated area, (2) total density of grasshoppers, and (3) the amount of bait applied to an area.

For control purposes, communities of grasshoppers can be classified as “bran acceptors” or “bran rejectors” depending on whether or not they consume treated baits (see chapter II.12 on bait acceptance). The larger the proportion of bran acceptors in the community, the greater the level of control by insecticidal baits. In turn, the species composition of grasshoppers is determined partly by vegetation. For example, some mixed-grass communities dominated by grasses will harbor a greater proportion of bran-rejector species than communities with abundant forbs (Quinn et al. 1991).

The effectiveness of insecticidal baits also depends on the density of grasshoppers in an area. Because insecticidal baits generally cause less mortality than sprays, baits can be ineffective when grasshopper densities are relatively high. For example, an insecticidal bait that causes only 60-percent mortality can reduce grasshopper populations below 10 per square yard only if initial densities are less than 25 per square yard.

There is some evidence that the amount of bait applied to rangeland also can limit the effectiveness of the treatments because much of the bait disappears quickly after application. For example, Mukerji et al. (1981) found that an increase in the amount of dimethoate-treated bran bait from 3.6 to 8 lb/acre caused an increase in mortality. Henry (1975) reported that most bran is consumed within a few hours of application.

In 1989, a 20-acre section of rangeland in the North Dakota Grasshopper Integrated Pest Management Project demonstration area was treated with 2 percent carbaryl bran bait at the rate of 2 lb/acre. After treatment, populations decreased 28 percent, but densities were still quite high at 25.8 grasshoppers/yd^2. After a second treatment of the insecticidal bait, populations declined an additional 47.3 percent. These results suggest that single applications of insecticidal baits at standard dosages may not produce the maximum possible control of grasshoppers because the bait is quickly consumed or lost. Besides grasshoppers, other insects may also compete for the bait. For example, Quinn et al. (1990) found that darkling beetles (Tenebrionidae), a dominant insect group on mixed-grass rangeland, probably consume treated bran bait.

Single and Multiple Applications of Bran Bait—A Case Study

In 1990, Foster et al. (unpubl.) conducted a detailed followup study to their 1989 work to determine if greater control of grasshoppers could be achieved with the application of higher dosages or multiple applications of insecticidal baits. In this study, the investigators applied flaky wheat bran containing carbaryl at 2 percent by weight to 40-acre, mixed-grass rangeland plots in North Dakota. The baits were applied with a Cessna Ag Truck operating at an altitude of 40–60 ft at 115 miles per hour (mi/hour) and equipped with a standard Transland 20244 spreader. Swath widths were 45 ft.

Three sets of plots received a single application of the carbaryl–bran bait at either 1.5, 3, or 4.5 lb/acre. One set of plots was treated with two successive applications of 1.5 lb/acre, and another set was treated with three successive applications of 1.5 lb/acre. The repeated treatments were applied 3 days apart. A final set of plots was left untreated. The six treatments were arranged in a randomized block design with four replicates per treatment. Pretreatment densities were used as the blocking variable. When the initial applications were made June 20–22, about 80 percent of the grasshoppers were in the nymphal stage.

The test showed that high dosages of the carbaryl–bran bait (3 and 4.5 lb/acre) caused greater reductions in grasshoppers after 2 days compared with the 1.5-lb/acre dosage (fig. II.15–1). The highest dosage, 4.5 lb/acre, caused a 48-percent reduction in populations of total grasshoppers after 2 days. Mortality in the single-application plots increased by an additional 7–14 percent after 7 days, perhaps because healthy grasshoppers cannibalized infected individuals.

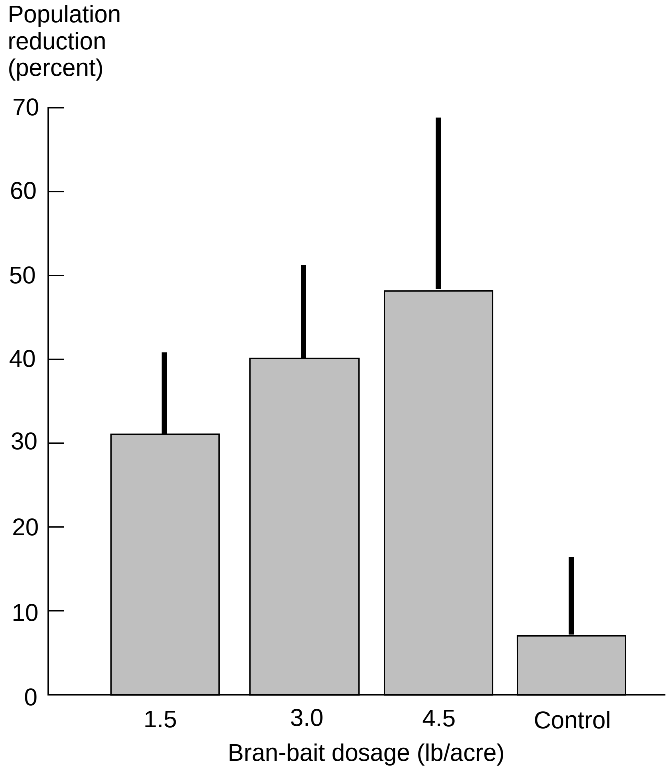

Figure II.15–1—Mean percent reduction in total grasshoppers after 2 days in plots treated with 1.5, 3, and 4.5 lb/acre of bran bait. Sample sizes for the 1.5, 3, 4.5, and control treatments were 12, 4, 4, and 4 plots, respectively. Bars indicate 1 standard error of the mean (SEM).

Successive applications of the insecticidal bait at 1.5 lb/acre caused progressive reductions in total grasshoppers (fig. II.15–2). For example, densities of grasshoppers declined by 52 percent in plots receiving the initial application of the 1.5 lb/acre treatment and declined by another 32 percent after the second application. The third application had no effect on grasshoppers.

Although repeated applications of insecticidal baits or higher dosages increased grasshopper mortality after 2 days, there was no difference in the effects of these treatments compared with a single application of 1.5 lb/acre after 7 days (fig. II.15–3). All treatments caused similar reductions after 7 days, whereas densities did not change in the control plots. Final densities of grasshoppers ranged from 6.3 to 15 per square yard in the treatment plots and were 23.8 per square yard in the control plots.

Uses of Multiple Applications of Insecticidal Baits

Foster et al. (unpubl.) found that multiple applications of 1.5 lb/acre had no real advantage over a single application at 1.5, 3, or 4.5 lb/acre. However, bran baits applied at lower dosages may be quickly consumed by a subset of grasshoppers and other insects, resulting in less control of some grasshopper species. Although there is a general relationship between the amount of bait applied and grasshopper mortality (see the chapter on multiple concentrations and rates of carbaryl–bran bait in this section), more bait is not necessarily better. Lower rates can give adequate control, particularly when grasshopper densities are relatively low (less than 25 per square yard).

Summary

The rather modest degree of overall control achieved by the insecticidal bait treatments in these tests was a result of the species composition of grasshoppers (fig. II.15–3). The presence of a high proportion of bran-rejector species diluted the effect of the treatments on total densities of grasshoppers. For example, treatments had no effect on *Aeropedellus clavatus*, the second most abundant species of grasshopper in the study plots. In contrast, treatments caused up to 96-percent reductions in densities of the most abundant species, *Aulocara elliotti*, a species that is known to consume baits.

An increase in the amount of bait can increase grasshopper mortality slightly, but this added control is not likely to be economical in many situations (see section II.3, “Sprays *versus* Baits”). Under certain conditions, however, it may be useful to increase the dosage of bran bait. For example, higher dosages can be used if the goal is to obtain high levels of grasshopper mortality (greater than 80 percent) in environmentally sensitive areas where insecticidal sprays cannot be used. These sensitive areas may include riparian habitats or sites with endangered plant and animal species.

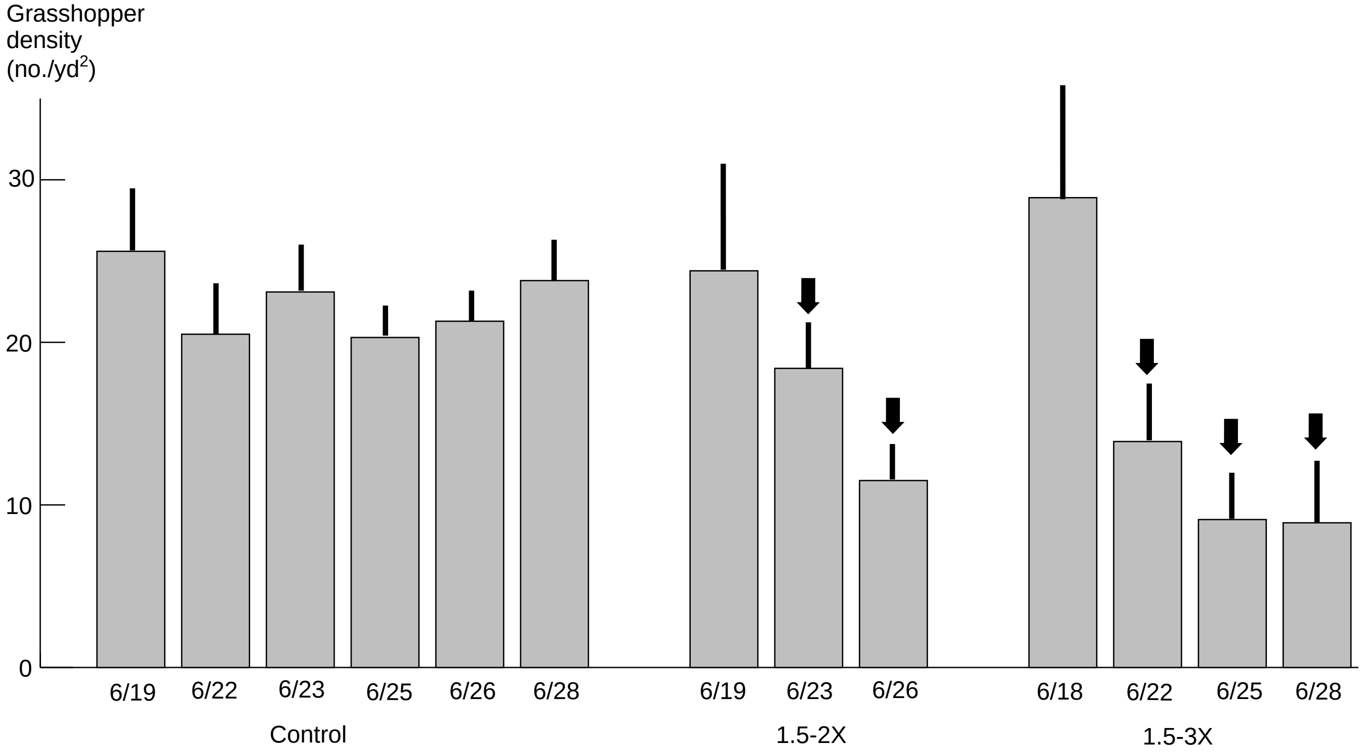

Figure II.15–2—Grasshopper densities (number/yd^2) in plots left untreated (control), treated two times with 1.5 lb/acre (1.5-2X), and treated three times with 1.5 lb/acre (1.5-3X). June 18–19 values represent pretreatment densities. Arrows indicate densities after treatments. Bars indicate 1 SEM.

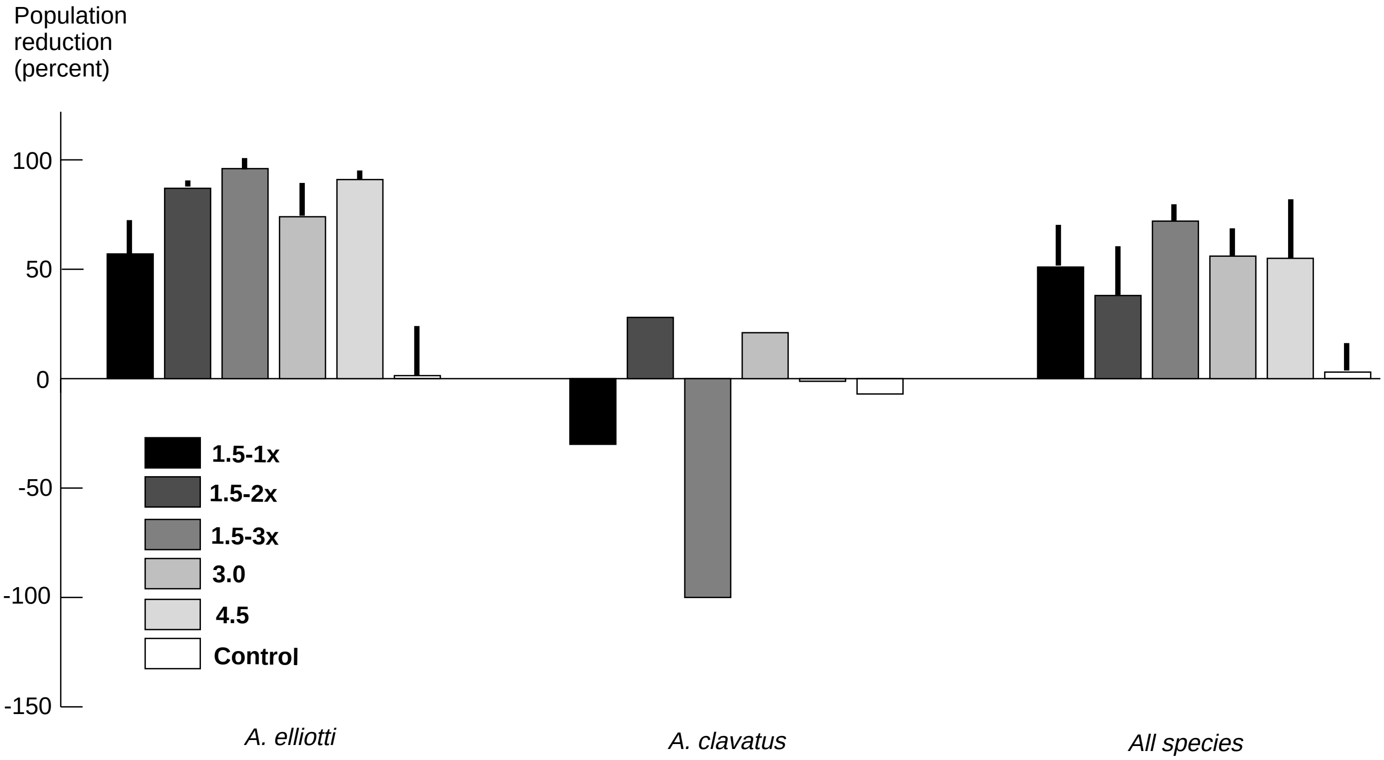

Figure II.15–3—Mean percent reduction in densities of *A. elliotti* (a bran acceptor), *A. clavatus* (a bran rejector), and all species combined, in treatment and control plots 7 days after initial treatments. A negative percent reduction indicates an increase in densities. Bars indicate 1 SEM. Standard errors for *A. clavatus* (not shown) ranged from 18.5 to 165.3.

References Cited

Ewen, A. B. 1990. A synthesis paper on the use of insecticides formulated as baits for grasshopper control in Canada and the USA. Boletin de Sanidad Vegetal (Fuera de series) 20: 83–89.

Henry, J. E. 1975. *Nosema locustae:* an alternative method of grasshopper control. In: Proceedings Advancements in Pesticides. Helena, MT: Montana Department of Health and Environmental Sciences: 16–28.

Jech, L. E.; Foster, R. N.; Colletto, D.; Walgenbach, D. D.; Roland, T. J.; Rodriquez, G. D.; Bohls, R.; Houston, R. D.; Meeks, W. K.; Queener, R. L.; Jackson, C. L.; Dines, J. L.; Puclik, J. L.; Scott, A. K. 1993. Field evaluation of diflubenzuron and carbaryl bran baits against grasshopper (Orthoptera: Acrididae) populations in South Dakota. Journal of Economic Entomology 86: 557–565.

Mukerji, M. K.; Ewen, A. B.; Craig, C. H.; Ford, R. J. 1981. Evaluation of insecticide-treated bran baits for grasshopper control in Saskatchewan (Orthoptera: Acrididae). Canadian Entomologist 113: 705–710.

Quinn, M. A.; Kepner, R. L.; Walgenbach, D. D.; Bohls, R. A.; Pooler, D.; Foster, R. N.; Reuter, K. C.; Swain, J. L. 1989. Immediate and 2nd-year effects of insecticidal spray and bait treatments on populations of rangeland grasshoppers. Canadian Entomologist 121: 589–602.

Quinn, M. A.; Kepner, R. L; Walgenbach, D. D.; Foster, R. N.; Bohls, R. A.; Pooler, P. D.; Reuter, K. C.; Swain, J. L. 1990. Effect of habitat and perturbation on populations and community structure of darkling beetles (Coleoptera: Tenebrionidae) on mixed-grass rangeland. Environmental Entomology 19: 1746–1755.

Quinn, M. A.; Kepner, R. L.; Walgenbach, D. D.; Bohls, R. A.; Pooler, P. D.; Foster, R. N.; Reuter, K. C.; Swain, J. L. 1991. Habitat characteristics and grasshopper community dynamics on mixed-grass rangeland. Canadian Entomologist 123: 89–105.

Reference Cited—Unpublished

Foster, R. N.; Quinn, M. A.; Reuter, K. C.; Jech, L. E.; Colletto, D.; Houston, R.; Puclik, M. J.; Scott, A.; Radsick, B. Comparison of single and multiple applications of carbaryl bran bait for controlling grasshoppers (Orthoptera: Acrididae) on rangeland. Crop Protection (in review).

II.16 Improving the Economics of Grasshopper Bait Application: Efficacy and Swath Comparison of an Experimental and Standard Aircraft Spreader

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W. Meeks, C. Jackson, J. Patterson, K. C. Reuter, and Mike W. Sampson

Using solid baits, particularly carbaryl–wheat bran bait, for controlling or suppressing grasshoppers on rangeland has gained renewed attention in recent years. During the 1950's, use of bait declined as use of effective small amounts of chemical sprays increased.

Renewed interest in the use of baits was a direct result of improvement in aerial application equipment and the development of calibration procedures that produced consistent results. Increasing concern for the environment and the environmental advantages inherent with baits over many chemical sprays spurred these improvements.

Grasshopper density management studies conducted in North Dakota in the mid-1980's relied on and successfully demonstrated these advances (Foster and Roland 1986). However, narrow swaths produced by the equipment used for aerial application of bait treatments in these studies demonstrated the competitive edge that was still associated with the wider swaths of aerially applied chemical sprays.

The narrow swath, while hindering the wide-scale use of baits from the air, led to the development and production of an experimental aircraft spreader with an improved swath width. Jack Henderson and the New Mexico State University designed and produced an improved spreader and incorporated further modifications during the late 1980's.

Field Studies

As part of the Grasshopper Integrated Pest Management (GHIPM) Project, we carried out field studies that looked at swath width, uniformity of bran flakes within the swath, and resulting efficacy of dispersed bait for grasshopper suppression on rangeland with the experimental spreader. During the tests, we used a Cessna Ag Husky for all flights with the modified experimental spreader. For studies with the standard spreader, a Transland 20244, a Cessna Ag Truck was equipped to prevent bridging (flow blockage) of the bran in the hopper and to promote uniform application (Foster and Roland 1986). We calibrated both spreaders according to U.S. Department of Agriculture (USDA), Animal and Plant Health Inspection Service (APHIS) guidelines for aerial contractors.

Bait was the D-Bug® Ag (Sidwell Enterprises, Inc., Parker, CO) formulation of carbaryl and wheat bran grasshopper bait containing 2 percent carbaryl by weight. Bait was applied at 1.42 lb/acre for the experimental spreader and at 1.54 lb/acre for the standard (Transland 20244) spreader.

Efficacy in the Field.—There were four treatment blocks of mixed-grass rangeland for each spreader trial. Pilots flew the blocks on July 19, 1989, northeast of Edgemont, SD. Application with the standard spreader was at 127 miles per hour (mi/hour) at an altitude of 50–75 ft with a

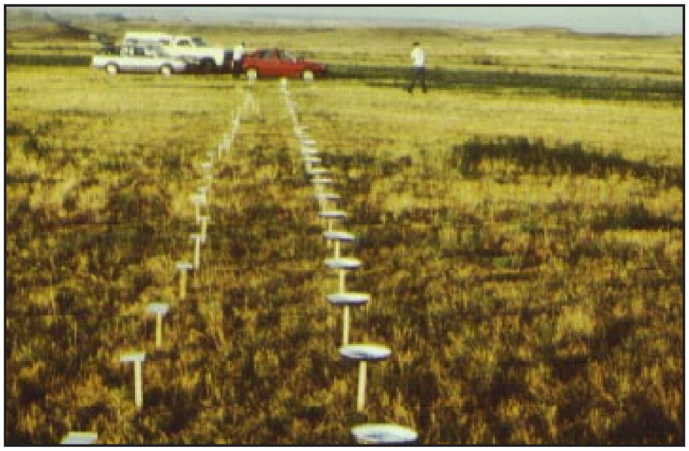


Figure II.16–1—Adhesive card and aluminum pan collection devices used to evaluate swath width and uniformity of application for the aircraft spreaders used in applying bran bait.

Figure II.16–2—Cessna Ag Husky with experimental bran bait spreader.

Figure II.16–3—Commercial Turbine Thrush with Transland 20244 standard spreader.

working swath of 45 ft. Application with the experimental spreader was at 120 mi/h at an altitude of 70–100 ft with a working swath of 100 ft. These swath assignments were based on widths determined in earlier studies with the standard and experimental equipment. When sprays are used, these aircraft are assigned working swaths of 75–100 ft depending on the type of formulations (USDA, APHIS 1994).

We measured grasshopper densities before and after treatment using 40 0.1-m^2 rings developed by Onsager and Henry (1977). Grasshopper densities from four untreated plots were used for comparison to determine natural change in the grasshopper population during the study and for comparison to treated populations. Posttreatment population levels were compared with pretreatment levels to determine the effectiveness of the bait to reduce grasshopper populations as dispersed by both spreaders.

Comparison of Swaths.—Another set of trials compared the uniformity and widths of swaths of the standard and experimental spreaders. Adhesive cards (unfolded sticky pink bollworm traps) (Foster et al. 1977) and aluminum cake pans collected particles of bran bait dispensed during the test flights. The total number of particles collected for each card or pan was converted to particles of bait per square foot to determine the uniformity of the swath, overall swath width, and effective or working swath width. Flights for these trials occurred on July 20, 1989, at an altitude of 30 ft. This altitude was chosen because the investigators were looking for information that might also be of use if bait were used on crops in the future. Applications on cropland typically occur at lower altitudes than on rangeland. Other flights at higher altitudes were studied to determine the effect of altitude on the uniformity of bait within the swath.

Among organizations or individuals who deal with aircraft applications, there is no widely accepted specific method or criteria for assigning operational swath widths. In this study we defined “effective swath width” as the width where collection devices captured at least 73 percent of the number of bran flakes expected per square foot. Extraordinary reductions in the rate of bran deposited took place when less than 73 percent of the expected rate actually did fall to the ground.

Results.—Pretreatment grasshopper densities ranged from 11.8 to 25 per square meter and averaged 20.2 grasshoppers/m^2 in the experimental spreader plots. In the standard spreader plots, grasshoppers ranged from 18.8 to 42.5 per square meter and averaged 27. Grasshoppers in the untreated check plots ranged from 20.3 to 29 and averaged 24.5 per square meter. The grasshopper density in the untreated check plots decreased .01 percent per day during the course of the study because of natural mortality.

At 24 and 48 hours after treatment, trials with both spreaders resulted in reducing grasshoppers below the general 1989 APHIS action level in 1989 of 8 per square yard (9.6 per square meter). There was no significant difference in grasshopper mortality between the spreaders (table II.16–1).

When compared to the standard spreader at an application altitude of 30 ft, the experimental spreader provided a significantly wider swath. Both the pan and adhesive-card particle collectors showed increases in overall and effective swath width (table II.16–2).

The experimental spreader showed an increase of between 125 and 132 percent for overall swath width and between 113 and 140 percent for effective swath width. Such significant increases strongly suggest that using the experimental spreader would make the choice of bait control more cost effective.

Table II.16–1—Efficacy of 2% carbaryl bran bait on grasshoppers when aerially applied with a standard Transland spreader and an experimental spreader near Edgemont, SD, 1989 (replicated 40-acre blocks)

Spreader	Application rate	Mean percent control at indicated interval after treatment[1] 2 days	4 days
	(Lb/acre)	*(Percent)*	
Experimental	1.42	39.4a	54.7a
Standard	1.54	41.7a	57.4a

[1]Adjusted for untreated check. Means followed by the same letter in a column do not differ significantly at the 5% level of confidence (Duncan's new multiple-range test).

Table II.16–2—Mean[1] swaths (overall and visual effective) of experimental and standard dry-material aircraft spreaders with aluminum pan and adhesive card collection devices (flown at 30-ft altitude)

	Swaths			
	Overall		Effective	
Spreader	Pan	Card	Pan	Card
		(Ft)		
Standard	60b	50b	35b	39b
Experimental	135a	116a	84a	85a

[1]Means in a column followed by the same letter do not differ significantly at the 5% confidence level (Mann–Whitney test).

The standard spreader demonstrated greater uniformity of bran bait particles at 30 ft within the effective swath than did the experimental spreader. At higher altitudes, the experimental spreader showed an increase in uniformity. This increase points to the need for more study that could show additional improvements in bait economics.

Key Findings and Conclusions

- Spreaders can be built that work with swaths equal to those used for liquid applications.

- The experimental spreader produced a working swath 2.2 to 2.4 times that of the standard spreader from an application altitude of 30 ft.

- Adhesive-card particle collectors accounted for a greater number of particles per square foot than did pan collectors. Cards also are more convenient to use.

- At an application altitude of 30 ft, the standard spreader gave greater uniformity of bran bait deposited than did the experimental spreader. With minimal improvement, the experimental spreader could offer increased uniformity.

- Using the experimental spreader at higher altitudes improved uniformity of depositing bait and may increase swath widths.

- Both spreaders performed equally well in terms of rangeland grasshopper control with baits.

- The experimental spreader was efficient and was an economical improvement compared to the standard spreader.

For More Information

A detailed report on the comparison of a standard and experimental aircraft spreader for bran bait is available from the USDA, APHIS, Methods Development Center, 4125 E. Broadway Road, Phoenix, AZ 85040. The report includes data on grasshopper species composition before and after treatment, grasshopper collection procedures, and techniques for determining density, swath overlap, particle-count data, and effects of aircraft altitude on bait coverage.

Selected References

Foster, R. N.; Roland, T. J. 1986. Grasshopper suppression: techniques for ultra low volume applications of carbaryl wheat bran baits. Bismarck, ND: North Dakota Cooperative Extension Service, North Dakota Agricultural Experiment Station, North Dakota Department of Agriculture: 68–73.

Foster, R. N.; Staten. R. T.; Miller, E. 1977. Evaluation of traps for pink bollworm. Journal of Economic Entomology 70: 289–291.

Onsager, J. A.; Henry, J. H. 1977. A method for estimating the density of rangeland grasshoppers (Orthoptera: Acrididae) in experimental plots. Acrida 6: 231–277.

USDA, APHIS. 1994. Prospectus No. 73-M-APHIS-94 for aerial application.

Selected Reference—Unpublished

Foster, R. N;, Walgenbach, D. D.; Henderson, J. A.; Rodriguez, G.; Jech, L. E.; Colletto, D.; Meeks, W.; Jackson, C.; Patterson, J.; Reuter, K. C. 1990. Improving the economics of grasshopper bait application: efficacy and swath comparison of an experimental and standard aircraft spreader. In: Cooperative Grasshopper Integrated Pest Management Project, 1989 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 173–183.

II.17 Efficacy of an Extended Swath With Carbaryl–Bran Bait

K. Christian Reuter, R. Nelson Foster, and Wendal J. Cushing

During 1992 and 1993, the U.S. Department of Agriculture's Animal and Plant Health Inspection Service (APHIS) conducted two separate studies each year, aerially treating separate rangeland areas with 2 percent carbaryl–bran bait at the rate of 1.5 lb/acre. In each study, a 45-ft application swath was compared to a 90-ft swath. APHIS attempted to create a 90-ft swath by increasing the aircraft's application height from 75 ft to 150 ft. Accordingly, the bait flow rate was increased to a level that maintained an application rate of 1.5 lb/acre. In theory, these adjustments would result in an increased swath (of the drifting bran bait), reducing the number of passes required by the aircraft to treat the acreage.

In 1992, APHIS applied bran bait at two sites in the Grasshopper Integrated Pest Management Project demonstration area in McKenzie County, ND. The treatment areas were approximately 1,085 acres with the 45-ft swath and 1,500 acres with the planned 90-ft swath in a location designated as the "Mead area." APHIS also treated about 1,740 acres with the 45-ft swath and about 1,753 acres with the planned 90-ft swath in a location designated as the "Crighton area." Ring counts and sweep-net samples at 10 sites in each of the treated and untreated areas were used to find grasshopper densities and species composition (see chapter II.2).

Mortalities resulting from the two swaths were not statistically different in the Mead area except at 4 days after treatment, where the 90-ft swath was superior. Results in the Crighton area showed that the 90-ft swath was statistically superior each time.

Upon examining the grasshopper species composition in the treatment areas, we noted that with the 45-ft swath in the Crighton area the dominant species was *Phlibostroma quadrimaculatum* at 24 percent of the pretreatment population. In the area treated with the 90-ft swath, this species accounted for only 9 percent of the pretreatment population. *P. quadrimaculatum* generally is a poor candidate for bran bait treatment as mortality is usually less than 25 percent (see chapter II.12 on bait acceptance). The higher proportion of a grasshopper species that does not readily eat bait in the 45-ft swath area may explain why the 90-ft swath consistently looked superior in the Crighton area.

In 1993, APHIS again applied bran bait at two sites in the demonstration area in McKenzie County. We treated 401 acres with the 45-ft swath and 408 acres with the 90-ft swath in a location designated as the "Corral Creek area." Also, we treated 422 acres and 425 acres with the 45-ft and 90-ft swaths, respectively, in a location designated as the "Wolf Coulee area."

Field personnel used ring counts and sweep-net samples at 10 sites in each of the treated and untreated areas to figure grasshopper densities and species composition. In both study areas, we found no statistical differences between the 45-ft and 90-ft swath at any time. In these studies, grasshopper species composition was very consistent between the treatment areas, containing dominant species that are susceptible to bait treatments.

These studies suggest the possibility to reduce aerial application costs with carbaryl–bran bait by increasing the application height and the bait flow rate to achieve an extended swath. It is certain that we did not get uniform coverage over the entire 90-ft swath. Visual observations in 1992 and 1993 showed the increased flight height only slightly widened the swath, and the bait did not cover the entire 90 ft. The data imply that, although the coverage was not uniform, the untreated gaps between swaths were compensated for by movement of grasshoppers to find sufficient particles of bait. Under different circumstances, gaps in bait coverage may or may not result in mortality equivalent to a uniformly covered application.

II.18 Equipment Modification, Swath Width Determination, and Calibration for Aerial Application of Bran Bait With Single-Engine Fixed-Wing Aircraft

R. N. Foster and T. J. Roland

Under certain conditions, bran bait is the best choice for controlling grasshoppers. Bait is commonly applied by ground equipment, but in many cases, rough terrain and/or extensive acreage make application by air necessary. Until recently, the acceptance of aerial application of bran bait has been hindered by the common occurrence of nonuniform application and the difficulty in calibrating the equipment accurately. Both problems are caused by uneven flow of bait from the hopper of the aircraft to the spreader.

This uneven flow usually results from what is commonly referred to as "bridging"—the formation of both a cavity in the lower portion of the bait load and an overlying bridge of bait. As bait flows from the bottom of the hopper to the spreader, the load in the hopper settles. Because the particles of bait are flat, they tend to overlap, layer, and lock together to form a bridge. That portion of the bait load that does not lock together flows to the spreader and is applied and leaves a cavity under the bridge. If the overlying bridge does not break and fall before all of the lower bait is applied, continuous flow of bait will be interrupted and nonuniform application will result.

Over several years, Foster and Roland (1986) solved these problems and demonstrated that bridging can be prevented so uniform aerial application is feasible. Nonuniform flow of bait can be detected by observation from the ground. If during application the observer watches the tips of the spreader and notices puffing or uneven flow of bran, bridging is probably occurring. This chapter will detail the required equipment modifications and procedures for establishing swath widths and consistent calibration and will identify potential problems commonly encountered during calibration and aerial application of bran baits.

Equipment Fabrication and Modification

Aerial application of bait requires the use of what are commonly called granular spreaders. These spreaders are used for aerial application of dry solid materials, such as fertilizers, herbicides, and seeds. Several different spreaders are available commercially, and some acceptable homemade types undoubtedly exist. To ensure a uniform application, each type of spreader must be evaluated with the type of aircraft on which it will be used. To date, the U.S. Department of Agriculture (USDA), Animal and Plant Health Inspection Service (APHIS) has evaluated and approved several aircraft and spreaders for aerial application of bran baits (table II.18–1).

Uniform flow of dry bait is a function of several factors, including the slope of the aircraft hopper, the physical shape (flatness) of the bait particles, the size of the opening of the gate seal assembly through which the bran is released from the hopper of the aircraft into the spreader, and the small amount of bait per acre that is usually desired for delivery. All of these factors contribute to bridging, which prevents a consistent and uniform flow of bait from the aircraft hopper to the spreader.

Three inexpensive, simple additions and modifications to the aircraft are required to ensure uniform delivery of bait. A ram air agitation system—consisting of a ram air tube, air agitation tube, and a vent tube air regulator—must be adapted to the aircraft.

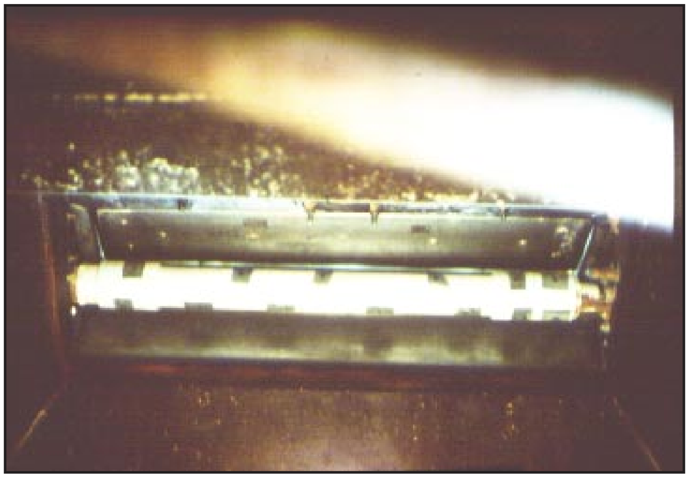
Air Agitation Tube

This tube directs air forced from the ram air tube to the inside lower area of the hopper. The moving air is forced up toward the bottom of the bait load and agitates the bait particles to prevent bridging. In addition, the air mixes with the bait particles to allow a uniform flow of material to the spreader.

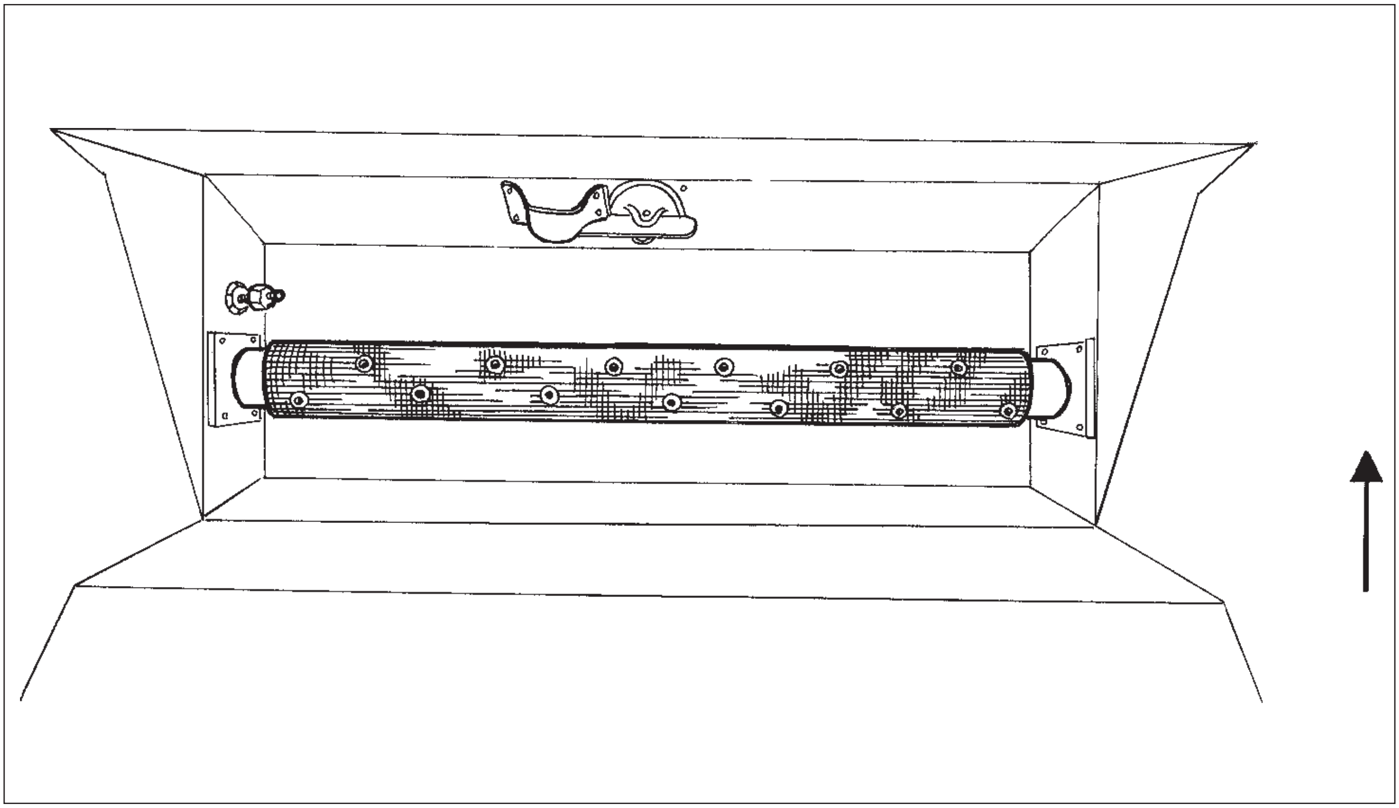
Table II.18–1—Aircraft/spreader combinations that have been certified and swath widths assigned for applying wheat bran bait

Aircraft make/model	Spreader make/model	Altitude	Swath
		(Ft)	
Cessna 188	Transland 20241/20244	50	45
Turbine Thrush	Transland 20250	50	45
Bull Thrush	Transland 22007	100	100

The air agitation tube can be built using Federal Aviation Administration-approved pipe and fittings. The pipe size shall have an inside diameter 1 to 1.5 in and shall be installed across the entire width of the hopper throat just above the gate opening (figs. II.18–1 and –2). A series of 1/4-inch-diameter, equally spaced holes is drilled across the upper side of the pipe and alternately angled to direct airflow to the fore and aft lower portion of the hopper walls. The number of holes can vary, but their accumulated area must not exceed 75 percent of the pipe's inside diameter area. Therefore, a 1-inch-diameter pipe should not have more than 12 holes, and a 1.5-inch pipe should not have more than 27 holes. All 1/4-inch holes are covered with window screen to prevent the entry of material into the air agitation tube.



Figures II.18–1 and –2—Air agitation tube installed across entire width of the aircraft hopper throat just above the gate opening.

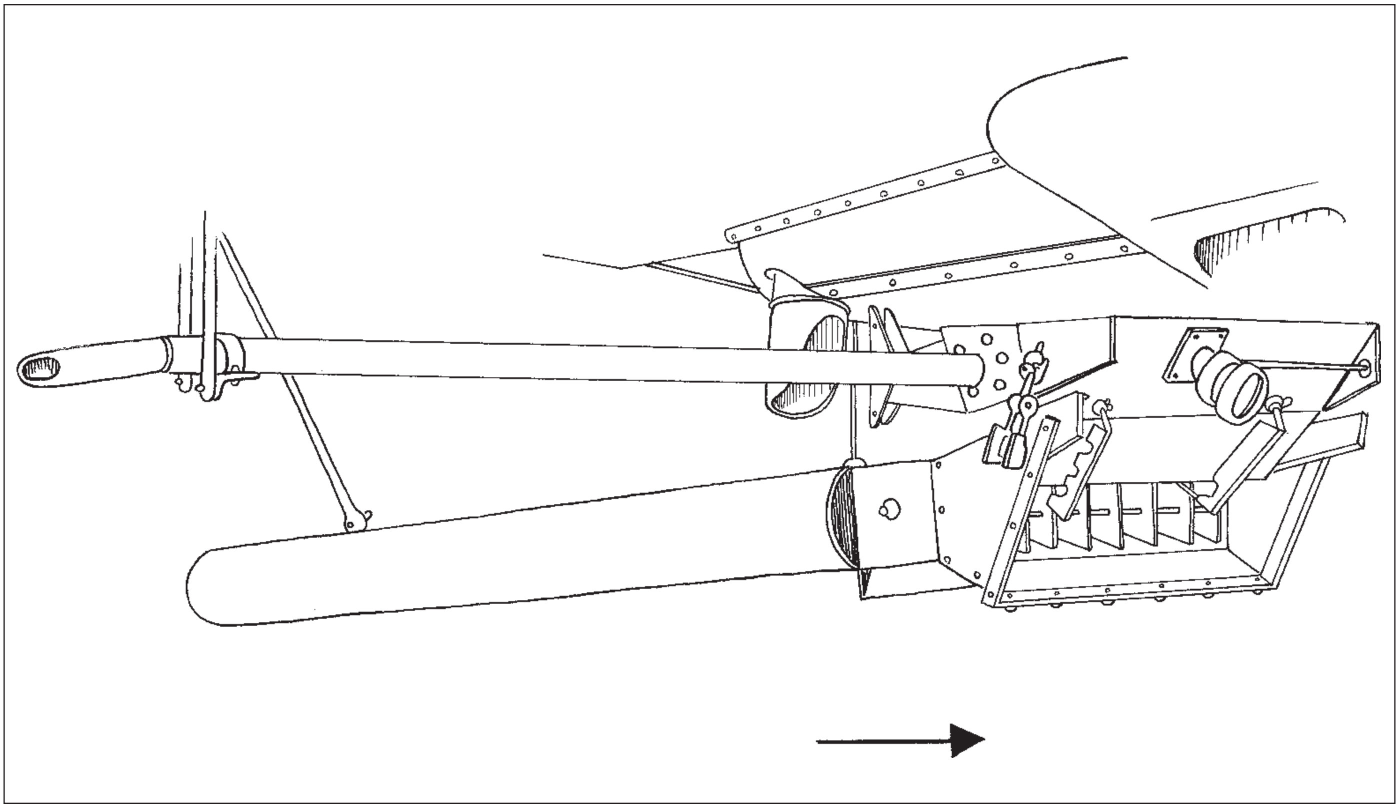


Ram Air Tube

This tube collects and directs forced air from outside the aircraft into the air agitation tube located in the bottom of the aircraft hopper. This supply of forced air can be provided in one of two ways.

1. Insert a pipe through the side opening of the hopper subtank with the spray valve removed and position the open end forward at approximately a 45-degree angle to the slipstream to allow for uninterrupted ram air during flight. The opposite end of the air agitation tube inside the hopper must be tightly sealed (figs. II.18–3 through –5).

Figures II.18–3 and –4—Ram air tube fastened to underside of aircraft provides forced air during flight to the air agitation tube.



2. Install a pipe tee at the proper location in the agitation tube and insert a pipe through the opening that supplies the pump for spray operations. Position the open end forward to allow for uninterrupted ram air during flight (fig. II.18–6). When this modification is used, the ends of the air agitation tube inside the hopper must be tightly sealed (fig. II.18–7).

Figure II.18–5—Ram air tube and air agitation tube before installation on aircraft.

Figure II.18–6—Front-mounted ram air tube for providing forced air to the air agitation tube during flight.

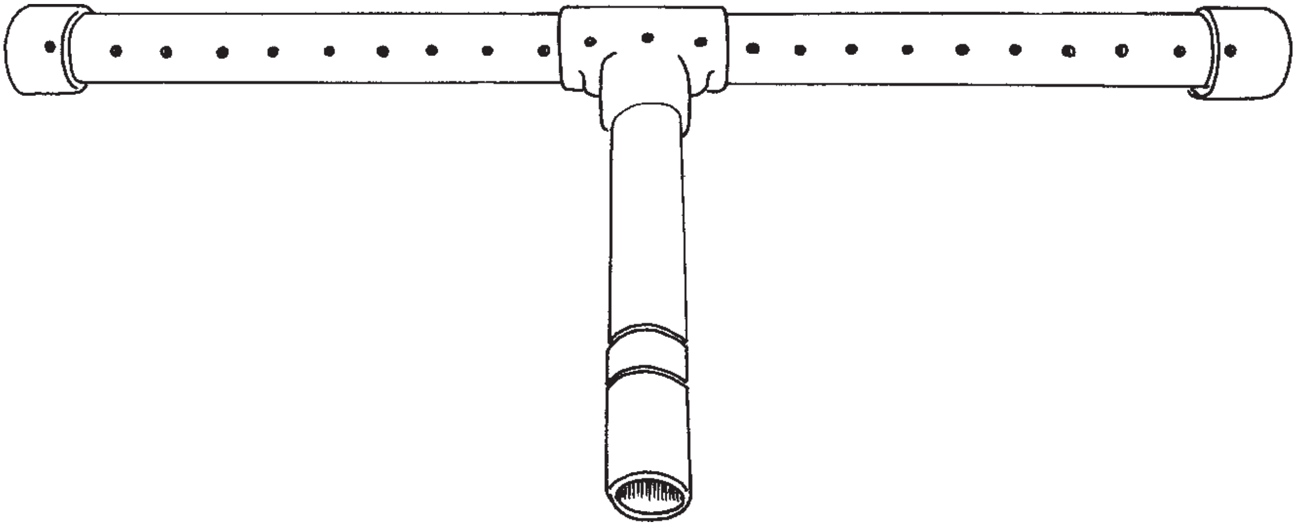


Figure II.18–7—Air agitation tube with both ends sealed when used with front-mounted ram air modification.

Vent Tube Airflow Regulator

The existing hopper vent tube can be modified easily to function as a flow regulator for the bait. The flow regulator works on the same principle as two holes in the top of an oil-can. When fluid is poured out of one hole, the opposite hole serves to prevent a vacuum from building up in the can. In the aircraft system, the hopper opening is similar to the pour hole in an oil-can. The vent tube is similar to the second hole in the oil-can. By simply restricting the amount of air that is allowed to enter the hopper vent tube, one can reduce the speed that bran is delivered through a fixed hopper-gate opening. Very minor changes in the amount of air allowed into the vent tube can cause major changes in the amount of bait delivered.

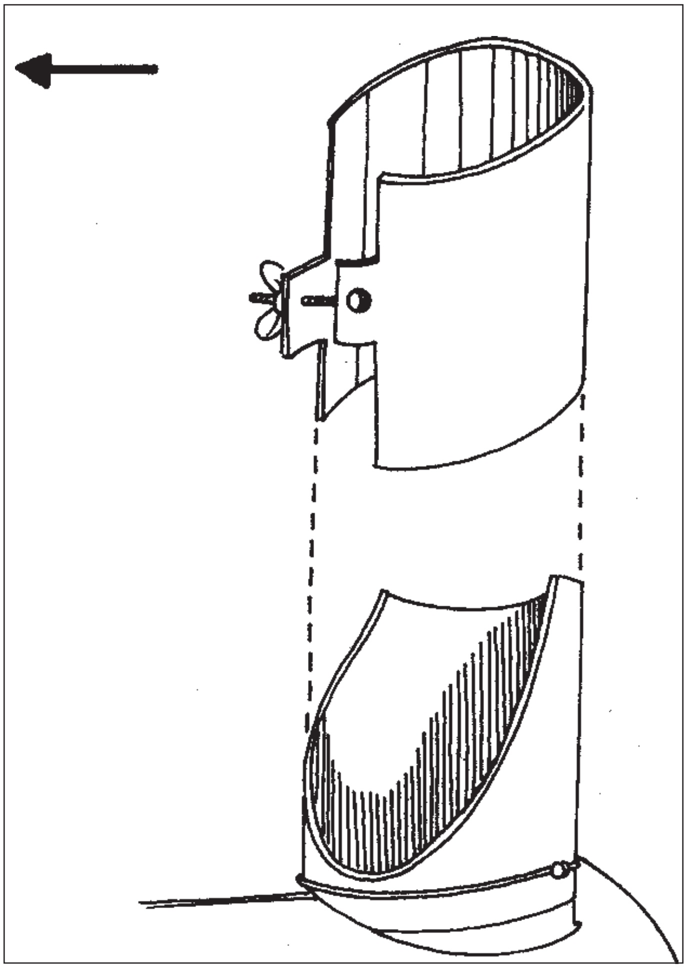


Figure II.18–8—A vent tube flow regulator fashioned from sheet metal is used to adjust the air flow through the vent tube to the aircraft hopper.

A sheet metal sleeve is fashioned and attached to the vent tube to allow adjusting the airflow through the vent tube to the aircraft hopper (fig. II.18–8). Other materials or duct tape can be used to produce similar results.

Other Requirements

The aircraft hopper-gate seal must be clean, dry (not sticky), and in good condition across its entire length to prevent an accumulation of material along the seal and edge of the gate when it is opened. An accumulation of bait on the gate seal can prevent uniform distribution into the spreader and, in some cases, can even promote bridging in the hopper. Linkage between the gate and its cockpit control handle must be in good condition or the gate may not stop in the same position each time it is opened. Gate stops are also required to ensure that the hopper gate is opened to exactly the same position each time. Screw-type stops are preferred.

Seal all openings where the ram air tube enters the subtank of the hopper. Doing this prevents leakage of bait from the aircraft and ensures a sufficient and constant amount of air entering the air agitation tube.

Remove all mechanical agitation components, nonstructured baffles, and other nonstructured obstructions from the hopper interior. Any unnecessary object can act as an anchor for the buildup of bait and thus promote bridging.

If present, the side-loader flapper valve inside the hopper should be sealed and covered to reduce protrusions. Doing that prevents dry material from entering the system when used for liquid application. Covering all protrusions reduces the chance of material buildup, which can promote bridging. The hopper interior must be thoroughly clean and dry to prevent the buildup of bait.

Determining Swath Width

The swath width for both liquid and dry bait applications will differ among types of aircraft. With baits, different types of spreaders on the same type of aircraft can produce different swath widths. Other differences among the aircraft, such as landing gear configuration, automatic flagman equipment, and weight, may also result in different swath widths.

Any combination of aircraft, spreader, and spreader attachments that has not been previously evaluated for swath widths must be characterized. (That is, a detailed study of the uniformity of particle deposition must be made.)

The hopper interior must be completely dry before loading the bait. A proven technique for ensuring this is to fly the aircraft for several minutes with the hopper empty and the hopper gate open.

Load a sufficient amount of bran bait into the hopper to conduct swath evaluations. For determining the swath width, the rate of bait flow (application rate) is unimportant as long as bait being dispensed by the aircraft can be seen in the air by observers from the ground. The hopper gate opening should be set wide enough to make certain that bridging is not occurring. A setting that allows for a gate opening of $^1/_4$ inch or more is usually sufficient.

Conduct swath evaluations in a relatively flat area free of obstructions. Collection devices, such as pans, paper plates, or sticky cards, should be placed in a line 200 ft long perpendicular to the planned flightline. Place collection devices at 5-ft intervals along the line.

Conduct all flights to determine swath widths during no-wind conditions or by flying into a wind that does not exceed 5 miles per hour (mi/hour). The aircraft must be in level flight and at the proper operating speed and altitude for at least 1,000 ft before reaching collection devices. To ensure that bait will hit the collection devices, open the hopper gate at 500 ft before reaching the collectors and leave it open until the aircraft has passed the devices by 1,500 ft.

After each flight, inspect all collection devices and count and record the number of particles in each device. The overall swath width is the distance between the extreme collection devices that caught at least 1 particle of bait. Collection devices in the middle portion of the overall swath will contain many more particles than the devices on either end.

In many cases, the overall swath width ends abruptly on either end and is very obvious. The effective or working swath width (overall swath width minus 10 ft) is the swath width that will be used in the calculations for calibration and during the actual application. The difference between the effective swath and the overall swath is the amount of overlap that will occur during application. Where abrupt ends are not obvious, calculate the average number of particles in the heaviest portion of the swath. For the amount of material being applied on a particular test flight, the average number is the desired amount of material that should be reaching the target. Working toward the extremes of the overall swath, the points are marked at which you find about half of the average number of particles. The distance between these two points is the usable working swath width. At least three good swath-width test flights are recommended.

Calibration

Calibration is simply comparing the amount of material that was applied to a given area for a given period of time during a test flight with what is desired to be applied to that area. Make adjustments in the system until agreement is reached. The wheat bran calibration worksheet at the end of this chapter will be helpful in determining calibration.

After determining the swath width and the groundspeed of the aircraft, determine the number of acres that will be treated in a minute. To do this, multiply the groundspeed times the swath width and divide by 495 (a constant). For example, 120 mi/hour times an 80 ft-swath divided by 495 equals 19.39 acres/min (table II.18–2). By multiplying the acres per minute times the amount of bait desired per acre, you can determine the amount of bait that should be applied in 1 minute. For example, if 1.5 lb of bait per acre is desired, then from the above example, 1.5 lb times 19.39 acres/minute equals 29.09 lb of bait, the amount that should be applied in 1 minute.

For the first flight, the gate opening should be set at $^1/_4$ inch. The shank of a $^1/_4$-inch drill bit can be used as a gauge. You will need an apparatus to drain and recover wheat bran from the aircraft hopper and a scale to weigh the bait. Weigh the bait to be loaded into the aircraft. Actual weight may vary slightly from that printed on the

Table II.18–2—Matrix to determine the number of acres treated per minute

	Working swath width (ft)								
Flying speed	50	55	60	65	70	75	80	90	100
Mi/hour	*Acres/min*								
75	7.58	8.33	9.09	9.85	10.61	11.36	12.12	13.64	15.15
80	8.08	8.89	9.70	10.51	11.31	12.12	12.93	14.54	16.16
85	8.59	9.44	10.30	11.16	12.02	12.88	13.74	15.45	17.17
90	9.09	10.00	10.91	11.82	12.73	13.64	14.55	16.36	18.18
95	9.60	10.56	11.52	12.47	13.43	14.39	15.35	17.27	19.19
100	10.10	11.11	12.12	13.13	14.14	15.15	16.16	18.18	20.20
110	11.11	12.22	13.33	14.44	15.56	16.67	17.78	20.00	22.22
120	12.12	13.33	14.55	15.76	16.97	18.18	19.39	21.82	24.24
130	13.13	14.44	15.76	17.07	18.36	19.70	21.01	23.64	26.26
140	14.14	15.56	16.97	18.38	19.80	21.21	22.63	25.45	28.28
150	15.15	16.67	18.18	19.70	21.21	22.73	24.24	27.27	30.30

Note: If the above table does not list the swath width or speed, use the following formula to determine acres per minute:

$$\frac{\text{Aircraft groundspeed (mi/hour)} \times \text{Swath width (ft)}}{\text{495 (a constant)}} = \text{Acres per minute}$$

bag. Use the actual measured weight. Load the hopper with approximately 50 lb of bait plus the amount of bait to be applied in 1 minute to ensure that you will not run out of bait during the calibration flight. If there is no bait left in the hopper after a flight, overapplication was occurring; appropriate adjustments must be made, and the flight must be repeated.

Make all calibration flights crosswind and dispense bait for 1 minute. Flying upwind will increase the rate of application, and flying downwind will decrease the rate of application. Use a stopwatch to determine the exact amount of time the hopper gate is open. Timing devices attached to the application system may increase the accuracy.

After the first calibration flight, drain and weigh all bait remaining in the hopper. Make sure bait that may have fallen into the spreader during draining is included. Subtract this weight from the weight loaded. Compare the amount of bait applied to what was desired to be applied. If the application rate per minute is below the desired rate, increase the gate opening and conduct another calibration flight.

If the application rate per minute exceeded the desired rate, do not change the gate opening. Cover about half of the hopper air vent. Use the fabricated airflow regulator or duct tape. Reducing or enlarging the vent opening changes the internal pressure in the hopper, decreasing or increasing the flow rate, respectively. Make a second calibration flight.

If after the second flight the flow per minute still exceeds the desired rate, further reduce the vent opening and conduct another calibration flight. Do this until the application rate equals the desired rate. Calibration accuracy should be within 10 percent of the desired rate. A minimum of five consecutive acceptable calibration flights at the same settings will assure accurate application.

Safety and Storage

Before initiating a treatment for grasshoppers or Mormon crickets with wheat bran bait, always read the label carefully. Keep wheat bran bait dry during storage in enclosed buildings, trailers, or vans to eliminate the risk of the bait's becoming unusable. Also, keep bait in a cool location. Hot storage for long periods of time may cause the bait to become rancid and reduce its effectiveness. Dispose of empty bags or containers according to State and Federal regulations printed on the label.

Potential Problems

The following lists identify some of the problems most commonly seen to occur with calibration and application of wheat bran baits.

Equipment

- Improper or no modifications or fabrication.
- Nonstructural hopper baffles not removed.
- Airholes not covered with screen on agitation tube.
- Hopper gate seal not clean and dry.
- Side-loader flapper valve inside hopper not sealed.
- Air and agitation tube connection and alignment not proper.
- Loose gate linkage.
- Gate-setting stop not in place.
- Gate-setting screw jack moves.
- Hopper doors not covered during rain.

Material

- Lumps in bait from commercial formulation.
- Strings and/or paper in bait from the container or bag.
- Rocks, pebbles, or other objects in bait.
- Clumped bait due to moisture.
- Weight printed on bag or container inaccurate.
- Different types of bran or bran sources.
- Different formulations of bait.

Methodology

- Failure to follow guidelines.
- Failure to open hopper gate firmly and consistently.
- Inaccurate weighing during calibration and application.
- Failure to read scales accurately.
- Bait left in throat of spreader when weighing during calibration.
- Bait left in hopper when weighing during calibration.
- Calibration loads inconsistent in weight.
- Unlevel load during calibration flights.
- Calibration runs not conducted crosswind.

Weather Conditions

- Damp or wet hopper due to condensation or rain.
- Calibration may change due to large humidity changes.

Conclusion

Accurate aerial application of wheat bran bait is no more difficult than applying chemical sprays. The problems associated with accurate calibration and consistent application of bran bait by air have been identified. Solutions to the problems and procedures for implementing the solutions have been developed and refined. Both solutions and procedures are inexpensive. With experience, accurate calibration and application of bran bait by air can now be expected.

Acknowledgments

The authors wish to thank Jack Henderson, retired USDA/APHIS chief pilot, whose early work and suggestions with baits were instrumental in arriving at the final design for ram and air agitation tubes. The authors are also indebted to Tim Lockley, USDA/APHIS, for illustrations used.

References

Foster, R. N.; Roland, T. J. 1986. Grasshopper suppression: techniques for ultra low volume applications of carbaryl wheat bran baits. Bismarck, ND: North Dakota Cooperative Extension Service, North Dakota Agricultural Experiment Station, North Dakota Department of Agriculture: 68–73.

USDA, APHIS, 1994. Prospectus No. 73-M-APHIS-94 for aerial application. Phoenix Methods Development Center. Phoenix, AZ.

Wheat Bran Calibration Worksheet

Date ____________________

Pilot ____________________

Aircraft make/model ____________________

Spreader make/model ____________________

Aircraft speed (mi/hour) ____________________

Assigned swath (ft) ____________________

Material applied ____________________

Desired rate per acre (lb) ____________________

Desired rate per minute (lb) ____________________

Acceptable range per minute (plus or minus 10 percent of desired)

Minimum ____________ lb

Maximum ____________ lb

Calibration Formula

(Speed _______ mi/hour × swath _______ ft) divided by 495 =
_______ acres per minute

Acres per minute ______ × rate per acre _____ lb =
_______ lb per minute

Calibration Worksheet, 6 replications

Load #____________________
Loaded ____________________ lb
Drained ____________________ lb
Applied ____________________ lb
Time ____________________ seconds
Rate ____________________ lb/acre
Percent ____________________ low–high
Adjustments:

Load #____________________
Loaded ____________________ lb
Drained ____________________ lb
Applied ____________________ lb
Time ____________________ seconds
Rate ____________________ lb/acre
Percent ____________________ low–high
Adjustments:

Load #____________________
Loaded ____________________ lb
Drained ____________________ lb
Applied ____________________ lb
Time ____________________ seconds
Rate ____________________ lb/acre
Percent ____________________ low–high
Adjustments:

Load #____________________
Loaded ____________________ lb
Drained ____________________ lb
Applied ____________________ lb
Time ____________________ seconds
Rate ____________________ lb/acre
Percent ____________________ low–high
Adjustments:

Load #____________________
Loaded ____________________ lb
Drained ____________________ lb
Applied ____________________ lb
Time ____________________ seconds
Rate ____________________ lb/acre
Percent ____________________ low–high
Adjustments:

Load #____________________
Loaded ____________________ lb
Drained ____________________ lb
Applied ____________________ lb
Time ____________________ seconds
Rate ____________________ lb/acre
Percent ____________________ low–high
Adjustments:

II.19 Ground Application of Bran Bait Insecticides

M. A. Boetel, B. W. Fuller, L. E. Jech, and R. N. Foster

Aerial insecticide application methods are most appropriate when extremely rough terrain and/or extensive acreages require treatment. However, smaller, isolated grasshopper outbreaks are often managed more economically using ground application equipment and techniques. A number of different application systems are available for both bran baits and conventional liquid insecticide formulations. For help selecting the appropriate insecticide formulation (liquid *v.* bait) see chapter II.3, "Sprays *versus* Baits."

In a 5-year cooperative effort 1987–91, several private and governmental agencies carried out field testing of bran bait application methods made modifications for improvement, and exposed farmers, ranchers, and Extension personnel in six States to these methods. Participants included Peacock Industries (Canada), the South Dakota Governor's Office of Economic Development, South Dakota State University, and the U.S Department of Agriculture, Animal and Plant Health Inspection Service's Plant Protection and Quarantine (USDA/APHIS/PPQ).

Bait Application Equipment

The Brie-Mar® Applicators Division of Peacock Industries (Saskatoon, SK) has developed three bran bait spreaders (models 10, 30, and 60). These spreaders are equipped with gasoline-powered pneumatic (air-driven) delivery systems that provide uniform flake distribution and can be set to deliver bran at various application rates. The spreaders have noncorrodible bran hoppers, are relatively inexpensive and easily operated, and require minimal maintenance. State and Federal cooperators in Colorado, Minnesota, Montana, North Dakota, South Dakota, and Wyoming have carried out extensive field evaluations of the units.

Model 10.—This unit is a shoulder-mounted backpack system that works well for small jobs, such as roadside ditch and yard or garden uses. It weighs 27 lb, holds 14 lb of bran, and can deliver 1.2 or 3 lb of bran per acre in 20- to 25-ft swaths with the operator walking at 3 miles per hour (mi/hour).

Model 30.—This bran spreader is designed for mounting on an all-terrain vehicle (ATV) or pickup truck, and can be used for bran applications in small and moderate-size grasshopper outbreak areas (isolated hot-spots in rangeland and pasture, roadside ditch areas, row-crop and forage field margins, large lawns, commercial vegetable gardens, and golf courses). This applicator can be used to treat outbreaks in very rough terrain where travel with a tractor or pickup truck may be difficult or impossible. Like the model 10, this system delivers bran flakes in a 20–25-ft swath. Its two-speed feed roll can deliver either 1 or 2 lb of bran per acre at 10 mi/hour, and it is capable of holding up to 45 lb of bran at a time.

Model 60.—This applicator is a larger unit that may be used for a range of different situations. It is designed for moderate-size outbreaks in areas where aerial treatment is not economically practical (roadside ditches, row-crop and forage field margins, and small to moderate acreages of pasture, rangeland, forage, and seedling row crops). Additionally, model 60 is well suited for conditions where the model 30 can be used (provided a pickup truck or tractor can traverse the terrain where applications must be made). This unit allows the operator to apply bran at 0.9, 2.1, 3, and 4 lb/acre in 40- to 45-ft swath widths at 10 mi/hour, and its hopper can hold up to 135 lb of bran flakes. In addition, bran output is turned on and off from within the pickup or tractor cab, and swath direction can easily be switched from right to left by manually moving the output tube. Using two spreaders (each applying in opposite directions) can double the swath width. This technique has been successful in the Grasshopper Integrated Pest Management Project demonstration area in North Dakota.

Bran Bait Applicator Calibration

Effective and economical insecticide applications require careful and accurate equipment calibration, and bran bait treatments are no exception. The following steps are essential for proper calibration of an applicator for broadcasting bran bait insecticide treatments.

1. Determine Swath Width.—Bran-spreader swath width should be measured before each bait application and as conditions (wind velocity and direction, terrain, or the material to be applied) change. Wind velocity is the most critical factor affecting bran-bait swath width, and neither calibration nor bait application should be conducted if winds are in excess of 5 mi/hour. If you are using a pickup- or ATV-mounted applicator

Figure II.19–1—The Brie-Mar bran spreader fits in the back of a pickup truck and will hold up to 135 lb of bran flakes. This spreader can treat up to a 45-ft swath width. (Photos courtesy of Peacock Industries; used by permission.)

(fig. II.19–1), measure swath width while the spreader is actually mounted on the vehicle and preferably under the same conditions that you will experience during bait applications. Swath width measurement and actual bait applications should be done by traveling directly into or against the prevailing wind.

The usual measurement consists of placing collection devices (paper plates work well) at even distances apart (5 ft apart is adequate for ground-operated units) in a grid pattern over a large block (see table II.19–1). The block should be several feet larger than the maximum range specified for the particular applicator model you are using (if using a Brie-Mar unit, refer to the "Bait Application Equipment" segment of this chapter for respective maximum swath width specifications of the different spreader models) to account for wind effects on the swath. If slight breezes exist during swath width assessment, drive a nail through the center of each paper plate and fasten it to the ground. After collection devices are in place, carry out two or three test runs to determine where bran bait distribution drops off (the drop-off point will be fairly abrupt under calm wind conditions). Count and record the bran flakes that land on plates after each test run. These counts will establish the effective bran swath width.

Table II.19–1—Distribution collection devices (paper plates) for bran spreader swath width determination

Row	Plate number									
no.	1	2	3	4	5	6	7	8	9	10
	Number of bran flakes collected									
1	3	5	4	10	8	7	8	4	3	0
2	5	7	11	11	12	6	6	5	5	1
3	4	10	8	9	11	10	7	5	4	1
4	6	9	11	7	7	9	5	6	3	0
5	4	4	12	4	8	10	7	4	4	1
Total	22	35	46	41	46	42	33	24	19	3

Note: Data in the table represent the number of bran flakes collected on individual paper plates (1–10) within rows (1–5). In practice, the spreader should move perpendicular to the direction of the rows.

In this trial run, bran flakes were distributed well between and including plates #1 and #9. Since there is a total of eight 5-ft increments between these plates, the effective swath width of this bran spreader is 8 × 5 = 40 ft.

2. Measure Applicator Delivery Rate.—This process consists of running the applicator in a timed interval at the rate that will be used in the field, collecting bran output, and determining its weight as a function of time. If you are using a Brie-Mar unit, the usual practice involves filling the hopper to about 50 percent full, running the engine at full throttle, turning on the output auger, attaching one nylon pantyhose leg to the bran output tube, and collecting bran output in at least 1-minute intervals. Repeat this step several times to obtain an accurate estimate of output. Weigh samples individually to measure bran output as weight per unit time (an example of output determination appears in table II.19–2).

Table II.19–2—Weight data from five timed (1-minute) samples for estimating bran applicator output per unit time

Sample	Weight (lb)
1	0.682
2	0.655
3	0.590
4	0.724
5	0.671
Total	3.322
Average	3.322 lb/min × 5 = 0.6644

3. Determine Vehicle Speed.—Precise determination of vehicle speed may sound much easier than it is in practice. When traversing rough terrain, most vehicle speedometer needles will bounce a lot and give inaccurate readings. Under such conditions, it may be necessary to install a digital tachometer, travel in a low gear, and establish a tachometer reading to go by rather than the speedometer needle. The appropriate tachometer reading used during bait application should be established in the actual area requiring treatment. First, measure a practice path of a given distance (minimum of 100 ft) for the vehicle to pass. Then, calculate the desired time to cover the practice path. Let’s say that you are trying to apply bran bait at a rate of 1.5 lb/acre. The following calculations will use the 0.6644 lb/min applicator delivery rate derived from the example in step 2 (your delivery rate will be slightly different). The following calculation will tell you how much time it should take to cover 1 acre at the 1.5-lb application rate:

$$\frac{1.5\text{ lb}}{1\text{ acre}} \times \frac{1\text{ minute}}{0.6644\text{ lb}} = \textbf{2.258 minutes}\text{ to cover 1 acre at 1.5 lb bran per acre}$$

The next step involves dividing the area in 1 acre (43,560 ft^2) by the bran applicator's swath width derived from step 1 (40 ft in our illustration). This calculation will provide you with the number of linear feet that you should travel in the time it takes to cover 1 acre (2.258 minutes in our example) while applying bran bait at the desired application rate (1.5 lb/acre in this exercise).

$$\frac{43{,}560\text{ ft}^2}{40\text{ ft}} = \textbf{1,089 linear feet}\text{ should be traveled in 2.258 minutes}$$

Convert the time in minutes to seconds:

$$2.258\text{ minutes} \times \frac{60\text{ seconds}}{1\text{ minute}} = \textbf{135.48 seconds}\text{ to travel 1,089 linear ft}$$

The target time to traverse your 100-ft test path is then calculated using cross-multiplication as follows:

$$\frac{1{,}089\text{ ft}}{135.48\text{ seconds}} = \frac{100\text{ ft}}{X\text{ seconds}}\text{ or, } X = (135.48\text{ seconds} \times 100\text{ ft}) \div 1{,}089\text{ ft}$$

therefore, X = **12.44 seconds** to travel 100 ft

The vehicle speed to target for traveling 100 ft in 12.44 seconds is determined using the following calculation:

$$\frac{100\text{ ft}}{12.44\text{ seconds}} \times \frac{60\text{ seconds}}{1\text{ minute}} = \textbf{482.32 feet per minute}$$

Vehicle speed in ft/minute should be converted to mi/hour, which will provide a rough estimate for a speedometer reading to target when making test runs. A useful conversion factor is that for each 1 mi/hour, a vehicle travels 88 ft/minute. The target speedometer reading is calculated using cross-multiplication:

$$\frac{88\text{ ft/minute}}{1\text{ mi/hour}} = \frac{482.32\text{ ft/minute}}{X\text{ mi/hour}}\text{ or, } X = (482.32\text{ ft/minute} \times 1\text{ mi/hour}) \div 88\text{ ft/minute}$$

therefore, X = **5.48 mi/hour** as a target speedometer reading.

After the targeted time to travel the practice path and target speedometer reading have been calculated, use a stopwatch to time trial passes of the vehicle covering the test path and make adjustments until the desired speed and associated tachometer reading are established. Once these final steps are completed, you are ready to carry out a properly calibrated bran bait insecticide treatment using ground application equipment.

II.20 Alaska's Cooperative Bait Program

Wayne Vandre and Don Quarberg

Situation

Recent agricultural land development in Delta Junction, AK, has created conditions favorable for epidemic outbreaks of grasshoppers where there were few outbreaks before. Cooperative Federal grasshopper control programs in these agricultural areas have not been possible because of a 10-mile no-spray buffer zone around peregrine falcon habitat. In addition, the lack of Environmental Protection Agency (EPA) registration for use of carbaryl on barley, a major cereal crop in the area, hampered individual control efforts.

With the help of the University of Alaska Cooperative Extension Service, agricultural producers in the Delta Junction area turned to integrated pest management (IPM) techniques to control grasshopper outbreaks. Using readily available materials, small-batch mixing equipment, and spreading equipment, farmer cooperatives demonstrated the success of a local IPM philosophy.

Baiting hatching beds with carbaryl-treated wheat bran has been an effective means of controlling grasshopper populations in other States. Wheat is not a common crop grown in Alaska other than for personal use, so wheat bran is not readily available for use in baits. The farmers' cooperative successfully demonstrated that locally grown barley could successfully be substituted for wheat bran as a bait. The owner of the Sevin® registration label, Rhone–Poulenc, has stated (personal communication) that coarse barley millings can be substituted for wheat bran in formulating the carbaryl bait. Thus, the use of locally grown barley allows Alaskan farmers to formulate an effective carbaryl bait economically.

Alaska conducted a cooperatively developed grasshopper baiting trial with the U.S. Department of Agriculture (USDA), Animal and Plant Health Inspection Service (APHIS) and USDA's Agricultural Research Service (ARS), Rhone–Poulenc (carbaryl manufacturer), and the University of Alaska–Fairbanks Cooperative Extension Service. The trial used locally grown cereal grains (dry rolled barley and oats) as bait substrates. USDA/ARS laboratory bait-acceptance trials indicated that Alaskan grasshopper species would eat the barley bait.

A producer cooperative can be especially important in areas of widespread grasshopper infestation where the demand for bait application may exceed capabilities for bait formulation, distribution, and application. The cooperative can play an important role in:

- obtaining carbaryl insecticides and bait substrate material;
- providing equipment for formulating, transporting and applying bait;
- deciding on areas to which the bait is applied; and
- maintaining communication among users, the public, and regulatory agencies.

Producer cooperatives already exist in many rural communities. A board of directors elected from the producer membership governs these coops. The Alaska Farmers Cooperative of Delta Junction is such an organization and served as the bait cooperative in this trial program.

Bait Production

ARS' Rangeland Insects Lab in Bozeman, MT, tested local Alaskan barley and also oat products and found them suitable as a bait substrate. Rhone–Poulenc granted temporary permission to use Alaskan-grown barley as a bait substrate for the trial.

The cooperative obtained a $^{1}/_{4}$-yd^3 cement mixer to mix and formulate the bait. Bait batch ingredients included 100 lb of dry rolled barley, mixed with 2 qt each of carbaryl (Sevin 4-Oil®) and diesel oil. This combination produced a 2 percent carbaryl bait formulation. While the cement mixer rotated at approximately 50 revolutions per minute, a 50:50 mix of carbaryl and diesel oil was sprayed into the mixer with a portable sprayer. Using an 80-degree flat fan nozzle operated at approximately 30 lb/in^2, spray operators adjusted the sprayer pressure as high as possible with minimal overspray and misting. A cardboard cover installed over the cement mixer opening reduced spray drift.

A preliminary trial using rolled barley and water colored with red food dye determined mixing time requirements. It took nearly 30 seconds to add the liquid. Three minutes of agitation thoroughly mixed the bait and carbaryl material.

The cooperative mixed bait on an as-needed basis, with surplus bait stored in a cool, dry, signed, and locked storage facility. No bait was stored longer than 48 hours before application. The cooperative rebagged formulated bait in plastic woven sacks, each containing approximately 50 lb. All bags were sewn shut and labeled as “CARBARYL BAIT—CAUTION” with copies of the carbaryl label attached.

The cooperative used Wilmar 500 fertilizer spin-spreaders calibrated with water-treated rolled barley, to decide application rates. A bait application rate of 36 lb/acre, or 0.7 lb/acre of carbaryl, achieved a distribution density of 40 particles/ft^2 of soil surface area. This rate is within the limits specified on the carbaryl label.

Barley particle size and density are variable depending on the adjustment of the roller mill, which processes the bait substrate. Procedures for calibrating spreaders are available at Alaska’s Cooperative Extension Service offices and through the State at State Office Fairbanks, Cooperative Extension Service, University of Alaska Fairbanks, Fairbanks, AK 99775-5200, (907) 474-6357.

Training and Certification Program

The Cooperative Extension Service developed a training course for carbaryl bait applicators somewhat similar to the pesticide certification training dministered by the APHIS Plant Protection and Quarantine unit. The 3-hour course addressed the topics of grasshopper life cycles; preferred food and egg-laying site conditions; scouting techniques; deciding economic thresholds; alternative controls; understanding the carbaryl label; personal and environmental safety; formulating, mixing, calibrating, and applying baits; timing and biological conditions affecting the success of baits; and evaluating the effectiveness of the bait.

An exam followed the course. Only those who successfully passed the exam could participate in the baiting program. Agricultural producers and interested participants from the public could take the course.

Evaluation and Results

All persons applying baits submitted information for recordkeeping. A survey questioned bait users about their opinions on weather conditions when the bait was used, length of time the bait remained available and effective, growth stage of treated grasshoppers, effects on nontarget species (other insects and birds), any personal health effects, and if they would use bait again.

According to survey responses, the bait was effective. Grasshoppers readily ate the bait, and the larger bait particles remained effective even after a rainfall. Only one applicator mentioned effects on nontarget species (a decline in ground beetles following bait application). Another reported the successful raising of three robin clutches that fed on treated grasshoppers. There were no reports or observations of adverse effects on human health.

Conclusion and Discussion

The results of this grasshopper control project show that early and effective reductions in grasshopper populations are possible using a formulated carbaryl–barley bait. The reduction or elimination of pesticide spray drift, the selectivity toward pest species, and the relative safety to human and environmental health all support the approval and recommendation of this bait as an effective IPM tool.

Crop damage from grasshoppers is expected in the Delta agricultural area in the future. Federal and State agencies should authorize and encourage further development of bait-application programs. An acceptable plan must be in place well before potential outbreak periods. If not in place, the long delay in organizing the program could result in the return to more conventional pesticide controls, such as aerial spray operations over large tracts of land.

The experience gained through this trial project and input from participants shows that there are certain conditions and/or alternatives for continued use and future success:

- Barley should be included as an approved bait substrate on the label for carbaryl. This substrate is effective and does not incorporate any significant changes when compared to wheat. Local availability and cost are positive factors toward adoption by farmer–applicators in Alaska.

- The manufacturer(s) of carbaryl could request a waiver or deletion of the label requirement for direct supervision by a government official. A category-specific training and certification program approved by EPA and the State regulatory agency, such as Alaska's Department of Environmental Conservation (DEC), could substitute for direct supervision. This training program would ensure that all applicators would become knowledgeable in bait formulation, calibration, and application procedures, and all health and safety issues.

- Another alternative to the direct supervision requirement would be to have the Alaska DEC or other State regulatory agency assume this role through the State-approved certification program. The built-in safety and reduced risk of this baiting program compared to other pesticide spray procedures calls for this procedural change.

- A primary component of all future activities is education. The pesticide applicator training and certification program developed and maintained by the Alaska DEC and the Cooperative Extension Service has proven to be effective in developing applicator competence and reducing or preventing pesticide incidents. The successful start of such a certification and training component in this project would be reviewed and improved to meet all education and regulatory objectives.

Public awareness of pesticide use and misuse in the environment continues to grow. This awareness has resulted in the adoption and use of IPM philosophy and procedures when pest problems arise. The successful development and results of the grasshopper baiting program in the Delta agricultural area have shown that it is possible to develop an effective, low-cost pest management program that reduces health risks to humans and wildlife and is environmentally safe.

II.21 Bran Bait or Liquid Insecticide Treatments for Managing Grasshoppers on Croplands Adjacent to Rangeland or Conservation Reserve Program Acreages

B. W. Fuller, M. A. Catangui, M. A. Boetel, R. N. Foster, T. Wang, D. D. Walgenbach, and A. W. Walz

The principal emphasis of rangeland grasshopper intergrated pest management (IPM) is to protect forage for domesticated animals and wildlife. Row crops (corn, soybeans, small grains) occur intermixed with rangeland in the northern Great Plains. The undisturbed rangeland soils provide highly suitable habitat for grasshoppers to lay eggs, potentially leading to outbreaks of grasshoppers at levels sufficient to cause devastating damage to the rangeland ecosystem. At these times, nearby row-crops may be severely damaged by grasshopper invasion from infested rangelands.

Even in locations that are predominantly dedicated to row-crop farming, grasshopper outbreaks are not uncommon. Grasshopper sources in row-crop areas typically are roadsides, grassed waterways, fencelines, and other field margin areas where soil containing grasshopper egg pods remain undisturbed. Additionally, parks, wildlife refuges, Native American reservations, and Conservation Reserve Program (CRP) acreages can be potential sources of grasshopper hot-spots.

Farmers are advised to treat immature (third-instar) grasshoppers at or near their hatching sites prior to further movements into the perimeter rows of cropland. Doing so can often alleviate the need to treat an entire row-crop field. Not only does this preventive effort save considerable money over the cost of whole-field treatment, it can greatly reduce potential negative impacts on nontarget organisms (beneficial insects and endangered species).

Choosing the proper treatment and application method are critical considerations to successful grasshopper IPM. For example, in environmentally sensitive areas (wilderness preserves, endangered species habitats, wetlands, and lands adjacent to bodies of water), treatment options may be limited.

Grasshopper IPM Project research has found both benefits and weaknesses associated with ground-applied liquid insecticides and bran bait treatments for control of grasshoppers on row crops near rangeland. Bran bait offers increased environmental benefits compared to conventional liquid treatments. For example, carbaryl–bran bait with 2 percent active ingredient (AI) by weight applied at 2 lb/acre offers 92 to 97 percent less active ingredient compared to conventional liquid formulations of carbaryl (0.5 to 1.5 lb AI per acre). Additionally, baits offer reduced cost for application, improved applicator safety, and minimized risk to many nontarget organisms.

Typically, liquid formulations provide quick broad-spectrum activity, uniform coverage, cost competitiveness, effective control, and residual activity. Liquid sprays also receive wide acceptance among farmers and ranchers. While many of these characteristics may appear favorable for grasshopper control, they may produce undesirable effects on beneficial insects and other nontarget species. Liquid application may pose added concerns for handling and applicator safety when compared to the safety of bran treatments. In addition, aerially applied liquid chemicals are far more prone to wind-related drift problems. Using liquid sprays is questionable where spray sites border or approach environmentally sensitive areas.

To choose the most suitable treatment, carefully review conditions (terrain, density of vegetation, wind direction and speed, temperature, and grasshopper species composition). The Grasshopper IPM (GHIPM) Project has attempted to identify treatments or application methods that can provide acceptable levels of grasshopper suppression in association with short- and long-term environmental factors. To further these efforts, research on grasshoppers at South Dakota State University and within the Project has addressed the use of bran bait and liquid applications in several related studies: row-crop and forage protection, optimizing the level of active ingredient in bran baits, and grasshopper suppression in CRP acreage.

Row Crop and Forage Protection

As mentioned earlier, controlling grasshoppers before their movement from hatching sites into nearby row crops is highly desirable. Studies of the use of bran baits on roadside areas were conducted in Colorado, Minnesota, Montana, North Dakota, South Dakota, and Wyoming. Little definable control was found in North Dakota and Montana with plot integrity questioned.

Problems with control were noted in Wyoming; however, in larger areas, treatment with carbaryl bait provided effective grasshopper population reductions (Lockwood and DeBrey 1990). Failure of bran bait applications to control grasshoppers satisfactorily was far more evident in eastern parts of South Dakota, where roadside areas had a much denser canopy (height of more than 0.75 m) and ground cover (at least 90 percent plants). This scenario contrasts the good to excellent control that bran baits have provided in several separate studies on large tracts of western South Dakota rangeland (Jech et al. 1993, Quinn et al. 1989, Wang and Fuller 1990 unpubl). These erratic results do not warrant a strong endorsement of roadside application for bran baits. As noted earlier, plot integrity may have played a significant role in the less-than-desirable levels of control.

Grasshopper behavior (preference for open canopy over shaded areas or reduced natural ability to search for food associated with the settling of bran flakes) may be important considerations in control efforts. Grasshoppers hatching several days following a bran application are not likely to suffer negative impact because baits lack residual control.

Despite these negative factors, bran baits remain a strong option when other methods are impossible to use. Even though populations are not always reduced to subeconomic levels at the site of a bran treatment, partial control may be sufficient to reduce further movement into adjacent row-crop areas.

Seedling corn (about 3 inches in height) was treated with chlorpyrifos–bran bait to control *Melanoplus bivittatus* immature (second-instar) grasshoppers with reductions of 40 to 50 percent that resulted in subeconomic pest densities (Boetel et al. 1990a). Under a more controlled setting, screen cages (1 by 1 by 0.5 m) were placed over seedling corn and artificially infested with 20 third-instar *M. sanguinipes.* One hundred percent control was achieved after a 24-hour period with several toxicant treatments on bran bait (Wang et al. 1991). Applications directly to seedling crop foliage throughout the field would appear to be a more suitable treatment method than bran applications that were limited to field margins.

Unlike most row-crop annuals, alfalfa does not require seedbed preparation or cultivation after its initial establishment. This lack of cultivation contributes to high grasshopper survival across alfalfa fields. Field borders surrounding alfalfa are potentially even more suitable for grasshopper egg laying because of their vegetative diversity (Pooler 1989 unpubl.) and the long-term absence of soil disruption by cultivation practices. Thus, even though grasshoppers are likely to be found throughout an alfalfa field, the highest densities may still exist in perimeter areas.

Bran bait, carbaryl 2 percent AI at 2 lb/acre, was compared to a liquid application of carbaryl (Sevin® XLR, 4E) at 1 lb/acre on alfalfa plots (400 by 800 m) to control grasshoppers. Numbers of fourth- and fifth-instar grasshoppers were 20 and 18 per square meter, respectively, in pretreatment density estimates. Counts 4 days after bran bait treatment were almost unchanged (20). Conversely, a 99.5-percent reduction in grasshopper density was observed in plots that received liquid applications of carbaryl. Dead grasshoppers were observed on the ground in bran-bait-treated plots. Invasion from perimeter areas was obvious, but bran baits were offering little or no residual control. While initially effective, bran baits proved a poor choice in alfalfa because of the lack of residual control.

Optimizing the Level of Active Ingredient in Bran Baits

The percent of active ingredient placed onto bran flakes played only a minor role in grasshopper mortality in several field and laboratory studies. Significant differences were not detected among 2- and 5-percent carbaryl-treated bran baits. Likewise, 1- and 3-percent chlorpyrifos treatment provided similar grasshopper control (Boetel et al. 1990b). These results suggest that the lower dose bran baits contain sufficient toxicants to control grasshoppers. Laboratory trials provided evidence that 0.0007 g of bran flake treated with 2-percent carbaryl was adequate to cause death. Thus, bran-accepting grasshopper species will not require feeding on multiple flakes or high percentages of toxicant to receive a lethal dose.

Grasshopper Suppression in CRP Acreage

The stable environment of CRP lands is similar to rangeland in that grasshopper populations can build up in this habitat and threaten nearby croplands. Failure of bran baits to control grasshoppers effectively in roadside studies resulted in efforts to use liquid applications. Liquid applications can be cost prohibitive on CRP lands, where little economic return is expected. Thus, studies using lower rates of several insecticides (carbaryl, chlorpyrifos, dimethoate, esfenvalerate, diflubenzuron) have been undertaken.

Primary emphasis was placed on the need for residual activity in the presence of constant invasion potential. Carbaryl at 0.5, 0.75, and 1 lb AI per acre offered excellent control up to 10 weeks after treatment. Using the lowest rate would offer a farmer-acceptable control with significant economic savings. Other compounds tested offered similar results; however, several years of data support the carbaryl findings.

References Cited

Boetel, M. A.; Fuller, B. W.; Bergland, D. R.; Walgenbach, D. D. 1990a. Comparative efficacy of grasshopper bran baits in corn and ditch borders, 1989. Insecticide and Acaricide Tests 15: 184.

Boetel, M. A.; Fuller, B. W.; Bergland, D. R.; Walgenbach, D. D. 1990b. Bran bait efficacy for control of grasshoppers in South Dakota, 1989. Insecticide and Acaricide Tests 15: 180.

Jech, L. E.; Foster, R. N.; Colletto, D.; Walgenbach, D. D.; Roland, T. J.; Rodriguez, G. D.; Bohls, R.; Houston, R. D.; Meeks, W. K.; Queener, R. L.; Jackson, C. L; Dines, J. L.; Puclik, M. J.; Scott, A. K. 1993. Field evaluation of diflubenzuron and carbaryl bran baits against grasshopper (Orthoptera: Acrididae) populations in South Dakota. Journal of Economic Entomology 86: 557–565.

Lockwood, J. A.; DeBrey, L. D. 1990. Rangeland grasshopper control with baits, 1989. Insecticide and Acaricide Tests 15: 271.

Quinn M. A.; Kepner, R. L.; Walgenbach, D. D.; Bohls, R. A.; Pooler, P. D.; Foster, R. N.; Reuter, K. C.; Swain, J. L. 1989. Immediate and 2nd-year effects of insecticidal spray and bait treatments on populations of rangeland grasshoppers. Canadian Entomologist 121: 589–602.

Wang, T.; Boetel, M. A.; Fuller, B. W.; Chambers, W. W.; Jenson, J. M. 1991. Efficacy of bran baits in controlling grasshoppers in South Dakota, 1990. Insecticide and Acaricide Tests 16: 177.

References Cited—Unpublished

Fuller, B. W.; Wang, T. 1990. Grasshopper bait methodology investigations in eastern South Dakota. In: Cooperative Grasshopper Integrated Pest Management Project, 1990 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 182–186.

Pooler, P. D. 1989. Factors influencing grasshopper oviposition site selection on South Dakota rangelands. M.S. thesis. Brookings, SD: South Dakota State University. 50 p.

Wang, T.; Fuller, B. W. 1990. Field evaluation of grasshopper bran baits in western South Dakota. In: Cooperative Grasshopper Integrated Pest Management Project, 1990 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 187–191.

II.22 Aircraft Guidance for Grasshopper Control on Rangelands

Gil Rodriguez and T. J. Roland

Guidance methods and systems for aerial application have evolved throughout the years from the most rudimentary to the most sophisticated. The purpose was to provide aircraft guidance for the proper distribution of agricultural chemicals to field crops. In order to achieve this, pilots had to develop a method of guiding the aircraft over the ground.

Initially the pilot attempted to fly evenly spaced passes over the field by free-flying—visually estimating the distance between passes. This procedure was not accurate, and better methods were developed as time went by. Free flying is still in use, but only on smaller fields, where it is easier for the pilot to estimate the distance between passes and keep track of the number of passes. The following is a list of guidance methods/systems in the approximate chronological order that they were developed and a brief description of each.

Flaggers

Ground personnel waving flags guide the aircraft. The flagger indicates to the pilot the starting point for each pass. When the aircraft is properly lined up, the flagger steps off the required distance to get in position for the next pass. There may be one or two flaggers—one flagger at one end of the field, or one at each end. Long runs may require multiple flaggers. Flags are easy to see because of their waving motion, and this method is more accurate than free flying. Multiple flaggers may vary distance and introduce error when stepping off the spacing between passes and cause skips.

Kytoons

Ground personnel holding kytoons (tethered balloons) guide the aircraft much the same way flaggers do. This method is useful when there are visual obstructions, such as trees, buildings, or terrain, and where long runs are required. Some disadvantages of this method are that kytoons tend to get out of control under certain meteorological conditions that cause the balloons to dive into or have their tethers get tangled in trees. There are also safety hazards involved, such as collisions with the aircraft and contact with electrical power-lines.

Mirrors

Ground personnel using mirrors to flash reflected sunlight at the pilot guide the aircraft. The pilot flies toward the flashing light. This method is especially effective on long passes over flat terrain with few or no landmarks since the flashes are visible over long distances. Two disadvantages of using mirrors are that they are difficult to aim when there is a large angle between the sun and the aircraft, and they won't work if clouds block the sun. An alternate backup guidance method would be required during these conditions.

Automatic Flagman

This system consists of a mechanical device attached to the upper inboard area of the aircraft wing. The equipment is loaded with paper flags or streamers that the pilot releases at the end of each pass to assist in establishing the next pass. This system is used independently or to supplement other guidance methods.

Smoker

In this guidance system, the pilot releases a puff of smoke into the airstream by injecting a small amount of paraffin oil into the aircraft exhaust system. This procedure enables the pilot to mark the last pass momentarily in order to set up for the next one, much as with the Automatic Flagman. The Smoker also assists the pilot in determining wind direction and drift. This system supplements other methods of guidance but is not useful when winds displace the smoke while the pilot makes the turn for the next pass.

LORAN-C

LORAN (an acronym for LOng RAnge Navigation) is a radio navigation system that uses time-synchronized pulsed signals from ground transmitting stations spaced several hundred miles apart. The stations are configured in chains of three to five that transmit with the same time-synchronized signals. Within each chain, one station is designated as the master, and the remainder are secondaries.

An aircraft-mounted LORAN-C receiver converts the "time difference" between the arrival of radio signals from the master and the secondaries into latitude/longitude coordinates. Navigational values such as distance and bearing to the treatment area are computed from the aircraft's present latitude/longitude (geographic location).

A computer software program called GRIDNAV provides aircraft guidance to the pilot during aerial application. The pilot enters the geographic coordinates for the first pass plus the desired swath width into the program before leaving on the mission. The GRIDNAV software automatically provides directional and spacing guidance for each pass and keeps track of the number of passes during the aerial application operation.

This system eliminates the need for ground personnel. Mountainous terrain, mineral deposits, and position of the aircraft with relation to the stations can affect the precision of the system. LORAN-C is unsuitable for applications that require swath widths of less than 60 ft. The system is especially useful for releasing sterile insects where swath width is much wider and accuracy less critical.

Global Positioning System (GPS)

GPS is a location system based on a constellation of satellites orbiting the Earth at high altitude. The Department of Defense developed GPS for military operations, and the system proved itself during the Gulf War in 1992. GPS presently is the most accurate navigational system in the world.

Geographic position is developed in much the same way as with LORAN-C. One difference is that GPS operates in three dimensions because the transmitting stations are satellites and are not located on the surface of the Earth. The distance between several satellites and the aircraft-mounted GPS receiver is measured by highly sophisticated equipment and converted to geographic coordinates.

Although GPS is still in a developmental stage for agricultural use, it is capable of providing aircraft guidance for aerial application in the same manner as LORAN-C. This system also eliminates ground personnel and is not affected by the physical conditions that affect LORAN-C. However, it must maintain line-of-sight contact with the satellites being used. A position error of 60–100 ft can be expected under normal conditions and can be reduced to 3–6 ft or less with differential correction. Differential correction is accomplished by placing a GPS receiver base unit at a known location and using it to determine exactly what errors the satellite data contain. The base unit then transmits an error correction to the GPS receiver in use, which can use that information to correct its position. A disadvantage of this system is that it requires an additional stationary receiver placed at a known location in order to achieve maximum accuracy.

GPS will expand its use for agricultural applications and already has proven its accuracy and use in rangeland grasshopper and cotton boll weevil control programs in the United States.

Conclusions

Aircraft guidance for aerial application has made significant progress through the years. The trend has been toward greater accuracy and the elimination of ground personnel. Eliminating the need for ground personnel also reduces the exposure of humans to pesticides. Accuracy is very important in reducing damage to the environment and to threatened and endangered plant and animal species.

III. Environmental Monitoring and Evaluation

Grasshopper control does not take place in a vacuum but in complex rangeland ecosystems. Researchers funded by the Grasshopper Integrated Pest Management Project carefully studied the effects of various control regimes on aquatic organisms, small mammals, birds, and bees. (Photo by R. Miller, submitted through chapter author James R. Fisher and reproduced by permission.)

III.1 Introduction

L. C. McEwen

Grasshopper integrated pest management (GHIPM) is the preferred alternative for grasshopper control listed in the 1987 Environmental Impact Statement for the 17 Western States with rangeland. In conducting the U.S. Department of Agriculture (USDA) cooperative grasshopper control programs, it is necessary to meet the requirements of environmental protection laws, especially the National Environmental Policy Act, the Endangered Species Act, and laws to protect surface and ground water.

Three of the registered methods for the cooperative programs use liquid insecticide formulations. Although the amount of active ingredient applied has been reduced by using ultralow-volume spray techniques, these pesticides can still affect the ecosystem. Grasshopper sprays blanket the rangeland habitat and expose nontarget animal life to the chemicals. Though the spray programs effectively reduce grasshopper densities in the short term, effects on nontarget species and rangeland ecology need to be evaluated. Some aspects deserve continued monitoring after USDA's GHIPM Project ended in 1994.

Use of dry baits for grasshopper control, with less potential for unintended effects on nontarget life, was investigated in the field. Grasshopper baits carrying chemical or biological control materials have great promise for use in environmentally sensitive areas. Also, new candidate grasshopper control methods and materials, such as diflubenzuron and *Beauveria bassiana*, were examined for effects on American kestrels (sparrowhawks) in field studies of nestlings and fledglings. These materials appear to have little, if any, direct toxicity to birds.

Several field and laboratory studies of GHIPM materials or methods have been conducted since the inception of the GHIPM Project in 1987. Birds have received the most attention because they are usually more susceptible than mammals to direct toxicity and to indirect ecological changes, such as loss of insect food. Studies have varied from determining total avian population response following large-scale grasshopper control programs (on areas greater than 10,000 acres) to physiological and behavioral measurements in individual birds sublethally exposed to GHIPM materials.

Two species of endangered fish have been studied intensively for toxicity of malathion and carbaryl. Effects on nontarget invertebrates (both aquatic and terrestrial) were also investigated. Other GHIPM Project-sponsored environmental impact studies included (1) avian and mammalian brain and blood cholinesterase measurements, (2) use of American kestrels and killdeer as bioindicators of possible effects on closely related endangered species, (3) effectiveness of bird predation for regulating grasshopper population densities, (4) postspray pesticide residue concentrations in environmental samples and biota (fauna and flora), (5) results of aquatic field monitoring of spray treatments, (6) small mammal live-trapping recapture tests, and (7) field experiments to investigate the indirect effects (loss of food base) on productivity of nesting birds associated with application of malathion and Sevin® 4-Oil liquid sprays and carbaryl bait. Preliminary results of golden eagle postfledging survival after aerial spray of Sevin 4-Oil to nest areas are also reported in this Environmental Monitoring and Evaluation section.

The important question of potential effects on endangered plant species and their insect pollinators is addressed in a summary of several studies. Authors also discuss untreated buffer-zone requirements to protect endangered plants, aquatic habitats, nests of endangered birds such as peregrine falcons, and other environmentally sensitive sites.

Knowledge of GHIPM relationships to nontarget life and rangeland ecology is critical for successful grasshopper population management. The days are long past when estimating the grasshopper kill was the only concern while other effects of a spray program were ignored. For many years, aldrin, dieldrin, and other organochlorine compounds were extremely efficient at killing grasshoppers, but USDA stopped using those pesticides in the mid 1960's because of their effects on nontarget life. Organochlorine pesticides harmed wild mammals, migratory birds, endangered raptors, reptiles, aquatic life, and western rangeland ecosystems (McEwen 1982).

Dieldrin, for example, is a stable compound that circulated through food chains and ecosystems for years and was highly toxic to all fish and wildlife. The Environmental Protection Agency criterion for chronic dieldrin contamination in fresh water is only 0.0019 parts per billion (Nimmo and McEwen 1994), but the bioconcentration factor in aquatic life can be 49,000 times the level of contamination in the water (Moriarity 1988). Animals exposed to sublethal organochlorine contamination may be unable to reproduce—particularly many fish species, fish-eating birds, and endangered raptors—and may also be more vulnerable to disease, pathogens, predators, and other stresses.

The insecticides currently registered for GHIPM programs are not only less toxic to terrestrial nontarget wildlife (McEwen 1982, Stromborg et al. 1984, Smith 1987) but also much less persistent in the environment than organochlorine chemicals. Today's grasshopper insecticides soon degrade into biologically inactive compounds that do not circulate through food chains (U.S. Department of Agriculture, Animal and Plant Health Inspection Service, 1987). The primary questions to be answered concerning the current control materials are (1) significance of sublethal toxic effects on birds, mammals, and fish, particularly cholinesterase inhibition; (2) degree of hazard to endangered fish, wildlife, and plants, and other species of concern; (3) indirect effects due to reduction of insect or invertebrate food supply; (4) effects on nontarget insects, including pollinators of endangered plants; and (5) evaluation of wildlife population effects related to wide area GHIPM treatments. The answers to these questions are more difficult to determine than the relatively simple wildlife carcass counts and pesticide residue analyses that were used to investigate the old organochlorine pesticides.

The current, more comprehensive, investigations of sublethal and indirect effects reflect the need to determine the complex ecological impacts of GHIPM on nontarget life. The findings support GHIPM strategy, including recognition that healthy, vigorous, rangeland ecosystems are the most permanent solutions to range grasshopper problems in the long term.

References Cited

McEwen, L. C. 1982. Review of grasshopper pesticides vs. rangeland wildlife and habitat. In: Peek, J. M.; Dalke, P. D., eds. Proceedings of the wildlife–livestock relationships symposium; 20–24 April 1981; Coeur d'Alene, ID. Moscow, ID: University of Idaho: 362–382.

Moriarity, F. 1988. Ecotoxicology (2d ed.). London and New York: Academic Press. 289 p.

Nimmo, D. W.; McEwen, L. C. 1994. Pesticides. In: Calow, P., ed. Handbook of ecotoxicology, vol. 2. Oxford, UK: Blackwell Scientific Publishers: 155–203.

Smith, G. J. 1987. Pesticide use and toxicology in relation to wildlife: organophosphorus and carbamate compounds. Resour. Publ. 170. Washington, DC: U.S. Department of the Interior, U.S. Fish and Wildlife Service. 171 p.

Stromborg, K. L.; McEwen, L. C.; Lamont, T. 1984. Organophosphate residues in grasshoppers from sprayed rangelands. Chemistry in Ecology 2: 39–45.

U.S. Department of Agriculture, Animal and Plant Health Inspection Service. 1987. Final environmental impact statement: Rangeland Grasshopper Cooperative Management Program. USDA-APHIS FEIS 87-1. Washington, DC: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 2–9, 2–10, 2–11.

III.2 Direct and Indirect Effects of Grasshopper Integrated Pest Management Chemicals and Biologicals on Nontarget Animal Life

NOTE: Acephate is no longer approved by EPA for rangeland grasshopper control.

L. C. McEwen, C. M. Althouse, and B. E. Petersen

Initially there were 16 objectives (11 terrestrial and 5 aquatic) for the environmental monitoring studies of the Grasshopper Integrated Pest Management (GHIPM) Project. Most of the terrestrial objectives were concerned with determining effects of the grasshopper control methods and materials on birds. Studies varied from total bird population response after spray operations or bait treatments to toxicology tests with individual birds.

Small-mammal population effects and toxicology were investigated with one chemical (acephate). Some limited small-mammal observations also were obtained in areas sprayed with malathion and Sevin® 4-oil. Aquatic objectives were to investigate toxic effects of malathion and carbaryl on endangered fish in tank tests and to determine effects of grasshopper spray programs on fish and aquatic invertebrates in the field.

Other objectives included (1) evaluation of hazards to endangered species through study of related surrogate species, (2) determination of the significance of bird predation as a biological control of grasshoppers in an IPM program, and (3) wildlife tests with the candidate materials *Beauveria bassiana* (a fungal organism) and diflubenzuron (an insect growth inhibitor). More than 20 papers have been published in peer-reviewed journals on the GHIPM Project's environmental monitoring work, and other papers are in press.

Direct Effects

Direct effects on nontarget fish and wildlife of GHIPM materials may be lethal or sublethal. Unlike the organochlorine pesticides, such as dieldrin, chlordane, heptachlor, and toxaphene, formerly used for range grasshopper control (and still in use in some parts of the world) the current GHIPM chemicals do not kill wildlife by direct toxicity (McEwen 1982). There may be some rare exceptions to this statement, such as individual small nestlings of passerine (bird) species that are unusually sensitive to carbaryl or malathion being directly sprayed on an open nest. On the whole, however, GHIPM Project-funded investigators have seen only a very few such possible cases in a large number of nest observations. And none of these bird deaths could be positively attributed to chemical control materials.

At the malathion ultralow-volume (ULV) application rate of 8 fl oz/acre (0.58 kg/ha) and the Sevin 4-Oil formulation rate of 20 fl oz/acre (1.44 kg/ha) (carbaryl active ingredient [AI] rate of 0.56 kg/ha), there is very little possibility of toxicity-caused mortality of upland birds, mammals, or reptiles, and none has been observed.

However, these pesticides are more toxic to aquatic life: direct overspray of small ponds kills many aquatic invertebrates and may kill sensitive fish species. The risk is lower in flowing streams because the chemical is transported downstream and diluted more rapidly. Consequently, nonspray buffer zones around aquatic habitat must be observed (see chapter III.8). Lower-level exposure from pesticide drift or runoff (in contrast to direct overspray) does not kill fish but can be lethal to certain aquatic invertebrates (Beyers et al. 1995; also see chapter III.6).

One of our main environmental monitoring objectives was to determine effects of grasshopper control treatments on rangeland bird populations. We investigated 13 different grasshopper control treatments with GHIPM materials (malathion, Sevin 4-Oil, carbaryl bait, or *Nosema locustae).* We studied effects on total bird populations by concurrently conducting extensive line transect counts (Emlen 1977) before and after insecticide application in both treatment and control (untreated) plots. Total birds (total individuals of all species) did not change ($P > 0.05$) in the posttreatment periods (George et al. 1995). Populations of one highly insectivorous species, the western meadowlark *(Sturnella neglecta),* did consistently decrease at 10 and 21 days posttreatment. We presumed that was due to reduced food availability because there was no evidence of toxic signs in the remaining meadowlarks, and no dead ones were found. Comparative avian population response to many different pesticides used or tested for grasshopper control can be found in a report by McEwen (1982).

Sublethal Effects

Sublethal exposure to GHIPM pesticides is highly probable for wildlife inhabiting sprayed rangeland. The routes of exposure include dermal from direct hit or by moving through sprayed vegetation, ingestion in food or drinking water, and inhalation. The effects of sublethal exposure can vary from biological insignificance to convulsions and near death followed by recovery. Severe toxic signs have not been observed in terrestrial wildlife following GHIPM treatments. The potential for sublethal toxic effects can be minimized by use of bait formulations. Dry bait formulations use less actual chemical per acre or hectare and limit the route of exposure primarily to ingestion of affected insects. In comparison, liquid sprays result in multiple exposure routes (dermal, inhalation, and ingestion of coated vegetation as well as insects). Consumption of bait (bran particles) by wildlife is negligible because of the small size of bran particles and the low treatment rates used for GHIPM (2 to 5 lb/acre or 2.2 to 5.6 kg/ha of bait containing 2 percent carbaryl).

Use of bait treatments provides an environmentally safe means of obtaining some reduction of grasshopper densities in environmentally sensitive areas (such as habitat for endangered plants or animals). Vesper sparrow survival, growth, and fledging rates were not affected by carbaryl bait treatments around the nest areas (Adams et al. 1994). Total bird numbers were not reduced in a large area treated for grasshopper control with carbaryl bait (George et al. 1992a). Bait treatments at GHIPM rates reduce the potential for aquatic contamination (less drift and less chemical). Baits also appear safe for bees and pollinators of endangered plants (see chapters III.4 and III.5).

Cholinesterase Inhibition

All three of the GHIPM chemicals—carbaryl, malathion, and acephate—are cholinesterase (ChE) inhibitors. In vertebrates, acetylcholinesterase and butyrylcholinesterase are essential for normal function of the nervous system. Severe inhibition (>60 percent) often leads to death of the animal (fig. III.2–1). Moderately severe inhibition (40–60 percent) affects coordination, behavior, and foraging ability and can lead to death from other stresses of survival in the wild, such as weather or predators. Effects of lower levels of brain ChE inhibition (<40 percent) are still an open question regarding biological significance (Grue et al. 1991). In our samples of birds and mammals from areas treated with carbaryl, malathion, or acephate, we have not found any animals with >40 percent brain ChE inhibition, and only a few individuals inhibited >20 percent (Fair et al. 1995, George et al. 1995, and Petersen et al., in prep).

Figure III.2–1—Several highly toxic pesticides were field-tested to determine efficacy for grasshopper control and effects on nontarget life. Those chemicals found to be too toxic and hazardous to wildlife were not registered for use on rangeland. Most of the chemicals not registered were severe cholinesterase inhibitors and caused paralysis and death of beneficial birds, such as these Wilson's phalaropes. (Photo by G. Powell of the U.S. Fish and Wildlife Service; reproduced by permission.)

In a study of fish exposed to light drift of carbaryl (Sevin 4-Oil), Beyers et al. (1995) detected no effects on brain ChE. Blood plasma ChE also can be used as an indicator of pesticide exposure: effects of malathion on kestrels and carbaryl (Sevin 4-Oil) effects on golden eagles were reported by Taira (1994).

These results suggest that ChE inhibition is not a problem for upland wildlife when GHIPM chemicals are applied but do not mean that attention to accuracy and rigor of applications can be relaxed. Beyers et al. (1994) found that in water, concentrations of carbaryl as low as 1.3 mg/L (p/m) and of malathion as low as 9.1 mg/L were lethal to fish. Young kestrels died from malathion exposures of only 30 mg per kg of body weight (McEwen et al. 1993 unpubl.), much lower than lethal dosages for other species of birds (>100 to >400 mg/kg, Smith 1987).

A recent study by Nicolaus and Lee (1999) suggested a formerly unrecognized effect of organophosphate exposure. Birds that fed on affected insects developed a strong aversion to those insect species and would no longer capture them for food, even after the insects were free of contamination. Thus surviving birds were indirectly denied major food sources.

Indirect Effects

The most frequently asked question about effects on wildlife of grasshopper control is, "What about the effects on birds of the loss of the insect food base?" Much of our environmental monitoring effort was directed at this problem.

A 3-year investigation of indirect effects of malathion on nesting birds was conducted in Idaho. After a year of pretreatment study, two areas of rangeland were sprayed with the standard 8 fl oz/acre (0.58 kg/ha) ULV formulation of malathion. Intensive studies were conducted to measure effects on the insect and invertebrate populations and on survival and growth of Brewer's sparrow *(Spizella breweri)* and sage thrasher *(Oreoscoptes montanus)* nestlings (Howe 1993, Howe et al. 1996 and 2000).

Although the total invertebrate availability was significantly reduced by the spray applications, nesting birds switched their diets to the remaining insects and reproduced as successfully as birds on untreated comparison plots (Howe et al. 1996 and 2000). Adults had to forage longer on sprayed plots, and nestlings showed a higher propensity for parasitic blowfly *(Protocalliphora braueri)* infestation (Howe 1991, 1992), both of which might affect survival in some situations. Those effects were not significant in this study. Prespray grasshopper densities were low (1–4 per square yard or square meter) on all plots and were significantly reduced in the postspray period. This probably made the food availability test more rigorous than an operational grasshopper control program, where prespray densities are much higher and even postspray grasshopper densities usually exceed 1 or 2 per square yard or square meter.

Effects of Sevin 4-Oil sprays on killdeer populations were investigated in North Dakota. Two large treated areas were studied. One was sprayed with the standard rate of 20 oz/acre of formulation (16 oz Sevin 4-Oil + 4 oz diesel oil), and the other area received a lower rate of 16 oz/acre (12 oz Sevin 4-Oil + 4 oz diesel oil). These rates translated to 0.56 and 0.45 kg/ha of carbaryl AI respectively. No toxic signs and no mortality were observed in the killdeer.

Effects on foraging and diet of the killdeer were examined by both direct observation and analysis of stomach contents (Fair et al. 1995a). The insect capture rate by foraging killdeer increased during the period when affected insects were easily available 2 days after treatment (Fair et al. 1995b). No other differences in food habits were detected.

A test of carbaryl bait effects on vesper sparrow *(Pooecetes gramineus)* nestling growth and survival was conducted in North Dakota. This study simulated the "hot spot" method of treating small grasshopper infestations with carbaryl bait. There was no difference in any of the productivity parameters between nests on treated and untreated sites (Adams et al. 1994). Adult sparrows on treated sites had to forage farther from the nests to obtain food but did so successfully. Grasshoppers comprised 68 percent of all food deliveries to nestlings even though grasshopper densities were <1 per square meter. The ability of birds to capture a preferred food, even when grasshopper densities are extremely low, supports the value of predation by birds as a preventive force against grasshopper increase in an IPM approach to grasshopper management (see chapter I.10, "Birds and Wildlife as Grasshopper Predators").

Biennial grasshopper infestations in southeastern Alaska provided an opportunity to examine bird population response to the extreme differences in grasshopper abundance and availability that occur naturally. Densities alternate between >25 per square yard in high years and <1 per square yard in low years. This phenomenon apparently occurs because of a synchronized 2-year life cycle of the *Melanoplus sanguinipes* grasshopper species in the population. Birds were counted on permanently marked transects in 2 high and 2 low years, and nesting success of Savannah sparrows *(Passerculus sandwichensis),* the most abundant bird species, was measured. Total bird populations did not differ among years ($P > 0.05$).

Nesting success showed a trend of lower clutch size and nestling growth rates in the low grasshopper years (1991 and 1993) but not significantly ($P > 0.05$) (Miller et al. 1994). Grasshoppers constituted >45 percent of the birds' diet numerically and an even greater proportion of biomass in the high grasshopper years (1990 and 1992)

(McEwen et al. 1993 unpubl., Miller and McEwen 1995). The birds also managed to search out and capture grasshoppers in the low years, indicating their preference for this important food source. However, the breeding birds were able to switch their main food items to other insects (beetles, Hemiptera, larvae of Lepidoptera and others) in the low grasshopper years.

Rangeland wildlife has adapted to variable food availability and environmental conditions over the long term. Evidence of this was observed in North Dakota studies. An extreme drought in 1988 resulted in avian nesting failures and population declines. The effects on bird populations did not carry over to the succeeding years, when precipitation was in the normal range (George et al. 1992b; see also chapter III.7).

Small Mammal Studies

Small mammals generally are not affected as much as birds in the same area where a pesticide application is made, probably because small mammals generally are not exposed to as much toxicant as birds are. Most small mammals are nocturnal and are often in underground burrows during and immediately after a treatment; thus there is more time for the chemical to dissipate before small mammals are exposed (fig. III.2–2). Deer mice *(Peromyscus maniculatus)* collected on a malathion-sprayed area had lower residues than birds from the same sites (McEwen et al. 1989 unpubl.). Many small-mammal species also are inherently more resistant to specific toxicants than birds (Nimmo and McEwen 1994).

Figure III.2–2—Kangaroo rat being released after capture in a live-trap for study on a rangeland-grasshopper control area. Small mammals were generally less vulnerable to pesticide effects than birds inhabiting sprayed areas. (Photo by L. C. McEwen of Colorado State University; reproduced by permission.)

Effects of acephate and methamidophos (an acephate metabolite) on small mammals were studied on short grass range in Colorado. Results have not been completely analyzed, but preliminary data indicate a decrease in populations of certain species due to a combination of greater sensitivity to chemical toxicity and reduced competitive ability with other species. Deer mice were twice as sensitive to methamidophos (the lethal dose to 50 percent, or LD_{50}, was 9 mg/kg) than the other two most common species, grasshopper mice *(Onychomys leucogaster)* and 13-lined ground squirrels *(Spermophilus tridecemlineatus)*. The LD_{50} for both the latter was 21 mg/kg (Stevens 1989). Field live-trapping studies indicated postspray decreases of deer mice but not of the grasshopper mice and ground squirrels. Data analysis and manuscripts are still in progress on these studies (Althouse et al. unpubl., McEwen et al., in prep.).

Limited live trapping studies on malathion-sprayed areas in North Dakota showed no posttreatment decreases in abundant populations, primarily deer mice, and studies of carbaryl-sprayed areas at other locations had a similar outcome (McEwen et al. unpubl. 1988). An investigation of malathion ULV (8 fl oz/acre or 0.58 kg/ha) applied in Nebraska found no effects on small-mammal populations (Erwin and Sharpe 1973).

Golden Eagle Study

Golden eagles *(Aquila chrysaetos)* are a protected species and also are designated as a “species of concern” by wildlife conservation and land management agencies. This species also has special significance for Native Americans. Golden eagles nest in remote rangeland areas and often are found on areas slated for grasshopper control. Because of these concerns and problems, a study was initiated on the Western North Dakota IPM Demonstration Area where nesting territories and spray blocks often overlap.

Active nests of golden eagles were located and randomly selected for Sevin 4-Oil treatments or left unsprayed in 1993 and 1994. Overall, 12 nest areas were sprayed with Sevin 4-Oil at 20 fl oz/acre (1.4 kg/ha) or 8 oz/acre AI

(0.56 kg/ha AI) carbaryl. Approximately 10 ha were treated around each nest. For comparison, the investigators left eight nest areas untreated. At these control nests, the spray plane flew the same pattern and length of time but did not release any spray. Some nests contained two nestlings and some, a single nestling. The total number of treated nestlings was 17, and untreated totaled 11. Treatments were made when the eaglets were 4–7 weeks of age.

When the nestlings neared fledging age (10–11 weeks) they were captured to (1) take biological measurements, (2) take a 4- to 5-mL blood sample, and (3) attach a radio transmitter for postfledging location and observations (telemetry) (O'Toole et al. 1999). Field work and data analysis are incomplete, but preliminary results can be reported.

In 1993, two untreated and three treated fledglings died from various causes unrelated to the treatments. In 1994, a better prey year, all 6 untreated and 10 treated fledglings survived. Postfledging telemetry studies indicated two behavior differences in the eagles from sprayed nest areas: "sprayed" eagles tended to perch longer and to preen more in afternoon observation periods. These results will be reported by O'Toole et al. (in prep.). All fledglings dispersed from their hatch areas by November each year (except for one, which left by December 3, 1994), and radio signals could no longer be detected in ground searches. Aerial telemetry searches were conducted in 1995 to obtain more information on movements and long-term survival rates.

Blood plasma ChE and other blood components were measured. Golden eagles were found to have a higher proportion of butyrylcholinesterase (75 percent) than acetylcholinesterase (25 percent) in plasma (Taira 1994). Blood samples from the treated nestlings had higher total ChE activity than untreated, but not significantly (*P* = 0.11). This was somewhat predictable in that blood samples were not taken until 3 to 5 weeks after exposure, and an overcompensation or "rebound effect" has been found in other species after light exposure to carbamates.

In summary, it appears that Sevin 4-Oil sprayed at the GHIPM rate offers little risk to nesting golden eagles. With global positioning system technology, spray planes could shut off and leave a small unsprayed area of a few acres or hectares around active nests, to leave the eagles completely unaffected. Similar studies of effects of malathion sprays (8 fl oz/acre or 0.58 kg/ha) for rangeland grasshopper control need to be conducted with young golden eagles.

References Cited

Adams, J. S.; Knight, R. L.; McEwen, L. C.; George, T. L. 1994. Survival and growth of nestling vesper sparrows exposed to experimental food reductions. The Condor 96: 739–748.

Beyers, D. W.; Sikoski, P. J. 1994. Acetylcholinesterase inhibition in federally endangered Colorado squawfish exposed to carbaryl and malathion. Environmental Toxicology and Chemistry 13: 935–939.

Beyers, D. W.; Keefe, T. J.; Carlson, C. A. 1994. Toxicity of carbaryl and malathion to two federally endangered fishes, as estimated by regression and ANOVA. Environmental Toxicology and Chemistry 13: 101–107.

Beyers, D. W.; Farmer, M. S.; Sikoski, P. J. 1995. Effects of rangeland aerial application of Sevin-4-oil on fish and aquatic invertebrate drift in the Little Missouri River, North Dakota. Archives of Environmental Contamination and Toxicology 28: 27–34.

Emlen, J. L. 1977. Estimating breeding season bird densities from transect counts. Auk. 94: 455–468.

Erwin, W.,J.; Sharpe, R. S. 1978. Effect of wide area ultra-low volume application of malathion on small mammal populations. Transactions of the Nebraska Academy of Science 5: 25–28.

Fair, J. M.; Kennedy, P. L.; McEwen, L. C. 1995a. Effects of carbaryl grasshopper control on nesting killdeer in North Dakota. Environmental Toxicology and Chemistry 14: 881–890.

Fair, J. M.; Kennedy, P. L.; McEwen, L. C. 1995b. Diet of nesting killdeer in North Dakota. Wilson Bulletin 107: 174–178.

George, T. L.; McEwen, L. C.; Fowler, A. C. 1992a. Effects of a carbaryl bait treatment on nontarget wildlife. Environmental Entomology 21: 1239–1247.

George, T. L.; Fowler, A. C.; Knight, R. L.; McEwen, L. C. 1992b. Impacts of a severe drought on grassland birds in western North Dakota. Ecological Applications 2(3): 275–284.

George, T. L.; McEwen, L. C.; Petersen B. E. 1995. Effects of grasshopper control programs on bird populations in western rangelands. Journal of Range Management 48: 336–342.

Grue, C. E.; Hart, A.D.M.; Mineau, P. 1991. Biological consequences of depressed brain cholinesterase activity in wildlife. In: Mineau, P., ed. Cholinesterase-inhibiting insecticides: their impact on wildlife and the environment. (Vol. 2 of the Chemicals in Agriculture series.) Amsterdam, London, New York: Elsevier: 152–209.

Howe, F. P. 1991. Two new host species for the parasitic blow fly *Protocalliphora braueri.* Wilson Bulletin 103: 520–521.

Howe, F. P. 1992. Effects of *Protocalliphora braueri* (Diptera: Calliphoridae) parasitism and inclement weather on nestling sage thrashers. Journal of Wildlife Diseases 28(1): 141–143.

Howe, F. P. 1993. Effects of a grasshopper insecticide application on diet, food delivery rates, growth, and survival of shrubsteppe passerines. Ph.D. dissertation. Fort Collins, CO: Colorado State University. 108 p.

Howe, F. P.; Knight, R. L.; McEwen, L. C.; George, T. L. 1996. Direct and indirect effects of insecticide applications on growth and survival of nestling passerines. Ecological Applications 6: 1314–1324.

Howe, F. P.; Knight, R. L.; McEwen, L. C.; George, T. L. 2000. Diet switching and food delivery by shrubsteppe passerines in response to an experimental reduction in food. Western North American Naturalist 60: 139–154.

McEwen, L. C. 1982. Review of grasshopper pesticides vs. rangeland wildlife and habitat. In: Peek, J. M.; Dalke, P. D., eds. Proceedings of the wildlife–livestock relationships symposium; 20–24 April 1981; Coeur d'Alene, ID. Moscow, ID: University of Idaho: 362–382.

Miller, C. K.; McEwen, L. C. 1995. Diet of nesting savannah sparrows in interior Alaska. Journal of Field Ornithology 66: 152–158.

Miller, C. K.; Knight, R. L.; McEwen, L. C. 1994. Responses of nesting savannah sparrows to fluctuations in grasshopper densities in interior Alaska. The Auk 111: 960–967.

Nicolaus, L. K.; Lee, H. 1999. Low acute exposure to organophosphate produces long-term changes in bird feeding behavior. Ecological Applications 9: 1039–1049.

Nimmo, D. W.; McEwen, L. C. 1994. Handbook of ecotoxicology-pesticides. In: Calow, P., ed. Handbook of ecotoxicology, vol. 2. Oxford, UK: Blackwell Scientific Publications. Chapter 8.

O'Toole, L. T.; Kennedy, P. L.; Knight, R. L.; McEwen, L. C. 1999. Postfledging behavior of golden eagles. Wilson Bulletin 111: 472–477.

Smith, G. J. 1987. Pesticide use and toxicology in relation to wildlife: organophosphorus and carbamate compounds. Resour. Publ. 170. U.S. Department of the Interior, Fish and Wildlife Service. 171 p.

Stevens, P. D. 1989. Acute toxicity and inhibition of cholinesterase activity in small mammals following exposure to methamidophos. M.S. thesis. Ft. Collins, CO: Colorado State University. 51 p.

Taira, T. 1994. Blood analysis of American kestrel and golden eagle nestlings exposed to malathion or carbaryl. M.S. thesis. Fort Collins, CO: Colorado State University. 80 p.

References Cited–Unpublished

Althouse, C. M.; McEwen, L. C.; Hoffman, L. A. Influence of prior northern grasshopper mouse *(Onychomys leucogaster)* and 13-lined ground squirrel *(Spermophilus tridecemlineatus)* live trap captures on succeeding intra and interspecific captures. Submitted to Journal of Mammalogy.

McEwen, L. C.; George, T. L.; Petersen, B. E.; Beyers, D. W.; Fowler, A. C.; Howe, F. P. 1989. Environmental monitoring. In: Cooperative Grasshopper Integrated Pest Management Project, 1988 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 82–102.

McEwen, L. C.; Petersen, B. E.; Beyers, D. W.; Althouse, C. M.; Fair, J. M.; Schleve, G. R.; Bednarski, L. T.; Taira, T. 1993. Environmental monitoring. In: Cooperative Grasshopper Integrated Pest Management Project, 1993 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 127–142.

McEwen, L. C.; Althouse, C. M.; Logan, J. A.; Stevens, P. D. (In preparation). Small mammal long-term population response to 2 rates of methamidophos spray on mixed shortgrass/*Atriplex* rangeland. Journal of Range Management or Environmental Toxicology and Chemistry.

O'Toole, L. T.; McEwen, L. C.; Knight, R. L. [In preparation.] Behavioral responses of post-fledging golden eagles *(Aquila chrysaetos)* to Sevin 4-Oil applications in the Little Missouri National Grassland, North Dakota. Environmental Toxicology and Chemistry.

Petersen, B. E.; DeWeese, L. R.; McEwen, L. C.; Palmer, D.; Ryder, R. A. Nesting success of lark buntings on rangeland treated with acephate. Submitted to Journal of Range Management.

III.3 Impact of Control Programs on Nontarget Arthropods

Mark A. Quinn

Introduction

Grasshopper control programs potentially can have a large impact on the rangeland ecosystem. Of particular concern are the effects of large-scale control programs on natural enemies of grasshoppers, pollinators of seed crops and endangered plant species, endangered species of vertebrates, and general biodiversity of grasslands. Here, I will be addressing two main questions: (1) What are the immediate and more long-term effects of grasshopper control treatments on nontarget species? and (2) Does the disruption in communities of nontarget arthropods affect the population dynamics of grasshoppers and the potential for outbreaks?

Effect of Grasshopper Control Treatments on Nontarget Arthropods

There is very little information on the effects of grasshopper control treatments on beneficial and other nontarget arthropods (animals with exoskeletons, such as insects, spiders, and crayfish). Insecticidal sprays can cause high mortality of grasshoppers, so it should be assumed that sprays can cause large reductions in other arthropod populations as well. The potential for a significant impact on nontarget arthropods is large because they are often very active when grasshopper control treatments are typically applied. For example, Quinn et al. (1993) showed a relationship between the presence of nymphal grasshoppers, the stage usually treated in control programs, and the activities of some groups of nontarget arthropods, such as ants, ground beetles, wolf spiders, sphecid wasps, and robber flies.

As part of the Grasshopper Integrated Pest Management (GHIPM) Project work in South Dakota, Quinn et al. (1990, 1991, 1993) studied the effects of large-scale aerial applications of bran bait containing 5 percent active ingredient (AI) carbaryl at 1.5 lb/acre (1.68 kg/ha), and ultralow-volume (ULV) malathion (91 percent AI) at 0.58 lb AI/acre (0.65 kg/ha) on nontarget arthropods of mixed-grass rangeland. Table III.3–1 lists the groups of nontargets that my colleagues and I collected with malaise (aerial) and pitfall (ground) traps before treatments were applied. Of all the groups of nontargets collected in malaise traps, only two are considered predators of grasshoppers—sphecid wasps (15 percent) and robber flies (3 percent). Both of these groups feed on a variety of insects and not just grasshoppers. The most abundant groups collected in malaise traps were ichneumonid wasps (32 percent) and moths (27 percent). Most of the ichneumonid wasps collected were Lepidoptera parasites.

Most of the groups of nontarget arthropods collected in the pitfall traps were grasshopper predators. The two most abundant groups were blister beetles (36 percent) and ants (31 percent). Blister beetle larvae may be significant predators of grasshopper egg pods (Parker and Wakeland 1957, Rees 1973). Ants feed on molting grasshoppers. Other abundant groups of nontarget arthropods were darkling beetles (11 percent), wolf spiders (8 percent), and ground beetles (7 percent).

Some groups of nontarget arthropods were affected by both the insecticidal bait and spray treatments (table III.3–2). Activities of darkling beetles, ground beetles, and field crickets were reduced by 49 percent to 89 percent after 1 week in plots treated with either the insecticidal bait or spray. The dominant species of darkling beetles and ground beetles were similarly reduced by the two treatments (Quinn et al. 1990, 1991). Populations of these groups did not change in the control plots over the same time period. These groups were most likely affected by the insecticidal bait because they either consumed the bait directly or because they fed on infected grasshoppers. Other groups were affected by the insecticidal spray, but not the bait. For example, activities of blister beetles and ichneumonid wasps were reduced by 59 percent and 56 percent, respectively, in the malathion spray plots but did not change in the bran bait or untreated (control) plots. Activities of two species of ground beetles, *Cratacanthus dubius* and *Discoderus parallelus*, were reduced by 81 percent and 66 percent, respectively, in the insecticidal bait plots but did not seem to be affected by the insecticidal spray.

Pfadt et al. (1985) conducted a study to determine the effects of ULV malathion at 8 fluid oz/acre (0.58 lb AI/acre) on nontarget organisms of shortgrass rangeland in Wyoming. Pfadt's team concluded that (1) aerial applications of insecticidal sprays are not likely to have a large impact on nontargets because most species are protected (in nests, soil, and plants), and (2) the only arthropods likely to be affected are those that inhabit

Table III.3–1—Relative abundance (percent) of nontarget arthropods collected with malaise and pitfall traps, July 2–8, 1986, at mixed-grass rangeland plots, Butte County, SD (adapted from Quinn et al. 1993)

Nontarget group		Feeding habits	Relative abundance
			Percent
Malaise traps			
Ichneumonidae	Ichneumonid wasps	Mostly moth parasites	31.6
Lepidoptera	Moths	Plant feeders (as larvae)	26.6
Sphecidae	Sphecid wasps	General predators*	14.7
Odonata	Damsel flies	General predators	9.4
Mutillidae/ Tiphiidae	Velvet ants/ tephiid wasps	Wasp, bee, and beetle parasites	9.3
Pompilidae	Spider wasps	Spider predators	5.8
Asilidae	Robber flies	General predators*	3.0
Chrysididae	Cuckoo wasps	Wasp and bee parasites	1.8
Halictidae	Halictid bees	Pollen feeders/bee parasites	1.4
Others			1.8
Pitfall traps			
Meloidae	Blister beetles	Pollen feeders/grasshopper egg predators*	35.9
Formicidae	Ants	Seed and plant feeders/general predators*	31.0
Tenebrionidae	Darkling beetles	General scavengers/detritus feeders	10.9
Lycosidae	Wolf spiders	General predators*	7.8
Carabidae	Ground beetles	General predators/plant feeders*	6.9
Gryllidae	Field crickets	General predators/plant feeders*	2.6
Buprestidae	Metallic wood-boring beetles	Plant feeders	1.6
Other spiders		General predators*	1.1
Others			2.2

*Feed on grasshoppers

Table III.3–2—Effect of carbaryl bran bait and malathion ULV spray on change in activities of nontarget arthropods between the pretreatment and 1 week posttreatment sampling intervals, Butte County, SD

Nontarget group	Trap	Treatment	% change ($\bar{x} \pm$ SEM[1])	n
Blister beetles	Pitfall	Bran bait	–10.1 ± 13.6	10
		Malathion	–58.5 ± 6.4	10
		Control	–35.1 ± 15.9	9
Ants	Pitfall	Bran bait	32.6 ± 43.6	7
		Malathion	–39.6 ± 3.0	9
		Control	509.3 ± 447.6	5
Darkling beetles	Pitfall	Bran bait	–89.3 ± 4.2	10
		Malathion	–80.9 ± 9.5	10
		Control	210.2 ± 132.4	8
Wolf spiders	Pitfall	Bran bait	–80.5 ± 4.9	10
		Malathion	–76.1 ± 4.1	10
		Control	–61.6 ± 13.2	9
Ground beetles[2]	Pitfall	Bran bait	–88.0 ± 4.6	10
		Malathion	–53.0 ± 8.4	9
		Control	41.8 ± 37.8	9
Field crickets	Pitfall	Bran bait	–82.5 ± 0.1	9
		Malathion	–49.3 ± 14.6	9
		Control	24.4 ± 64.2	6
Ichneumonid wasps	Malaise	Bran bait	143.9 ± 68.7	10
		Malathion	–56.1 ± 6.9	10
		Control	71.1 ± 35.6	8
Sphecid wasps	Malaise	Bran bait	0.1 ± 18.1	10
		Malathion	–17.5 ± 13.7	10
		Control	32.8 ± 61.9	8
Spider wasps	Malaise	Bran bait	–1.8 ± 24.4	10
		Malathion	–9.9 ± 39.7	10
		Control	50.0 ± 57.5	8
Robber flies	Malaise	Bran bait	39.8 ± 27.7	10
		Malathion	–29.5 ± 30.2	9
		Control	–44.9 ± 13.3	7

[1] Standard error of the mean.

[2] Does not include *Amara impuncticollis*, which was not present in traps before treatments but was present after treatments.

foliage during the day. For example, this study showed that the ant *Formica obtusopilosa,* which is commonly found foraging on flowers, was affected by the insecticides. However, colonies of all ant species were not affected. Pfadt's results also indicated that immature Ephemeroptera (mayflies) and Odonata (dragonflies and damselflies) in ponds may have been affected by the malathion.

Swain (1986 unpubl.) conducted a study on desert grassland in New Mexico to determine the effects of malathion ULV (8 oz/acre–0.58 lb AI/acre), carbaryl (0.54 lb AI/acre), and 2 percent (AI) carbaryl bran bait (1.5 lb/acre) on nontarget arthropods. Her study showed that mean abundance of most groups of nontargets declined immediately after treatments. In particular, all treatments seemed to affect populations of ants and only the insecticidal sprays affected populations of spiders.

Swain (1986) and Quinn et al. (1990, 1991, 1993) found that large-scale application of insecticidal sprays and baits had little long-term impact on the groups of nontargets examined. For example, my team found that activities of four dominant species of ground beetles and three dominant species of darkling beetles rebounded to the pretreatment levels 1 year after treatment. Only one species of darkling beetle, *Eleodes tricostatus*, may have been affected 1 year after treatment. Quinn et al. (1993) also found that field crickets, ichneumonid wasps, and blister beetles, as groups, rebounded to or above the pretreatment levels 1 year after treatment.

Pollinators, such as honey bees and solitary bees, are important components of rangeland and adjacent cropping systems. Although the effects of large-scale control treatments on bees have not been examined thoroughly, insecticidal sprays should be presumed to exert a serious impact on bee populations because they are particularly susceptible to commonly used insecticides (carbaryl, malathion). The effects of insecticides on native bees and rare rangeland plants are reviewed in chapters III.4 and III.5 in this section of the User Handbook.

In summary, large-scale applications of nonselective insecticidal sprays can cause large reductions in populations of nontarget species of arthropods immediately after treatment. Species that are active during treatments or that feed on infected grasshoppers are particularly susceptible. These include ground beetles, darkling beetles, blister beetles, spiders (especially wolf spiders), field crickets, foraging bees, and ants. In contrast, insecticidal baits affect only species that consume the baits directly or prey that have consumed the baits. These species include darkling beetles, ground beetles, field crickets, and ants.

Although reductions in nontarget arthropods can last throughout the year of application, there is little evidence that grasshopper control treatments cause any long-term effects on nontargets. Besides the resiliency of populations, there may be numerous other explanations for this lack of evidence of long-term treatment effects. Inadequate sample sizes and large population variability inevitably lead to a conclusion that treatments have no effect, when in fact, one may exist. No studies of nontarget arthropods have examined the possibility of making such an error (by conducting a statistical power analysis). An additional problem with existing studies is that they frequently assess effects on whole families and not species. When lumping of species is done, species emerging after treatments can dilute the effects of treatments and cause one to find no treatment effect when one actually exists (Quinn et al. 1993). Thus, these studies must be viewed with caution.

Effect of Control Treatments on Grasshopper Outbreaks

In general, nonselective insecticides can cause pest resurgence when they disrupt populations of natural enemies. Similarly, large-scale grasshopper control programs can potentially *enhance* grasshopper outbreaks by killing off grasshopper predators and parasites or by affecting their behavior. Although it seems clear that insecticide applications can affect natural enemies of grasshoppers, at least in the short term, it is less clear that reductions in natural enemies automatically affect grasshopper population dynamics.

Several chapters in this User Handbook address the effects of natural enemies on grasshoppers. Results from studies summarized in these chapters indicate that grasshoppers are attacked by a wide variety of predators and parasites and that grasshopper mortality can be quite high, at least on a local level. For example, birds can reduce grasshopper densities by 30 to 50 percent (see chapter I.10 on "Birds and Wildlife as Grasshopper Predators"). Parker and Wakeland (1957) estimated that an average of 19 percent of grasshopper egg pods were destroyed by predators but that at the local level, mortality may be as high as 100 percent. Parasitism rates of grasshoppers can also be quite high at the local level (exceeding 50 percent), although they do not usually exceed 10 percent (Lavigne and Pfadt 1966, Rees 1973). As discussed by Capinera (1987), the collective effects of all the different mortality factors may add up to an overall large effect on grasshoppers. It seems clear that we should not underestimate the effects of grasshopper natural enemies and that we should work to preserve these organisms.

There is some evidence that grasshopper populations are regulated by natural enemies (particularly birds) under certain conditions (see chapter VII.14 on "Grasshopper Population Regulation"). In effect, natural enemies may be responsible for keeping grasshopper populations at low levels. Once the natural enemies are removed (for example, by nonselective insecticides), then grasshopper populations can no longer be regulated and outbreaks can occur. Once grasshoppers reach high densities, natural enemies are no longer able to suppress their populations. Unfortunately, few studies have examined the role of natural-enemy reductions, caused by nonselective insecticides, on subsequent grasshopper outbreaks.

In a review of grasshopper population dynamics over several years, Lockwood et al. (1988) found that the duration and stability of grasshopper outbreaks were greater in northern Wyoming, compared with southern Montana, and suggested that the more intensive grasshopper control programs in Wyoming may have contributed to this. In a study of the effects of an insecticidal spray (malathion) and bait (carbaryl on bran) on grasshopper and nontarget arthropod populations, Quinn et al. (1989, 1991, 1993) found that populations of most dominant grasshopper species, four species of ground beetles, and numbers of other nontargets rebounded to or above pretreatment levels a year after treatment. An exception was *Ageneotettix deorum*. Densities of this species remained low a year after treatment. These results indicate that some nontarget arthropods and grasshopper species are very resilient. Clearly, until more is known about the effects of natural enemies on grasshopper population dynamics and the effects of grasshopper control programs on resiliency of natural enemies, scientists and land managers should act to preserve these communities.

References Cited

Capinera, J. L. 1987. Population ecology of rangeland grasshoppers. In: Capinera, J. L., ed. Integrated pest management on rangeland: a shortgrass prairie perspective. Boulder, CO: Westview Press: 162–182.

Lavigne, R. J.; Pfadt, R. E. 1966. Parasites and predators of Wyoming rangeland grasshoppers. Sci. Monogr. 30. Laramie, WY: University of Wyoming and Wyoming Agricultural Experiment Station. 31 p.

Lockwood, J. A.; Kemp, W. P.; Onsager, J. A. 1988. Long-term, large-scale effects of insecticidal control on rangeland grasshopper populations (Orthoptera: Acrididae). Journal of Economic Entomology 81: 1258–1264.

Parker, J. R.; Wakeland, C. 1957. Grasshopper egg pods destroyed by larvae of bee flies, blister beetles, and ground beetles. Tech. Bull. 1165. Washington, DC: U.S. Department of Agriculture: 1–29.
Pfadt, R. E.; Marston, N. L.; Lawson, F. A. 1985. Effect of ULV malathion grasshopper spray on non-target arthropods. Sci. Monogr. 48. Laramie, WY: University of Wyoming and Wyoming Agricultural Experiment Station. 31 p.

Quinn, M. A.; Kepner, R. L.; Walgenbach, D. D.; Bohls, R. A.; Pooler, P. D.; Foster, R. N.; Reuter, K. C.; Swain, J. L. 1989. Immediate and 2nd-year effects of insecticidal spray and bait treatments on populations of rangeland grasshoppers. Canadian Entomologist 121: 589–602.

Quinn, M. A.; Kepner, R. L.; Walgenbach, D. D.; Foster, R. N.; Bohls, R. A.; Pooler, P. D.; Reuter, K. C.; Swain, J. L. 1990. Effect of habitat and perturbation on populations and community structure of darkling beetles (Coleoptera: Tenebrionidae) on mixed-grass rangeland. Environmental Entomology 19: 1746–1755.

Quinn, M. A.; Kepner, R. L.; Walgenbach, D. D.; Foster, R. N.; Bohls, R. A.; Pooler, P. D.; Reuter, K. C.; Swain, J. L. 1991. Effect of habitat characteristics and perturbation from insecticides on the community dynamics of ground beetles (Coleoptera: Carabidae) on mixed-grass rangeland. Environmental Entomology 20: 1285–1294.

Quinn, M. A.; Kepner, R. L.; Walgenbach, D. D.; Foster, R. N.; Bohls, R. A.; Pooler, P. D.; Reuter, K. C.; Swain, J. L. 1993. Grasshopper stages of development as indicators of nontarget arthropod activity: implications for grasshopper management programs on mixed-grass rangeland. Environmental Entomology 22: 532–540.

Rees, N. E. 1973. Arthropod and nematode parasites, parasitoids, and predators of Acrididae in America north of Mexico. Tech. Bull. 1460. Washington, DC: U.S. Department of Agriculture. 288 p.

Swain, J. L. 1986. Effect of chemical grasshopper controls on nontarget arthropods of rangeland in Chaves County, New Mexico. M.S. thesis. Las Cruces, NM: New Mexico State University. 102 p.

III.4 Direct and Indirect Effects of Insecticides on Native Bees

D. G. Alston and V. J. Tepedino

NOTE: Acephate is no longer approved by EPA for rangeland grasshopper control.

The successful reproduction of plants in both natural and agricultural ecosystems is highly dependent upon adequate populations of pollinators. The role of bees as pollinators in natural ecosystems, such as rangelands, is less obvious to the casual observer. The fact is that the majority of rangeland plants require insect-mediated pollination. Native, solitary bee species are the most important pollinators on western rangelands (Tepedino 1979).

Indiscriminate use of broad-spectrum insecticides is likely to cause changes throughout the rangeland community. In addition to controlling the targeted pest (grasshoppers), rangeland insecticides can have direct and indirect effects on nontarget insects and related animals (see also III.3). Potential negative effects of insecticides on pollinators are of special concern because a decrease in their numbers has been associated with decline in fruit and seed production of plants. And this decline may have dramatic repercussions throughout the rangeland food chain. Some of the possible negative effects to the ecosystem include changes in future vegetation patterns via plant competition, reduction in seed banks, and influences on the animals dependent upon plants for food.

Direct effects are those that are lethal in nature and cause direct mortality that can be attributed to use of insecticides. Indirect or sublethal effects are much more difficult to document. They generally act over a longer period of time and can result in negative effects on reproductive potential, lifespan, activity levels, body size, and behavior of current and future generations.

Important Characteristics of Native Bees

When choosing the timing of insecticide applications to rangelands, one should consider some important characteristics of native bees, of the insecticide applied, and of the growth cycle of native plants. The typical solitary bee overwinters in its nest and emerges as an adult the following spring to early summer (fig. III.4–1). Adult females are exclusively responsible for feeding the young and thus play the major role in plant pollination while foraging for nectar and pollen.

There is tremendous variation among bee species in the length of time that adults are active and foraging (fig. III.4–1). The seasonal activity period of solitary bees may extend from spring through early fall due to multiple generations per year and continual availability of blooming plants. Therefore, land managers cannot assume that simply avoiding the application of insecticides on rangeland during the major time of plant bloom will avoid endangering the native bee population.

Exposure of bees to insecticides is also influenced by foraging behavior and flight distance. For most native bees, our knowledge of foraging behavior is limited to information on flower associations, such as a particular species that has been seen collecting the pollen and/or nectar of certain plants. The leaf-cutting habit of the alfalfa leafcutter bee makes it particularly susceptible to residues of contact insecticides on plant foliage. Contaminated leaves, mud, water, or resins used for nest construction may result in detrimental effects to the young. Bees' flight range can greatly affect their exposure to insecticides. Extensive flight distances between nests and flowering plants increase their foraging time and make them more vulnerable to insecticides (see III.8).

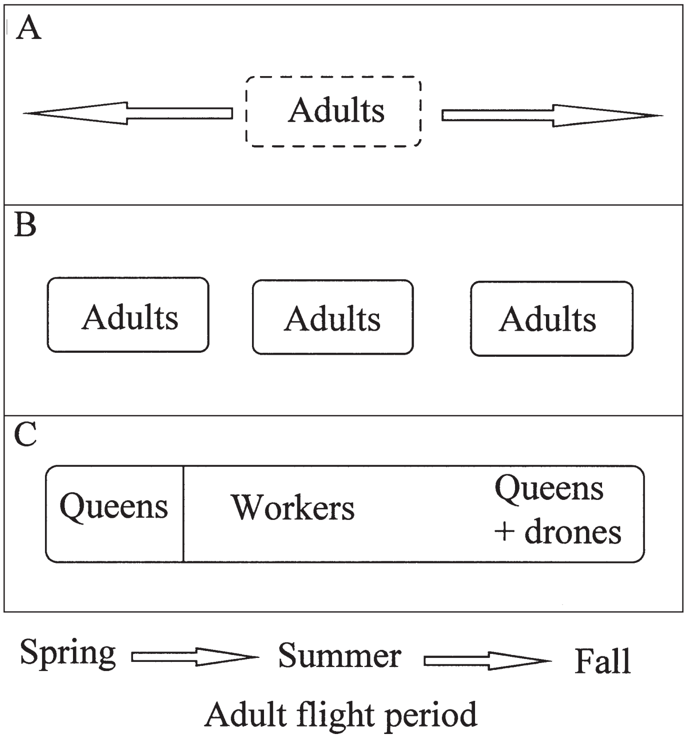


Figure III.4–1—Adult flight periods for three general life cycles of native bees: (A) Single generation per year, e.g., *Nomia* or *Osmia;* dotted lines indicate that flight period can shift in time depending on species. (B) Two or more generations per year, e.g., *Megachile* or *Ashmeadiella.* (C) Social, e.g., *Bombus.*

Body size of native bees also may affect susceptibility to insecticides in field situations. The greater surface-to-volume ratio of small bees increases their relative exposure to contact insecticides (Johansen 1972). Studies in a Montana forest (Flavell et al. 1975) found that, although the total bee population was not reduced following an application of the insecticide trichlorfon, the percentage of smaller bees (predominantly solitary species) present in the forest was significantly reduced. If this same effect is found in other ecosystems, then the greater susceptibility of smaller bees to insecticides is of particular concern for western rangelands.

Important Characteristics of Insecticides

Pesticide formulation strongly influences toxicity. Dusts and wettable powders tend to be more hazardous to bees than solutions or emulsifiable concentrates, while granular and bait formulations are generally low in hazard. Application technique is also important in determining toxicity; aerial spraying offers less opportunity for avoidance behavior and greatly increases drift (National Research Council of Canada 1981).

Currently, only broad-spectrum insecticides (acephate, carbaryl, and malathion) are registered for use on rangelands for grasshopper control. All three have received a high toxicity rating for their negative effects on bees (National Research Council of Canada 1981, Johansen and Mayer 1990, Johansen et al. 1983), and, therefore, are not registered for use on blooming crops or weeds if commercial bees are visiting the treatment area. Yet these insecticides are being sprayed on rangelands when native plants are in bloom and being visited by pollinators. Contact sprays can be very toxic to small, native bees because of direct contact with the insecticide or insecticide residue. Therefore, insecticides that are more selective in activity are highly desirable to reduce negative effects on bees.

One insecticide with promise for selectivity is carbaryl incorporated into bran flakes. Because such flakes act only upon ingestion, they are much more selective than contact formulations (Peach et al. 1994). Bees likely would encounter bran bait only when gathering pollen and nectar from open upright flowers into which particles of bait have fallen. Ingestion of the insecticide would have to occur in order for the bee to receive a toxic dose.

Lethal Effects

The direct, or lethal, effects of insecticides on bees have been the focus of much research. The majority of toxicological information has been obtained for three distantly related species: *Apis mellifera,* the honey bee; *Nomia melanderi*, the alkali bee; and *Megachile rotundata,* the alfalfa leafcutting bee. Toxicological data for the latter two species are of greater relevance to natural situations because of these bees' solitary nesting lifestyle and the primary role of adult females in foraging activities and provisioning the young. The greatest body of toxicity literature exists for the honey bee, but unfortunately these data have proved of limited use in prediction of toxicity to many species of native bees because of the major differences in lifestyle, behavior, physiology, and size.

On western rangelands where native plants are rare or their populations threatened, bait formulations of carbaryl have been suggested as a possible alternative to contact sprays. Liquid formulations of carbaryl can be quite toxic to all three bee species previously mentioned when bees directly contact insecticides or insecticide residues (Johansen and Mayer 1990). In contrast, under laboratory conditions, only extremely high doses of ingested carbaryl resulted in toxic effects to alfalfa leafcutting bee larvae when incorporated into the pollen provision either as liquid (Guirguis and Brindley 1974) or as bran bait (Peach et al. 1994). Such high rates of carbaryl are much greater than a bee would encounter in the field.

There were also no lethal effects of carbaryl bran bait on adult alfalfa leafcutting bees, even when they were fed a sustained diet of honey solution contaminated with carbaryl bait for up to 40 days (Peach et al. 1994). Other studies have found that young adult bees of this species (up to 4 days old) readily detoxify topically applied carbaryl, but this ability rapidly declines after day 4 (Lee and Brindley 1974).

Sublethal Effects

Other effects of insecticides to bees may not be as obvious. The long-term sublethal effects of insecticides to bees that would be most likely to lower visitation rates to flowers, and thereby reduce plant reproductive success, include negative changes in longevity of bees, adult activity levels, and number, size, and sex ratio of offspring produced. Such chronic effects could occur from the slow poisoning of the young through ingestion of contaminated pollen and exposure of foraging bees to insecticides through translocation in nectar. Although sublethal effects of insecticides can be subtle, in the long run they may have as great a weakening effect on bee populations as the mortality caused by direct toxicants.

Although few studies have addressed the subtle effects of insecticides on bees, some detrimental effects have been found. Female alfalfa leafcutting bees treated with contact applications of organophosphate insecticides showed reduced longevity and lower nesting rates and egg production than bees not treated (Torchio 1983, Tasei and Carre 1985, Tasei et al. 1988).

Approximately 40 percent of larvae of this bee fed provisions contaminated with deltamethrin could not successfully complete development (Tasei et al. 1988). However, studies with carbaryl bran bait found no sublethal effects on adults or larvae (Peach et al. 1994). There seems to be little reason for concern that any carbaryl eaten by foraging adult females from the nectar of open flowers will affect any aspect of reproduction. Again, it appears that the use of carbaryl bran bait on rangelands is a relatively safe option for pollinators (fig. III.4–2).

Figure III.4–2—Domestic bees often need protection during grasshopper conrol treatments using chemical sprays. Beekeepers can move the bees out of the application area, or control-program managers can leave a sufficient buffer zone to protect the bees. Applications of bran bait normally will be of little concern for beekeepers. (APHIS file photo.)

Implications for Management of Grasshoppers on Western Rangelands

Because of the multiple-use concept employed by managers of public lands, there is certain to be continual conflict among different users of the lands. The U.S. Department of Agriculture, Forest Service and the U.S. Department of the Interior, Bureau of Land Management have the unenviable task of making land-management decisions based on wide-ranging demands and input from recreational use and preservation of biodiversity to logging, mining, and grazing. Because of the current status of pest management technology, it is likely that use of insecticides for control of grasshoppers on western rangelands will continue for some time. Despite this current situation of conflict, there does appear to be some alternative in choice of insecticides that are more selective in their effects to nontarget plants and animals.

One such selective insecticide that appears well suited for use on rangelands is carbaryl bran bait. Demanding laboratory and greenhouse tests performed with the alfalfa leafcutting bee, a solitary nester, found no lethal or sublethal effects on adults and only minimal effects on larvae when doses much higher than would be encountered in the field were incorporated into their pollen provisions. However, there are more limitations to choosing carbaryl bran bait as a rangeland pest control tool. Because not all grasshopper species feed equally well on the bait (see II.12), proper identification of grasshopper species is especially important.

Although carbaryl bran bait may be a relatively safe option for a representative solitary bee, no one should feel comfortable with this assessment until there is further research on other pollinator species' susceptibility to various insecticides. Such research is critical for the preservation of insect biodiversity, as well as the biodiversity of the plants whose flowers cannot reproduce sexually without insect visits.

References Cited

Flavell, T. H.; Tunnock, S.; Barry, J. W.; Ekblad, R. B.; Ciesla, W. M. 1975. Western spruce budworm: a pilot control project with carbaryl and trichlorfon—environmental monitoring of aquatic organisms, birds, and insect pollinators. March 1978. Rep. 787-5. Missoula, MT: U.S. Department of Agriculture, Forest Service.

Guirguis, G. N.; Brindley, W. A. 1974. Insecticide susceptibility and response to selected pollens of larval alfalfa leafcutting bees, *Megachile pacifica* (Panzer)(Hymenoptera: Megachilidae). Environmental Entomology 3: 691–694.

Johansen, C. A. 1972. Toxicity of field-weathered insecticide residues to four kinds of bees. Environmental Entomology 1: 393–394.

Johansen, C. A.; Mayer, D. F.; Eves, J. D.; Kious, C. W. 1983. Pesticides and bees. Environmental Entomology 12: 1513–1518.

Johansen, C. A.; Mayer, D. F. 1990. Pollinator protection: bee and pesticide handbook. Cheshire, CT: Wicwas Press. 212 p.

Lee, R. M.; Brindley, W. A. 1974. Synergist ratios, EPN detoxification, lipid, and drug-induced changes in carbaryl toxicity in *Megachile pacifica.* Environmental Entomology 3: 899–907.

National Research Council of Canada. 1981. Pesticide–pollinator interactions. Ottawa, ON: National Research Council of Canada. 190 p.

Peach, M. P.; Alston, D. G.; Tepedino, V. J. 1994. Bees and bran bait: is carbaryl bran bait lethal to alfalfa leafcutting bee (Hymenoptera: Megachilidae) adults or larvae? Journal of Economic Entomology 87: 311–317.

Tasei, J. N.; Carre, S. 1985. Effets du traitement de luzerne en fleurs *(Medicago sativa* L.) avec de la deltamethrine et de la phosalone sur l'abeille solitaire: *Megachile rotundata* F. (Hym., Megachilidae). Acta Oecologica 6: 165–173.

Tasei, J. N.; Carre, S.; Moscatelli, B.; Grondeau, C. 1988. Recherche de la D.L. 50 de la deltamethrine (Decis) chez *Megachile rotundata* F. abeille pollinisatrice de la luzerne (*Medicago sativa* L.) et des effets de doses infralethales sur les adultes et les larves. Apidologie 19: 291–306.

Tepedino, V. J. 1979. The importance of bees and other insect pollinators in maintaining floral species composition. Great Basin Naturalist Memoirs 3: 139–150.

Torchio, P. F. 1983. The effects of field applications of naled and trichlorfon on the alfalfa leafcutting bee, *Megachile rotundata* (Fabricius). Journal of the Kansas Entomological Society 56: 62–68.

III.5 The Reproductive Biology of Rare Rangeland Plants and Their Vulnerability to Insecticides

NOTE: Acephate is no longer approved by EPA for rangeland grasshopper control.

Vincent J. Tepedino

The Western United States is an area of high plant and animal diversity. Many of the plants on this vast expanse of mountain, plain, and desert occur nowhere else in the world (Cronquist et al. 1972, Barbour and Billings 1988). Currently about 150 of these plant species are so rare that they have been listed under the Endangered Species Act as either threatened or endangered. Four are shown in figure III.5–1 (a–d). Most of these rare plants have been found on public rangelands (fig. III.5–2).

Figure III.5–1—Rare rangeland plants. A = Blowout penstemon (Nebraska), B = Dwarf bear-poppy (Utah), C = Dudley Bluffs twinpod (Colorado), D = San Rafael cactus (Utah).

Preserving rare plant species means removing or reducing threats to existing individuals and ensuring that those individuals can reproduce. Plants reproduce both asexually and sexually. For example, the rare plants *Cycladenia humilis* var. *jonesii* in Utah and *Mirabilis macfarlaneii* in Idaho and Oregon both reproduce sexually by seeds and asexually by the production of rhizomes. However, in seed plants, sexual reproduction is the predominant method. All rare plants that my associates and I studied and described in this chapter reproduce sexually. Sexual reproduction is particularly important because it enables plants to generate and maintain in their offspring the genetic variability necessary to cope with unusual circumstances. In contrast, asexual reproduction produces only copies of the parent plant, not variations on the theme.

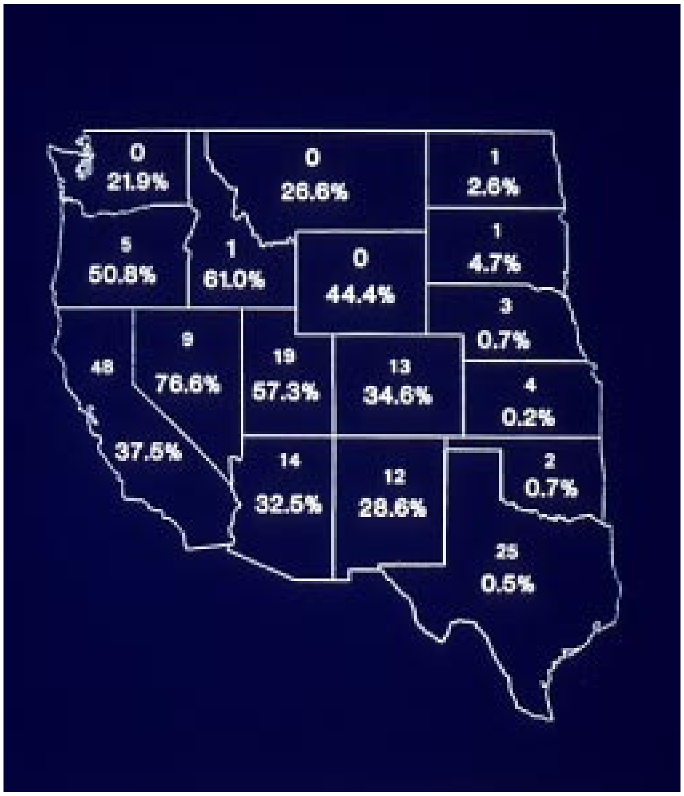


Figure III.5–2—Number of threatened and endangered plant species listed under the Endangered Species Act as of August 1993 (U.S. Fish and Wildlife Service 1993, upper figure) and percent total area administered by the Bureau of Land Management and Forest Service (lower figure), by State, in the West.

In seed plants, sexual reproduction depends on the movement of mature pollen from the anthers to a receptive stigma (pollination). To complete the process, pollen grains must germinate and send pollen tubes down the style to fertilize one or more ovules in the ovary (fertilization). Sexual reproduction may take place between individuals, or individuals may fertilize themselves if they are self-compatible, meaning their stigmas are receptive to their own pollen.

Because plants are immobile, they require “go-betweens” to move pollen from anthers to stigma. Such assistance comes mostly from insects–although wind, water, gravity, and other animals may occasionally be agents of pollination for some species. Although butterflies, moths, flies, ants, and beetles may pollinate flowers as they visit them to eat pollen and/or nectar, the truly essential pollinators for North American flowering plants are bees.

The bees to which we refer are not honeybees, which are of Eurasian origin, but native bees, which have evolved in North America. The North American bee fauna is quite diverse. In the State of Wyoming alone, there are more than 600 species (Lavigne and Tepedino 1976). In the Western United States, there are well over 2,500 species. Many of these bees are quite specialized in the plants that they visit and pollinate. For example, *Perdita meconis*, an uncommon bee that pollinates the endangered dwarf bearclaw poppy, *Arctomecon humillis*, visits only plants in the genera *Arctomecon* and *Argemone* for pollen.

Most bees that visit rare plants are solitary rather than social (the familiar honeybee). Like social bees, solitary bee females care for their offspring. Individual females carefully construct nests without the aid of workers, usually in the ground (fig. III.5–3) or in dead wood (fig. III.5–4). These nests will hold and protect the young bees and the food provided for them. The nesting material varies from species to species and may be quite specific. For example, for certain species, the ground must have a certain slope or soil moisture content or texture (Cane 1991).

Figure III.5–3—Entrance/exit holes at a nest-site of a ground-nesting bee.

Bees provision these nests with pollen and nectar molded into a loaf (fig. III.5–4) for the young to eat. Adults also eat nectar and pollen while foraging. In addition, bees may forage for water or other extraneous materials needed to construct the nest, such as leaf pieces (fig. III.5–5), resin, mud, etc., (Stephen et al. 1969). Adult females must launch many foraging expeditions from their nest-sites to obtain these resources. Frequently the best nesting substrate is not in the same area as food or other necessities, and bees must travel some distance to obtain nest materials.

Unfortunately, bees are generally vulnerable to most commonly used insecticides, including those that are approved for use to control grasshoppers on Federal rangelands: acephate, carbaryl, and malathion (Johansen et al. 1983). Bees that are forced to travel widely to gather their resources are most vulnerable because they must forage over larger areas and are therefore more likely to encounter a spray area. If bees are vulnerable, so may be the plants that depend on them for pollination services. Because of the potential vulnerability of both bees and plants, the U.S. Department of the Interior's U.S. Fish and Wildlife Service (FWS) and the U.S. Department of Agriculture's (USDA) Animal and Plant Health Inspection Service must hold joint consultations before aerially treating rangelands with insecticides. Usually, insecticide-free safety zones called buffers must be left around rare plant populations to reduce effects on both plant and pollinators.

Figure III.5–4—The nest of a twig-nesting bee, split open to expose feeding larvae, their food provisions, and the partitions between cells.

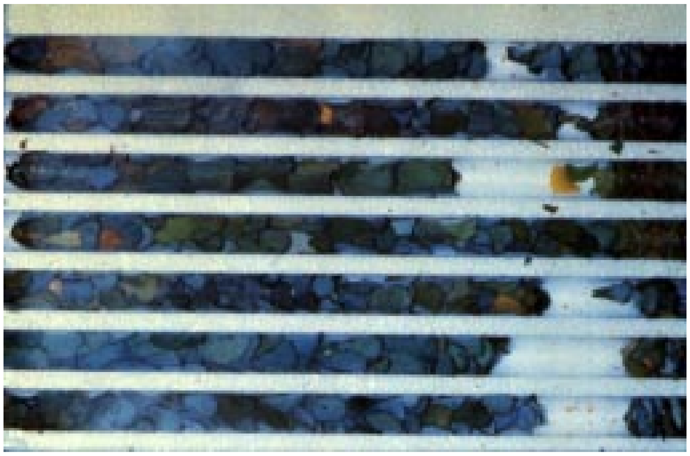


Figure III.5–5—Several leafcutter bee nests in an artificial domicile, exposed to show the numerous cells enfolded in leaves.

Questions about optimal buffer zone size and vulnerability of rare plant reproduction to insecticides are important. If flowers normally self-fertilize automatically, then grasshopper spraying programs are unlikely to be of consequence because pollinators will not be necessary for reproduction. Thus, scientists first must determine whether the flowers of the plant species in question are capable of self-fertilization, and, second, if self-fertilization is automatic. We also must determine whether fruit and seed set are improved by cross-pollination and identify the agents of pollination. When this is accomplished, we will have described the breeding system of the plant and will have some idea about the life history of its pollinators.

The size of the buffer zone that should be left around rare plant populations that rely exclusively on insect pollination depends on how far bees fly to obtain their resources. Presently, a buffer zone of 3 miles is being left around rare plant populations, but this is provisional in that it is based on best guesses rather than accurate estimates. By experimentation, we can help resolve questions about the value of buffer zones and whether they should be expanded or contracted in size.

Conducting a Study

To uncover general patterns in the reproductive biology of rare plants on western rangelands, I elected to study the breeding systems and pollinators of a large number of species rather than to conduct very detailed studies on a few species.

I gave study priority to rare plant species on actively grazed public rangelands (fig. III.5–6) in counties with high probabilities of having large numbers of grasshoppers, and thus of being sprayed. The approximate locations of the species studied are shown in figure III.5–7. With two exceptions *(Penstemon harringtonii* in Colorado and *Castilleja aquariensis* in Utah), all are listed as threatened or endangered under the Federal Endangered Species Act.

Figure III.5–6—Cattle grazing at a Brady pincushion cactus site (Arizona).

To describe the plant breeding system, we conducted a series of experiments using mesh bags or cages to prevent insects from visiting the flowers. Individual flowers, entire inflorescences (flower clusters), or entire plants (where necessary) were bagged or caged just prior to the onset of flowering (fig. III.5–8). Each of the following treatments was applied to a different flower: for self-pollination, flowers were hand-pollinated with the pollen of another flower on the same plant; for cross-pollination, flowers were hand-pollinated with pollen from a flower on a distant plant; to test for automatic self-pollination, flowers were left untreated; and, as a control, some flowers were left unbagged (open-pollinated). My associates and I carried out a complete series of treatments, one of each, on each of 15 to 25 experimental plants. Treatments were randomized on each plant to remove any effects of order or position on fruit or seed set.

We observed and collected naturally occurring pollinators as they visited the flowers during several time periods each week. Insects were pinned and identified later using the insect collections at the USDA, Agricultural Research Service, Bee Biology and Sytematics Laboratory in Utah, and the collection at Utah State University.

Estimating the distances a bee typically flies on its foraging trips proved very difficult because of its size, the speed at which it moves, and the size of the area to be monitored. Because native bees are too small to track with radio collars or electronic chips, as many mammals and birds can be, other methods were necessary. We used both direct (A below) and indirect (B, C, D) methods:

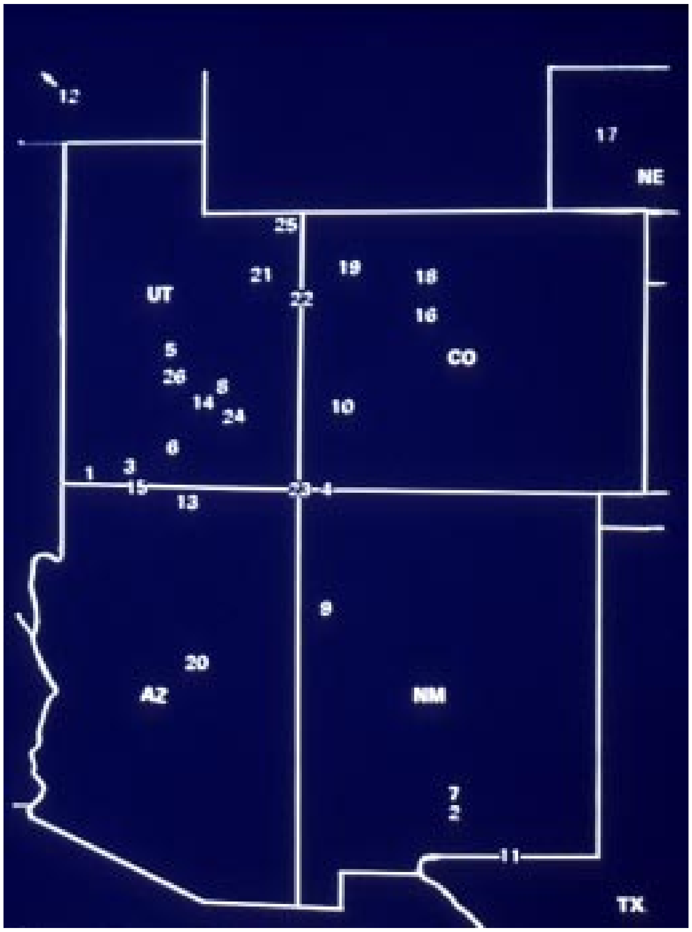


Figure III.5–7—Locations of specific threatened and endangered plants studied from 1988 to 1993. 1 = dwarf bear-poppy, 2 = Sacramento prickly-poppy, 3 = Welsh's milkweed, 4 = Mancos milkvetch, 5 = Heliotrope milk-vetch, 6 = Aquarius paintbrush, 7 = Sacramento Mountains thistle, 8 = Jones' cycladenia, 9 = Zuni fleabane, 10 = clay-loving wild-buckwheat, 11 = McKittrick pennyroyal, 12 = McFarlane's four-o'clock, 13 = Brady pincushion cactus, 14 = San Rafael cactus, 15 = Siler pincushion cactus, 16 = Harrington beard-tongue, 17 = blowout penstemon, 18 = Penland beard-tongue, 19 = Dudley Bluffs twinpod, 20 = Arizona cliffrose, 21 = shrubby reed-mustard, 22 = Uinta Basin hookless cactus, 23 = Mesa Verde cactus, 24 = Wright fishhook cactus, 25 = Ute ladies'-tresses, 26 = last chance townsendia.

(A) Foraging bees were captured, marked on the thorax with a dot of water-resistant paint that was nontoxic to plants and insects, released, and then searched for on subsequent days at other plant populations at set distances from the marking site (fig. III.5–9 and 10).

(B) Nontoxic fluorescent powders (pollen analogs or imitators) were placed in "donor" flowers, where they would be picked up and spread by foraging bees, and were searched for in the evening with a black light in other flowers at different distances from the donors.

Figure III.5–8—Fitting a cage over a cactus plant to exclude insects.

Figure III.5–9—Coaxing a bee into a marking tube.

Figure III.5–10—The coaxed bee marked on the thorax.

Figure III.5–11—An artificial bee "condominium" offers bees cheap housing.

(C) Trap-nests (artificial nests that bees will use, figure III.5–11) were placed at different distances from donor flowers, and the provisions of the cells made therein were examined for fluorescent powder.

(D) A "mobile garden," a pickup truck with a bed full of blooming potted plants, was used to attract marked bees that had earlier foraged on flowers dusted with fluorescent powders (see above) (fig. III.5–12). The "mobile garden" was parked at different distances from areas where bees had been marked and flowers had been dusted. My associates and I then recorded marked bees visiting plants in the garden or any flowers with fluorescent powder deposited on them.

Figure III.5–12—The oldest floating "mobile garden" in Arizona.

Study Results

Three clear patterns were evident from the data. First, rare plants do not tend to be automatic self-fertilizers. Indeed, just the opposite is the case. With the exception of two species *(Astragalus montii* in central Utah and *Schoencrambe suffrutescens* in eastern Utah), all species are primarily outcrossing (table III.5–1). Many are also self-compatible, meaning pollen moved from one flower to another on the same plant will sometimes cause fertilization, but in most cases the fruits and seeds produced are inferior either in number or size to those produced as a result of cross-pollination. In any case, pollinators also are needed to cause this type of self-pollination, which is not automatic.

The second pattern is that the most abundant visitors to the flowers of these plants are almost always native bees (table III.5–1). In some cases, bee pollination is supplemented by other animals. For example, in New Mexico the Sacramento Mountains thistle *(Cirsium vinaceum)* also is pollinated by several species of hummingbirds, flies, and butterflies.

Table III.5–1—Summary of the reproductive characteristics of 26 species of rare plants

Common name	Species name	Status	State	BrSys	I	Pollinators	L
Dwarf bear-poppy	*Arctomecon humilis*	E	UT	CR SI	Y	Bees, many	N
Sacramento prickly-poppy	*Argemone pleiacantha pinnatisecta*	E	NM	CR PS	Y	*Dialictus*	?
Welsh’s milkweed	*Asclepias welshii*	T	UT	?	Y	Bees, wasps	?
Mancos milk-vetch	*Astragalus humillimus*	E	CO NM	CR SC	Y	Bees, many	N
Heliotrope milk-vetch	*Astragalus montii**	T	UT	AS SC	?	*Osmia*	N
Aquarius paintbrush	*Castilleja aquariensis**		UT	CR SI	Y	*Bombus*	?
Sacramento Mountains thistle	*Cirsium vinaceum*	T	NM	CR PS	Y	Various	?
Jones cycladenia	*Cycladenia humilis* var. *jonesii**	T	UT	CR SI	Y	Bees, many	?
Zuni fleabane	*Erigeron rhizomatus*	T	NM	CR PS	Y	Various	N
Clay-loving wild-buckwheat	*Eriogonum pelinophilum*	E	CO	CR SC	Y	Various	?
McKittrick pennyroyal	*Hedeoma apiculatum*	T	NM TX	CR SC	Y	Halictidae	N
MacFarlane’s four-o’clock	*Mirabilis macfarlanei**	E	ID OR	CR PS	Y	Bees, many	?
Brady pincushion cactus	*Pediocactus bradyi*	E	AZ	CR SI	Y	*Dialictus*	N
San Rafael cactus	*Pediocactus despainii*	E	UT	CR SI	Y	Bees, many	N
Siler pincushion cactus	*Pediocactus sileri*	E	AZ UT	CR SI	Y	Bees, many	N
Harrington beardtongue	*Penstemon harringtonii*		CO	CR PS	Y	Bbees, many	?
Blowout penstemon	*Penstemon haydenii*	E	NE	CR PS	Y	Bees, many	N
Penland beardtongue	*Penstemon penlandii*	E	CO	CR SC	Y	Bees, many	N
Dudley Bluffs twinpod	*Physaria obcordata*	T	CO	CR SI	Y	Bees, many	N
Arizona cliffrose	*Purshia subintegra*	E	AZ	CR PS	Y	Bees, many	Y
Shrubby reed-mustard	*Schoencrambe suffrutescens**	E	UT	AS SC	?	Halictidae	N
Uinta Basin hookless cactus	*Sclerocactus glaucus**	T	CO UT	CR SI	Y	Bees, many	Y
Mesa Verde cactus	*Sclerocactus mesae-verdae**	T	CO NM	CR PS	Y	Halictidae	N
Wright fishhook cactus	*Sclerocactus wrightiae*	E	UT	CR SI	Y	Halictidae	N
Ute ladies’-tresses	*Spiranthes diluvialis**	T	CO UT	CR SC	Y	*Bombus*	N
Last chance townsendia	*Townsendia aprica*	T	UT	CR PS	Y	*Osmia*	N

T = threatened, E = endangered. BrSys describes the plant’s breeding system: CR = cross-pollinated, AS = automatic self-pollination, SI = self-incompatible, SC = self-compatible; PS = partially self-compatible. I = insect pollinated, Y = yes. Pollinators: genus or family of bee given when possible, many = several bee taxa, various = several animal taxa. L = evidence that fruit or seed set is being limited by inadequate pollination, N = no, Y = yes; * = uncommonly visited species.

The third pattern is that the flowers of about one-third of the plant species studied received few visits (table III.5–1). For several species, insect visitation was so low that we were forced to abandon the original pollinator observation and collection schedules. In these cases insects were simply captured whenever possible. Such low numbers of flower visitors are of concern, especially for rare plants that can produce seeds only when visited by pollinators.

These experiments also can be used to indicate species that may be producing fewer than the highest number of seeds, perhaps because of insufficient pollinator visits. Species whose seed production is low are of special concern because they may not be producing enough new individuals to replace those that are dying. Fortunately, only *Purshia subintegra* in central Arizona and *Sclerocactus glaucus* in eastern Utah gave any indication of underpollination. Because these two species set significantly fewer seeds in open-pollinated treatments than in cross-pollinated treatments, these plants should be studied further to determine if underpollination is common.

My results in estimating distances traveled by foraging bees were surprising. While it was easy to recapture bees in the general vicinity in which they were marked, or to detect fluorescent powders in flowers in the general area of the donor flowers, it was very difficult to find either marked bees or fluorescent particles at distances beyond a few dozen yards from the marking point. The record for distance moved was about a quarter mile (400 m) from a donor flower in a study of *Pediocactus sileri* in northern Arizona (Peach et al. 1993).

Implications for Chemical Sprays

To say that most plants reproduce sexually and that most depend on insects to pollinate them does not necessarily mean that rare plants do so. Indeed, prior to this study, there were reasons to suspect that rare plants were more likely than common plants to automatically self-pollinate and less likely to require insect visitors to achieve sexual reproduction (Tepedino 1979, Karron 1991). If this were true, then insecticide spraying for grasshoppers would have little effect on reproduction by rare plants, and land managers would not need to be concerned about the potential effects on the plants' pollinators.

The results obtained in this study show that rare plants on rangelands do not commonly self-pollinate. Almost all species studied set seed only when native bees visit their flowers. Because these bees are likely susceptible to liquid insecticide sprays, land managers should consider the implications of some reduction in pollinators as a result of spraying. Significant reduction of pollinators is likely to reduce the seed production of rare plants.

In addition, land managers should consider that many of the insect pollinators may be vulnerable to insecticides at any time of the year. Unless there is a perfectly synchronized, one-generation-per-year specialist pollinator for a plant, and my associates and I found none of those, the conservative approach—until more is known—is to avoid spraying within the buffer zone around each rare plant population at any time. However, if the plan is to use carbaryl bran bait (2 percent active ingredient), a nonliquid treatment, no buffer zones are needed (see III.4).

Overall, the pollinator situation on Federal rangelands may not be as perilous as some scientists had feared. Despite past spraying history, there is little indication that rare plants on rangelands are currently producing fewer seeds than they are capable of producing. While this is a conclusion that cries out for additional corroboration, it is also encouraging to find that seed production of open-pollinated flowers of rare plants do not seem to be pollinator limited. In most cases, visitation rates of bees to flowers, and by implication, bee numbers, appear to be sufficient to support maximum seed production. It is probable that bee numbers and seed production of native forbs have not been impacted because large-scale insecticide spray programs to control or suppress populations of grasshoppers on rangeland are not usually applied in the same areas in successive years. This policy must continue if rangeland pollinators are to have ample time to recover from spray episodes. Other researchers working in Canadian forests have shown that bee numbers will usually return to prespray levels in 1 to 3 years, depending upon the species of bee and the insecticide used (Plowright and Thaler 1979, Kevan and LaBerge 1979, Wood 1979, Miliczky and Osgood 1979). Recovery times and patterns for rangeland pollinators also should be studied.

Scientists regard the absence of evidence for long-distance movement of pollen grain analogues (fluorescent powders) less as evidence that native bees do not move long distances than as an indication of a logistical problem in testing. It is simply impossible for one or two people effectively to cover the area that must be censused. A complicating factor is that every study to look at pollen dispersal has reported drastic reductions in pollen deposition with distance (Handel 1983). By the time one samples flowers more than 33 ft (10 m) from the source, the number of pollen grains deposited is minimal. Again, this does not mean that pollen flows only over very short distances but that investigators are faced with detecting a very small needle in a very large haystack.

Other studies of bee movement and gene flow are of little help because they are invariably conducted over relatively short distances (Handel 1983). Pollen can, however, move long distances. Kernick (cited in Levin 1984) noted that several species of crop plants must be isolated by as much as 1.24 miles (2 km) to maintain varietal purity. Several other studies have examined the homing ability of solitary species of bees. They have shown that bees are capable of returning to their nests from distances of up to 5 miles (Fabre 1925, Rau 1929 and 1931; reviews by Packer 1970 unpubl. and Roubik 1989). While such experiments in no way tell us the distance that a bee normally flies on a typical foraging trip, they help to put an upper bound on bees' movements.

Conclusions

Although much valuable information has been obtained on both plants and their pollinators, much remains to be done. There are four areas in which additional research should be encouraged. First, the pollination biology of other plant species listed under the Endangered Species Act must be studied. The Grasshopper Integrated Pest Management Project has supported studies of 26 species in 13 families (see table III.5–1) or roughly 17 percent of the plant taxa in the Intermountain West which are listed under the Endangered Species Act. Thus, we feel confident in concluding that, in general, the flowers of rare plants must be pollinated by native bees to produce seeds. However, unless administrators and land managers are willing to assume that all rare plants must be managed as if they required bee pollinators, the reproductive biology of the remaining species must be studied.

Second, to make informed recommendations about the size of buffer zones to be left around rare plant populations, better information is needed on the distances pollinators and/or pollen travel. Laboratory methods that demonstrate genetic differences between the enzymes produced by different plants can be used, together with theoretical population genetic models, to provide information on gene flow between plant populations separated by a range of distances and on the genetic isolation of selected plant populations (Slatkin 1985 and 1993, Slatkin and Barton 1989). Long-distance pollinator movement can be documented by showing that certain forms of particular enzymes, which are primarily or exclusively restricted to one population, have moved to other populations. Indeed, these techniques can be used to give a rough approximation of the average number of individual plants per generation that are the result of pollen migration between populations.

Third, information is needed on the toxic effects to native bees of the liquid insecticides commonly used to treat rangeland grasshoppers. Current knowledge has been obtained from studies of the honey bee and the alfalfa leafcutter bee (both introduced species) and the alkali bee because they are cultured for crop pollination and are easily obtainable. Little is known about how susceptible the 2,500-plus species of rangeland bees are to insecticides because their populations are too small, or too difficult to obtain, to yield adequate sample sizes for experimentation of this kind. Prior to studying the toxicology to native species, it will be necessary to build up their populations to a sufficient size for experimentation by raising them in large field cages or greenhouses.

Fourth, decisionmakers must be advised when it is safe to spray. As noted earlier in this chapter, such decisions cannot be made by simply using flowering phenology records for the rare plant species because its pollinators may be active at other times of the year. Information must be available on the flight times of adult pollinators and on their activity patterns for the potential season of spraying. Thus far, activity patterns for pollinators of only one rare plant species have been studied (Peach et al. 1993).

References Cited

Barbour, M. G.; Billings, W. D., eds. 1988. North American terrestrial vegetation. New York: Cambridge University Press. 434 p.

Cane, J. H. 1991. Soils of ground-nesting bees (Hymenoptera: Apoidea): texture, moisture, cell depth and climate. Journal of the Kansas Entomological Society 64: 406-413.

Cronquist, A.; Holmgren, A. H.; Holmgren, N. H.; Reveal, J. L. 1972. Intermountain flora, vol. 1. New York: Hafner Publishing Co. 270 p.

Fabre, J. H. 1925. The mason bees. Garden City, NY: Garden City Publishing Co. 315 p.

Handel, S. N. 1983. Pollination ecology, plant population structure, and gene flow. In: Real, L., ed. Pollination biology. New York: Academic Press: 163–211.

Johansen, C. A.; Mayer, D. F.; Eves, J. D.; Kious, C. W. 1983. Pesticides and bees. Environmental Entomology 12: 1513–1518.

Karron, J. D. 1991. Patterns of genetic variation and breeding systems in rare plant species. In: Falk, D. A.; Holsinger, K. E., eds. Genetics and conservation of rare plants. New York: Oxford University Press: 87–98.

Kevan, P. G.; LaBerge, W. E. 1979. Demise and recovery of native pollinator populations through pesticide use and some economic implications. In: Caron, D. M., ed. Proceedings of the fourth international symposium on pollination. Misc. Publ. 1. College Park, MD: Maryland Agricultural Experiment Station: 489–508.

Lavigne, R.; Tepedino, V. J. 1976. Checklist of the insects in Wyoming. I. Hymenoptera. Agric. Exp. Stn. Resour. Jour. 106. Laramie, WY: University of Wyoming. 61 p.

Levin, D. A. 1984. Immigration in plants: an exercise in the subjunctive. In: Dirzo, R.; Sarukahn, J., eds. Perspectives on plant population ecology. Sunderland, MA: Sinauer Associates: 242–260.

Miliczky, E. R.; Osgood, E. A. 1979. The effects of spraying with Sevin-4-oil® on insect pollinators and pollination in a spruce–fir forest. Tech. Bull. 90. Orono, ME: University of Maine and Maine Agricultural Experiment Station: 1–21.

Peach, M. L.; Tepedino, V. J.; Alston, D. G.; Griswold, T. L. 1993. Insecticide treatments for rangeland grasshoppers: potential effects on the reproduction of *Pediocactus sileri*(Englem.) Benson (Cactaceae). In: R. Sivinski; Lightfoot, K., eds. Proceedings of the southwestern rare and endangered plant conference. Misc. Pub. 2. Santa Fe, NM: New Mexico Forestry and Resources Conservation Division: 309–319.

Plowright, R. C.; Thaler, G. R. 1979. The effect of biocides on forest pollination in New Brunswick. In: Caron, D. M., ed. Proceedings of the fourth international symposium on pollination. Misc. Publ. 1. College Park, MD: Maryland Agricultural Experiment Station: 483–487.

Rau, P. 1929. Experimental studies in the homing of carpenter and mining bees. Journal of Comparative Psychology 9: 35–70.

Rau, P. 1931. Additional experiments on the homing of carpenter-and mining-bees. Journal of Comparative Psychology 12: 257–261.

Roubik, D. W. 1989. Ecology and natural history of tropical bees. New York: Cambridge University Press. 514 p.

Slatkin, M. 1985. Rare alleles as indicators of gene flow. Evolution 39: 53–65.

Slatkin, M. 1993. Isolation by distance in equilibrium and non-equilibrium populations. Evolution 47: 264–279.

Slatkin, M; Barton, N. 1989. A comparison of three indirect methods for estimating average levels of gene flow. Evolution 43: 1349–1368.

Stephen, W. P.; Bohart, G. E.; Torchio, P. F. 1969. The biology and external morphology of bees. Corvallis, OR: Oregon State University and Oregon Agricultural Experiment Station. 140 p.

Tepedino, V. J. 1979. The importance of bees and other insect pollinators in maintaining floral species composition. Great Basin Naturalist Memoirs 3: 139–150.

U.S. Fish and Wildlife Service. 1993. Endangered and threatened wildlife and plants. U.S. Fish and Wildlife Service. 50 CFR 17.11 and 17.12. Washington, DC: U.S. Government Printing Office: 1993-342-447/94033. 40 p.

Wood, G. W. 1979. Recuperation of native bee populations in blueberry fields exposed to drift of fenitrothion from forest spray operations in New Brunswick. Journal of Economic Entomology 72: 36–39.

Reference Cited—Unpublished

Packer, J. S. 1970. The flight and foraging behavior of the alkali bee *Nomia melanderi* (Ckll.) and the alfalfa leafcutter bee *Megachile rotundata* (F.). Ph.D. dissertation. Logan, UT: Utah State University. 119 p.

III.6 Grasshopper Treatment Effects on Aquatic Communities

D. W. Beyers and L. C. McEwen

Concern about potential for adverse effects on endangered species from inadvertent exposure to insecticides was partially responsible for initiation of the Grasshopper Integrated Pest Management (GHIPM) Project. Investigation of effects of grasshopper control operations on aquatic communities was one aspect of the Project and had two major emphases.

The first emphasis was evaluation of the toxicity of carbaryl and malathion to two federally endangered fishes that inhabit rivers of the Colorado River Basin (the Colorado River and tributaries in Wyoming, Colorado, Utah, New Mexico, and Arizona). The second area of research involved environmental monitoring of the effects of operational grasshopper insecticide applications on aquatic invertebrates and fish in ponds and streams. Results of these studies provide information on potential effects of pesticide application practices and allow evaluation of adequacy of no-spray buffer zones around aquatic habitats.

Toxicity Testing With Endangered Fishes

The Colorado pikeminnow *(Ptychocheilus lucius)* and bonytail *(Gila elegans)* are large minnows historically found throughout the Colorado River Basin. Populations of both species have declined as a result of interactions with introduced fishes, construction of dams, and habitat modification. Young Colorado pikeminnow and bonytail occupy shallow, low-velocity, near-shore nursery habitats. These habitats have low rates of water exchange, and pesticides deposited in them may persist in sufficient concentration and duration for toxic effects to occur.

The timing of grasshopper control programs coincides with the presence of potentially sensitive early life stages of Colorado pikeminnow and bonytail in nursery habitats. But the infrequency and low application rate of pesticide use in Federal grasshopper control programs present a minor risk to these endangered fishes in comparison to other hazards, such as cropland chemicals, instream flow changes, and introduced (exotic) species. Nevertheless, data are needed on the IPM chemical effects.

Because of uncertainty in predicting the sensitivity of Colorado pikeminnow and bonytail to carbaryl and malathion, Beyers et al. (1994) estimated toxicity of these chemicals using methods recommended by U.S. Environmental Protection Agency and the American Society for Testing and Materials. The toxicity of technical carbaryl, Sevin® 4-Oil, and technical malathion was estimated by determining (1) 96-hour median lethal concentrations, and (2) concentrations that affected survival and growth in 32-day early life-stage tests (Beyers 1993, Beyers et al. 1991 and 1994).

One concern responsible for initiation of toxicological studies was that Colorado pikeminnow or bonytail might be supersensitive to carbaryl or malathion. To evaluate this possibility, we compared the sensitivity of Colorado pikeminnow and bonytail to other commonly studied fishes. We concluded that Colorado pikeminnow and bonytail were 2 to 10 times more sensitive to carbaryl than fathead minnow *(Pimephales promelas)* but were about as sensitive to malathion as fathead minnow (Beyers et al. 1994, Mayer and Ellersieck 1986). Some pesticide formulations are more toxic than their technical compounds; however, toxicity of Sevin 4-Oil (49 percent carbaryl) is approximately one-half that of technical carbaryl. No synergistic or antagonistic toxic effects due to formulation of carbaryl as Sevin 4-Oil were observed.

Results of standardized toxicity tests provided quantitative description of toxicant effects, but the tests did not simulate chemical exposure conditions likely to occur in the field. Therefore, we conducted studies of brain acetylcholinesterase (AChE) inhibition in order to estimate toxicant effects at a scale consistent with the duration of exposure and concentration range typically observed in the field. AChE activity was measured in Colorado pikeminnow after 24-hour *in vivo* exposure to technical carbaryl or malathion (Beyers and Sikoski 1994).

A comparison of the potency of the 2 toxicants showed that technical carbaryl was about 13 times more toxic than malathion to Colorado pikeminnow. Toxicant concentrations that significantly affected AChE activity were 15 times lower for carbaryl and 4 times lower for malathion than concentrations that affected growth or survival in 32-day early life-stage tests. These differences were attributed to development of physiological tolerance over the 32-day period used for early life-stage tests, and greater sensitivity of biochemical processes (AChE inhibition) compared to whole-organism responses (growth or survival).

Environmental Monitoring

Insecticides used to control grasshopper infestations pose a potential hazard to fish and invertebrates because, although no-spray buffer zones are observed around aquatic habitats, pesticide may be deposited by drift or mobilized from upland areas by runoff. We investigated effects of several aerial grasshopper control pesticide applications within the Little Missouri National Grasslands in western North Dakota (Beyers et al. 1995, Beyers and Myers 1996).

Environmental monitoring in aquatic habitats involved collection of water samples for pesticide analysis and study of sublethal and lethal effects on invertebrates and fish. In pond studies, we used enclosures called mesocosms to divide a portion of a pond into independent experimental units. Each mesocosm contained sediment, plants, and invertebrates that occurred naturally in the pond. We monitored survival of invertebrates within mesocosms for up to 4 days after pesticide application. *In situ* toxicity tests using naturally occurring invertebrates were also conducted with mesocosms.

The effects of pesticide application on river-dwelling organisms in the Little Missouri River were investigated on two separate occasions. Potential effects on aquatic invertebrates were investigated by quantifying daytime invertebrate drift. Normally, aquatic invertebrate drift in rivers is low. However, when pesticides are introduced, catastrophic drift may occur as invertebrates attempt to avoid toxicant exposure or suffer toxic effects (Wiederholm 1984). Sublethal effects on fish in the Little Missouri River were evaluated by studying fish-brain AChE inhibition. AChE activity of flathead chub *(Platygobio gracilis)* collected from control and treatment sites before and after pesticide application was measured.

Results of monitoring showed that when the standard 500-ft (152-m) no-spray buffer was employed, trace amounts of pesticide were always detected in aquatic habitats. The amount of deposition was dependent on the size of the aquatic habitat; smaller ponds had higher pesticide concentrations. Detection of trace amounts of pesticides does not necessarily result in biological effects on aquatic organisms.

We intensively studied six ponds but found evidence of direct mortality of pond-dwelling organisms in only one. On this occasion, a 0.6-acre (0.23-ha) pond containing abundant amphipods was monitored during an application of Sevin 4-Oil. All amphipods in treatment enclosures died within 24 hours of pesticide application. Subsequent collections confirmed that the amphipod population in the pond had declined. Amphipods are known to be extremely sensitive to carbaryl and malathion (Mayer and Ellersieck 1986). Other taxa in the pond appeared to be unaffected by the application.

Studies in the Little Missouri River during a drought year (1991), when discharge and the dilution potential of the river was low, detected an increase in invertebrate drift during the first 3 hours after pesticide application (Beyers et al. 1995). This increase was primarily composed of Ephemeroptera, especially Heptageniidae. There was no change in drift at the reference site. Subsequent sampling during the day of pesticide application showed that the increase in invertebrate drift was transient and undetectable after 3 hours.

The biological significance of increased invertebrate drift due to pesticide application is uncertain but probably of minimal consequence. The increase in invertebrate drift was mostly due to Ephemeroptera; other taxa were unaffected. Because a relatively small portion of the Little Missouri River was within the spray block (3.2 river-miles or 5.2 river-km), mortality was probably compensated by recolonization from unaffected organisms living in the substrate or upstream. Thus only a portion of the invertebrate community may have been affected, and the likelihood of rapid recovery of affected populations was high. Analyses of brain AChE activity in flathead chub showed that fish were not affected by the pesticide application. Similar monitoring studies conducted during a year when precipitation was above average (1993) did not detect any increase in aquatic invertebrate drift or effects on fish (Beyers et al. 1995). The overall conclusion was that these grasshopper control operations had no biologically significant affect on aquatic resources.

A factor that may reduce the potential for toxic effects to aquatic organisms is the natural degradation of carbaryl and malathion. Both pesticides hydrolyze (decompose chemically) rapidly in waters with pH >7 (Beyers and

Myers 1996). All aquatic habitats monitored in North Dakota had pH's greater than 7. Although the amount of pesticide deposited in aquatic habitats may be potentially toxic to some aquatic life, the short duration of the exposure can reduce or eliminate toxic effects.

Our investigations were designed to detect AChE inhibition or invertebrate mortality within 96 hours of pesticide application. If toxic effects were manifested over a longer time scale it is unlikely that effects would have been detected by our investigations. Toxicity endpoints other than death of aquatic organisms (such as swimming ability, avoidance of predators, feeding behavior, and reproductive effects) also are receiving attention by others in the field of aquatic ecotoxicology (Nimmo and McEwen 1994).

A Note on Quality Assurance for Pesticide Monitoring

One of the reasons why carbaryl and malathion are used to control grasshopper infestations is that they degrade relatively rapidly in the environment. Short persistence assures less potential for nontarget effects; however, these qualities complicate sampling for pesticide analysis because, if precautions are not taken, degradation may continue to occur after a sample has been collected and pesticide concentration estimates will be in error.

An important aspect of quality assurance (QA) that can be used to guard against this eventuality is fortification (spiking with measured pesticide amounts) of similar environmental samples. Prior to pesticide application, samples for fortification should be collected at the same localities where pesticide monitoring samples will be collected. A known amount (for example, 1 mL) of a fortification standard should be added to each QA sample. To prevent investigator bias, QA samples should not be identified any differently than posttreatment monitoring samples. QA samples should be handled and submitted for chemical analysis along with other monitoring samples. In general, QA samples should be fortified to approximately 10 times the detection limit reported by the analytical laboratory and the number of QA samples should be about 10 percent of total number of samples submitted for analysis. If only a few monitoring samples are being collected (fewer than 10), then at least 2 QA samples should be submitted. Fortification standards should be obtained from the laboratory that will be conducting the analytical work (see Chapter III.9). When reporting results of pesticide monitoring, percent recovery from fortified samples also should be reported. The importance of including QA samples cannot be overstated: they provide the only method for judging accuracy of reported results.

References Cited

Beyers, D. W. 1993. Toxicity of carbaryl and malathion to Colorado squawfish and bonytail. Ph.D. dissertation. Fort Collins, CO: Colorado State University. 92 p.

Beyers, D. W.; Myers, O. B. 1996. Use of meta-analysis to predict degradation of carbaryl and malathion in freshwater for exposure assessment. Human and Ecological Risk Assessment 2: 366–380.

Beyers, D. W.; Sikoski, P. J. 1994. Acetylcholinesterase inhibition in federally endangered Colorado squawfish exposed to carbaryl and malathion. Environmental Toxicology and Chemistry 13: 935–939.

Beyers, D. W.; Carlson, C. A.; Tessari, J. D. 1991. Solid-phase extraction of carbaryl and malathion from pond and well water. Environmental Toxicology and Chemistry 10: 1425–1429.

Beyers, D. W.; Keefe, T. J.; Carlson, C. A. 1994. Toxicity of carbaryl and malathion to two federally endangered fishes, as estimated by regression and ANOVA. Environmental Toxicology and Chemistry 13: 101–107.

Beyers, D. W.; Farmer, M. S.; Sikoski, P. J. 1995. Effects of rangeland aerial applications of Sevin 4-Oil on fish and aquatic invertebrate drift in the Little Missouri River, North Dakota. Archives of Environmental Contamination and Toxicology 28: 27–34.

Mayer, F. L., Jr.; Ellersieck, M. R. 1986. Manual of acute toxicity: interpretation and data base for 410 chemicals and 66 species of freshwater animals. Resour. Publ. 160. Washington, DC: U.S. Department of the Interior, U.S. Fish and Wildlife Service. 506 p.

Nimmo, D. W.; McEwen, L. C. 1994. Pesticides. In: Calow, P., ed. Handbook of ecotoxicology, vol. 2. London, Edinburgh, Boston: Blackwell Scientific Publications: 155–203.

Wiederholm, T. 1984. Responses of aquatic insects to environmental pollution. In: Resh, V. H.; Rosenberg, D. M., eds. The ecology of aquatic insects. New York: Praeger Publishers: 508–557.

III.7 Bioindicator Species for Evaluating Potential Effects of Pesticides on Threatened and Endangered Wildlife

L. C. McEwen, B. E. Petersen, and C. M. Althouse

Monitoring pesticide applications for possible effects on wildlife is an integral part of pesticide registration and regulation and of a successful grasshopper integrated pest management (GHIPM) system. During grasshopper outbreaks, U.S. Department of Agriculture cooperative grasshopper control programs have treated as much as 13.1 million acres (5.3 million ha) of rangeland in a single season (U.S. Department of Agriculture, Animal and Plant Health Inspection Service 1987).

Large numbers of insectivorous birds may inhabit, or congregate in, areas where these insecticide applications are made. One grasshopper egg bed found in Otero County, CO, encompassing 2 acres (0.8 ha), was populated by "about 200 western horned larks and lark buntings," which were seen feeding heavily on the grasshopper nymphs (Wakeland 1958). An effective GHIPM program should retain the natural controls on grasshoppers and not disrupt the rangeland ecosystem, including threatened and endangered species.

Wiens and Dyer (1975) reported breeding-season bird densities averaging approximately 0.8 to 1.3 birds/acre (1.9 to 3.3 birds/ha) on rangeland. Johnson et al. (1980) summarized avian densities for grassland–sagebrush habitats as averaging 1.2 to 5.0 breeding birds/ha. Therefore, large numbers of birds and other wild vertebrates can be exposed to a chemical during a single pesticide application (McEwen 1987). In areas not monitored during an application, mortality, and particularly sublethal effects, caused by pesticides can be overlooked because mortality "usually affects only part of the fauna, is scattered in space and time, and generally occurs where there is no biologist to record it" (Stickel 1975).

Toxicity evaluation has employed the use of white rat species in a laboratory setting utilizing test animals that are common species, easily bred, maintained, and handled. Controlled tests are pertinent for determining baseline data and comparing relative toxicity of chemicals. However, to understand pesticide effects in the natural environment, all the intricate interactions of cover, weather, food, exposure routes, and animal behavior, must be considered. Toxicity tests in the laboratory can only predict ecotoxicity in the field setting within broad limits.

An intermediate step between laboratory and field investigations is the use of caged or penned vertebrates located within an application block as used by Kreitzer and Spann (1968). However, it was found that the cage-in-field method resulted in less exposure to the pesticide than free-ranging wildlife received and actually protected the experimental animals from possible predation related to sublethal effects (Heinz et al. 1979).

Sublethal effects can be observed in the controlled environment of laboratory investigations, and researchers often surmise that "a sublethal effect seen in the laboratory would also occur in the field and that this effect would result in mortality or reproductive problems" (Heinz 1989). These effects can also be misleading or overlooked. For example, Grue et al. (1982) found that free-living starlings differed from captive birds by losing weight after dosing with dicrotophos, an organophosphate (OP) insecticide. Field investigations are a necessary step in evaluating the overall effects of large-scale pesticide applications.

It has been recognized that data on effects of OP's and other classes of pesticides are incomplete (Grue et al. 1983, Kirk et al. 1996). The Avian Effects Dialogue Group (1994) set forth some recommendations for more effective techniques in gathering data. Several issues of concern were studies on focal avian species, study sites, carcass searching, population changes, modeling, use of radio telemetry, and dissemination of information.

Species of critical concern are usually unavailable for any hands-on laboratory or field toxicity studies, thus making the need for surrogate species a necessity. Lower and Kendall (1990) suggested some criteria for selecting a sentinel species (one in which effects may be interpreted as indicators of similar disturbances in other species) when evaluating synthetic compounds, such as pesticides in the field. This approach has several limitations.

For example, can the toxicity of a chemical to a chicken, duck, or quail predict toxic effects on a falcon or eagle? How do the differences in a species' physiology, food, habitats, and ecology affect the animal's exposure and reaction to the chemical? When threatened or endangered (T and E) species may be at risk, they of course, cannot be collected for chemical analysis, pathology examination, or food-habits study. Thus, the next best approach is to estimate potential effects on T and E species by study of closely related sentinel species.

The American kestrel *(Falco sparverius)* has been shown to be more sensitive to anticholinesterase insecticides than other avian species (such as quail and ducks) used to establish toxicity (Rattner and Franson 1984, Wiemeyer and Sparling 1991). Consequently, the kestrel is a conservative bioindicator of possible effects on the related peregrine falcon *(Falco peregrinus).*

Our environmental monitoring team's studies have utilized the American kestrel and killdeer *(Charadrius vociferus),* as surrogates for other Falconiformes and Charadriidae, such as the peregrine falcon and mountain plover *(Charadrius montanus),* respectively. Kestrels and killdeer are representative of their genera, are widely distributed, and are found in much greater numbers than their endangered relatives.

The American and European kestrels have been utilized in toxicology studies for many years (Wiemeyer and Lincer 1987). Studies of the American kestrel, the smallest and most abundant falcon throughout North America, have progressed from laboratory toxicity tests to field ecotoxicology investigations over the past 20 years. Since kestrels are commonly present on rangelands where grasshopper outbreaks occur, they are excellent subjects for examining direct and indirect effects of control programs. Kestrel use of nest boxes (fig. III.7–1) and tolerance of disturbance and observers makes it possible to investigate all stages of their life cycle. Henny et al. (1983) examined productivity of free-ranging kestrels using nest boxes beginning in 1978 for investigating the adverse effects of the pesticide heptachlor in Oregon's Columbia River Basin.

Figure III.7–1—Kestrel nest box used on rangeland. Access to the eggs and nestlings is through a hinged side of the box. Field crews can check nests periodically to determine egg hatchability, growth measurements, and survival of young, and to affix leg bands and attach transmitters. (Photo by L. C. McEwen of Colorado State University; reproduced by permission.)

On rangelands, population densities of American kestrels may be restricted by the lack of natural tree cavities for nesting sites. Investigation of pesticide effects could be difficult to document because of small sample sizes of kestrels, but nesting populations can be increased by adding artificial nest box structures. Frocke (1983) summarized the use of nest boxes in avian management and research; cavity-nesting species have exhibited a readiness to use, and possibly a preference for, nest boxes over natural cavities. Kestrels are very adaptable and will easily accept the use of human-made nest boxes.

Kestrels favor open-space sites for hunting, so establishing new nest sites in these open areas for experimental purposes can be effective. Although Loftin (1992) found in Florida that nest boxes placed in pastures or areas away from known kestrel use were ineffective in increasing American kestrel populations, we did not find this to be true. We had >50 percent use of all nest boxes in six different geographic locations from Colorado to Alaska. However, in some areas, it took 2–3 years to reach maximum use of boxes. (Plans and directions for construction and placement of nest boxes are given in chapter I.11 of this Handbook.)

Seven years of production data have been compiled on nesting American kestrels during the Grasshopper Integrated Pest Management (GHIPM) Project. Approximately 560 nest boxes were in place by the sixth year among 6 locations: the 2 GHIPM demonstration areas in Idaho and North Dakota, Alaska, Wyoming, and 2 parts of Colorado—the northwestern section and in the Front Range (fig. III.7–2). Data on clutch size, hatchability, and numbers of nestlings fledged were collected annually (table III.7–1).

Productivity is presented as baseline data for each location and compared between years. Mean clutch sizes did not vary among locations, but yearly differences were observed ($P < 0.05$). Alaskan kestrels surpassed birds

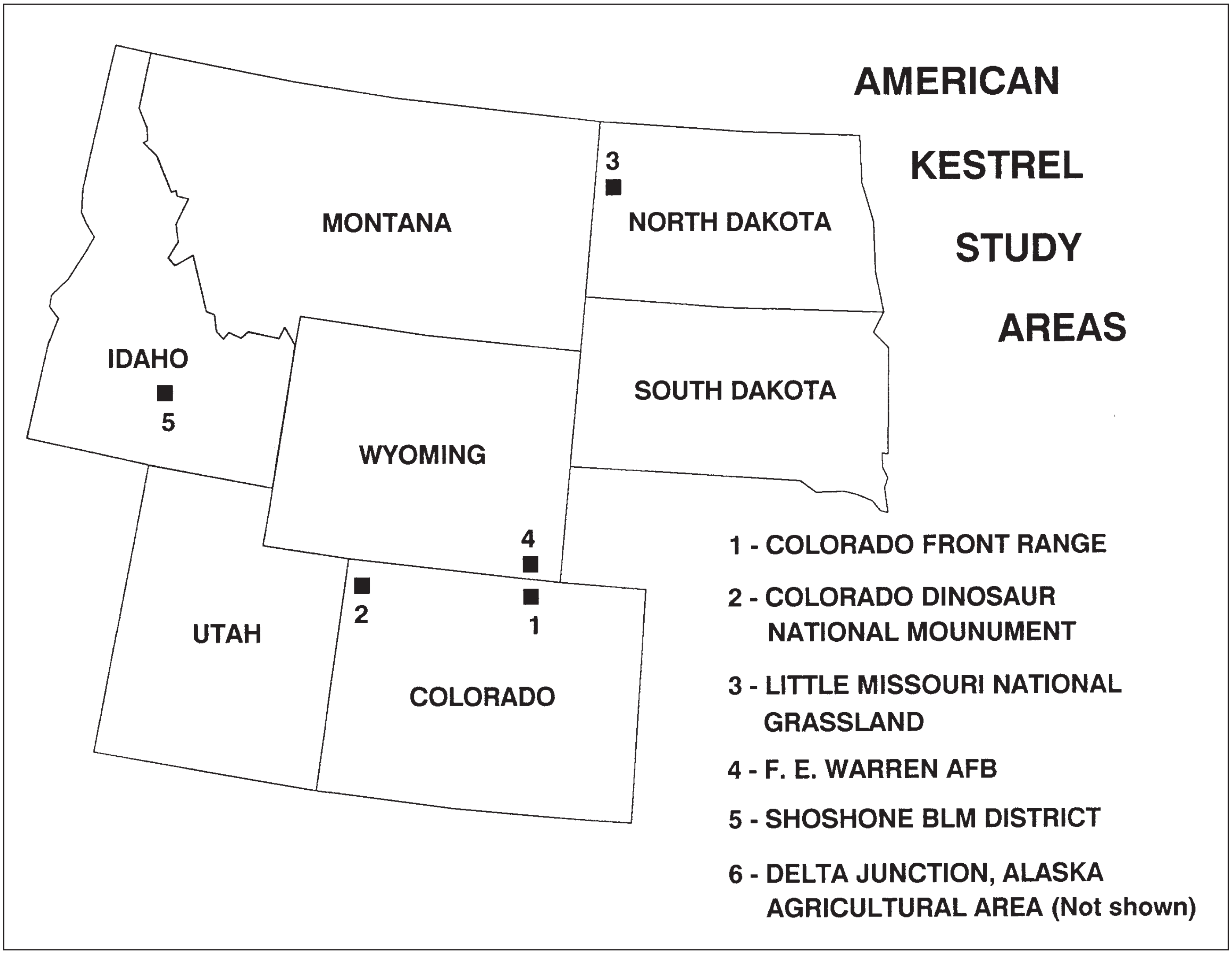


Figure III.7–2—Locations of kestrel study areas where >500 nest boxes have been placed (total of all areas). Key: 1 = Colorado, Front Range; 2 = Colorado, Dinosaur National Monument; 3 = Little Missouri National Grasslands; 4 = F. E. Warren Air Force Base; 5 = Bureau of Land Management's Shoshone District. (A sixth location, an agricultural area in Delta Junction, AK, is not shown.)

Table III.7–1—Variation in nesting productivity of American kestrels in the GHIPM demonstration areas and other treatment and reference areas during 1988–94

Location and years	Mean no. of nests/yr	% of nests hatched[1]	% of nests fledged[2]	Mean no. fledged per nest attempt
Alaska				
1990–93	33	85–97	82–97	3.5–4.3
Colorado, Front Range				
1988–94	26	61–88	55–81	2.0–2.9
Colorado, northwestern				
1988–94	24	81–89	79–84	2.9–3.1
Idaho				
1988–93	62	60–90	48–81	1.8–3.5
North Dakota				
1988–94	83	58–88	50–70	1.5–3.0
Wyoming				
1989–94	12	31–100	19–100	0.6–3.8

[1] Hatched nest: ≥ 1 egg hatched.
[2] Fledged nest: ≥ 1 young fledged.

from all other areas sampled in mean number of eggs hatched and young fledged in 1990 through 1993, but the differences were not statistically significant ($P > 0.05$).

Lower kestrel productivity in Idaho and North Dakota coincided with drought years and with the one extreme high-precipitation year in the Dakotas but otherwise was similar for most years (table III.7–1). The results illustrate the variability in kestrel nesting success due to natural factors and emphasize the importance of having concurrent untreated nest boxes for observation when investigating possible pesticide effects on nests in sprayed areas. Comparison of comparable untreated nests with sprayed nests over the same time period, is necessary to differentiate effects of weather, predation on nestlings by great horned owls *(Bubo virginianus)*, and other natural factors from pesticide treatment effects.

In 1990–94, a limited number of nest boxes in several locations, excepting Idaho, were used to study sublethal effects on kestrel nestlings and fledglings of (1) *Beauveria bassiana*, a fungus bioinsecticide; (2) carbaryl, a carbamate (sprays and bran-bait treatments); (3) malathion, an organophosphate; and (4) diflubenzuron (Dimilin®), an insect growth regulator. These results are presented in separate sections.

Field Applications

A carbaryl bran-bait treatment was examined at the Delta Agricultural Project in Alaska where five kestrel nest sites with heavy grasshopper infestation were selected for study of the effects of carbaryl bait. At the time of application, nestlings were approximately 18–22 days of age. Three of these nests had 2 percent carbaryl bran-bait applied at approximately 2.2 lb/acre on 40 acres (16.2 ha) adjacent to the nest box entrances, and 2 nests were left untreated. No adverse effect was noted on the treated nests, and all kestrel nestlings fledged normally. It was also found that numbers of breeding birds in North Dakota on line transects before and after application did not differ when controlling grasshoppers with carbaryl bait (George et al. 1992).

Possible effects on killdeer from spray applications of two formulations of Sevin® 4-Oil (20 or 16 fl oz/acre, with each containing 4 fl oz of diesel oil; active ingredient [AI] of carbaryl was 8 and 6.4 fl oz/acre or 0.56 and 0.45 kg/ha, respectively) were investigated in North Dakota during 1992. Brain AChE activities were monitored at 2, 8, and 21 days after applications and found not to differ from normal (Fair et al. 1995). Whole body carbaryl residues were low (averaging <0.1 to 1.4 p/m [parts per million]) but significantly ($P < 0.05$) greater for birds collected from the sprayed areas compared to birds from unsprayed surrounding locations. No toxic signs were observed in any killdeer. On the treated areas, birds captured invertebrate prey at rates significantly higher than on reference areas at 2 and 8 days after spraying (Fair 1993) presumably due to the availability of dying insects.

Acute Oral Dosing Treatments and Procedures

Growth, nestling and fledgling survivability, and postfledging movements of young wild kestrels were measured in the field after exposure to an acute sublethal oral dose of one of the following standard or experimental IPM materials: *Beauveria bassiana*, diflubenzuron, carbaryl, malathion, or their formulation carriers (diesel or corn oil). A minimum of four young per brood were used in these studies. The remaining nestling(s), if any, in each box served to maintain a normal brood size and provided an untreated comparison to the dosed birds. Their ages varied from 8 to 16 days when nestlings were randomly selected and given a single dose of one of the following: corn oil, pesticide formulation, the petroleum-based oil used in the formulation (carrier oil or #2 diesel fuel), or the technical material. Behavior and growth data were collected every 4 days following dosing. Surviving test nestlings were fitted with transmitters at 26–31 days of age (fig. III.7–3). After fledging, all birds were located daily or every other day until transmitters failed or young moved too far from the nest box area to be located.

Beauveria bassiana Sublethal Test

This investigation was conducted in the short-grass prairies of north-central Colorado during 1992. Thirteen nest boxes containing 55 young were tested (table III.7–2). Two of the nests were given challenge dosages of 5 µL

Figure III.7–3—Young kestrel with small transmitter attached for the study of postfledging behavior, movements, and survival. (Photo by B. E. Petersen of Colorado State University; reproduced by permission.)

Table III.7–2—Survival of American kestrel nestlings dosed with *Beauveria bassiana* formulation, carrier oil, corn oil, or untreated in north-central Colorado, May–August 1992

	Beauveria formulation[1]	Carrier oil[2]	Corn oil[2]	Untreated control
No. nestlings dosed	14	13	13	15
No. nestlings survived	11	12	13	15
No. fledglings with radios	11	12	13	2
No. fledglings survived	10	10	12	2

[1] Contains formulation oil and *Beauveria bassiana* spores. Dosage was based on 500,000 spores/µL and 1 µL/g of body weight.

[2] Dosages based on 1 µL/g of body weight.

(microliters)/gram of body weight for the formulation and carrier oil; for the main test, broods were dosed at 1 µL/gram of body weight. No statistical significance was detected in either growth rates or behavior data among treated and untreated groups (*P* > 0.05). Transmitters were attached to 38 kestrels. Data were collected on survival and movements of 28 of those birds (10 radio attachments failed). No detectable differences in survival or movements were found among treated and untreated kestrels.

Seven treated fledglings, ages 31–42 days, were collected for examination. Two additional fledglings were found dead and also the remains of one eaten by predators. Necropsies were performed on all collected birds at the Colorado Veterinary Teaching Hospital; no visible gross pathology was detected.

Diflubenzuron Sublethal Test

This investigation was conducted in north-central Colorado during 1993–94. Forty nest boxes containing 170 young were used (table III.7-3). Two of the nests were given preliminary challenge dosages of 64 mg/kg of body weight of technical diflubenzuron (Dimilin) to estimate toxicity, if any. (In English measure, this is the equivalent of 0.0009 oz diflubenzuron per pound of body weight). All following dosages will be given in metric units as used in toxicology. Kestrel broods in the main study were dosed at 10.2 mg/kg.

No statistical differences were detected in nestling growth rates, behavior data, or survival among treated and untreated birds (*P* > 0.05). Although no differences were found in nestlings, possible effects on fledgling survival were seen the first year. Transmitters were attached to 42 fledgling kestrels. During 1993 approximately half the fledgling kestrels dosed with diflubenzuron formulation died or were lost, warranting a second year of research. In 1994, however, more than 70 percent of the 43 kestrels fitted with transmitters survived, and no differences were observed between treated and control fledglings.

Several treated fledglings, ages 27 to 45 days, were found dead due to predation or other causes. Necropsies were performed on all the dead birds, and no gross pathology was detected.

Carbaryl Sublethal Test

American kestrel nestlings in nest boxes on the North Dakota GHIPM demonstration area were administered sublethal acute oral doses of Sevin 4-Oil formulation in 1992 to determine effects on growth and postfledging survival. Two 10-day-old nestlings were given 200 mg/kg body weight of Sevin 4-Oil (40.5 percent carbaryl or 81 mg/kg AI) to establish a lethal dosage. Brain acetylcholinesterase (AChE) activity was depressed 80 percent at death in 27–35 minutes. Four additional nestlings all survived Sevin 4-Oil dosages of 30–100 mg/kg.

Table III.7–3—American kestrel nestling and fledgling survival after dosing with technical or formulation diflubenzuron, diesel oil #2, corn oil, or untreated in north-central Colorado during 1993–94

	Diflubenzuron		Diesel	Corn	No
	Technical	Formulation	oil #2	oil	treatment
No. nestlings dosed	[1]40	40	40	39	11
No. nestlings survived	32	33	34	32	10
No. fledglings with radios	25	27	27	6	—
No. fledglings survived	22	19	21	3	—

[1] One bird dosed with technical diflubenzuron was collected prior to radio transmitter fitting.

Sublethal dosages then were given to 32 nestlings (8 to 14 days old). Sixteen were dosed at 15 mg/kg and 16 at 30 mg/kg with Sevin 4-Oil. Sixteen additional nestlings were given corn oil at 2 µL/g of body weight as untreated controls subjected to the same handling procedures. Blood samples were collected from the nestlings for analysis of plasma cholinesterase activity at 1 hour, 24 hours, and 7 to 14 days after dosing. Radios were placed on 30 of the nestlings for study of postfledging movements and survival. Twenty-one of the nestlings and fledglings were collected at 10 to 38 days after treatment for brain AChE activity measurements, carcass residue analysis, and necropsy. Carbaryl residues were no longer detectable in the carcasses, but three had 0.08–0.15 p/m in their gastrointestinal tracts (analyzed separately). No gross pathology was found.

None of the 21 birds had significant inhibition of brain AChE activity or any signs of gross pathology. The lack of brain AChE inhibition was not unexpected because of the sublethal dosage levels and the rapid reversibility of carbaryl inhibition. Blood plasma samples showed mild AChE inhibition at 1 hour after treatment (averages = 4 percent at 15 mg/kg and 12 percent at 30 mg/kg). Recovery from the low degree of plasma AChE inhibition was evident in all carbaryl-dosed nestlings by 24 hours after treatment.

Malathion Sublethal Test

American kestrel nestlings in North Dakota were administered sublethal acute oral malathion dosages in 1993 and 1994. To establish the sublethal treatment dosages, it was first necessary to determine the acute oral lethal levels by conducting preliminary range-finding toxicity tests. Based on reported malathion toxicity to other avian species, dosages ranging from 49 to 500 mg/kg were administered to seven nestlings, and all dosages were found to be lethal. In further tests, it was determined that lethal malathion dosages began at 20 to 40 mg/kg (Taira 1994). These results indicated that young kestrels are much more sensitive to malathion toxicity than many other bird species for which LD_{50}'s (lethal dose to 50 percent of the birds) range from >100 to >400 mg/kg (Smith 1987). Part of this sensitivity may be age related, but scientists do not know the acute oral LD_{50} of malathion for adult American kestrels.

Young birds in 17 nest boxes were given malathion at 1 of 2 dosage levels: 5 or 20 mg/kg. An equal number were given corn oil or left untreated. Posttreatment blood samples were taken for plasma AChE and butyrylcholinesterase (BChE) assay from each bird at 1 hour, 24 hours, and between 7 and 14 days after treatment. At the 20 mg/kg dosage, both AChE and BChE were severely inhibited (77.1 and 71.6 percent respectively) at 1 hour posttreatment (table III.7-4). AChE activity was still inhibited 60.3 percent at 24 hours. BChE recovered more quickly, showing 21.9 percent inhibition at 24 hours. Nestlings dosed with 5 mg/kg were not as strongly affected but had plasma AChE inhibition of 45.4 percent and BChE inhibition of 60.8 percent at 1 hour. These results support the conclusion from the range-finding tests that young kestrels are more sensitive to malathion than many other avian species (Taira 1994).

Nestlings that were casualties in the malathion range-finding tests were analyzed for carcass residue concentrations. Whole-carcass residues ranged from 0.38 p/m in the lowest-dosed bird (49 mg/kg) to 46.5 p/m in the highest-dosed nestling (500 mg/kg). Gastrointestinal tracts (including contents) were analyzed separately, and residues varied from 12.1 p/m to 4,860 p/m corresponding to dosage levels. Only 6 of the sublethally dosed nestlings/fledglings were recovered for analysis. Residues were not detectable except in one carcass, which contained 0.21 p/m of malathion.

Summary and Conclusions

Field studies of bioindicator species are a useful approach for estimating potential ecotoxicological effects of pest control operations on threatened or endangered (T and E) species or other wildlife species of special concern. Species selected as bioindicators should be widely distributed and relatively abundant in the habitat types subjected to pest controls. Species closely related to T and E species also may be considered “surrogates” for those species and for others of concern.

In our environmental monitoring studies, we have investigated effects on American kestrels as bioindicators for peregrine falcons (and other small raptors) and effects on killdeer as bioindicators for mountain plovers. Our data on total bird populations in treated and untreated rangeland sites also could be examined in retrospect if questions arise concerning other species such as long-billed curlews, burrowing owls, ferruginous hawks, loggerhead shrikes, or rare species of sparrows.

From our GHIPM work, these two conclusions can be drawn:

(1) Young kestrels are more vulnerable to toxicity of malathion and anticholinesterase pesticides than many other avian species. Therefore, nonspray buffer zones

Table III.7–4—Mean percentage of plasma cholinesterase (ChE) activity in malathion-dosed kestrel nestlings compared to control ChE activity

	Dosages					
	5 mg/kg			20 mg/kg		
Posttreatment collection time	ChE	Total AChE[1]	BChE[2]	ChE	Total AChE	BChE
1 hour	51.1	54.6	39.2	24.2	22.9	28.4
24 hours	74.8	73.8	80.5	46.4	39.7	78.1
7 days	94.0	94.5	91.6	89.0	86.9	101.8
14 days	98.3	100.8	88.2	94.6	97.0	84.7

[1] Acetylcholinesterase.

[2] Butyrylcholinesterase.

around active nests of the closely related peregrine falcon should always be observed when liquid pesticide formulations are applied. However, bait formulations of IPM chemicals and biologicals are safe and pose no significant hazard even if used in the immediate vicinity of the nests. Acute dosages of diflubenzuron or *Beauveria bassiana* formulations indicate very low direct toxicity to young kestrels. These materials would have no direct effects on nontarget terrestrial wildlife but might reduce the insect food base in some cases. These findings should also apply to other nesting raptors on rangeland.

(2) Studies of Sevin 4-Oil grasshopper sprays (16 or 20 fl oz/acre) indicated little or no effect on killdeer (Fair et al. 1995). Cholinesterase activity was not significantly inhibited, whole-body carbaryl residues were low (<0.1 to 1.4 p/m), and food-habits studies showed that the birds maintained adequate diets. No gross pathology was found on necropsy of the killdeer. Whole body lipids were measured as an indicator of body condition and did not differ between killdeer from sprayed and unsprayed sites.

These results indicate that Sevin 4-Oil applications at 20 fl oz/acre (0.56 kg/ha carbaryl AI) or lower pose little hazard to the closely related mountain plover, a Category 1 species that may be listed in the future as endangered. However, areas known to be in the immediate vicinity of mountain plover nests should be excluded from spray applications because of the variation in individual bird response to synthetic chemical compounds. Bait formulations would be the least hazardous method of grasshopper control in mountain plover habitat.

References Cited

Avian Effects Dialogue Group. 1994. Final report of the AEDG, 1988-1993. Washington, DC: RESOLVE, Center for Environmental Dispute Resolution. 156 p.

Fair, J. M. 1993. The effects of carbaryl grasshopper control on nesting killdeer *(Charadrius vociferus).* M.S. thesis. Fort Collins, CO: Colorado State University. 41 p. + appendices.

Fair, J. M.; Kennedy, P. L.; McEwen, L. C. 1995. Effects of carbaryl grasshopper control on nesting killdeer in North Dakota. Environmental Toxicology and Chemistry 14: 881–890.

Frocke, J. B. 1983. The role of nestboxes in bird research and management. In: Snag habitat management: proceedings of the symposium. Gen. Tech. Rep. RM-99. Ft. Collins, CO: U.S. Department of Agriculture, Forest Service, Rocky Mountain Forest and Range Experiment Station: 10–13.

George, T. L.; McEwen, L. C.; Fowler, A. 1992. Effects of a carbaryl bait treatment on nontarget wildlife. Environmental Entomology 21: 1239–1247.

Grue, C. E.; Powell, G.V.N.; McChesney, M. J. 1982. Care of nestlings by wild starlings exposed to an organophosphate pesticide. Journal of Applied Ecology 19: 327–335.

Grue, C. E.; Fleming, W. J.; Busby, D. G.; Hill, E. F. 1983. Assessing hazards of organophosphate pesticides to wildlife. Transactions of North American Wildlife and Natural Resources Conference 48: 200–220.

Heinz, G. H. 1989. How lethal are sublethal effects? Environmental Toxicology and Chemistry 8: 463–464.

Heinz, G. H.; Hill, E. F.; Stickel, W. H.; Stickel, L. F. 1979. Environmental contaminant studies by the Patuxent wildlife research center. In: Kenega, E. E., ed. Avian and mammalian wildlife toxicology. Spec. Tech. Publ. 693. Philadelphia: American Society for Testing and Materials: 9–35.

Henny, C. J.; Blus, L. J.; Stafford, C. J. 1983. Effects of heptachlor on American kestrels in the Columbia Basin, Oregon. Journal of Wildlife Management 47: 1080–1087.

Johnson, R. R.; Haight, L. T.; Riffey, M. M.; Simpson, J. M. 1980. Brushland/steppe bird populations. In: Workshop proceedings: management of western forests and grasslands for nongame birds. Gen. Tech. Rep. INT-86. Ogden, UT: U.S. Department of Agriculture, Forest Service, Intermountain Forest and Range Experiment Station: 98–113.

Kirk, D. A.; Evenden, M. D.; Mineau, P. 1996. Past and current attempts to evaluate the role of birds as predators of insect pests in temperate agriculture. Current Ornithology 13: 175–269.

Kreitzer, J. F.; Spann, J. W. 1968. Mortality among bobwhites confined to a heptachlor contaminated environment. Journal of Wildlife Management 32: 874–878.

Loftin, R. W. 1992. Use of nest boxes by the Florida kestrel. Florida Field Naturalist 20: 57–88.

Lower, W. R.; Kendall, R. J. 1990. Sentinel species and sentinel bioassay. In: McCarthy, J. F.; Shugart, L. R., eds. Biomarkers of environmental contamination. Boca Raton, FL: CRC Press: 309–332.

McEwen, L. C. 1987. Function of insectivorous birds in a shortgrass IPM system. In: Capinera, J. L., ed. Integrated pest management on rangeland: a shortgrass prairie perspective. Boulder, CO: Westview Press: 324–333.

Rattner, B. A.; Franson, J. C. 1984. Methyl parathion and fenvalevate toxicity in American kestrels: acute physiological responses and effects of cold. Canadian Journal of Physiology and Pharmacology 62: 787–792.

Smith, G. J. 1987. Pesticide use and toxicology in relation to wildlife: organophosphorous and carbamate compounds. Resour. Publ. 170. Washington, DC: U.S. Department of the Interior, U.S. Fish and Wildlife Service. 171 p.

Stickel, W. H. 1975. Some effects of pollutants in terrestrial ecosystems. In: McIntyre, A. D.; Mills, C. F., eds. Ecological toxicology research. New York: Plenum Publishing Corp.: 25–74.

Taira, T. 1994. Blood analysis of American kestrel and golden eagle nestlings exposed to malathion or carbaryl. M.S. thesis. Ft. Collins, CO: Colorado State University. 80 p.

U.S. Department of Agriculture, Animal and Plant Health Inspection Service. 1987. Rangeland Grasshopper Cooperative Management Program: final environmental impact statement. Washington, DC: U.S. Department of Agriculture, Animal and Plant Health Inspection Service. 221 p.

Wakeland, C. 1958. The high plains grasshopper. Tech. Bull. 1167. Washington, DC: U.S. Department of Agriculture, Agricultural Research Service. 168 p.

Wiemeyer, S. N.; Lincer, J. L. 1987. The use of kestrels in toxicology. In: Bird, D. M.; Bowman, R., eds. The ancestral kestrel. Raptor Res. Rep. 6. Ste. Anne de Bellevue, PQ: Raptor Research Foundation, Inc., and Macdonald Raptor Research Centre of McGill University: 165–178.

Wiemeyer, S. N.; Sparling, D. W. 1991. Acute toxicity of four anticholinesterase insecticides to American kestrels, eastern screech-owls and northern bobwhites. Environmental Toxicology and Chemistry 10: 1139–1148.

Wiens, J. A.; Dyer, M. I. 1975. Rangeland avifaunas: their composition, energetics, and role in the ecosystem. In: Smith, D. R., coord. Proceedings of the symposium on management of forest and range habitats for nongame birds; 6–9 May 1975, Tucson, AZ. Gen. Tech. Rep. WO-1. Washington, D.C.: U.S. Department of Agriculture, Forest Service: 146–182.

III.8 Buffer Zones: Their Purpose and Significance in Grasshopper Control Programs

L. K. Winks, L. C. McEwen, R. N. Foster, Mike W. Sampson, Michael Green, and V. J. Tepedino

NOTE: Acephate is no longer approved by EPA for rangeland grasshopper control.

A buffer zone is a distance or space around an environmentally sensitive area that acts as a deterrent to harm and/or disturbance of that area and its plant and animal life. For Federal cooperative grasshopper control or suppression operations, buffer zones are strips or areas of land left untreated and free of grasshopper suppression chemicals or materials.

Such zones, also called buffers, are pesticide-free areas established to protect (1) species listed or proposed as threatened or endangered (T and E) under the Federal Endangered Species Act, (2) designated or proposed critical habitats of T and E species, (3) aquatic sites (water or wetlands) of all types, and (4) other areas such as residences, parks, campgrounds, schools, cropland, apiaries and insectaries, and habitat for other sensitive species. Before any lands are treated in large-scale U.S. Department of Agriculture (USDA)-sponsored cooperative grasshopper management programs, land management agencies meet with USDA's Animal and Plant Health Inspection Service (APHIS) to consider all aspects of an operational plan to protect the T and E species and sensitive sites in the proposed treatment area.

Land-management agencies typically include the U.S. Department of the Interior's Bureau of Land Management and U.S. Fish and Wildlife Service (FWS) and USDA's Forest Service. An APHIS-prepared biological assessment opens the required consultations, and agencies discuss and negotiate buffer-zone requirements until agreement is reached among APHIS and the affected land-management agencies. At times, discussions and negotiations also involve State agencies.

The agencies determine buffer-zone specifics using existing Federal guidelines, the most recent information, and the best judgment of their personnel. The written agreement reached is expressed in detail in the FWS biological opinion for the site-specific environmental assessment. In practice, optimal treatment of a control block also depends on the experience of the project manager and the skill and experience of the spray pilots or ground applicators and on their observance of buffer boundaries and wind and weather conditions.

Generalized Buffer Zone Requirements

There are two general types of insecticide used for grasshopper control: liquid ultralow-volume (ULV) chemical sprays and insecticide-impregnated wheat-bran flakes. Requirements for use are more stringent for liquid ULV sprays than for bait application because ULV sprays are less selective in action, are more prone to drift, and contain more active ingredient (AI).

For treating grasshoppers in large-scale rangeland programs, APHIS not only follows chemical labeling recommendations but at times adds more restrictions based on environmental concerns. APHIS and other agencies base their current recommendations and mitigation (softening of effects) on guidelines contained in the Rangeland Grasshopper Cooperative Management Program and the Final Environmental Impact Statement (EIS) (U.S. Department of Agriculture, Animal and Plant Health Inspection Service 1987). APHIS also relies on changes agreed to by the FWS and content of the biological opinion. In addition, APHIS considers information that has come from its Grasshopper Integrated Pest Management (GHIPM) Project, which began in 1987.

Protecting areas of water on rangeland is important in grasshopper control programs. Present EIS guidelines state that liquid ULV sprays should not be applied within 500 feet (152 m) of aquatic habitat (reservoirs, lakes, ponds, seasonal pools, springs, streams, rivers, swamps, bogs, marshes, and potholes) or where leaching or surface runoff is likely, or when precipitation seems imminent. In recent years, there has been unresolved discussion about the definition of wetlands, and whether or not dry intermittent creek beds, wet meadows, and seasonally dry potholes qualify under the definition.

Aquatic habitat buffers also apply to areas treated with some baits. When chemical baits are used, the width of the no-treatment zones around aquatic habitats is 200 feet (61 m). When baits are used, buffer zones are smaller, and more of the area harboring grasshoppers can be treated. Bran baits containing the biological control agent *Nosema locustae* can be used without buffer zones. Some pest managers believe that being able to treat a larger proportion of the area lengthens the time period before the site is reinfested.

Baits do have limitations: damp or wet weather hampers use, not all grasshopper species will eat dry baits, baits are more expensive to apply than liquid ULV sprays, and baits provide a lower level of control of susceptible species compared to liquid sprays (see chapter II.12). However, baits do make it possible to reduce the size of buffer zones, obtain some suppression of grasshoppers that otherwise would be untreated using ULV sprays, and minimize insecticide effects on nontarget species.

After no-treatment and no-spray zones for sensitive areas are identified and mapped, the APHIS State plant health director or the authorized APHIS representative should verify the treatment locations in a pretreatment reconnaissance flight with the spray pilot(s). Boundaries should be clearly and adequately marked, preferably with large peices of fluorescent orange material. There should be confirmation of the no-treatment sites. Records and maps also should be signed by APHIS representatives and pilots and dated after the pretreatment flights. The pilots(s) must clearly understand locations and boundaries of buffer zones.

When called for during chemical spray operations, spray-deposit dye cards should be placed within the buffer zones to detect drift or inadvertent treatment of no-spray sites. Lack of spray deposit will verify that buffer zones did prevent exposure to sensitive areas being protected. With bran baits, cards containing adhesive or small pans placed in the buffer zones will detect inadvertent treatment.

Aircraft utilizing an electronic guidance system (Loran C or Global Positioning System) will aid greatly in identifying buffer zones and increasing the accuracy of applying sprays or baits (fig. III.8–1). When acceptable electronic guidance is available and used, ground flagging to mark the areas can be reduced or eliminated. Some guidance systems also are combined with a printed record of the flight showing exact locations of areas treated. A printed record adds to accountability and quality assurance. In the future, Federal agencies may require detailed printed records of insecticide applications in treatment areas.

APHIS has found that only rarely is part of a treatment block treated a second year in a row. Typically, APHIS may treat a block of land only once every several years.

Figure III.8–1—In the era before global positioning systems, agricultural pilots had to turn the nozzles of their spray equipment on and off manually. Pilots did this when they spotted “flagmen” who stood on the ground at the edge of spray plots or buffer areas. It was virtually impossible to adjust the on/off decision in light of near-ground wind, so insecticide drift was common. Naturally, flagmen were exposed to toxicants just like the target pests! Now, however, computerized equipment on the spray planes can automatically starts and stops the flow of pesticides using sophisticated mapping and geostationary satellite coordinates.

Buffer Zones for Endangered Plants

Buffer zones for T and E plants are important, not because of a direct effect of insecticides on plants but to protect any insect pollinators that might be necessary for reproduction of the plants. The only insecticides (malathion, acephate, and carbaryl) registered and approved by APHIS for use in grasshopper control on Federal lands are not known to be toxic to plants at the rates used. The insecticides are toxic to some flower-visiting insects, however.

Is it common for T and E plants to need insect pollinators? The T and E plant species studied during the GHIPM Project demonstrated a dependency on insects, particularly native bee species, to move pollen from one flower to another (chapter III.5). Reproductive success of 24 of 26 plant species studied during the project is greatly increased by the presence of native bees. Grasshopper control efforts must be designed to prevent or minimize insecticide exposure to active pollinators of T and E plants.

The question of adequate buffer-zone size is extremely complex. How can pest managers define “adequate size” in a T and E context? The answer to this question depends on several factors including:

- The distance bee pollinators move between their nesting sites and flower populations,
- The distances over which bees forage for food from flowers, and
- The distances bees must move to gather other needs such as mud, leaf pieces, resin, etc., that are important for nest construction.

The brief answer to questions of adequate size is that scientists and pest managers really do not know what is adequate. One way to determine the size of buffer zones is to base the size on the protection needed; however, determining the protection needed often can be difficult. Some studies to determine at least partial answers to the question of size have not been successful (chapter III.5). For the most part, bees appear to act in ways that increase their foraging efficiency. When possible, bees nest close to the flowers they visit for pollen and nectar. Sometimes bees cannot do so because the proper nest sites are absent. Sometimes bees also forage farther than usual because flower density is low or because other resources are not available at nesting sites.

Studies noted in chapter III.5 did show that many species of bees are capable of flying several miles to return to their nests. Whether bees do this routinely is not known. Without a complete knowledge of insect pollinator behavior, the common (and some scientists believe the safest) approach is a conservative one. A buffer zone of 3 miles’ (4.8 km) radius usually is employed around T and E plant populations when using liquid insecticides.

The 3-mile buffer zone can be reduced or eliminated if information shows that the species in question is a self-pollinator or reproduces asexually or if the spray is not a potential problem to the pollinator species. Obviously, if no pollinators are needed, there is no effect on the T and E plants from the use of insecticides.

When using the common formulation of 2 percent carbaryl bran bait or other dry baits to treat grasshoppers, it is unlikely that the control program would need any buffer zone (chapter III.4) even with bees present. Because they do not eat bran baits, bees are not directly exposed to the insecticide.

Change in Peregrine Falcon Buffer Zones

The former standard buffer for peregrine falcon *(Falco peregrinus)* aeries (nests), hack sites (release of young peregrines after acclimation and supplemental feeding), and other release or habitat sites was a 10-mile no-treatment or drift radius (for aerial applications). It is now possible to establish buffer zones that are less arbitrary and correspond to the foraging area of the birds–often a long, narrow strip such as a valley or canyon. The foraging areas must be determined by a review team including one representative each from APHIS, FWS, the State conservation agency, and the land manager (or landowner if private land).

Aerial insecticide treatments then can be applied to within 1 mile (1.6 km) of the nest or release site. The boundaries of known foraging areas have a 500-ft (152-m) no-treatment zone. Bait applications with ground equipment can be made to within 0.5 mile (0.8 km) of a nest or release site and within 200 feet (61 m) of foraging areas. Reduced peregrine falcon buffer zones have not been widely used yet in grasshopper control programs, so the zones' use and effect should be part of the project monitoring plan.

Examples of Effective Uses of Buffer Zones

Piping plovers *(Charadrius melodus)*, an endangered species, nest on the sandy shoreline of Lake Sakakawea adjacent to grasshopper control areas in North Dakota. In 1989, a "hot spot" carbaryl bait treatment (2 lb/acre of 2 percent carbaryl bran bait–0.04 lb/acre AI) was applied to land immediately adjacent to a breeding pair of piping plovers with two small chicks and their no-treatment buffer zone (200 ft) near the nest site. Periodic posttreatment observations verified normal development and behavior of the chicks and adults (McEwen and Fowler unpubl.).

In 1991, a 19,200-acre (7,770-ha) area was sprayed with Sevin® 4-Oil at the standard IPM rate. APHIS sprayed liquid Sevin in the block–excluding a 0.5-mile (0.8-km) strip along the lake shore that was treated with carbaryl bait (2 lb/acre–2 percent actual ingredient). APHIS applied the bait and left a 200-ft (61-m) untreated strip at the water line. Observations on the nesting plovers indicated no effect, and breeding piping plovers were found at the same site in the following year (McEwen unpubl.).

This piping plover site is an especially difficult treatment situation because it is near reseeded crested wheatgrass *(Agropyron cristatum)*. Large areas of nearby native range have been reseeded to crested wheatgrass. The plant's clumpy growth form, with bare ground between plants, tends to promote high pest grasshopper densities. Many grasshopper species prefer bare ground for laying eggs. Also, large expanses of crested wheatgrass lose nearly all the bird species associated with native grasses (Reynolds and Trost 1980) that would be preying on the grasshoppers. Part of the loss of breeding birds is based on poor nesting habitat associated with crested wheatgrass.

The authors also have used and evaluated buffer zones around other aquatic sites in western North Dakota. These zones were in relation to large-scale Sevin 4-Oil treatments in 1991 and 1993 adjacent to the Little Missouri River. The standard aquatic buffer zones of 500 ft (152 m) were in place. In both years, carbaryl was detected in the river.

In 1991, a drought year, the maximum concentration of carbaryl detected was 0.085 parts per million (p/m); in 1993, a wetter year, it was 0.013 p/m. These low concentrations were found 1–2 hours after treatment and then rapidly declined (Beyers et al. 1995). Samples at 48 hours contained less than 0.0005 p/m, well below the concentrations generally known to begin affecting other invertebrates (0.002–1.90 p/m) and fish (1.95–39 p/m) (Johnson and Finley 1980). The only biological effect was an increase in the number of Ephemeroptera (mayflies) in the immediate (1–3 hr) postspray drift samples in 1991.

Natural events had greater impact on the aquatic invertebrates in the river in 1991 than did the insecticide. Monitoring of brain acetylcholinesterase (AChE) activity in flathead chubs *(Platygobio gracilis)* collected from the treatment area showed no inhibition, indicating no adverse carbaryl effects. Measurement of AChE activity is a method of detecting toxic effects of pesticides. It was concluded that the light drift of Sevin 4-Oil into the Little Missouri River was biologically insignificant (Beyers et al. 1995).

A study of golden eagle *(Aquila chrysaetos)* response to Sevin 4- Oil treatments around active nests was initiated in 1993 and is still underway (1995) in North Dakota. Nest areas were treated in June 1993 and 1994 when the young eagles were 4–7 weeks of age. Each young eagle was captured at fledging (10–11 weeks of age) so field crews could take biological measurements and blood samples and attach radio transmitters for postfledging observations. Telemetry is used to determine movements, behavior, survival, and dispersal from the natal (hatching) areas. Preliminary results indicate no differences in survival, movements, and dispersal between young golden eagles from sprayed and unsprayed territories.

Eagles from treated nests tended to be less active in afternoon and evening time periods and preened more (Bednarski and McEwen 1994, Bednarski unpubl.). Fledglings from treated areas had slightly higher ($P = 0.11$) blood plasma cholinesterase activity, a normal "rebound" or overcompensation effect commonly seen in birds after a light exposure to an inhibiting pesticide (Taira 1994), Taira and McEwen unpubl.). Territory maintenance, nesting activity, and productivity of the mature pairs of golden eagles in the sprayed and untreated areas are being followed 1 and 2 years after treatment.

Preliminary findings suggest that buffer zones of 500 ft (152 m) or possibly 200 ft (61 m) around the actual nest site will be adequate for protection when treating with Sevin 4-Oil. Further studies may show that buffer zones could be even smaller or possibly eliminated. The large foraging area (± 50 mi^2 or 129 km^2) characterizing an average territory of a breeding pair of golden eagles need not be of concern. A small area (± 5 acres or 2 ha) around each nest easily could be left untreated, without the human disturbance caused when placing flags, by using an electronic guidance system. The human disturbance of people on foot in the immediate vicinity of the nest should be avoided and could cause more problems than the treatment itself. Again, restrictions of the biological assessment and biological opinion will control program design and operation.

Although the effects of carbaryl on nesting golden eagles have been examined during the GHIPM Project, there has been no study of the effects of malathion on golden eagles. A study utilizing malathion also should be done because it was found that another raptor species, the American kestrel *(Falco sparverius)*, is very sensitive to malathion toxicity in the nestling stage (Schleve et al. 1993 unpubl., McEwen et al. 1994 unpubl.).

Potential Consequences of Buffer Zones

Treatment-free buffer zones may appear to be an obvious way to protect sensitive areas. Although liberal use and size of zones may seem safest, unneeded or exaggerated protection may reduce the effectiveness (efficacy) of grasshopper control programs. Buffers have varying impacts on treatment program efficacy, depending on the specific goals of the program (minimum economic level of control or maximum control) and where in the cycle the current grasshopper population exists. While designed to protect nontargets, buffer zones also can provide protection for pests the program seeks to control.

One concern with buffers occurs when the grasshopper population is expected to be about the same or greater in the following year. When the control effort is crisis in nature, maximum control of damaging grasshoppers is the goal. Untreated zones in a treated block may contribute to extending or expanding the problem by harboring grasshoppers, especially when grasshopper populations are cycling upward. In some cases, a large number or size of buffer zones can result in an immediate loss in the integrity of the spray block (less efficacy of treatment). These zones may result in the need for additional treatments and may expose larger tracts of land to pesticide treatments later. Fewer long-term control problems should result from untreated buffer zones when the grasshopper population is expected to decline.

Regardless of the grasshopper population cycle, blocks with large numbers of irregular buffer zones may result in increased treatment difficulties during the actual spray operation. The increased difficulty may be reflected in an increased cost of the application contract. Increased cost may result from marking each zone on the ground to ensure its identify from the aircraft applying the treatment. Marking is required if accurate electronic guidance is not available to the applicator. Additionally, costs associated with environmental monitoring (if required) of the buffer zones also may substantial. Together, these additional costs may be very significant. Coupled with leaving enough of the problem grasshopper population in the buffer zones possibly to reinfest treated areas, these additional costs could reduce the length of the economic benefit of the treatment. There even may be cases where the total buffer-zone acreage or the associated additional costs are so high as to negate the value of a particular treatment.

Buffers around water are the most frequently encountered treatment-free areas within a spray block. However, it is not unusual for grasshoppers to exist at high densities near rivers, streams, lakes, or ponds. In some cases, these areas around water harbor the highest densities of grasshoppers in the entire proposed treatment area. The entire grasshopper population, including that in buffer zones, must be considered for the most economically, biologically sound program to result.

One area of concern for use of buffers is in small, isolated infestations identified as historic hot-spots. In such areas, buffers that prevent effective treatment could be a threat to the concept of treating localized areas before grasshoppers can spread to larger acreages. Large numbers of uncontrolled grasshoppers in buffers–within areas where preventative hot-spot treatment is the foundation of an areawide program–could prevent full implementation of the concept and seriously jeopardize the overall program.

In many cases, a specifically customized treatment may provide the protection needed for a sensitive area while addressing most of the pest population. An example of a customized treatment would be the use of ground-applied bait adjacent to waterways, with an application direction away from the water. If performed properly, such a treatment could be conducted within a few feet of the water. Conscientious consideration–on a case-by-case basis by all participants–should provide an economically, biologically, and environmentally acceptable treatment solution in almost all situations.

Additional research and more knowledge may, in the future, justify modifications to buffer zones and the agreements between Federal agencies and land managers. Until the knowledge is available to call for modifications, the guidelines set forth in the 1987 EIS and guidelines specified for T and E species will dictate how buffer zones are established for grasshopper control programs.

Conclusions

Buffer zones play a vital role in protecting the environment during grasshopper control programs on public lands. APHIS and land-management agencies regularly share information about T and E species, aquatic areas, and sensitive areas necessary to provide effective buffer zones. Currently, APHIS uses the guidelines contained in the 1987 EIS when conducting treatment programs for rangeland grasshopper control and suppression. As noted in the EIS, buffer zones may be subject to revision as new information comes to light.

APHIS bases its treatment programs on sound biological knowledge. At no time does APHIS intentionally jeopardize nontarget species in a treatment block. Buffer zones reflect the desire to provide protection as needed. Customized treatment programs could help resolve difficult situations, especially when grasshopper populations are building and presence of buffers within treatment areas could lead to reinfestation.

References Cited

Bednarski, L. T.; McEwen, L. C. 1994. Response of post-fledging golden eagles *(Aquila chrysaetos)* to Sevin-4-oil application. In: Abstract book, 15th annual meeting, Society of Environmental Toxicology and Chemistry; [date of meeting unknown]; Denver, CO. [Place of publication unknown]: Society of Environmental Toxicology and Chemistry: 249.

Beyers, D. W.; Farmer, M. S.; Sikoski, P. J. 1995. Effects of rangeland aerial applications of Sevin®-4-Oil on fish and aquatic invertebrate drift in the Little Missouri River, North Dakota. Archives of Environmental Contamination and Toxicology 28: 27–34.

Johnson, W. W.; Finley, M. T. 1980. Handbook of acute toxicity of chemicals to fish and aquatic invertebrates. Resour. Publ. 137. Washington, DC: U.S. Department of the Interior, U.S. Fish and Wildlife Service. 98 p.

Reynolds, T. D.; Trost, C. H. 1980. The response of native vertebrate populations to crested wheatgrass planting and grazing by sheep. Journal of Range Management 31: 122–125.

Schleve, G.; McEwen, L. C.; Clements, W. E. 1993. Sublethal effects of carbaryl and malathion on nestling American kestrels. In: Abstract book, 14th annual meeting, Society of Environmental Toxicology and Chemistry; [date and place of meeting unknown]. [Place of publication unknown]: Society of Environmental Toxicology and Chemistry: 87.

Taira, T. 1994. Blood analysis of American kestrel and golden eagle nestlings exposed to malathion or carbaryl. M.S. thesis. Ft. Collins, CO: Colorado State University. 80 p.

U.S. Department of Agriculture, Animal and Plant Health Inspection Service. 1987. Rangeland Grasshopper Cooperative Management Program: final environmental impact statement. Washington, DC: U.S. Department of Agriculture, Animal and Plant Health Inspection Service. 221 p.

References Cited–Unpublished

Bednarski, L. T. Response of post-fledging golden eagles *(Aquila chrysaetos)* to Sevin® 4-Oil application. M.S. Thesis. Ft. Collins, CO: Colorado State University. [In preparation.]

McEwen, L. C. and A.C. Fowler. 1989. Observations on breeding piping plover *(Charadrius melodus)* and chicks adjacent to a site treated with carbaryl bait for grasshopper control. Unpublished data.

McEwen, L. C. 1991. Observations on piping plover *(Charadrius melodus)* nesting success near a site treated with carbaryl bait and Sevin 4-Oil for grasshopper control. Unpublished data.

McEwen, L. C.; Petersen, B. E.; Beyers, D. W.; Althouse, C. M.; Bednarski, L. T.; Taira, T.; Schleve, G. R. 1994. Grasshopper Integrated Pest management Program Environmental Monitoring and Evaluation. In: Cooperative Grasshopper Integrated Pest Management Project, 1994 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 131–150.

Taira, T.; McEwen, L. C. Blood analysis of American kestrel and golden eagle nestlings exposed to malathion or carbaryl. [Manuscript has been submitted to Environmental Toxicology and Chemistry.]

III.9 Environmental Monitoring of Grasshopper Control Programs

Michael T. Green

Environmental monitoring is the measurement of the effect on the environment of pesticides used for pest control. Monitoring is required by law, is the policy of the Animal and Plant Health Inspection Service (APHIS), and provides useful information for pest-control programs. Monitoring has been, and will continue to be, an important part of grasshopper control operations.

Why Monitor?

Monitoring is required by the National Environmental Policy Act (NEPA) to document the implementation of mitigative (moderating) measures, such as buffers around sensitive sites. In APHIS, we monitor to compare residue levels and nontarget effects resulting from treatments with predictions made in the risk analyses in environmental impact statements written for programs such as grasshopper control.

Sometimes monitoring is conducted under the Endangered Species Act (ESA) to demonstrate protection of threatened and endangered (T and E) species or habitats that are critical for those species. Whether or not to monitor is specified in protection measures agreed to during consultations between APHIS and the U.S. Department of the Interior’s U.S. Fish and Wildlife Service (FWS).

Not only is environmental monitoring APHIS policy, it also provides valuable information for APHIS. Information gained from monitoring leads to a greater understanding of the effects of the program on the environment, information that has proven itself useful numerous times. Information gained also is valuable as a tool for assessing the effects of future programs, for educating the public regarding the effects of programs on public health and the environment, and for defense of the program in case of claims or of litigation over purported adverse effects.

In grasshopper programs, monitoring is done mostly out of concern for effects on nontarget plants and animals. Monitoring often is required around sensitive sites (habitats of T and E species, wildlife refuges, aquatic habitats, areas of human occupancy, and other sites of concern to the public) and to demonstrate that standard operating procedures or protective or mitigation measures are adhered to. In addition, monitoring is used to fill gaps in knowledge regarding the fate and transport of program chemicals or biological control treatments.

The Monitoring Plan

Environmental monitoring should be thought of as integral to every grasshopper treatment. APHIS’ Environmental Monitoring Team (EMT), within Plant Protection and Quarantine (PPQ), designs the monitoring plans for APHIS programs. EMT should be contacted in the early planning stages for each new control program, such as during the preparation of the site-specific environmental assessment (EA). EMT also should be contacted if treatments are planned for new areas already covered by a previously existing EA and no new EA is being prepared.

The APHIS State Plant Health Director (SPHD) or officer organizing the program should also involve the PPQ environmental monitoring coordinator when contacting EMT. If a site-specific EA is prepared, it should state whether or not monitoring will be conducted and then describe the type of sensitive sites to be monitored. EMT—in coordination with the SPHD, the environmental monitoring coordinator, and the FWS if T and E species are involved—will determine whether any sites should or should not be monitored. If monitoring is required, then EMT personnel will write the monitoring plan.

The monitoring plan will describe where and when sampling will take place, what will be sampled, and how many samples should be collected. The types of samples collected might include flowing or stationary water, soil, sediment, fish, insects, vegetation, and dye cards that measure airborne drift. Trained personnel (environmental monitors) will carry out the monitoring plan and send samples for residue analysis to APHIS’ National Monitoring and Residue Analysis Laboratory (NMRAL) in Gulfport, MS. The results from the laboratory are analyzed by EMT and interpreted with the aid of field notes and data collected at the time of treatment and sample collection. These data are reported in monitoring reports by EMT at the end of the treatment season. Addresses and phone numbers are listed on the next page.

Addresses and Phone Numbers
USDA–APHIS–PPQ
National Monitoring and Residue Analysis
 Laboratory (NMRAL)
3505 25th Avenue, Building 4
Gulfport, MS 39501
(228) 863–8124
(228) 867–6130 FAX

USDA–APHIS–PPQ
Environmental Monitoring Team
4700 River Road, Unit 150
Riverdale, MD 20737–1237
(301) 734–7175
(301) 734–5992 FAX

Monitoring Tools

There are many tools environmental monitors use to collect samples from the environment. It is important to make a list of the equipment necessary before starting environmental monitoring. NMRAL will send supplies overnight if necessary. The basic tools are dye cards, which are used to measure airborne drift of chemicals and pans or gypsy moth sticky traps to collect drifting bait.

Water is collected by dipping a container into the water body or continuously sampled with a peristaltic pump, depending on the sampling question of interest, the type of water body being monitored, and the chemical being sampled. Soil corers sometimes are used to collect soil; vegetation is collected by (gloved) hand. Water samples must be stabilized by lowering the pH with a special kit, and all samples must be frozen as soon as possible after collecting. This process requires having a large freezer nearby, even at relatively remote sites, and preferably dry ice or an ice bath in which to place bagged, labeled samples in the field. EMT and NMRAL are available to help with questions about collecting sites and methods.

Monitoring plans and techniques require considerable forethought and planning. It is critical, therefore, to get EMT involved early on in any operation, so that an environmental monitoring plan can be written, distributed, and worked into the overall cooperative control operation.

Chemicals in the Water?

The chemical labels for ultralow-volume (ULV) malathion, carbaryl, and carbaryl bait plainly state the risks to aquatic animals. The 2000 Cheminova label for Fyfanon® ULV malathion states, “This product is toxic to fish, aquatic invertebrates, and aquatic life stages of amphibians. For terrestrial uses, do not apply directly to water, or to areas where surface water is present. . . . Drift and runoff may be hazardous to aquatic organisms near the application site.” The labels for carbaryl spray and carbaryl bait are similar. For this reason, a 500-ft no-treatment buffer for aerially applied ULV pesticides and a 200-ft buffer for bait applications have been adopted as operational procedures in grasshopper programs.

The technology for detecting chemical residues is such that malathion residues can now be detected in water down to about 1/100th (0.01) of a microgram per liter (µg/L). In a pond 1 acre in size and 1 foot deep, the amount of malathion necessary to create residues near 0.05 µg/L is only about 0.03 fluid oz, or 0.38 percent of the original application (8 fluid oz/acre). Thus, if 99.5 percent of the spray lands on its target or in the buffer, and just 0.5 percent of it reaches a 1-ft-deep 1-acre pond, then the resulting residues would be detectable. The calculations for carbaryl are similar. At 1.0 µg/L, small aquatic crustaceans and aquatic stages of insects become susceptible. These organisms are more tolerant of carbaryl residues, showing sensitivity near 1 to 5 µg/L. Fish are from 10 to 1,000 times more tolerant of malathion and carbaryl than are aquatic invertebrates.

The chemical label states the risks of the pesticides to aquatic organisms and that drift and runoff could be harmful to them. The self-imposed buffers in the grasshopper program are probably sufficient in most cases to prevent harmful residues. Regardless, monitoring is recommended to be sure aquatic ecosystems are unaffected by program activities. Dye cards at the water’s edge and water samples will help program managers detect and quantify any residues reaching the water and suggest when buffers might need to be enlarged to minimize residues further.

Although carbaryl and malathion are the most commonly used pesticides in the grasshopper program, other pesticides (such as Dimilin®) might be adopted in the future. Most pesticides that would be effective at grasshopper control probably also will require a no-treatment buffer and residue monitoring around water bodies.

Conclusions

Environmental monitoring is a method of assessing effects of the grasshopper control program on nontarget animals and plants. Monitoring sometimes is required to bring the program in compliance with Federal statutes such as the ESA and the NEPA. APHIS also has the policy of monitoring the environment around pest eradication and control programs such as the cooperative rangeland grasshopper control program.

Whether or not monitoring is required depends on the site, the presence of T and E species, protected areas, wetlands, and other factors. EMT will help determine if monitoring is advisable for specific grasshopper control operations and should be contacted as early as possible during the planning of such operations.

Information gained through monitoring has been of considerable value to the program in the past, and monitoring will continue to be an important part of grasshopper programs in the future.

IV. Modeling and Population Dynamics

Weather stations in the field, like this one near Young, AZ, supply valuable information used in grasshopper phenological studies. (Agricultural Research Service photo by James R. Fisher.)

IV.1 What Modeling Is and How It Works

Jerome A. Onsager

A range manager and a modeler have at least four traits in common. Both respect intuition and experience, both are subject to bias, both are exposed to risk, and both do the best they can with the information that is available. Those range managers who believe that two or more heads can solve a problem better than one are encouraged to read on about modeling. In a recent book about modeling insect populations, Goodenough and McKinion (1992) describe a model as “a representation of a real system,” and then define a system as “a collection of a number of elements or components which are interconnected to form a whole.”

How does modeling work? First, modeling uses mathematical symbols and processes to express relationships that, as scientists and land managers, we think we understand or that seem reasonable. The knowledge or logic is greatly condensed into extremely efficient statements called formulae. This usually is possible only after a lot of clear thinking, problem definition, and trial-and-error evaluations have taken place. Next, the formulae are imbedded in a computer program. Doing this requires a rigid format for reasoning that requires each user to consider every important element. Finally, the user provides as many details as possible about as many elements or components as necessary, after which the model calculates a likely representation of response by the system.

The least complex systems contain few elements and are open to few outside influences. A simple example is a hydraulic jack. If one assumes no leaks and essentially 100-percent efficiency, each stroke of the handle yields a result that can be predicted exactly. Rangeland obviously represents an opposite extreme of complexity, with its multitude of physical forces plus plants and animals of all sizes, each affecting each other in ways that often are unknown. As land managers and scientists, we do not pretend that we can precisely model the entire system, but we are confident that we can model some elements to a useful degree.

The chapters in this section all discuss interrelationships among elements or components of rangeland ecosystems that are important to grasshopper management. A small proportion of that prose already has been translated into mathematical language and is being used in the grasshopper model portion of Hopper (the decision support tool that is described in VI.2). Examples include the time and rate of grasshopper development as a function of temperature, forage consumption as a function of grasshopper size and density, and expected responses of grasshopper populations to management tactics.

For a variety of reasons, the overwhelming majority of the following chapters is not yet available in management-oriented models. In some cases, like soil temperature–egg development relationships, the information was acquired only recently. In other cases—like relationships between weather, host plant quality, grasshopper food consumption, and grasshopper population dynamics—causes and effects have not yet been precisely quantified. In still other cases, like predicting outbreaks, scientists and land managers cannot yet calculate which one of several likely events will eventually occur. The information nevertheless is being presented in narrative form, intended both to establish the current state of knowledge about grasshopper population dynamics and to expedite future modeling efforts.

For additional insights about what modeling is and how it works, you are encouraged to study appendix A of the Hopper Users’ Guide (VI.2). Also, chapters in section VII discuss models that probably will be developed in the near future.

Reference Cited

Goodenough, J. L.; McKinion, J. M. 1992. Basics of insect modeling. Monogr. 10. St. Joseph, MI: American Society of Agricultural Engineers. 221 p.

IV.2 Grasshopper Egg Development: the Role of Temperature in Predicting Egg Hatch

J. R. Fisher, W. P. Kemp, F. B. Pierson, and J. R. Wight

Hatch, the emergence of a nymph from the egg, is an important phenomenon in the life of a grasshopper. The embryo, the developmental stage that precedes the nymph, is the longest living stage, often lasting more than 10 months. The timing of hatch is important to grasshopper management because the timing of management activities is linked to nymphal emergence from eggs in the soil.

Most North American grasshoppers have one generation per year. Eggs are usually laid (oviposited) during late summer and early fall and hatch the following spring. There are usually five developmental stages (instars) that are present over a period of about 45 days during the late spring to early summer. Grasshoppers can usually be found as adults in the summer months up to late September, depending on the occurrence of the first hard frost.

Development and distribution of grasshoppers is largely governed by temperature. Each species has adapted to temperatures and other conditions of its habitat. The ancestors of modern grasshoppers were probably general feeders and lived in areas that had mild temperatures (>32 °F) all year. Over time, climate and habitat changed, as did food resources. Each species adapted, migrated, or perished.

Overwintering Adaptations

A number of adaptations have been described for insects that occur in the temperate regions. Most insects that spend the winter as a nymph or an adult have adapted by inreasing the amounts of complex sugars or glycerols (antifreeze-like compounds) in their blood. As winter approaches, these insects seek out areas such as the bases of plants, crevices on the outsides of buildings, soil cracks and crevices, nooks under rocks or tree bark, or even the insides of buildings. These insects overwinter in a dormant state (stupor) called quiescence or aestivation. They are inactive but will become active whenever the temperature in their microhabitat warms enough to support physiological processes: you may recall flies flying around on a warm day in January. However, these insects will go back to the quiescent state when the temperature cools.

Another adaptation to environmental adversity is a phenomenon called diapause. Diapause commonly occurs either in the embryonic stage, the late larval stages, or the pupal stage. Diapause is like quiescence, but instead of a stupor brought on by cold temperature, diapause is a state of suspended animation of nearly all physiological processes. That state has been genetically programmed in the insect over evolutionary time.

There are two kinds of diapause. Facultative diapause is brought on by certain environmental conditions and may only happen to individuals that are exposed to that condition or set of conditions. Obligatory diapause occurs to nearly every individual of a population at the same stage of development regardless of climatic or photoperiodic conditions. With either kind, once an insect is in the state of diapause, it stays in that state, no matter what kind of climate is encountered, until a certain event or events occur. These events can be a specific sequence of moisture regimes (such as contact moisture), temperature, photoperiod, time, or combinations thereof.

Overwintering in Grasshoppers

Grasshoppers lay eggs in the soil. In the act of laying eggs: first, a female grasshopper digs a hole in the soil with the tip of her abdomen to the depth of 0.4–1.0 inch (1–2.5 cm); second, she secretes a viscous material to line the hole (this becomes the pod); third, she places the eggs in the pod; and last, she plugs the pod with a frothy substance. Subsequently, the pod is covered with fine soil; the female places nearly each grain of soil with her hind legs. Temperature at pod depth in the soil is critical to the development of an embryo.

Most species of rangeland grasshoppers have one generation per year and have an embryonic diapause that occurs several weeks after the eggs are laid and usually lasts until the ground is frozen or freezing temperatures are common. Through diapause, these grasshoppers avoid hatching in the late summer and fall, when conditions would be unfavorable for growth and development. Diapause is the primary reason why most North American grasshoppers have only one generation per year.

For most species of the genus *Melanoplus,* embryonic diapause is facultative. With *Melanoplus sanguinipes*, a major pest grasshopper of rangeland and crops in the Western United States, diapause may last from 0 days to more than 200 days when eggs are held at room temperature. Environmental conditions, such as photoperiod length (daylight length) and temperatures experienced by the female, have been mentioned as possible factors that influence the occurrence and length of diapause in this species. However, in North America north of latitude 36° (Las Vegas, NV), *M. sanguinipes* eggs appear to require either some diapause or cold quiescence before winter because no partial or whole second generation has been reported.

Aulocara elliotti, the bigheaded grasshopper, is a grass-feeding specialist and rangeland pest that has, in the northern tier of the Western United States, an obligatory diapause. The diapause occurs when an individual *A. elliotti* embryo is about 60 percent developed; this stage is reached within 8 days after egg laying if the daily temperatures average about 86 °F (30 °C). If the temperatures average only about 68 °F (20 °C), *A. elliotti* eggs will take about 14 days to reach 60-percent development. *Ageneotettix deorum,* the whitewhiskered grasshopper, another grass-feeder on rangelands, appears to have an obligatory diapause similar to that of the bigheaded grasshopper.

Termination of Embryonic Diapause

Some persons aware of the process of embryonic diapause may think that diapause is "broken" (terminated or completed) by exposure to cold winter temperatures. This idea is partially true. With some insects, the amount of time spent in embryonic diapause has been found to be controlled by a hormone called the diapause hormone (DH). Hormones in insects are much the same as hormones in humans; each has a specific purpose and each can enhance or reduce the actions of certain other hormones. DH is initially at a high level (titer) in diapausing eggs. A high titer prevents a growth promoting hormone, esterase A (EA), from doing its job. With some insects, time decreases the activity of DH. In other insects, cool temperatures (around 37–59 °F [3–15 °C]) promote an increase in EA titers and activity and a regression of titers of DH.

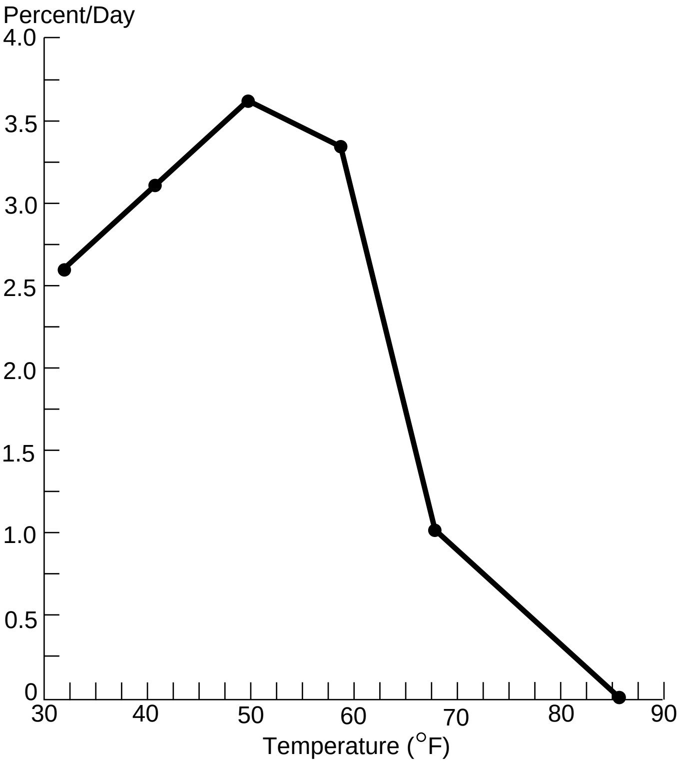


Figure IV.2–1—Generalized illustration of the percent of diapause completed per day when a diapausing embryo is exposed to certain cool temperatures.

Figure IV.2–1 illustrates, in general, the amount of diapause completed per day by a hypothetical insect that requires cool temperature to terminate diapause. This illustration was compiled by the authors after an extensive review of embryonic diapause of a number of insect species from temperate climates that spanned three orders—Lepidoptera (moths and butterflies), Coleoptera (beetles), and Orthoptera (grasshoppers, roaches, walking sticks, crickets). This illustration could represent, in a circumstantial way, the amount of DH dissipated daily at the temperatures represented.

The time between diapause initiation and termination is often called diapause development; not much is developing, but hormonal action and some metabolism are going on. Figure IV.2–1 shows that the fastest diapause development times (>3.0 percent per day) would occur near 45–54 °F (7–12 °C). This is true for the grasshoppers *Aulocara elliotti* and *Ageneotettix deorum* and possibly other rangeland grasshoppers. To put this in perspective, the following example helps explain the meaning of figure IV.2–1. If the daily temperatures averaged 50 °F, diapause development would occur in increments of about 3.5 percent per day. To determine the amount of time needed to complete diapause at 50 °F, divide 100 percent by 3.5 percent. The result—29 days—is the period of development needed to have complete diapause.

North of 40° latitude (Salt Lake City, UT), this ideal temperature range (the range of fastest diapause development, 45–54 °F) occurs in the months of September, October, and November. Of course, we are considering average temperature; most nights are colder, and many daylight hours are much warmer. Even so, for many species, diapause usually is terminated by early to mid-November (> 90 days after the end of egg laying by most grasshoppers).

Spring Egg Hatch (Postdiapause Development)

Once diapause terminates, normal embryonic development will proceed whenever temperatures exceed 50 °F (10 °C). This is called the developmental threshold (DT), the temperature below which nearly all metabolic processes cease (quiescence). At temperatures above the DT, metabolic processes proceed at increased rates with increasing temperatures (the higher the temperature, the faster the metabolism) until a lethal temperature, usually >106 °F (41 °C), is reached. The increases in metabolic processes translate into a rate of development for the embryo. Table IV.2–1 shows the postdiapause embryonic development rate in relation to soil temperatures for four pest species of grasshoppers. These development relationships were derived from several of our experiments with egg development and hatch.

Predicting *Aulocara elliotti* Hatch

To predict the hatch of an insect such as *Aulocara elliotti*, two key pieces of information are needed: when diapause terminates and the rate of embryonic development. Because these are insects that hatch at spring temperatures, grasshoppers are extremely temperature dependent. They also have an obligatory diapause that stops development until certain temperature requirements are met. Most insects take very little time to resume normal metabolism once the DT is reached. But if they are in diapause, time exposed to temperatures above the DT does not contribute to development. Thus, it is important to know when diapause terminates. Knowledge of the rate of embryonic development at various nonlethal and nonquiescent temperatures is necessary if daily or hourly temperature averages are used as drivers for a model that predicts hatch.

Table IV.2–1—Days needed for a grasshopper egg to hatch when exposed to various constant soil temperatures

Temperature		Days to hatch			
°F	°C	*Melanoplus sanguinipes*	*Melanoplus bivittatus*	*Melanoplus differentialis*	*Aulocara elliotti*
50	(10)	—	595	250	602
59	(15)	33	26	49	135
68	(20)	15	13	27	36
77	(25)	10	9	18	15
86	(30)	7	6	14	11
95	(35)	6	5	11	10
104	(40)	5	4	9	9

Aulocara elliotti Diapause Termination

We determined the time of diapause termination (completion) for *A. elliotti* by collecting egg pods from the field periodically from early October through the spring of 1990–91 and 1992–93. We subjected the egg pods to temperatures of 86 °F in the laboratory for 120 days. At that time (120 days), we determined how many had hatched, how many were dead, or how many were still alive.

In Figure IV.2–2, live eggs can be interpreted to still be in diapause. From these studies, we found that more than 70 percent of the eggs hatched and thus had completed diapause by the collection on Julian date (JD) 317 (Nov. 13) (fig. IV.2–2). However, note that more than 30 percent had hatched from collections on JD 287 (Oct. 14) in 1992 and by JD 300 (Oct. 27) in 1990. By the collection date 334 (Nov. 30), in both seasons nearly 100 percent of the eggs that survived to hatch had terminated diapause. When we considered these results and the normal variability in vital life events for most animals and, in particular, *Aulocara elliotti*, we decided to begin our hatch predictions by accumulating above-DT temperature units from JD 303 (Oct. 30).

Aulocara elliotti Rate of Embryonic Development

Table IV.2–2 shows the days needed for hatch and the rate of development of an embryo of *Aulocara elliotti* when held, after diapause, at constant temperatures from 59 °F (15 °C) to 108 °F (42 °C). The observed median is from our actual data. But, to predict hatch from an actual temperature base, we needed to create a model (equation) from our data that represented the embryo's reaction to a continuum of temperatures. For this we went to simple high school algebra and derived a rate model, an equation that fitted a sine curve because the data appeared similar to a sine curve. The rate of development per day is the reciprocal of the predicted median days to hatch.

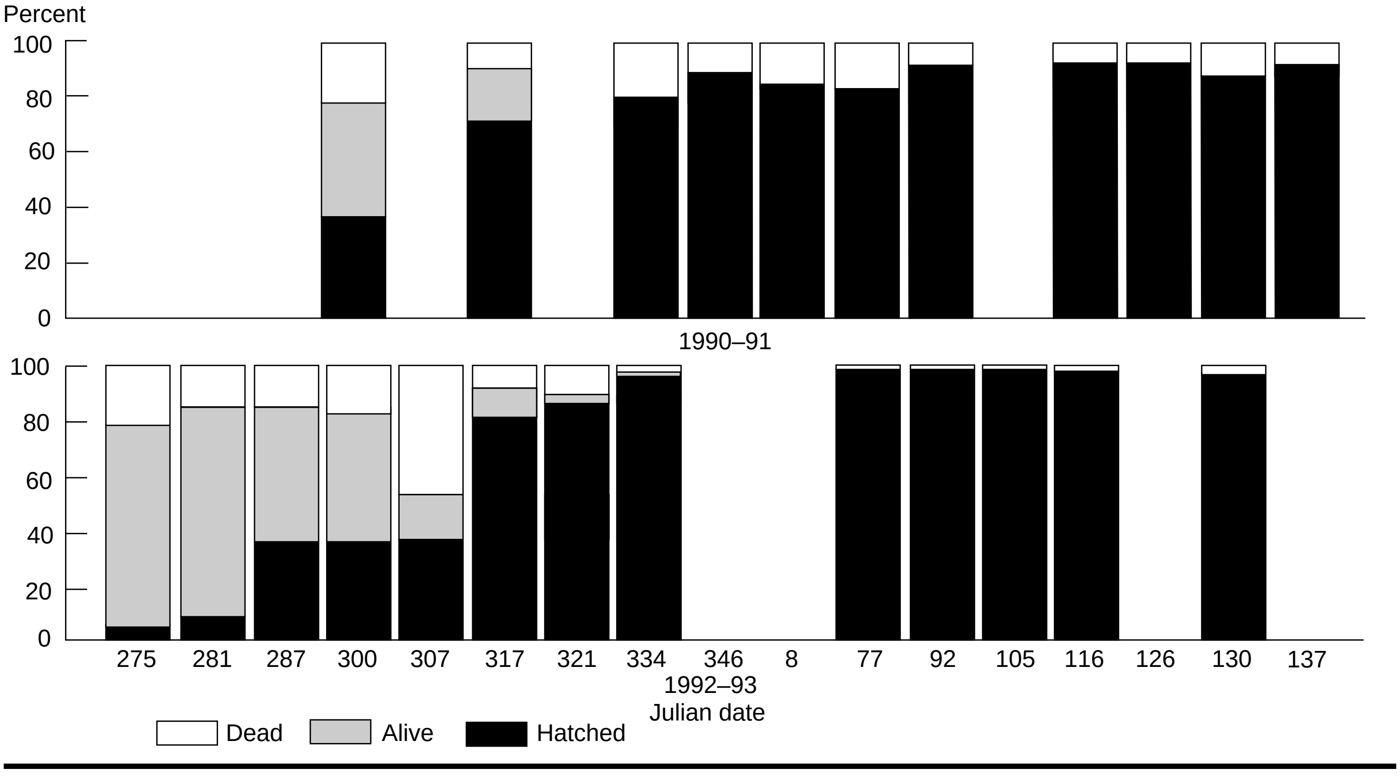


Figure IV.2–2—Proportion of hatch (alive *v.* dead eggs) of *Aulocara elliotti* collected in the field from October to the spring of 1990–91 and 1992–1993 when exposed to 86 °F (30 °C) for 120 days after collection.

Table IV.2–2—Observed median days to hatch and predicted median days to hatch and rate of embryonic development per day for *Aulocara elliotti* eggs after diapause, when held at various constant temperatures

Temperature °F	(°C)	Median (observed)	Median (predicted)	Rate of development/ day
				Percent
59	(15)	136.00	92.9	1.01
64	(18)	56.00	59.17	1.7
75	(24)	21.08	25.38	4.0
81	(27)	15.18	17.42	5.8
86	(30)	16.29	12.50	8.0
91	(33)	9.66	9.46	10.6
97	(36)	7.28	7.8	12.8
102	(39)	6.00	6.42	15.6
108	(42)	5.98	5.70	17.5

Prediction of Hatch of *Aulocara elliotti*

Most air and soil temperatures are monitored for a daily high–low record or an average hourly record. For this study we used an hourly record of soil temperature from egg-pod level, three quarters of an inch (2 cm) below the surface of the soil. A straightforward prediction of hatch could be made by taking the hourly temperature after JD 303 (Oct. 30) and placing it in the rate of development equation and tallying the amount of development for each hour over a 24-hour period and then tallying this predicted development over each day of the winter and spring. However, this calculation does not take into account the variation that is omnipresent for every metabolic process among individuals in a species. This problem was corrected by using another model that accounted for the variation in development times found for each group of eggs tested at the various constant temperatures.

Through some computer software (PMDS, Version 5) we were able to take the two models mentioned earlier and the temperature data and derive predictions for hatch for two sites in southwestern Montana over 2 years (table IV.2–3). Site MH1 is at 4,412 ft (1,345 m) above sea level, and site MH2 is at 5,075 ft (1,547 m) above sea level. The two sites are about 2 mi (3.2 km) apart. To see how accurate our predictions were, each day from late April through mid-July in each year we collected first-instar grasshoppers at each of the sites (MH1 and MH2) (table IV.2–3).

Model Efficiency

Accuracy of these models is best noted when the prediction of 50-percent hatch is indicated. If you examine table IV.2–3, you will notice that the predicted 50-percent hatch was within 1 day or less of the actual first-instar samples for three of the four comparisons. With MH1 for 1992, the 50-percent hatch was predicted to occur only 7 days beyond actual. In both years, MH2 actual hatch did not start until at least 10 days later than at MH1. Temperatures at the higher altitude were cooler; thus, hatch was later.

Utility and Implications of These Models

The sensitivity of these models is remarkable. We feel that accuracy in the predictions was obtained by (1) knowing a starting time to begin our temperature accumulation for hatch (diapause termination), (2) taking temperature at pod level (microclimate of the egg), (3) knowing an estimate of the variation in hatch of species at an array of temperatures, and (4) knowing the rate of development of the postdiapause embryo at an array of above-quiescent, below-lethal temperatures.

Our two sites had a difference of 650 ft (198 m) in altitude. At the higher altitude site, hatch was later—at least 10 days. Many areas within a management district will vary in altitude, land aspect, distance from mountains, and more. These features cause changes in microclimate. When these microclimatic differences are tallied over a 5- to 6-month period, their influence on embryonic development may be significant.

Most range managers do not have access to records of soil temperatures at 0.4 inch to assist with prediction of hatch at a site. However, air-temperature records at 1 ft (30.4 cm) or 3 ft (91.4 cm) are common, and instrumentation to assist in maintaining records is reasonably priced and readily available. We have developed a simulation model with the objective to predict soil temperature accurately at 1–2 cm by using air temperature at 3 ft

Table IV.2–3—*Aulocara elliotti* egg hatch, by percentage and Julian date, at two Montana sites (actual sampling *v.* model predictions)

Site, year		Percent of egg hatch					
	Initial	1	5	25	50	75	90
MH1							
1991							
Sweep sample	133	144	149	153	156	159	164
Model	130	140	145	153	157	161	165
MH1							
1992							
Sweep sample	111	119	122	126	128	131	134
Model	93	97	122	129	135	139	142
MH2							
1991							
Sweep sample	154	154	158	163	168	171	176
Model	144	147	155	163	168	171	176
MH2							
1992							
Sweep sample	120	120	128	139	143	145	149
Model	98	125	129	139	143	148	152

above the ground (see V.9). Thus, by using the soil temperature simulation model and our *A. elliotti* hatching models that are based on soil temperature at 1–2 cm, air-temperature data banks that have been kept over a number of years at any site may be able to accurately predict when hatch of this species would begin (this work is in progress). Accurate soil temperature prediction from air temperatures used with these models for hatch would assist with the timing of survey assessment of populations and with the timing for consideration of management options.

Suggested References

Danks, H. V. 1987. Insect dormancy: an ecological perspective. Biol. Surv. Canada Monogr. Ser. 1. Ottawa, ON: Biological Survey of Canada. 439 p.

Downer, R.G.H.; Laufer, H. 1983. Endocrinology of insects. New York City: Alan R. Liss, Inc. 707 p.

Fisher, J. R. 1993. Location of *Aulocara elliotti* egg pods in a crested wheatgrass field in Montana. Journal of the Kansas Entomological Society 65: 416–420.

Fisher, J. R. 1994. The effect of temperature on the post-diapause development and survival of embryos of three species of *Melanoplus* (Orthoptera: Acrididae). Annals of the Entomological Society of America 87: 604–608.

Fisher, J. R.; Kemp, W. P.; Pierson, F. B. 1996. Hatch of the bigheaded grasshopper, *Aulocara elliotti* (Thomas) (Orthoptera: Acrididae). Environmental Entomology 25: 1158–1166.

Hewitt, G. B. 1985. Review of factors affecting fecundity, oviposition and egg survival of grasshoppers in North America. Bull. ARS-36. Washington, DC: U.S. Department of Agriculture, Agricultural Research Service. 35 p.

Kemp, W. P. 1986. Thermoregulation in three rangeland grasshopper species. Canadian Entomologist 118: 335–343.

Kemp, W. P.; Sanchez, N. E. 1987. Differences in post-diapause thermal requirements for eggs of two rangeland grasshoppers. Canadian Entomologist 119: 653–661.

Logan, J. A.; Weber, L. A. 1991. Population model design system (PMDS). A user's guide. Blacksburg, VA: Virginia Polytechnic Institute and State University, Department of Entomology and Department of Forestry. 72 p.

Shotwell, R. L. 1941. Life histories and habits of some grasshoppers of economic importance of the Great Plains. Tech. Bull. 774. Washington, DC: U.S. Department of Agriculture. 47 p.

Tauber, M. J.; Tauber, C. A.; Masaki, S. 1986. Seasonal adaptations of insects. New York City: Oxford University Press. 411 p.

IV.3 Grasshoppers and Vegetation Communities

Anthony Joern, William P. Kemp, Gary E. Belovsky, and Kevin O'Neill

Important links exist between grasshoppers and the vegetation community. Vegetation communities provide the backdrop against which all grasshopper activities occur and determine the availability and distribution of all resources required by grasshoppers. Many critical questions concerning the relationship between vegetation communities and grasshopper communities remain unanswered, even unasked. Given the potential importance of such relationships to both forecasting of changes in grasshopper populations and grasshopper management activities, we summarize the currently available insights relevant to integrated pest management (IPM) activities on rangeland.

In this chapter, we stress that much unfinished research remains on critical questions concerning these communities. At the same time, we also stress that scientists understand a great deal, at least in terms of framing the appropriate questions. We will review the problem at two levels:

(1) At the macroscale level, how do grasshopper assemblies change as vegetational communities shift along environmental gradients? Do the dynamics underlying grasshopper community structure change; and, if they do, what are the consequences to the development of management tactics?

(2) On a more detailed, microscale level, how do grasshoppers actually use the structural and spatial components of their environments? What constitutes a resource in this sense and how do changing vegetational communities alter the quality or availability of resources for grasshoppers?

An extremely broad array of vegetation community types exists within the roughly 753 million acres of the Western United States classified as range (U.S. Department of Agriculture, Forest Service 1972). These plant community types, which range from inland deserts to alpine meadows, contain a collection of insects that often compete with humans for resources. Annual forage losses to grasshoppers alone often exceed 20 percent of the total annual production of rangelands in the Western United States (Hewitt 1977; Hewitt and Onsager 1982, 1983). Of the nearly 600 grasshopper species nationwide (Hewitt and Onsager 1982), 200 exist on rangelands (Onsager 1987, p. 60–66), and about 25 regularly reach economically damaging densities (Hewitt and Onsager 1983).

Unfortunately, management plans typically treat rangeland grasshoppers as a more or less homogeneous group from Montana to Arizona. Current rangeland pest management strategies seldom, if ever, consider differences among either vegetation patterns or grasshopper communities of varying species composition (Capinera 1987, Onsager 1987). These differences are ignored, even though important differences in biology exist among coexisting rangeland species (Kemp and Onsager 1986, Joern 1987, Kemp and Sanchez 1987, Onsager 1987).

Macroscale Patterns: Grasshopper and Vegetation Classifications

In recent years, plant ecologists have developed an environmental classification system based on the concept of habitat type (HT). Pioneered by Daubenmire (Daubenmire 1978), the methods for identifying HT's are those developed for identifying plant communities. HT's consist of discrete and repeatable vegetational units that characterize various resources, including forage or timber. Land managers use HT's to help predict responses to natural and human perturbations (such as fire, grazing or harvesting) (Mueggler and Stewart 1980, Pfister et al. 1977). The HT concept is being used increasingly in the management of forests and rangelands by the U.S. Department of Agriculture's Forest Service and Natural Resources Conservation Service (formerly the Soil Conservation Service) and the U.S. Department of the Interior's Bureau of Land Management. The HT concept has appeal in terms of resource management because it recognizes habitat heterogeneity yet reduces the complex vegetation landscape to a set of discrete groupings. Sites within the same HT thus can be managed in similar ways, and agencies can develop management strategies that are more rational from an ecological viewpoint.

If HT's can be used to classify sites satisfactorily into discrete groups based on the potential to produce resources and responses to management activities, it seems logical that HT's also will differ in their ability to sustain specific insect communities. A number of studies of mid- and large-scale communities have been conducted on species richness and diversity of both plants and grasshoppers (Vestal 1913, Otte 1976). Otte (1976), for example, observed that the Sonoran Desert of Arizona exhibited a significantly richer grasshopper fauna than the floristically “similar” Monte Desert of Argentina. Total niche space could not adequately account for these differences. Other regional studies that consider the gross distribution of grasshoppers have concentrated heavily on the presence of either grasshoppers (Isely 1937) or plants (Anderson 1973), with anecdotal inclusion of plants in the former and grasshoppers in the latter. Smaller scale studies (Banfill and Brusven 1973, Scoggan and Brusven 1973) that attempt to relate vegetation type to grasshopper community complexity typically lack the sampling intensity within given plant communities required to make regional inferences.

In a replicated study of patterns of plants and grasshoppers on Montana rangeland, Kemp et al. (1990a) found that the presence and relative dominance of about 40 individual grasshopper species changed with HT. The environmental gradients (precipitation and elevation) and plant species compositions of the different habitats determined grasshopper presence and dominance (fig. IV.3–1). In comparisons among plant communities associated with grasshopper communities along a natural elevational gradient, the native *Stipa comata–Bouteloua gracilis* HT (lower elevation and drier) and *Festuca idahoensis–Agropyron spicatum* HT (higher elevation and wetter) contained very different species complexes (Kemp et al. 1990a). Species like *Phlibostroma quadrimaculatum* and *Xanthippus corallipes* were found only in the drier habitats, whereas species such as *Melanoplus dawsoni* were found only in wetter sites.

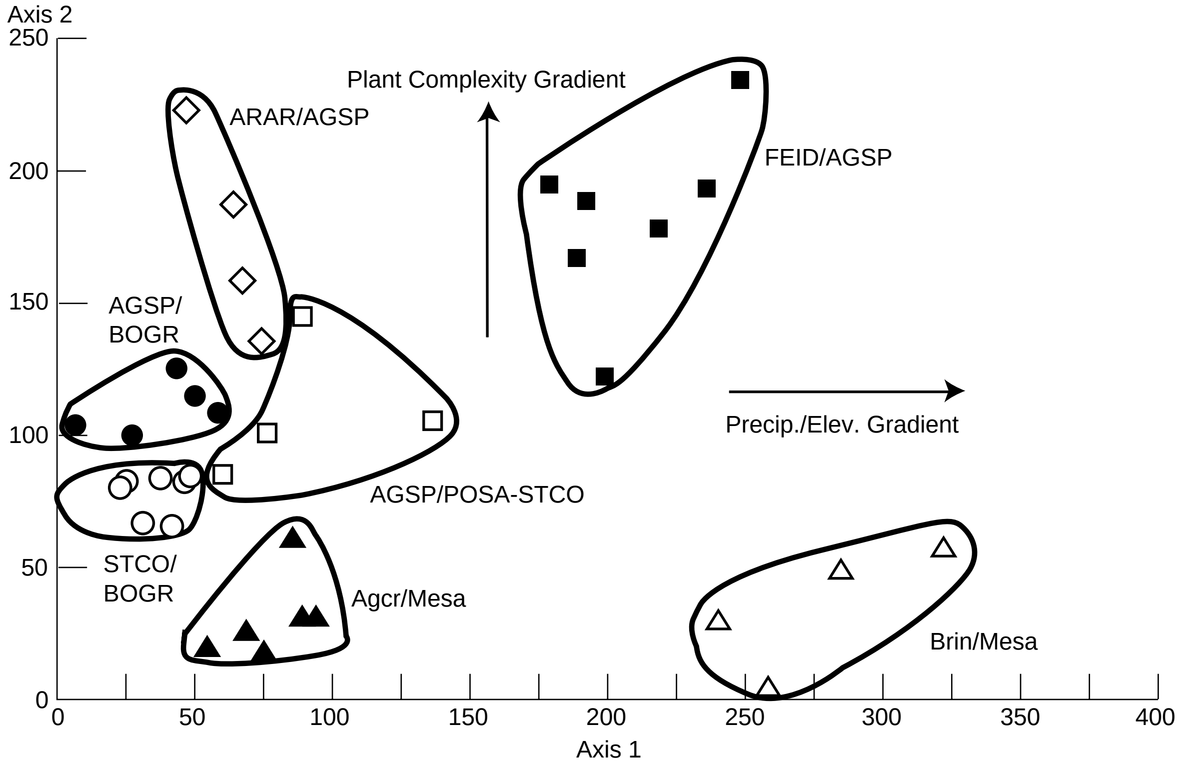


Figure IV.3–1A—Ordination of plant communities using detrended correspondence analysis of a range of habitat patches found in Gallatin Valley, MT, in 1988. A precipitation–elevation gradient is mostly responsible for spreading out the sites along the x axis, while a plant complexity gradient explains the spread along the y axis. Habitat codes relate to dominant plant species: Agcr=*Agropyron cristatum*, AGSP=*Agropyron spicatum*, ARAR=*Artemesia arbuscula*, BOGR=*Bouteloua gracilis*, Brin=*Bromus inermis*, FEID=*Festuca idahoensis,* Mesa=*Medicago sativa,* POSA=*Poa sandbergii,* STCO=*Stipa comata*. (Adapted from Kemp et al. 1990a.)

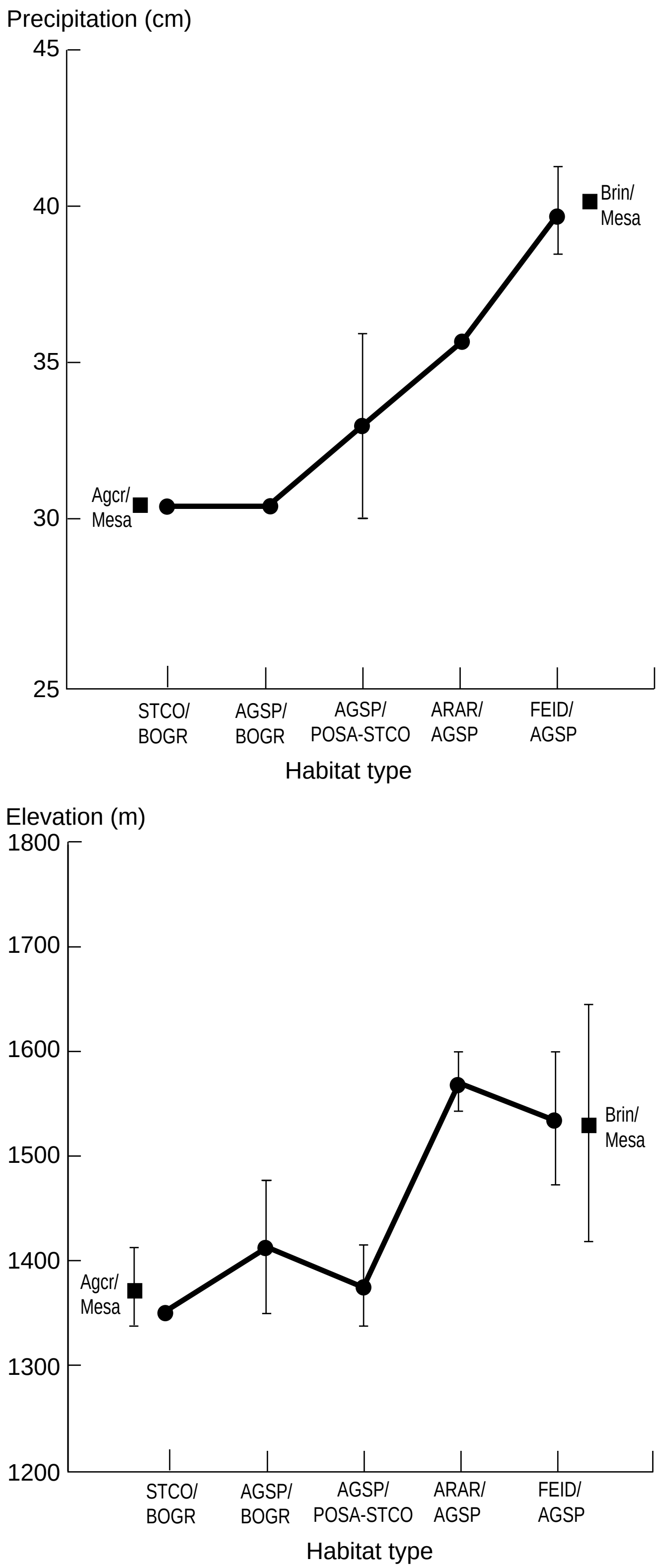


Figure IV.3–1B—Mean values (± 2 SE) for precipitation and elevation for a range of HT's surveyed for grasshoppers and vegetation in Gallatin Valley, MT, 1988. (Adapted from Kemp et al. 1990a.)

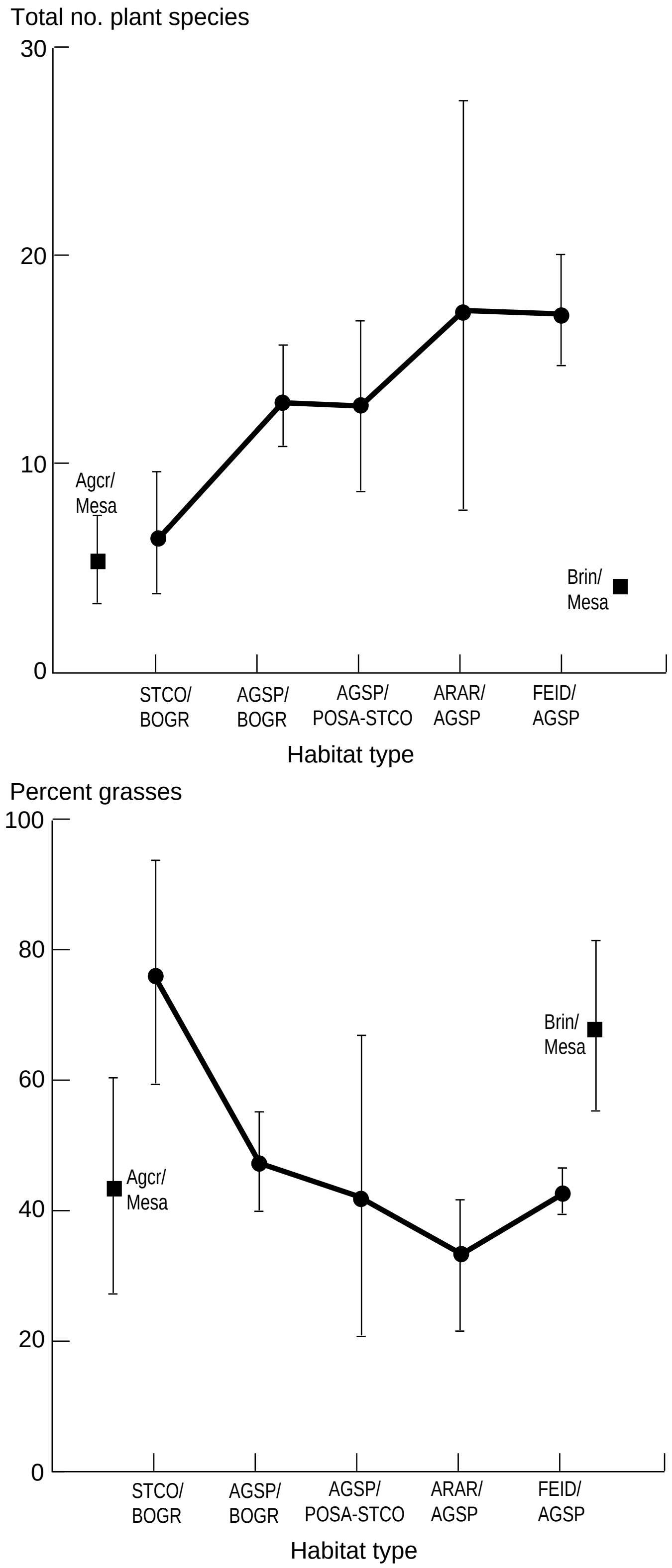


Figure IV.3–1C—Mean values (± 2 SE) for the total number of plant species and percent grasses for a range of HT's surveyed for grasshoppers and vegetation in Gallatin Valley, MT, 1988. (Adapted from Kemp et al. 1990a.)

Further, over a range of HT's, more than 10 common grasshopper species exhibited significant affinities for either end of the precipitation–elevation gradient.

Recent investigations in southern Idaho using mapping by ecological condition (another form of HT mapping) revealed a historic association between increased grasshopper densities and ecological disturbance, especially shrub loss from wildfires and other causes (Fielding and Brusven 1993a). Grasshopper assemblages from areas dominated by annual vegetation exhibited higher densities, lower species diversity, and broad diet breadths (Fielding and Brusven 1993b). Grasshopper assemblages from areas of perennial grasses largely contained grass-feeding species and exhibited high species diversity. Sagebrush–grass-dominated areas exhibited high grasshopper species diversity and lower densities. Therefore, land managers should not expect grasshopper communities to exhibit the same species composition from place to place when vegetational or environmental gradients exist.

Additional support for the use of HT to make inferences concerning invertebrate herbivore communities comes from avian research. In a study of HT's (as defined herein), Harvey and Weaver (1976) found very distinct use patterns among approximately 50 bird species in the northern U.S. Rocky Mountains. The presence or absence of bird species differed seasonally and in space among replicated stands of six HT's.

On a small scale, HT and differences found among grasshopper communities over a range of HT's will influence the need for and expected success of some research and management activities on rangelands. Rangeland grasshopper species typically vary in their susceptibility to biological control agents such as *Nosema locustae* (Henry 1971, Ewen and Mukerji 1979). Grasshoppers also differ in their willingness to feed on bran bait, which is often used for applying such biologicals (Onsager et al. 1980). For example, both *M. sanguinipes* and *Ageneotettix deorum* are known to accept bait (Onsager et al. 1980). However, Quinn et al. (1989) found that densities of *Trachyrhachys kiowa*, a species known to reject bait (Onsager et al. 1980), were unaffected by the bait treatment. Therefore, communities composed of significant proportions of grasshopper species that either will not accept bran bait, are not susceptible to *N. locustae,* or both will not be vulnerable to this type of control plan.

Additionally, exploiting the relationship between HT and grasshopper species composition offers entomologists and ecologists a way of simplifying experimental design problems. For example, two problems could occur if a series of test and control plots designed to assess efficacy of a particular treatment were selected without regard to HT. First, the target grasshopper communities could be completely different among sites and therefore respond differently to the treatment. If this happens, decision makers may draw conclusions based upon misleading evidence. Second, the assessment of block, treatment, and interaction effects in standard Analysis of Variance type experimental designs could be confounded by other indirect influences of HT on grasshopper community complexity and sampling. Such confounding would severely limit interpretations of cause and effect in this hypothetical case, a serious problem because the investigator would be unaware of the confound.

The perception of what processes might lead to different insect community structure among HT's will also influence research directions on natural processes that affect insect populations. The effectiveness of natural enemies in stabilizing pest populations may vary among HT's and disturbance levels. Joern (1988) has shown that electivities (food choices) of the grasshopper sparrow *(Ammodramus savannarum)* for particular grasshopper species are dependent upon their relative abundance. Perhaps rangeland habitats of lower plant diversity also harbor less diverse communities of predators and parasitoids, as seems to be the case for cultivated systems (Russell 1989). Further, when food is a limiting factor (Belovsky 1986), we expect that grasshopper community composition will vary among HT's, the difference depending on the varying intensities of interspecific competition.

Within the rangelands of the Western United States, the relationships between grasshopper community composition, HT, and long-term population trends become important. Certain HT's may serve as indicators of impending general population increases or declines. Such HT's warrant continuous monitoring, even during years where general densities are low. These sites could comprise a regional early warning system for grasshopper population eruptions. Alternately, other HT's may rarely support high grasshopper densities.

The HT concept deserves additional emphasis in both pest management and insect ecology (Kemp et al. 1990b). The recognition of vegetational communities confers to the problem of pest management a discreetness that helps managers design appropriate remedies. The HT concept also helps identify links between a site and its biotic (species interactions) and abiotic (weather) attributes.

In terms of insect pest management, the use of the HT concept could allow managers to describe units in several different but related layers to facilitate the use of Geographic Information Systems. At any given moment, all of the aspects described in the preceding paragraphs will influence what insect species can occupy sites within a given HT. Further, all of these factors will contribute to the susceptibility to pest outbreaks (short-term increases in densities) or infestations (long-term, sustained high-level densities). If pest managers can employ "type" communities as indicators of current or future pest conditions, preventative rather than reactive management activities can be used. While it is obvious that reactive efforts will always be necessary in certain areas, the HT concept could also help managers anticipate the location of insect outbreaks in space and time.

Microscale: Vegetation Structure and Resources

Vegetation communities described earlier rely strictly on taxonomic (species) relationships. However, grasshoppers typically react solely to the resources supplied by the composite plant assemblage and seldom employ the same cues used by biologists or range managers to recognize plant communities. From a grasshopper's perspective, plant community means more than just a list of coexisting plant species. How the plant species present in a community are spaced relative to one another define the entire physical environment encountered by a grasshopper. For example, microclimatic features such as air temperature, wind speed, and incoming solar radiation levels are intricately entwined with the structural profile of the environment.

What are the consequences of these differences in perspective? If macroscale analyses correctly predict grasshopper assemblages, what are such analyses actually measuring from the grasshopper's viewpoint? Will such insights at this level contribute to developing successful control strategies? In this chapter, we identify critical resources that are needed by grasshoppers and that vary as plant communities change. These resources likely explain the large-scale patterns.

In choosing microhabitats or a range of microhabitats, grasshoppers must choose sites in which they can make a living. Actual microhabitat selection by grasshoppers represents a compromise among multiple factors that determine habitat suitability as shown in figure IV.3–2 (Joern 1979, 1985). Important attributes that define microhabitat suitability and correlate well with grasshopper microhabitat selection include (reviewed in Joern 1982) microclimatic variables (temperature, light intensity, humidity), food availability, structural qualities, oviposition sites, substrate characteristics that render an individual cryptic (hidden), or biotic features (presence of competitors or predators). Dynamics (availability and use) of each of these resource attributes underlie macroscale patterns and become important in developing grasshopper integrated pest management (IPM) tactics. Understanding each may provide the appropriate clues to devise sound practices that work in concert with naturally occurring processes. We provide several representative examples to indicate the impact of specific resources on habitat use or the reciprocal (effect of habitat structure on resource availability and use). In this sense, we emphasize elements of habitat structure determined by the plant community. Remember, a lot of research remains before scientists fully understand these issues.

Food Resources.—Plant community structure and taxonomic composition combine to define food availability. For some grasshopper species, especially for grasshoppers that exhibit restricted food preferences, the habitat becomes good or bad depending on the presence or absence of preferred food plants: nothing else may matter (Joern 1983). For grasshopper species that eat a variety of food plants, the relative abundance of grasshopper species varies according to the array of suitable food plants (including quality and productivity). Because the broad-scale habitat patterns described above include shifts in

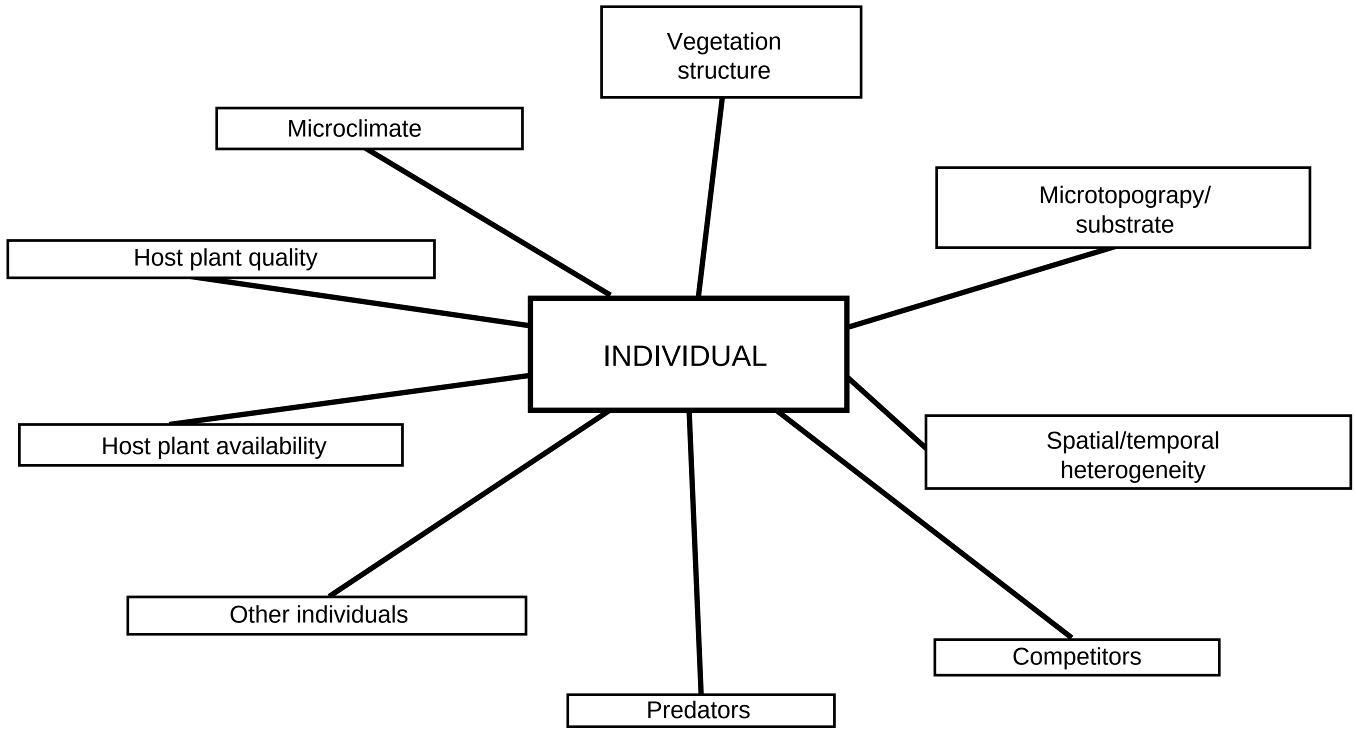


Figure IV.3–2—Environmental pressures that direct behavioral responses in patterns of resource use. (Adapted from Joern 1987.)

both the absolute and relative abundances of both plant and grasshopper species, responses at the grasshopper community level may relate to local food-use patterns. However, feeding responses by grasshopper assemblies to plant communities are not entirely species independent. Average diet breadth for entire grasshopper assemblies (an estimate of the range of plant species eaten) increases as average precipitation increases (fig. IV.3–3). Sites with low average precipitation (deserts and desert grasslands) contain fewer plant species, and grasshoppers tend to eat mainly more predictable plant species (Otte and Joern 1977), even though the diversity of plant species on a daily basis can be very high when present. At sites with higher average precipitation, average diet breadths increase, probably because more plant species exist at more predictable levels.

Structural Relationships.—Grasshoppers often position themselves in space based on structural aspects of the environment and exhibit clear species-specific differences (Joern 1979, 1981, 1982). For example, squat-looking species, such as *Ageneotettix deorum*, typically exist in open patches with little or no vegetation canopy compared with morphologically elongated species that live *on* vegetation *(Mermiria bivittata* or *Paropomala wyomingensis)*. For entire grasshopper assemblages, species partition available microhabitats in such a way that coexisting species tend to use microhabitats very differently (Joern 1979, 1982, 1986).

Grasshoppers exhibit the behavioral ability and visual sharpness to use structural and spatial cues to select microhabitats. Vegetation-inhabiting grasshoppers move toward vertical rather than horizontal cues (Williams 1954, Mulkern 1969). Presumably, ground-dwelling species are less responsive to these cues, but definitive studies have not yet been done. In addition to responding to vertical structure, many grasshoppers select microhabitats so that they blend with the background (Gillis 1982). Active microhabitat selection based on clearcut physical

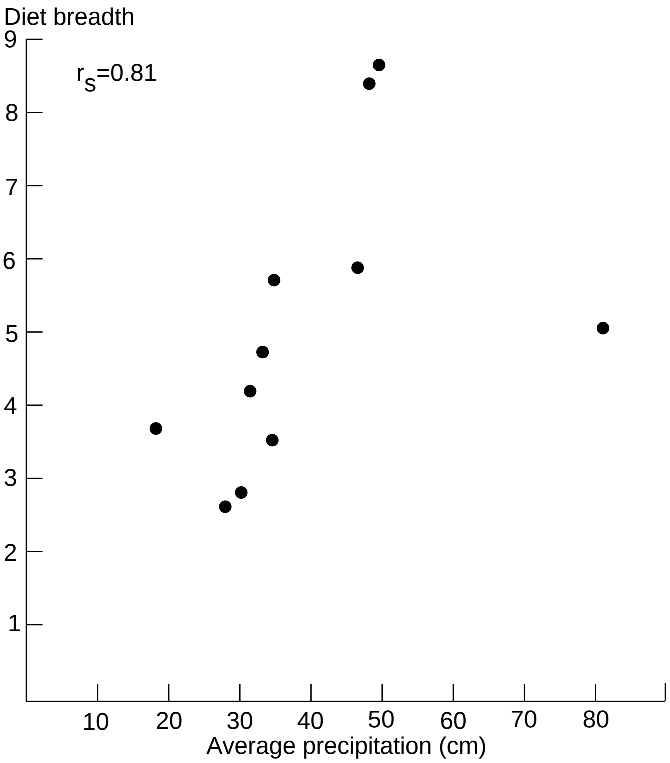


Figure IV.3–3—Average diet breadth of grasshoppers from sites across the United States that differ in total average precipitation. (Adapted from Yang and Joern 1994a and b.)

features of the environment supports our contention that structural resources provide important clues to understanding grasshopper distributions on a larger scale. These structural components derive directly from the vegetation community.

Thermal Attributes.—Body temperature underlies most physiological and biochemical processes associated with patterns of resources use. For example, developmental rates, food-processing capabilities, reproductive activity, life-cycle characteristics, and metabolic activity all are temperature-driven processes (Chappell and Whitman 1990). Perhaps more importantly for grasshopper IPM, many population processes are temperature dependent (Hilbert and Logan 1983, Begon 1983, Kemp and Onsager 1986, Kemp and Dennis 1989). Any factor that alters accumulated temperature by grasshoppers (either too little or too much) can profoundly influence population responses (Kingsolver 1989, Dunham et al. 1989).

As small animals with little control of body temperature (coldblooded animals), grasshoppers must rely on external heat sources and sinks to control body temperature. As with most insects, incoming solar radiation, windspeed, and air temperature coupled to anatomical features set the limits on grasshopper body temperatures (fig. IV.3–4A). Physical structure in the habitat directly affects each of these attributes. If grasshoppers were unable to thermoregulate, their body temperatures would track the temperature of the surrounding environment. However, using behavioral means, grasshoppers readily manipulate their body temperatures within a limited range, resulting in characteristic daily thermoregulation patterns (fig. IV.3–4B) (Joern 1981b, Kemp 1986).

Vegetation structure and topography interact with regional weather to determine the “microclimatic resources” that grasshoppers encounter for thermoregulation. Air temperature and incoming solar radiation levels ultimately determine a grasshopper’s energy budget (Dunham et al. 1989, Kingsolver 1989, Grant and Porter 1992). The number of hours of sunlight per day, the likelihood of cloud cover, or the effect of the vegetation canopy ultimately restrict access to solar radiation and can significantly alter the number of hours per day that a grasshopper can achieve optimal body temperatures. These restrictions limit the ability of the grasshopper to find, eat, and assimilate food and then allocate nutrients. As such, demographic responses will be shifted, not because of food quality, but because the grasshopper cannot take in and use the maximal levels.

Final Comments

Given the importance of the plant community as a provider of resources, it should not be surprising if grasshopper species and resulting communities correspond with vegetation changes in some predictable manner. Preliminary studies described in this section strongly suggest this possibility. IPM programs should refine and then exploit these relationships. For managers responsible for particular land parcels, detailed maps will provide insight about where to concentrate IPM efforts. For individuals responsible for larger areas, perhaps on a regional basis, vegetation-based analyses will provide a framework for efficient monitoring because survey efforts can be parceled more precisely.

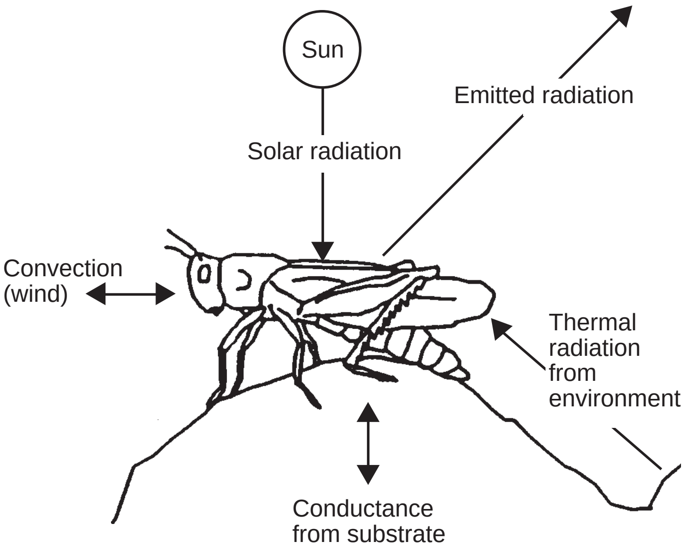


Figure IV.3–4A—Generalized heat-exchange pathways for a grasshopper on the ground. (Reproduced from Chappell and Whitman 1990; used by permission of John Wiley and Sons.)

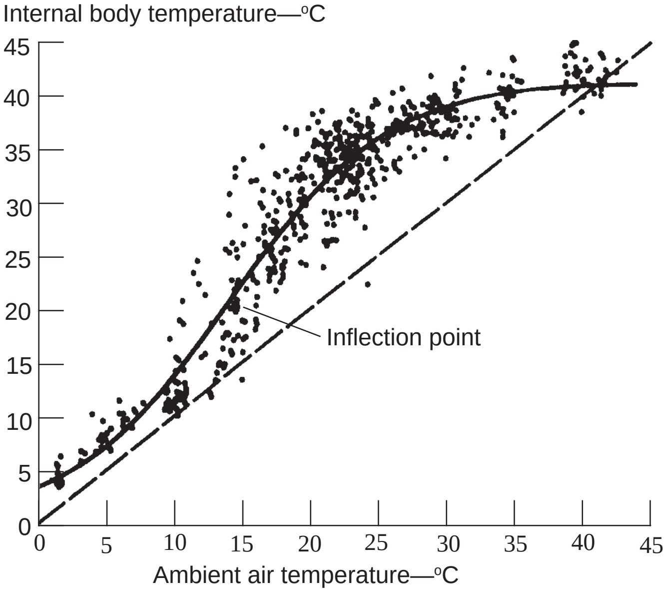


Figure IV.3–4B—The relationship between internal body temperature and ambient air temperature for *Aulocara elliotti* females over a broad range of ambient temperatures. (Adapted from Kemp 1986.) The solid line represents temperatures predicted from a logistic equation of the body temperature relationship based on ambient air temperature and incoming solar radiation. The dashed line indicates the situation where body and ambient air temperatures are equal.

However, important additional information that could help design effective IPM strategies derives from specific details associated with how grasshoppers actually use resources. We presented some representative but not exhaustive examples to clarify exactly what we mean here. Both forecasting efforts as well as cultural control (including grazing rotations to manipulate vegetation structure) can benefit from such insights. Finally, the behavioral responses that affect resource use and the resulting ecological patterns are truly complex. Scientists and land managers are just beginning to understand these interactions.

References Cited

Anderson, N. L. 1973. The vegetation of rangeland sites associated with some grasshopper studies in Montana. Bull. 668. Bozeman, MT: Montana State University and Montana Agricultural Experiment Station.

Anderson, R. V.; Tracy, C. R.; Abramsky, Z. 1979. Habitat selection in two species of short-horned grasshoppers: the role of thermal and hydric stresses. Oecologia 38: 359–374.

Banfill, J. C.; Brusven, M. A. 1973. Food habits and ecology of grasshoppers in the Seven Devils Mountains and Salmon River Breaks of Idaho. Melanderia 12: 1–21.

Belovsky, G. E. 1986. Optimal foraging and community structure: implications for a guild of generalist grassland herbivores. Oecologia 70: 35–52.

Begon, M. 1983. Grasshopper populations and weather: the effects of insolation on *Chorthippus brunneus*. Ecological Entomology 8: 361–370.

Capinera, J. L. 1987. Integrated pest management on rangeland: a shortgrass prairie perspective. Boulder, CO.: Westview Press. 426 p.

Chappell, M. A.; Whitman, D. W. 1990. Grasshopper thermoregulation. In: Chapman, R. F.; Joern, A., eds. Biology of grasshoppers. New York: John Wiley and Sons: 143–172.

Daubenmire, R. 1952. Forest vegetation of northern Idaho and adjacent Washington, and its bearing on the concepts of vegetation classification. Ecological Monographs 22: 301–330.

Daubenmire, R. 1959. A canopy cover method of vegetational analysis. Northwest Science 33: 43–64.

Daubenmire, R. 1978. Plant geography: with special reference to North America. New York: Academic Press.

Daubenmire, R.; Daubenmire, J. B. 1968. Forest vegetation of eastern Washington and northern Idaho. Tech. Bull. 60. Pullman, WA: Washington State University and Washington Agricultural Experiment Station.

Dunham, A. E.; Grant, B. W.; Overall, K. L. 1989. Interfaces between biophysical and physiological ecology and the population ecology of terrestrial vertebrate ectotherms. Physiological Zoology 62: 335–355.

Ewen, A. B.; Mukerji, M. K. 1979. Susceptibility of five species of Saskatchewan grasshoppers to field applications of *Nosema locustae* (Microsporida). Canadian Entomologist 111: 973–974.

Fielding D. J.; Brusven, M. A. 1993a. Spatial analysis of grasshopper density and ecological disturbance on southern Idaho rangeland. Agricultural Ecosystems and Environment 43: 31–47.

Fielding D. J.; Brusven, M. A. 1993b. Grasshopper (Orthoptera: Acrididae) community composition and ecological disturbance on southern Idaho rangeland. Environmental Entomology 22: 71–81.

Gillis, J. E. 1982. Substrate color matching cues in the cryptic grasshopper, *Circotettix rabula* (Rehn and Hebard). Animal Behavior 30: 113–116.

Grant, B. W.; Porter, W. P. 1992. Modeling global macroclimatic constraints on ecotherm energy budgets. American Zoologist 32: 154–178.

Harvey, S.; Weaver, T. 1976. The avifauna of six Montana vegetation types. Proceedings of the Montana Academy Sciences 38: 36–42.

Henry, J. E. 1971. Experimental application of *Nosema locustae* for control of grasshoppers. Journal of Invertebrate Pathology 18: 389–394.

Hewitt, G. B. 1977. Review of forage losses caused by rangeland grasshoppers. Misc. Pub. 1348. Washington, DC: U.S. Department of Agriculture, Agricultural Research Service. 22 p.

Hewitt, G. B.; Onsager, J. A. 1982. Grasshoppers: yesterday, today, and forever. Rangelands 4: 207–209.

Hewitt, G. B.; Onsager, J. A. 1983. Control of grasshoppers on rangeland in the United States—a perspective. Journal of Range Management 36: 202–207.

Hilbert, D. W.; Logan, J. A. 1983. A population system model of the migratory grasshopper *(Melanoplus sanguinipes)*. In: Lauenroth, W. K.; Skogerbee, G. V.; Flug, M., eds. Analyses of ecological systems: state of the art in ecological modeling. Amsterdam: Elsevier Scientific Publishing: 323–334.

Isely, F. B. 1937. Seasonal succession, soil relations, numbers, and regional distribution of northeastern Texas acridians. Ecological Monographs 7: 318–344.

Joern, A. 1979. Resource utilization and community structure in assemblages of arid grassland grasshoppers (Orthoptera: Acrididae). Transactions of the American Entomological Society 105: 253–300.

Joern, A. 1981a. Guild structure in grasshopper assemblages based on food and microhabitat resources. Oikos 37: 93–104.

Joern, A. 1981b. Importance of behavior and coloration in the control of body temperature by *Brachystola magna* Girard (Orthoptera: Acrididae). Acrida 10: 117–130.

Joern, A. 1982. Vegetation structure and microhabitat selection in grasshoppers (Orthoptera: Acrididae). Southwestern Naturalist 27: 197–209.

Joern, A. 1983. Host plant utilization by grasshoppers (Orthoptera: Acrididae) from a sandhills prairie. Journal of Range Management 36: 793–797.

Joern, A. 1986. Resource partitioning by grasshopper species from grassland communities. In: Proceedings of Pan American Acridological Society 4th triennial meeting; 29 July–August 2 1985, Saskatoon, SK. Detroit, MI: Pan American Acridological Society: 75–100.

Joern, A. 1987. Behavioral responses underlying ecological patterns of resource use in rangeland grasshoppers. In: Capinera, J. L., ed. Integrated pest management on rangeland: a shortgrass perspective. Boulder, CO: Westview Press: 137–161.

Joern, A. 1988. Foraging behavior and switching by the grasshopper sparrow *Ammodramus savannarum* searching for multiple prey in a heterogeneous environment. American Midland Naturalist 119: 225–234.

Kemp, W. P. 1986. Thermoregulation in three rangeland grasshoppers. Canadian Entomologist 118: 335–343.

Kemp, W. P.; Dennis, B. 1989. Development of two rangeland grasshoppers at constant temperatures: developmental thresholds revisited. Canadian Entomologist 121: 363–371.

Kemp, W. P.; Onsager, J. A. 1986. Rangeland grasshoppers (Orthoptera: Acrididae): modeling phenology of natural populations of six species. Environmental Entomology 15: 924–930.

Kemp, W. P.; Sanchez, N. E. 1987. Differences in post-diapause thermal requirements for eggs of two rangeland grasshoppers. Canadian Entomologist 119: 653–661.

Kemp, W. P.; Harvey, S. J.; O'Neill, K. M. 1990a. Patterns of vegetation and grasshopper community composition. Oecologia 83: 299–308.

Kemp, W. P.; Harvey, S. J.; O'Neill, K. M. 1990b. Habitat and insect biology revisited: the search for patterns. American Entomologist 36: 44–48.

Kingsolver, J. G. 1989. Weather and the population dynamics of insects: integrating physiology and population ecology. Physiological Zoology 62: 314–334.

Mueggler, W. F.; Stewart, W. L. 1980. Grassland and shrubland habitat types of western Montana. Gen. Tech. Rep. INT-66. Ogden, UT: U.S. Department of Agriculture, Forest Service, Intermountain Forest and Range Experiment Station. 154 p. with map.

Mulkern, G. B. 1969. Behavioral influences on food selection in grasshoppers (Orthoptera: Acrididae). Entomologia Experientia. Applicata 12: 509–523.

Onsager, J. A., ed. 1987. Integrated pest management on rangeland: state of the art in the sagebrush ecosystem. ARS-50. Washington, DC: U.S. Department of Agriculture, Agricultural Research Service.

Onsager, J. A.; Henry, J. E.; Foster, R. N.; Staten, R. T. 1980. Acceptance of wheat bran bait by species of rangeland grasshoppers. Journal of Economic Entomology 73: 548–551.

Otte, D. 1976. Species richness patterns of New World desert grasshoppers in relation to plant diversity. Journal of Biogeography 3: 197–209.

Otte, D.; Joern, A. 1977. On feeding patterns in desert grasshoppers and the evolution of specialized diets. Transactions of the Philadelphia Academy of Sciences 128: 89–126.

Pfister, R. D.; Kovalchik, B. L.; Arno, S. F.; Presley, R. C. 1977. Forest habitat types of Montana. Gen. Tech. Rep. INT-34. Ogden, UT: U.S. Department of Agriculture, Forest Service, Intermountain Forest and Range Experiment Station. 174 p.

Quinn, M. A.; Kepner, R. L.; Walgenbach, D. D.; Bohls, R. A.; Pooler, P. D.; Foster, R. N.; Reuter, K. C.; Swain, J. L. 1989. Immediate and 2nd-year effects of insecticide spray and bait treatments on populations of rangeland grasshoppers. Canadian Entomologist 121: 589–602.

Russell, E. P. 1989. Enemies hypothesis: a review of the effect of vegetational diversity on predatory insects and parasitoids. Environmental Entomologist 18: 590–599.

Scoggan, A. C.; Brusven, M. A. 1973. Grasshopper–plant community associations in Idaho in relation to the natural and altered environment. Melanderia 12: 22–33.

U.S. Department of Agriculture, Forest Service. 1972. The nation's resources—a forest–range environmental study. Rep. 19. Washington, DC: U.S. Department of Agriculture, Forest Service, Forest Range Task Force. 147 p.

Vestal, A. G. 1913. Local distribution of grasshoppers in relation to plant associations. Biological Bulletin 25: 141–180.

Yang, Y.; Joern, A. 1994a. Compensatory feeding in response to variable food quality by *Melanoplus differentialis*. Physiological Entomology 19: 75–82.

Yang, Y.; Joern, A. 1994b. Influence of diet, developmental stage and temperature on food residence time in *Melanoplus differentialis*. Physiological Zoology 67: 598–616.

Williams, L. H. 1954. The feeding habits and food preferences of Acrididae and the factors that determine them. Transactions of the Royal Entomological Society, London 105: 423–454.

IV.4 Host Plant Quality and Grasshopper Populations

Anthony Joern

Understanding how grasshopper populations respond to food availability and quality may contribute critical components to models predicting outbreaks. In this chapter, I examine the relationships between demographic features of grasshopper population biology (growth rate, developmental rate, survival, and reproductive output) in the context of host plant quality. Because these relationships can be readily modeled and easily monitored under field conditions, models developed to forecast grasshopper outbreaks could incorporate this information for better accuracy (see chapters IV.1 and VI.2).

Like all range herbivores, grasshoppers require a diet that provides adequate protein, energy, and water plus trace nutrients and minerals. Sometimes, requirements include unique needs, such as a specific amino acid or sterol to complete development or fuel a specific biochemical pathway. After paying the cost to acquire and process food input, grasshoppers then allocate remaining nutrients to fuel physiological and biochemical processes. This allocation process determines developmental rate, growth, survival, and reproductive output. Host plant quality varies seasonally, among years and among habitats. Toxic substances in plants may hinder nutrient acquisition by either slowing feeding rate, reducing digestibility, requiring detoxification, or otherwise making the diet suboptimal. Each of these effects reduces the availability of nutrients for other grasshopper needs. Investigators need to understand how variable plant nutritional quality affects central features of grasshopper biology and population dynamics. Managers must assess range quality for grasshoppers in addition to standard measures applied to the effects of livestock, wildlife, or other range activities. Information on plant quality for grasshoppers can then be used to forecast population changes.

A grasshopper does not typically encounter optimal food items in a normal day's foraging. To obtain needed nutrients, an individual grasshopper may sample a variety of leaves from a few to many plant species that vary in levels of each critical nutrient category (see IV.7). After grasshoppers locate and consume the best possible diet, how does that diet drive population dynamics of a particular species? Do different grasshopper species respond to nutrient availability in the same fashion? In this chapter, I also describe basic grasshopper responses to diets of different quality in order to provide a framework for assessing grasslands from the grasshopper's perspective. So, from a manager's perspective, a good sense of available food quality and quantity will provide some useful "rules of thumb" for assessing potential problems. What features can be factored into these decisions? Such insights will contribute to forecasting capabilities (see VI.2 and VII.14).

A General Framework to the Problem

Range grasshopper populations, as with many insect herbivores, often fluctuate in response to variable plant quality. As suggested in several comprehensive reviews (White 1978, 1984, 1993; Mattson and Haack 1987; Joern and Gaines 1990; Jones and Coleman 1991), nutrients often limit grasshopper populations, and any environmental condition that increases plant quality will increase population growth in insect herbivores. Environmental stress routinely causes plant quality to shift as plants respond to drought, temperature, nutrient availability, or tissue loss to feeding (herbivory) (Mattson and Haack 1987, Jones and Coleman 1991). Natural environments seemingly fluctuate as a matter of course and multiple stresses capable of altering plant quality abound (see IV.5).

Following initial arguments of White (1978, 1984), the link between plant quality and climatic variation may explain many of the statistical links between climatic variation and variability in grasshopper densities. Moderately stressed host plants exhibit increased plant quality in two ways: food quality goes up, and there is also an increase in the quantity of high-quality leaf material relative to grasshopper population densities. These two improvements in host material contribute to increased grasshopper densities. By explicitly including density dependence, I am extending White's framework.

Variation in plant quality results from many sources. Available soil nutrients and environmental stress (drought, for example) can significantly change plant quality (Levitt 1972, Mooney et al. 1991). Stress (broadly defined) can result in increased total-N (protein) (Mattson and Haack 1987), increased total soluble protein and free amino acids (Wisiol 1979, White 1984), or altered levels of energy-containing compounds, such as total nonstructural carbohydrates (TNC) or free sugars like sucrose (Levitt 1972). Herbivore feeding can alter subsequent plant quality by forcing reallocation of mineral and energy resources within the plant (Coley et al. 1985, Bazzaz et al. 1987, Chapin et al. 1987, Mooney et al. 1991). Variable plant quality resulting from these combined effects significantly influences insect herbivore populations: As plant quality increases, insect populations increase (Mattson and Haack 1987, Berryman 1987, Joern and Gaines 1990, Jones and Coleman 1990). Growth, developmental rates, survival, and reproduction rates, or some combination of these demographic forces, vary according to these shifts in plant quality.

How Variable Is Plant Quality in Nature?

Range plants routinely undergo significant stresses from many sources, especially drought and herbivory (grazing animals). These stresses ultimately alter the nutritional plant quality available to grasshoppers. Thus, grasshoppers experience a wide range of “nutritional environments” within and among years. Many readily measured attributes contribute to food quality variation—plant species-specific differences, plant growth stage, or environmental conditions (especially water and nutrient availability, which affect physiological function). Similarly, different grasshopper species or developmental stages for a particular species often exhibit variable nutritional needs. Care is required when directly specifying quality based on simple plant chemical measures. However, direct measures of key plant chemical classes provide an unambiguous baseline for comparison.

Knowledge of nutritional requirements for dominant species at a site simplifies monitoring changes in plant quality to predict possible grasshopper responses. My examples will illustrate the main responses that can be expected for dominant nutritional classes. From a land manager’s perspective, an estimate of shifts in plant quality may help when assessing range condition and how that condition is changing from the standpoint of feeding by both grasshoppers and cattle. Low-cost chemical assays exist to help managers assess plant quality on rangeland.

Total Nitrogen.—The amount of total nitrogen (g N per g dry green plant material) indicates protein availability: percent protein $\sim 6 \times$ (percent of total N). Total N varies significantly among plant species, seasonally and among years at a given site, while important differences are often observed among sites in the same year and season. Forbs typically exhibit higher total N levels than grasses, all else being equal. However, forbs also include many secondary compounds that may act as feeding deterrents or toxins. As a rule of thumb, 1 percent total N becomes a lower limit to support grasshopper growth and development satisfactorily, although notable exceptions exist (such as *Phoetaliotes nebrascensis).* After starting at high levels (≥ 4–5 percent total N) when growth just begins in spring, total N concentrations often drop to about 1 percent (or lower) in late July or early August for northern grasslands. A moderate rebound typically occurs in early September. However, in some years, when conditions are particularly favorable, total N may never drop to 1 percent. Also, certain plants may exhibit high N levels, and others, low concentrations. A grasshopper faces such variation as it searches for good food.

Total Nonstructural Carbohydrates (TNC).—These compounds represent the immediate products of recent photosynthesis and show a more irregular seasonal pattern than that observed for nitrogen. TNC represent an immediate energy source for grasshoppers. While carbohydrates affect grasshopper growth, the availability of proteins tends to be more significant in limiting it.

Total Free Sugars and Total Free Amino Acids.—These nutritional components change in ways similar to total N and TNC, respectively, and may be important as feeding cues as well as nutrients. Both can vary with environmental stress (see IV.5 and IV.7). The amino acid proline provides a good example. Proline can either provide a good source of amino acids or can be metabolized as an energy source. It often increases in plants under drought stress, presumably to aid plant osmoregulation (maintain water balance) (Wisiol 1979, Behmer and Joern 1994). Along with the common free sugar sucrose, proline significantly stimulates feeding in some grass-feeding grasshoppers during phases of their life cycle when nutritional resources are limited.

How Does Altered Host Plant Quality Affect Feeding?

Feeding includes searching for acceptable food, selecting foods from among several choices, and then digesting the food. The grasshopper actively controls each of these phases in the feeding cycle (for more details see IV.7).

Food intake provides resources for all subsequent physiological processes. In general, higher quality food leads to larger individual meals but lowered overall time spent feeding, increased time in the gut, and increased digestibility. Each individual grasshopper requires less total food when feeding on higher quality tissue, and high-quality plants lose less total tissue per grasshopper. However, individual plants vary in quality. Overall grasshopper feeding becomes context dependent. For example, a poor-quality host plant by itself may lose much leaf mass to support a grasshopper (it takes more tissue to provide adequate nutrients) but will not be fed upon as much when it grows alongside high-quality plants. Thus, potential loss to an individual plant shifts depending on the alternate plants available to the grasshopper.

Accumulating evidence suggests that most grasshoppers mix food to balance diets. Some species select from a great number of host plants. Grasshoppers that feed on multiple host plants often exhibit higher survival and fecundity (reproductive ability) than those fed single food plants. *Melanoplus sanguinipes*, for example, does not do nearly as well when fed either grass or forbs alone as when fed both grasses and forbs. In experiments with other grass-feeding grasshopper species, *M. sanguinipes* often surpasses other species in food gathering when grasses and forbs are present but loses if forbs are absent (Chase and Belovsky 1994). In a similar vein, some grasshoppers often mix turgid with wilted tissue of the same food plant, typically resulting in increased fecundity (Lewis 1984).

It appears that few host plants provide a completely balanced diet for most grasshopper species and that grasshoppers can adjust behaviorally to optimize diets (Simpson and Simpson 1990). Very few species exist that are truly specialists and feed on a single host plant species. If we can learn what is required for balanced diets by economically important grasshopper species and remove that balance, then we may be able to manipulate plant communities to decrease grasshopper populations. In the case of *M. sanguinipes*, controlling densities of preferred forbs may prove important, both to alter individual growth and reproduction as well as to shift the competitive balance with other species.

How Does Plant Quality Affect Key Demographic Attributes?

Key demographic parameters, such as survival, fecundity, developmental rate, and growth, significantly respond to changes in plant quality. Poor-quality food results in poor demographic performance and vice versa (Bernays et al. 1974). Total food availability directly affects these factors (Mulkern 1967, Mattson and Haack 1987, Joern and Gaines 1990). From a grasshopper's viewpoint, plant quality surely includes both nutritional and defensive properties of the host plant.

Evidence indicates that different species of host plants influence fecundity (Pfadt 1949; Pickford 1958, 1962, 1966). For example, *Camnula pellucida* performed poorly (developmental rate, nymphal and adult survival, and fecundity) when fed native vegetation in Canada compared to spring and summer wheat (Pickford 1962). Egg production makes significant demands on the grasshopper's nutritional economy and depends significantly on protein and energy obtained from the diet. Nutrient stores cannot supply the reproductive process for long. *M. sanguinipes* laid few eggs when fed wheat seedlings low in nitrogen (Krishna and Thorsteinson 1972). Similarly, when *Locusta migratoria* females fed on low-protein diets, egg production dropped and terminal eggs were resorbed (McCaffery 1975). Similar results have been observed for other species. In addition, extreme drought often results in a decrease in the food's quality and quantity, decreasing reproduction in a number of grasshopper species. Such results become important for understanding grasshopper population dynamics, as reproductive changes can drive population change.

Fecundity in common range grasshoppers varies in response to both protein and carbohydrates. While lifespan has some effect on fecundity and is also dependent on food quality, total N significantly affects reproductive output. Dramatic species differences exist. While these different patterns are yet unexplained, they should alert managers to the potential problem of generalizing results from a small set of species to all grasshopper species.

Grasshopper survivorship is sensitive to food plant quality. As with fecundity, species-specific survivorship varies according to host plant eaten (Pickford 1962, Mulkern 1967, Bailey and Mukerji 1976, Joern and Gaines 1990). For example, *A. deorum* lives longest in experiments with highest N-levels in the leaves of its primary food plant. To emphasize the importance of species-specific differences, *P. nebrascensis* exhibits the opposite response to plant quality as seen in *A. deorum.* Furthermore, in a third species, *M. sanguinipes*, total N only minimally affects survival. But *M. sanguinipes* requires a mixture of grasses and forbs, indicating that a varied diet is important for this species.

How Does Plant Quality Affect Spatial Distribution of Grasshoppers?

While grasshopper integrated pest management (IPM) is primarily concerned with overall densities, the distribution of grasshoppers in time and space offers important insights into grasshopper demographic responses. Often, individual patches of range reach very high grasshopper densities while most of the remaining range experiences low densities. It is not generally clear why these distributional patterns arise. Grasshoppers forage in a variable environment, with plant quality often changing over short distances. If some plant patches reach higher quality levels than others, local grasshopper densities may increase as individuals move into the patch and remain (Heidorn and Joern 1987). In typical rangeland situations, grasshoppers often move onto adjacent areas after haying, possibly in response to a significant removal of quality food material. However, because haying changes so many environmental features, reasons other than loss of available high-quality food may explain this movement.

Trap Strips as a Management Tool

It seems clear that any range management technique that increases plant quality in a patchy fashion may increase local grasshopper densities. By adding fertilizer to areas to enhance plant growth, land managers can expect increased grasshopper densities. While untested, a promising idea is to develop treatable trap strips by fertilizing sufficiently large patches to reduce overall densities elsewhere. If trap strips remained ungrazed, they would also provide superb nesting habitat for grassland birds and thereby further support control. Optimal spacing and size for these strips is not known, nor is the year-to-year dynamics of grasshopper populations on or near these proposed strips. For example, will grasshoppers lay more eggs leading to greater buildups? Will hot spots develop from such treatments? Will increased grasshopper density greatly reduce food on these trap strips, leading to movement away from the trap? Or will density-dependent mortality kick in and greatly reduce the infestation? Will bird predators seek out these high-density patches and greatly reduce numbers? While each of these issues hold promise or concern for grasshopper IPM, insufficient data currently exist to predict responses accurately. I feel, however, that clever managers will find ways to incorporate these approaches using trial-and-error techniques coupled with accurate records and thoughtful interpretations. While such manipulations have been poorly studied, I believe that they hold great practical promise for developing innovative grasshopper IPM programs.

Final Comments

My major take-home message in this section concerns how alteration of plant quality can affect grasshopper population processes. In quick summary, most host plants that are routinely consumed by grasshoppers vary significantly in nutritional quality, over any time or space scale that may interest land managers. Often, host plant quality responds directly to stresses induced by climatic variation. Moderate amounts of environmentally induced stress typically increase the quality of grasshopper food, especially with regard to protein.

In response to changing host plant quality, grasshoppers alter feeding patterns as well as allocation of assimilated food. All key demographic variables respond to altered plant quality, although managers must remember that all grasshopper species do not respond in the same fashion. Grasshopper IPM programs must be pegged to the amount of forage eaten by individual grasshoppers, the significance of these losses, and the number of grasshoppers that are eating relative to available forage. Grasshopper population processes become important only in the context of long-term issues: those programs that keep grasshopper populations at low levels will incur less forage loss over the long term. But the interaction is two sided and dynamic: variability in both host plant quality and grasshopper demographic responses interact to drive forage loss.

References Cited

Bailey, C. G.; Mukerji, M. K. 1976. Consumption and utilization of various food plants by *Melanoplus bivittatus* and *Melanoplus femurrubrum*. Canadian Journal of Zoology 54: 1044–1050.

Bazzaz, F. A.; Chiarello, N. R.; Coley, P. D.; Pitelka, L. F. 1987. Allocating resources to reproduction and defense. Bioscience 37: 58–67.

Behmer, S. T.; Joern, A. 1994. The influence of proline on diet selection: sex-specific feeding preferences by the grasshoppers *Ageneotettix deorum* and *Phoetaliotes nebrascensis* (Orthoptera: Acrididae). Oecologia 98(1): 76–82.

Bernays, E. A.; Chapman, R. F.; Horsey, J.; Leather, E. 1974. The inhibitory effect of seedling grasses on feeding and survival of acridids. Bulletin of Entomological Research 64: 413–420.

Berryman, A. A. 1987. The theory and classification of outbreaks. In: Barbosa, P.; Schultz, J. C., eds. Insect outbreaks. New York: Academic Press: 3–30.

Chapin, F. S. III; Bloom, A. J.; Field, C. B.; Waring, R. H. 1987. Plant responses to multiple environmental factors. Bioscience 37: 49–57.

Chase, J. M.; Belovsky, G. E. 1994. Experimental evidence for the included niche. American Naturalist 143: 514–527.

Coley, P. D.; Bryant, J. P.; Chapin, F. S. III. 1985. Resource availability and plant anti-herbivore defense. Science 230: 895–899.

Heidorn, T. J.; Joern, A. 1987. Feeding responses and spatial distribution of grasshoppers (Acrididae) in response to nitrogen fertilization of *Calamovilfa longifolia*. Functional Ecology 1: 369–375.

Joern, A.; Gaines, S. B. 1990. Population dynamics and regulation in grasshoppers. In: Chapman, R. F.; Joern, A., eds. Biology of grasshoppers. New York: John Wiley and Sons: 415–482.

Jones, C. G.; Coleman, J. S. 1991. Plant stress and insect herbivory: toward an integrated perspective. In: Mooney, H. A.; Winner, W. E.; Pell, E. J., eds. Response of plants to multiple stresses. New York: Academic Press: 249–280.

Krishna, S. S.; Thorsteinson, A. J. 1972. Ovarian development of *Melanoplus sanguinipes* (Fab.) (Acrididae: Orthoptera) in relation to utilization of water soluble food proteins. Canadian Journal of Zoology 50: 1319–1324.

Lewis, A. C. 1984. Plant quality and grasshopper feeding: effects of sunflower condition on preference and performance in *Melanoplus differentialis*. Ecology 65: 836–843.

Levitt, J. 1972. Responses of Plants to Environmental Stress. New York: Academic Press. 697 p.

Mattson, W. J., Jr.; Haack, R. A. 1987. The role of drought in outbreaks of plant-eating insects. Bioscience 37: 110–118.

McCaffery, A. R. 1975. Food quality and quantity in relation to egg production in *Locusta migratoria migratorioides*. Journal of Insect Physiology 21: 1551–1558.

Mooney, H. A.; Winner, W. E.; Pell, E. J., eds. 1991. Response of plants to multiple stresses. New York: Academic Press. 422 p.

Mulkern, G. B. 1967. Food selection by grasshoppers. Annual Review of Entomology 12: 59–78.

Pfadt, R. E. 1949. Food plants as factors in the ecology of the lesser migratory grasshopper *(Melanoplus mexicanus)*. Bull. 29. Laramie, WY: University of Wyoming and Wyoming Agricultural Experiment Station.

Pickford, R. 1958. Observations in the reproductive potential of *Melanoplus bilituratus* (Wlk.) (Orthoptera: Acrididae) reared on different food plants in the laboratory. Canadian Entomologist 90: 483–485.

Pickford, R. 1962. Development, survival and reproduction of *Melanoplus bilituratus* (Wlk.) (Orthoptera: Acrididae) reared on different food plants. Canadian Entomologist 94: 859–869.

Pickford, R. 1966. Development, survival and reproduction of *Camnula pellucida* (Scudder) (Orthoptera: Acrididae) in relation to climatic conditions. Canadian Entomologist 98: 158–169.

Simpson, S. J.; Simpson, C. L. 1990. Mechanisms of nutritional compensation by phytophagous insects. In: Bernays, E. A., ed. Plant–insect interactions. Vol. II. Boca Raton, FL: CRC press: 111–160.

White, T.C.R. 1978. The relative importance of shortage of food in animal ecology. Oecologia 33: 71–86.

White, T.C.R. 1984. The availability of invertebrate herbivores in relation to the availability of nitrogen in stressed food plants. Oecologia 63: 90–105.

White, T.C.R. 1993. The inadequate environment: nitrogen and the abundance of animals. Berlin: Springer-Verlag. 425 p.

Wisiol, K. 1979. Clipping of water-stressed blue grama affects proline accumulation and productivity. Journal of Range Management 32: 194–195.

IV.5 Environmental Factors That Affect Plant Quality

Anthony Joern

Variation in host plant quality arises from many sources. Environmental stress, primarily a response to varying soil nutrients, light, and water, affects physiological responses by plants in a species-specific manner. This variation provides a remarkable array of available plant quality to insect herbivores (Coley et al. 1985, Chapin et al. 1987, Mooney et al. 1991). In addition, herbivore feeding (both insect and mammalian) further alters the nutritional quality of leaf material, both soon after feeding and in the future. Why do these responses occur? Plants reallocate minerals and energy in response to stress, and the consequence is considerable variation over time in the foliar concentrations of primary nutrients. These nutrients include levels of nitrogen-containing compounds, such as protein, energy-containing compounds, such as nonstructural carbohydrates (including free sugars), or specific chemical constituents, such as individual amino acids. Clearly, grasshoppers seldom face a simple "nutritional environment" when searching for food to satisfy crucial needs.

As variable plant quality often influences grasshopper population dynamics, can range managers predict how plant quality varies in time and space? For managers charged with long-term planning, which sites typically exhibit higher host plant quality? Will stress explain observed spatial patterns in plant growth and foliar quality? Will identification of stressed areas help identify grasshopper problem areas? Answers to how grasshopper food resources vary in time and space will provide important insights to aid in both forecasting grasshopper population change and formulating appropriate management strategies.

In this chapter, I briefly outline how environmental stress affects plant response at several levels. Once plant responses are recognized, managers can more effectively incorporate these responses into strategic plans, including forecasting models and economic assessments.

Plants are integrated units, and plant stress cannot be evaluated except in that context. Photosynthesis (light and carbon dioxide [CO_2] capture), which occurs in leaves and to a lesser extent in stems, is coupled with nutrient and water uptake through roots to provide all essential raw materials for plant growth, development, and reproduction. As in animals, different plant tissues and organs contribute different functions, and a plant must balance the action of each to promote healthy, whole-plant function. Available resources fall short of the amount needed to facilitate all life activities, so plants are forced to allocate scarce resources (fig. IV.5–1). Significant tradeoffs exist because the plant cannot supply resources to all of its parts simultaneously, given the competition for resources in a limited environment. This scenario is the notion of "source–sink" relationships (Turgeon 1989). A *source* provides limited resources (roots provide the plant with nitrogen), and a *sink* gets first priority for use of limited resources (the leaf needs nitrogen for photosynthesis). Note the cyclical nature of the relationship. Some resources are obtained by the plant through absorption of nutrients through the roots, and energy-containing and structural compounds are produced by photosynthesis. The available nutrient pool obtained in this fashion is then allocated to those tissues housing the most critical metabolic activity at the time—the sink(s). As conditions change, new sinks develop, and the allocation patterns can be altered quickly.

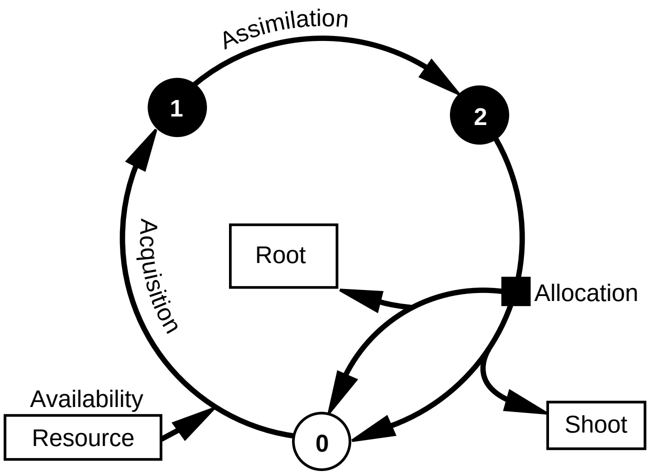


Figure IV.5–1—Simple, diagrammatic metabolic paths that indicate steps in obtaining and then allocating limiting resources among plant tissues. (Adapted from Sharpe and Rykiel 1991.)

Ultimately, these resource allocation “decisions” determine the fate of the whole plant in terms of survival, total biomass production, and long-term reproductive fitness. Because unlimited external resources seldom exist, plants cannot operate at maximal rates. The difference between optimal and actual rates of function defines the level of stress experienced by the plant (Mooney et al. 1991). As stress from such factors is imposed, it triggers a cascade of responses: the plant rebalances to the new conditions. Plants are exposed to a wide range of abiotic (weather) factors that directly reduce growth. These variable conditions include drought, flooding, mineral deficiencies or imbalances, temperature extremes, and air pollution (Jones and Coleman 1991). From the herbivore’s point of view, these cascading responses alter the nutritional quality and distribution in leaves.

As indicated in figure IV.5–2, Jones and Coleman (1991) provide an effective framework for quickly illustrating both the types of plant responses to environmental stress as well as anticipated herbivore reactions to altered plant quality. Herbivory (livestock and insects) feeding on plants cannot be strictly separated from other stresses because losses in leaf, root, or stem material stress plants as much as physical or chemical factors.

Plant Responses

Both internal and external features control plant responses. Internally, individual genotypic differences and phenological development can exert significant effects on plant use and allocation of resources. For example, the plant genotype limits rates of acquisition, sets priorities for partitioning among plant parts, modifies allocation to biochemical processes, and determines the magnitude of other related responses, such as the amount of defensive compound that can be produced (examples in Jones and Coleman 1991).

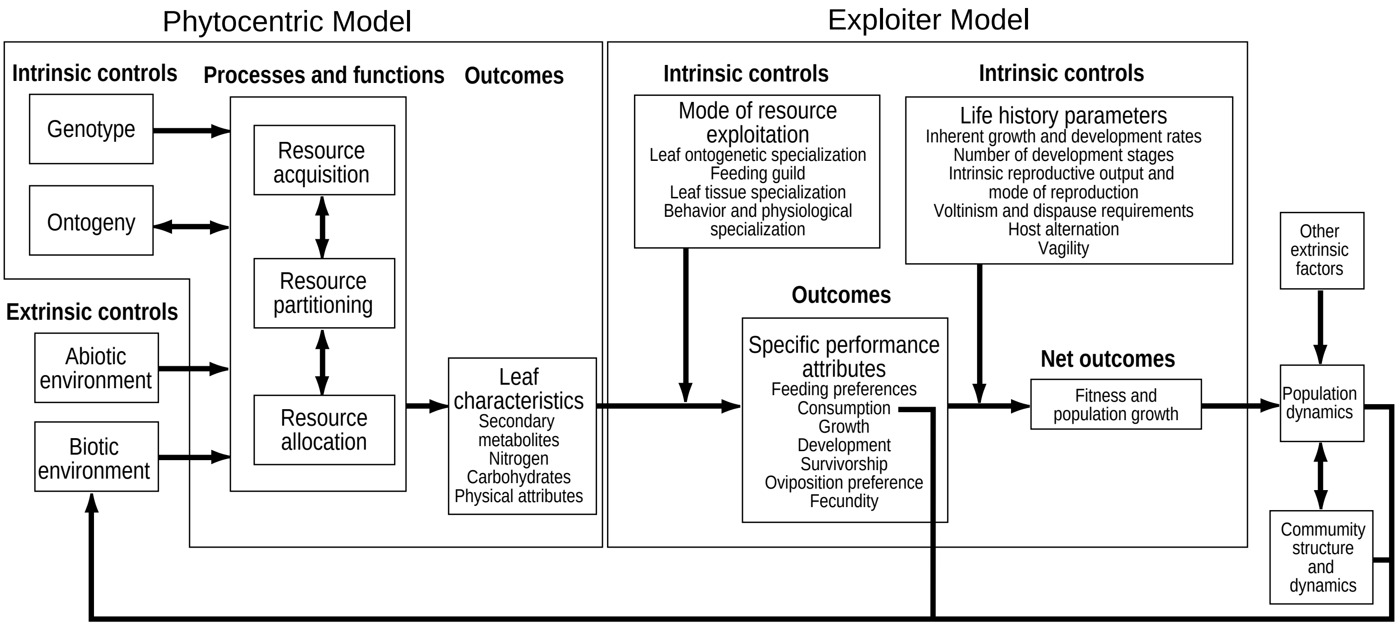


Figure IV.5–2—A conceptual framework of the linkages and feedbacks between plant allocation processes and herbivore consumers. (Adapted from Jones and Coleman 1991.)

Fast- versus slow-growing species typically exhibit very different patterns of nutrient allocation (Coley et al. 1985), largely due to the value of individual leaves. In fast growers, individual leaves are relatively less important than in slow-growing plants. Fast growers allocate a higher proportion of resources to growth and less to defense (chemicals and leaf structures that deter herbivory). The converse exists for slow-growing species. Seasonal plant growth modifies the capacity and demand for resources and sets partition and allocation priorities as plants grow and mature.

While intrinsic features clearly modify the strength of plant responses, external environmental features typically exert more influence on plant responses. Resources required by grasshoppers vary in both time and space; because some resources are limited, a plant is typically playing catchup. Within the limits imposed by genotype and phenological stage, plants attempt to obtain limited nutrients selectively. This process allows the plant to maintain a carbon-nutrient balance somewhere near the optimum for plant function. For example, plants limited by nutrients or water often allocate more resources to build root tissue to increase root surface area and increase nutrient absorption from the soil. As a result, leaf tissues receive fewer resources. The message here is that plants continuously respond to shifts in resource availability, resulting in significant changes in leaf quality.

In adjusting to variable resource availability, biochemical, anatomical, and physiological shifts also occur in the leaves. From an herbivore's point of view, both defensive secondary metabolites (described below) and nutritional features change. As stated earlier, the growth strategy of the plant (whether it is a fast or slow grower) dictates the response.

Secondary Metabolites.—Secondary metabolites in plants comprise a long list of compounds produced at various steps in the metabolic pathway that are not directly related to regulating photosynthesis or other primary metabolic pathways, such as respiration. So, while sucrose or enzymes are considered primary metabolites, a variety of chemical compounds such as alkaloids or phenolics are termed secondary. This term does not mean that these metabolites are unimportant for plant function or success—quite the contrary. According to Coley et al. (1985), fast-growing plant species under stress should exhibit extensive variability in secondary metabolite production because growth is a higher priority than the production of defensive compounds. Conversely, allocation to secondary defensive compounds becomes a high priority in slow growers because leaf tissue must continuously be defended, even under stress. Finally, plant life form correlates well to the presence of and nature of plant defenses. Secondary metabolite defenses are much more common in forbs than grasses (Mole and Joern 1993, *contra* Redak 1987).

A diversity of chemical compounds serves to defend plants. In some plants, the defensive chemical also routinely serves a number of functions, while in other cases a plant uses different chemicals under different stress conditions (Coley et al. 1985, Jones and Coleman 1991). In addition, different forms of stress (drought, pollution, or nutrient deficiency) result in a diversity of responses as plant allocations vary with the stress. For example, drought responses are particularly complex. They alter acquisition of both carbon and nutrients, they disrupt transport function, and they cause secondary metabolite concentration to vary because water concentration in leaves varies.

Nitrogen.—Among all of the leaf nutritional characteristics that significantly respond to environmental stress and influence grasshoppers, nitrogen content is one of the most important. Many environmental stresses induce the mobilization of nitrogen in plants. This mobilization results in increased levels of total nitrogen as well as specific amino acids and proteins (Stewart and Larher 1980, Rhodes 1987). Drought and nutrient stress typically result in increased carbon-to-nitrogen ratios, often accompanied by altered amino acid composition (Stewart and Larher 1980, McQuate and Connor 1990), as shown in table V.5–1. Similarly, increased plant water stress (too little or too much water) often results in altered free amino acid composition. Free amino acids such as proline often increase in grasses with moderate water stress (Barnett and Naylor 1966, Hsiao 1973, Wisiol 1979, Bokhari and Trent 1985, Zuniga and Corcuera 1987), possibly because proline acts as an osmoregulator (Stewart and Lee 1974) or as storage for nitrogen and carbon (Barnett and Naylor 1966). [An osmoregulator serves to help maintain water balance within the plant.]

Table IV.5–1—Amino acids exhibiting increased concentrations in soybean leaves in response to increasingly severe water deficits (adapted from McQuate and Connor 1990)

Plant water deficit	Amino acids exhibiting increase
0 to –0.5 MPa[1]	Isoleucine, leucine, lysine, phenylalanine, tryptophan
–0.5 to –1.0 MPa	Cystine, glutamine, histidine, threonine, tyrosine, valine
–1.0 to –1.5 MPa	Proline
–1.5 to –2.0 MPa	Arginine, asparagine, glycine

Note: Reduction of leaf water potential is the decrease observed in water-deficient plants compared to well-watered individuals. Glutamic acid, alanine, aspartic acid, and serine do not change concentration in response to water deficit.

[1] Presure units for plant water deficit are in megapascals (MPa).

Significant shifts in resource allocation often cause variation in many important nutritive chemicals. These chemicals include soluble nitrogen and free amino acids, nonstructural carbohydrates, and chemical defense molecules (Perry and Moser 1974, McKindrick et al. 1975, Chapin and Slack 1979, Mooney and Gulmon 1982, Bernays 1983). Total nonstructural carbohydrates (TNC) respond to environmental changes, such as grazing, temperature, water potential of soil and leaves, nutrient status of the soil, and maturity state of the plant (Ryle and Powell 1975, Bokhari 1978, Caldwell et al. 1981, Hayes 1985). Foliar carbon–nitrogen ratios can shift dramatically in response to grazing, water, and nitrogen fertilization (Bokhari 1978, Caldwell et al. 1981, Bryant et al. 1983, Mattson and Haack 1987).

Impact to Plant Quality From Biotic Sources

Interactions with herbivores, pathogens, and symbionts (organisms living in close association with the plant that confer a positive impact, such as nitrogen-fixing bacteria in root nodules of many legumes) often significantly influence allocation schedules in plants, thus altering plant quality. In most North American grasslands, plants experience extraordinary pressure from cattle or sheep grazing, which severely reduces above- and belowground biomass. Thus, many range plants routinely suffer moderate to extreme stress from leaf loss from mammalian herbivores in addition to leaf losses from grasshoppers. In these cases, ecological interactions take place above versus below ground, mediated through the plant by changing allocation schedules.

Often, loss of either above- or belowground tissue alters the commitment to the other. For example, loss of leaf material from herbivores above ground results in reduced root mass. Root-grazing by a variety of nematodes and insect larvae leads to lower leaf mass above ground (Geiger and Servaites 1991, Mooney and Winner 1991). The soil surface effectively partitions the grazing system into these two components. Plants mediate interactions between aboveground versus belowground herbivores because herbivory in one compartment changes overall plant quality, often increasing herbivore load in the other compartment (Seastedt 1985, Seastedt et al. 1988). To range managers, management of plant loss in both compartments becomes critical because grazing pressure above ground can increase root quality to belowground feeders and thereby increase feeding on those tissues. Such complex responses further decreases the chance that plants will recover quickly from moderate to heavy grazing.

Such biotic interactions between plants and their herbivores are numerous. Some examples include mychorrhizal fungal or nitrogen-fixing bacterial associations with the roots, both of which increase nutrient acquisition rates by plants (Powell and Bagyaraj 1984, Arora 1991). Conversely, organisms causing plant diseases often lower rates of photosynthesis, respiration, and nutrient uptake as well as shift allocation schedules between roots and leaves, as do root-feeding nematodes (Ingham and Detling 1984).

In spite of significant grazing pressure, some plant species cope readily while others do not. Plants of different life forms (grasses versus forbs) typically differ in their tolerance to foliage loss. Plant life form influences regrowth characteristics based on the protection or redundancy of primary growth tissue or the possession of such tissues that are typically missed by herbivores (Dahl and Hyder 1977). In grasses, the primary growing tissue is often found at the soil surface, below the level normally grazed by herbivores. In this sense, it is protected. Other adjustments that plants make to grazing include higher photosynthetic rates, reduced foliage longevity, low proportion of reproductive shoots, and faster rates of leaf replacement (Caldwell et al. 1981, Archer and Tieszen 1983). Species with the same life form (grasses), however, often can exhibit striking differences in response to herbivory.

How do grasses cope with herbivory? Caldwell et al. (1981) assessed physiological responses by two *Agropyron* bunchgrass species *(A. desertorum* and *A. spicatum)* that evolved with and without significant likelihood of herbivory. These grasses exhibited significant differences in tolerance to grazing, *A. desertorum* being more tolerant. Otherwise, these species exhibit similar growth timing and thus experience the same physical and climatic environment. Following grazing, *A. desertorum* rapidly established a new canopy with three to five times the photosynthetic surface than *A. spicatum* with the same available resources. *A. desertorum* exhibited a lower investment of nitrogen and biomass per unit of photosynthetic area, more tillers, more leaves per bunch, and shorter stems. In addition, this species exhibited greater flexibility of resource allocation following grazing by reallocating more resources to shoot growth at the expense of root growth. This process quickly achieved preclipping root–shoot balance. Nitrogen required for regrowth came from uptake rather than reserve depletion. Carbohydrate pools in the shoot system of both species remained low following severe defoliation. Interestingly, when competing plant species were removed, even the poorly coping *A. spicatum* could tolerate extreme defoliation (Mueggler 1972).

Final Comment

Take-home messages from these examples reinforce the major point of this section: resource allocation schedules for limited nutrients in plants largely dictate responses by mediating source–sink relationships. Consequently, any abiotic or biotic factor that alters these relationships will change the allocation schedules, resulting in an altered nutritional environment for both mammalian grazers and insect herbivores, such as grasshoppers. An understanding of the general framework underlying source–sink and within-plant allocation provides the insight for anticipating favorable versus unfavorable conditions for both plants and herbivores. After all, range resource managers are really managing the vegetation resource, not the consumers per se. Such a realization will undoubtedly alter the way that humans devise strategies to manage grasshopper control programs.

References Cited

Archer, S.; Tieszen, L. 1983. Effects of simulated grazing on foliage and root production and biomass allocation in an arctic tundra sedge *(Eriophorum vaginatum)*. Oecologia 58: 920–102.

Arora, D. K., ed. 1991. Soil and plants. New York: Marcel Dekker. 720 p.

Barnett, N. M.; Naylor, A. W. 1966. Amino acid and protein metabolism in Bermuda grass during water stress. Plant Physiology 41: 122–123.

Bernays, E. A. 1983. Nitrogen in defence against insects. In: Lee, J. A.; McNeill, S.; Rorison, I. H., eds. Nitrogen as an ecological factor. Oxford, UK: Blackwell Scientific Press: 321–344.

Bokhari, U. G. 1978. Nutritional characteristics of blue grama herbage under the influence of added water and nitrogen. Journal of Range Management 31: 18–22.

Bokhari, U. G.; Trent, J. D. 1985. Proline concentrations in water stressed grasses. Journal of Range Management 38: 37–38.

Bryant, J. P.; Chapin, F. S. III; Klein, D. R. 1983. Carbon–nutrient balance of boreal plants in relation to vertebrate herbivory. Oikos 40: 357–368.

Caldwell, M. M.; Richards, J. H.; Johnson, D. A.; Nowak, R. S.; Dzrec, R. S. 1981. Coping with herbivory: photosynthetic capacity and resource allocation in two semiarid *Agropyron* bunchgrasses. Oecologia 72: 14–24.

Chapin, F. S.; Slack, M. 1979. Effect of defoliation upon root growth, phosphate absorption and respiration in nutrient limited tundra graminoids. Oecologia 42: 67–79.

Chapin, F. S., III; Bloom, A. J.; Field, C. B.; Waring, R. H. 1987. Plant responses to multiple environmental factors. Bioscience 37: 49–57.

Coley, P. D.; Bryant, J. P.; Chapin, F. S., III. 1985. Resource availability and plant antiherbivore defense. Science 230: 895–899.

Dahl, B.; Hyder, D. 1977. Developmental morphology and management implications. In: Sosebee, R., ed. Rangeland plant physiology. Denver, CO: Society for Range Management: 257–290.

Geiger, D. R.; Servaites, J. C. 1991. Carbon allocation and response to stress. In: Mooney, H. A.; Winner, W. E.; Pell, E. J., eds. Response of plants to multiple stresses. New York City: Academic Press: 103–127.

Hayes, D. C. 1985. Seasonal nitrogen translocation in big bluestem during drought conditions. Journal of Range Management 38: 406–410.

Hsiao, T. C. 1973. Plant responses to water stress. Annual Review of Plant Physiology 24: 519–570.

Ingham, R. E.; Detling, J. K. 1984. Plant–herbivore interactions in North American mixed grass prairie. III. Soil nematode populations and root biomass on *Cyonomys ludovicianus* colonies and adjacent uncolonized areas. Oecologia 63: 307–313.

Jones, C. G.; Coleman, J. S. 1991. Plant stress and insect herbivory: toward an integrated perspective. In: Mooney, H. A.; Winner, W. E.; Pell, E. J., eds. Response of plants to multiple stresses. New York City: Academic Press: 249–280.

Mattson, W. J., Jr.; Haack, R. A. 1987. The role of drought in the outbreaks of plant-eating insects. Bioscience 37: 110–118.

McKindrick, J. D.; Owensby, C. E.; Hyde, R. M. 1975. Big bluestem and indiangrass vegetative reproduction and annual reserve carbohydrate and nitrogen cycles. AgroEcosystems 2: 75–93.

McQuate, G. T.; Conner, E. F. 1990. Insect responses to plant water deficits: II. Effects of water deficits in soybean plants on the feeding preference of Mexican bean beetle larvae. Ecological Entomology 15: 433–445.

Mole, S.; Joern, A. 1993. The foliar phenolics of Nebraska sandhills prairie graminoids: between years, seasonal and interspecific variation. Journal of Chemical Ecology 19: 1861–1874.

Mooney, H. A.; Gulmon, S. L. 1982. Constraints on leaf structure and function in reference to herbivory. Bioscience 32: 198–206.

Mooney, H. A.; Winner, W. E. 1991. Partitioning response of plant stress. In: Mooney, H. A.; Winner, W. E.; Pell, E. J., eds. Response of plants to multiple stresses. New York City: Academic Press: 129–141.

Mooney, H. A.; Winner, W. E.; Pell, E. J., eds. 1991. Response of plants to multiple stresses. New York: Academic Press. 422 p.

Mueggler, W. F. 1972. Influence of competition on the response of bunchgrass wheatgrasses to clipping. Journal of Range Management 25: 88–92.

Perry, L. J.; Moser, L. E. 1974. Carbohydrate and organic nitrogen concentrations within range grass parts at maturity. Journal of Range Management 27: 276–278.

Powell, C. L.; Bagyaraj, D. J., eds. 1984. VA mycorrhiza. Boca Raton, FL: CRC Press. 234 p.

Redak, R. 1987. Forage quality: secondary chemistry of grasses. In: Capinera, J. L., ed. Integrated pest management on rangeland: a shortgrass perspective. Boulder, CO: Westview Press: 38–55.

Rhodes, D. 1987. Metabolic responses to stress. In: Davies, D. D., ed. The biochemistry of plants. London: Academic Press: 201–241.

Ryle, G.J.A.; Powell, C. E. 1975. Defoliation and regrowth in the graminaceous plant: the role of current assimilate. Annals of Botany 39: 297–310.

Seastedt, T. R. 1985. Maximization of primary and secondary productivity by grazers. American Naturalist 126: 559–564.

Seastedt, T. R.; Ramondo, R. A.; Hayes, D. C. 1988. Maximization of densities of soil animals by foliage herbivory: empirical evidence, graphical and conceptual models. Oikos 51: 243–248.

Sharpe, P. J.; Rykiel, E. J., Jr. 1991. Modeling integrated responses of plants to multiple stresses. In: Mooney, H. A.; Winner, W. E.; Pell, E. J., eds. Response of plants to multiple stresses. New York City: Academic Press: 205–224.

Stewart, G. R.; Larher, F. 1980. Accumulation of amino acids and related compounds in relation to environmental stress. In: Miflin, B. J., ed. The biochemistry of plants. Vol. 5. Amino acids and derivatives. London: Academic Press: 609–635.

Stewart, C. R.; Lee, J. A. 1974. The role of proline accumulation in halophytes. Planta 120: 279–289.

Turgeon, R. 1989. The sink–source transition in leaves. Annual Review Plant Physiology and Plant Molecular Biology 40: 119–138.

Wisiol, K. 1979. Clipping of water stressed blue-grama affects proline accumulation and productivity. Journal of Range Management 37: 28–30.

Zuniga, G. E.; Corcuera, L. J. 1987. Glycine–betaine accumulation influences susceptibility of water-stressed barley to the aphid *Schizaphis graminum*. Phytochemistry 26: 367–369.

IV.6 *Melanoplus sanguinipes* Phenology North–South Across the Western United States

J. R. Fisher, W. P. Kemp, and J. S. Berry

Distribution and abundance of an insect species are affected by its habitat requirements, such as food and/or climatic resources. As requirements become more specific, distribution and abundance become more limited. For instance, *Melanoplus bowditchi*, a grasshopper found in many Western States, is limited to the range of its primary host plants, silver sagebrush and sand sagebrush (Pfadt 1994). In fact, the relative abundance of these plants will determine if you can even find *M. bowditchi*. Distribution of the bigheaded grasshopper, *Aulocara elliotti*, appears to be limited by climatic conditions. It feeds mainly on grasses and sedges but is restricted to States west of longitude 95° W, where it is particularly abundant in the more arid areas (Pfadt 1994). But *M. femurrubrum*, a general feeder (polyphagous), is distributed throughout North America from coast to coast and from northern British Columbia to northern Guatemala (Pfadt 1994).

Melanoplus sanguinipes, the lesser migratory grasshopper, is polyphagous and distributed in North America from Alaska to Mexico and from coast to coast (Pfadt 1994). It is the most economically important species on Western U.S. rangeland and was partially responsible for the grasshopper “plagues” of the 1930’s. Given the territory covered by *M. sanguinipes*, it appears that this species has a remarkable ability to adapt to a multitude of environmental and climatic conditions.

Egg Development and Hatch

All North American grasshoppers of economic importance lay eggs in the soil in pods in the late summer and fall (see Pfadt 1994 for more details). Egg development is important because the timing of hatch in the spring affects the timing of all subsequent stages of grasshopper growth. Hatch can be delayed by diapause, by temperatures below 50 °F (10 °C)—the threshold of developmental activity for most pest grasshoppers, by lack of soil moisture, and by placement of the egg pod in the soil; placement affects temperature and moisture. Likewise, hatch can be accelerated by temperatures above 50 °F and by soil moisture.

For instance, in southwestern Montana, embryos of *M. sanguinipes* develop faster at all temperatures above 50 °F than embryos of *A. elliotti* (see IV. 2). Yet *A. elliotti* hatchlings typically appear earlier in the spring than *M. sanguinipes* hatchlings (Kemp and Sanchez 1987), mainly because the pods of *A. elliotti* are nearer the surface of the soil and are generally laid in areas devoid of vegetation. Heat reaches the *A. elliotti* eggs earlier in the spring, and thus they begin to develop earlier than *M. sanguinipes* eggs, which are placed 0.4 inch (1 cm) deeper in the soil and among grass clumps (in areas cooler than bare areas) (Fisher 1993, Kemp and Sanchez 1987).

M. sanguinipes and most other economically important grasshopper species on rangeland have an embryonic diapause. Diapause can be defined as a genetically controlled physiological state of suspended animation that will revert to normal working physiological processes and growth only after occurrence of a specific event or a specific sequence of events. There are two major types of diapause: obligatory (occurs in every individual in a population at the same stage regardless of prevailing conditions) and facultative (not always occurring in every individual in a population and usually dependent upon specific environmental conditions).

In *M. sanguinipes*, the embryonic diapause is facultative. It often occurs when the embryo is about 80 percent developed. Diapause may last for several weeks or several months. With *M. sanguinipes*, we have found that less than 50 percent of any given population (group from a specific place) appears to exhibit long-term diapause (where, at room temperature [about 72 °F or 22 °C], hatch does not occur for at least 2 months). However, for nearly all populations we have studied, the minimum time for eggs to hatch when incubated at 86 °F (30 °C) has been 4 weeks. This 4-week minimum may indicate a very short diapause because embryos from a lab-reared nondiapause strain take only 18–21 days from laying to hatch at 86 °F. We do know that with all natural populations that we have tested, exposure of eggs to cool temperature, particularly 40–52 °F (4–12 °C), for at least 15 days has decreased the subsequent time needed at 86 °F for an embryo to hatch.

Phenological Studies of *M. sanguinipes*

During the spring through summer of 1992 and 1993, we studied the phenology (seasonal growth and development as it relates to climate) of *M. sanguinipes* at selected sites in Arizona, Utah, and Montana. Each week, we took samples at each site and determined the life stages of *M. sanguinipes*. The results are illustrated in figure IV. 6–1. This research revealed that the progression of growth by *M. sanguinipes* is a similar function of temperature at Bonita, AZ, and at Augusta, MT. The major difference is the calendar time when comparable events occur. Thus, it may be, at least in terms of what controls development in relation to temperature, that the genetic makeup of *M. sanguinipes* does not differ across its range.

Traveling north–south through the Western States, particularly during March, it is easy to notice phenological differences in plant maturity. In Montana, Idaho, Wyoming, and northern Utah, there will be snow on the ground, often freezing nighttime temperatures, and little or no green vegetation. As one moves south from about Salt Lake City, UT (40° N.), some greening is found west of the mountains in Utah and Nevada, and dramatic changes can be found south from Cedar City, UT (about 38° N.), where it is often cool and frosty, to the border of Arizona and Mexico (near 31° N.), where cacti are blooming and cotton has already been planted.

The first hatchlings at 32° N. (Bonita, AZ) were found in early to mid-March, whereas, the same event at New Harmony, UT (near 37° N.), did not occur until early to mid-May. Near 48° N. (Augusta, MT), hatchlings were not found until the first of June. Adults appear to live longer at the more southern sites—as much as 5 months at Bonita, AZ, compared to 3 months at Three Forks, MT.

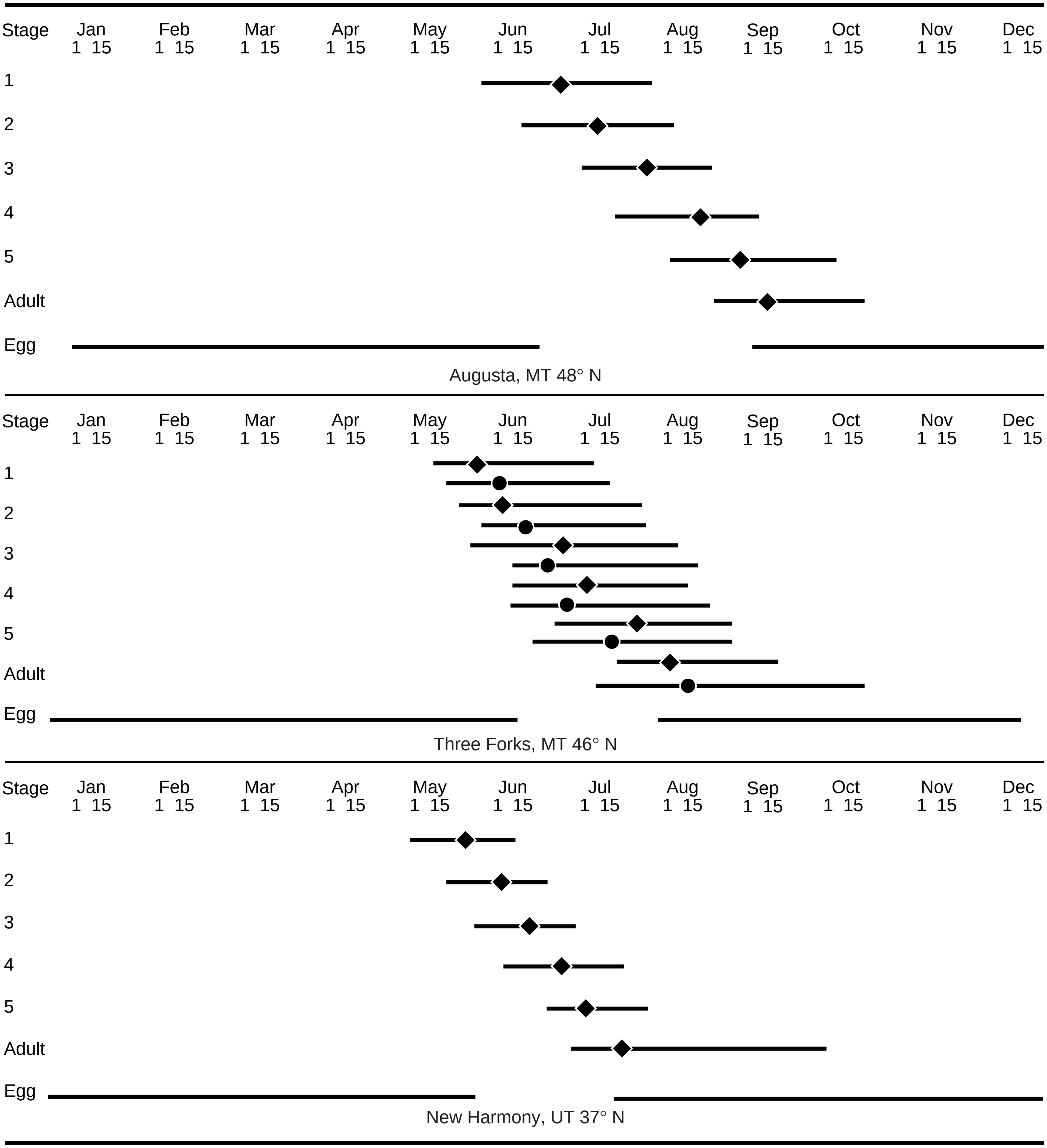
The effects that accompany latitudinal differences in climate appear to have a greater overall effect on grasshopper growth and development than altitude. However, the sites that are illustrated here vary from about 3,800 ft (1,160 m) to 6,000 ft (1,830 m) in altitude. Two other sites that have been examined over the past 2 years have been Rubys Inn, UT, at more than 7,600 ft (2,316 m) and Pinedale, WY, at more than 7,200 ft (2,200 m). Over the 2 years of these studies at the two high altitude sites, we never collected enough *M. sanguinipes* to derive phenological diagrams. Phenology at high altitudes (> 6,000 ft [1,830 m]) within the same latitude may be different. For instance, Rubys Inn, UT, is at the same latitude as New Harmony, UT. But when hatchlings were showing in New Harmony (early May), there was still 2 inches of snow on the ground at Rubys Inn. Of course, it should be noted that an altitude of 7,000 ft (2,133 m) or greater in the Western United States at latitudes south of 40° N. is the beginning of the subalpine zone and at latitudes north of 40° N. is the subalpine to alpine zone.

When dealing with rangeland sites at altitudes higher than 7,000 ft, you should remember that mountainous areas have local temperature patterns. If you need to know phenology of grasshoppers in these areas, then temperature needs to be recorded and monitored over time to produce a data base. However, much of the rangeland in the Western United States where we would expect a need for grasshopper integrated pest management is at altitudes below 6,000 ft (1,830 m), and thus, what is mentioned here is applicable.

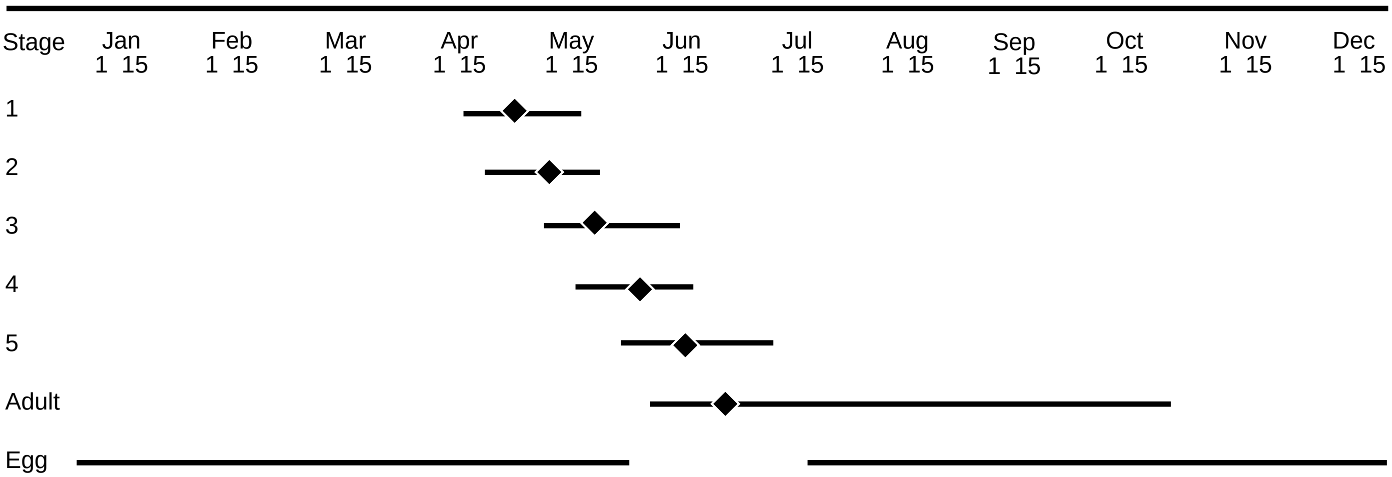
Sampling for phenological development at many of the sites in 1992 was difficult due to wet weather and low populations of *M. sanguinipes*. However, three sites, Three Forks, MT, and San Carlos and Bonita, AZ, were sampled enough times and had high enough populations to derive phenological diagrams. Therefore, we were able to compare phenological development for 2 years at those three sites (fig. IV.6–1). Occurrence of most stages was a few days later in 1993 than in 1992 even though the two seasons were quite different. In 1992, there was a dry spring and a wet summer south of 40° N. and an average (normal) spring and summer north of 40° N. In 1993, areas south of 40° N. suffered an extremely wet spring with a hot, dry summer while areas north of 40° N. had a cold, wet spring and summer.

The reason why there was not much difference in grasshopper development between the 2 years at each of the three areas is speculative. However, grasshopper nymphs are mobile and can seek warm microhabitats, such as bare, south-sloping areas during the day or under leaves at the base of plants at night, to adjust their internal temperature (thermoregulation). Thus, they can maintain metabolism at optimum levels (Kemp 1986, Hardman and Mukerji 1982). In cool weather, grasshoppers can increase their body temperature through basking (sunbathing) or sitting in areas that maximize collection of radiant heat. In warm to hot weather, grasshoppers keep their body temperature cooler than ambient air by seeking

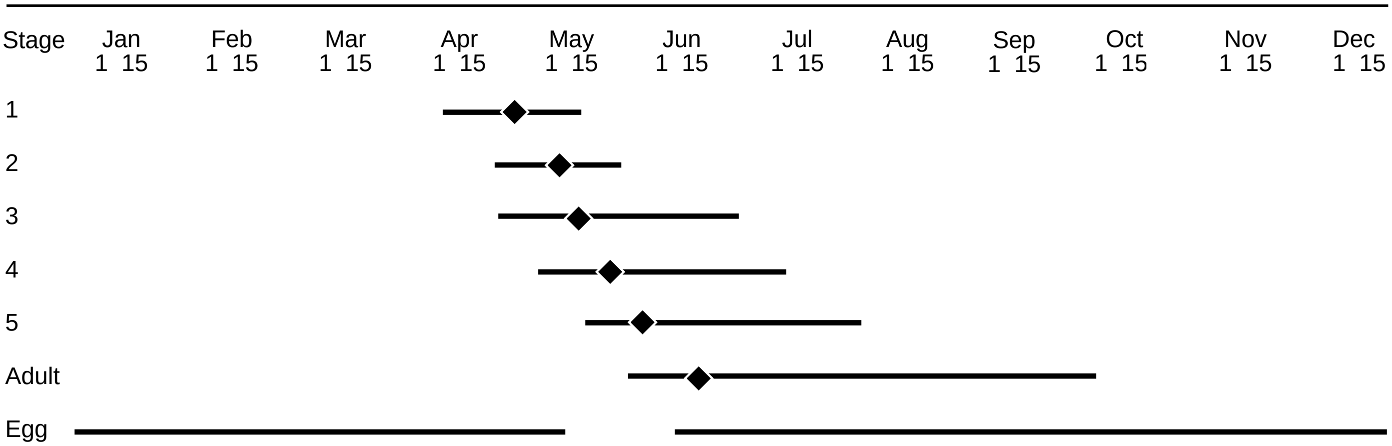
Figure IV.6–1—Phenological occurrence of life stages for *Melanoplus sanguinipes* at two sites in Montana and six sites south of 40° N., 1993 (◆) and 1992 (●)[1]



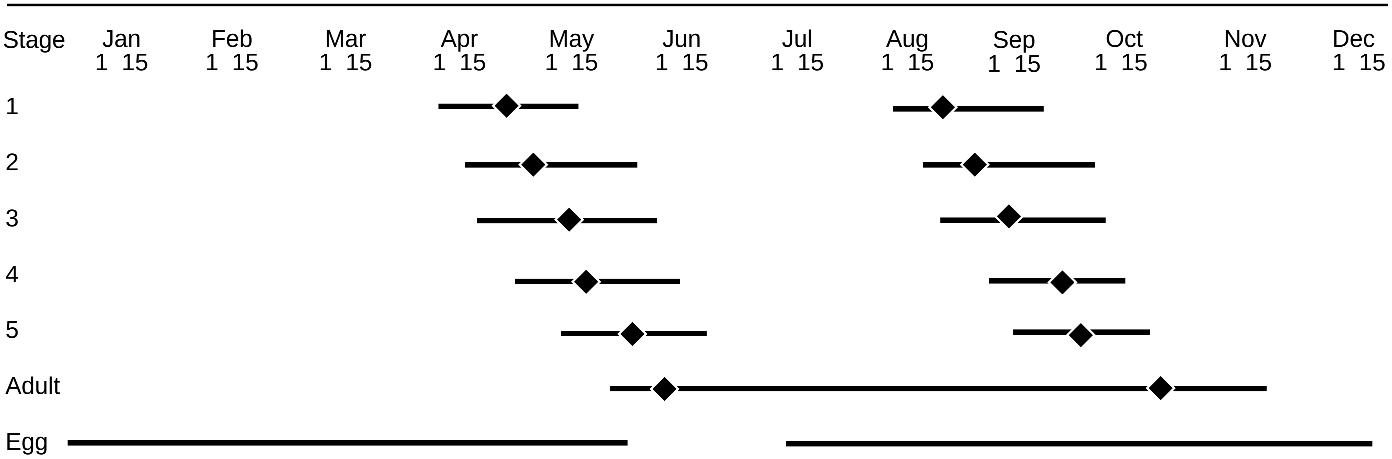
[1] The position of the diamond (◆) or circle (●) indicates the peak occurrence of the stage. The long solid lines indicate occurrence only.



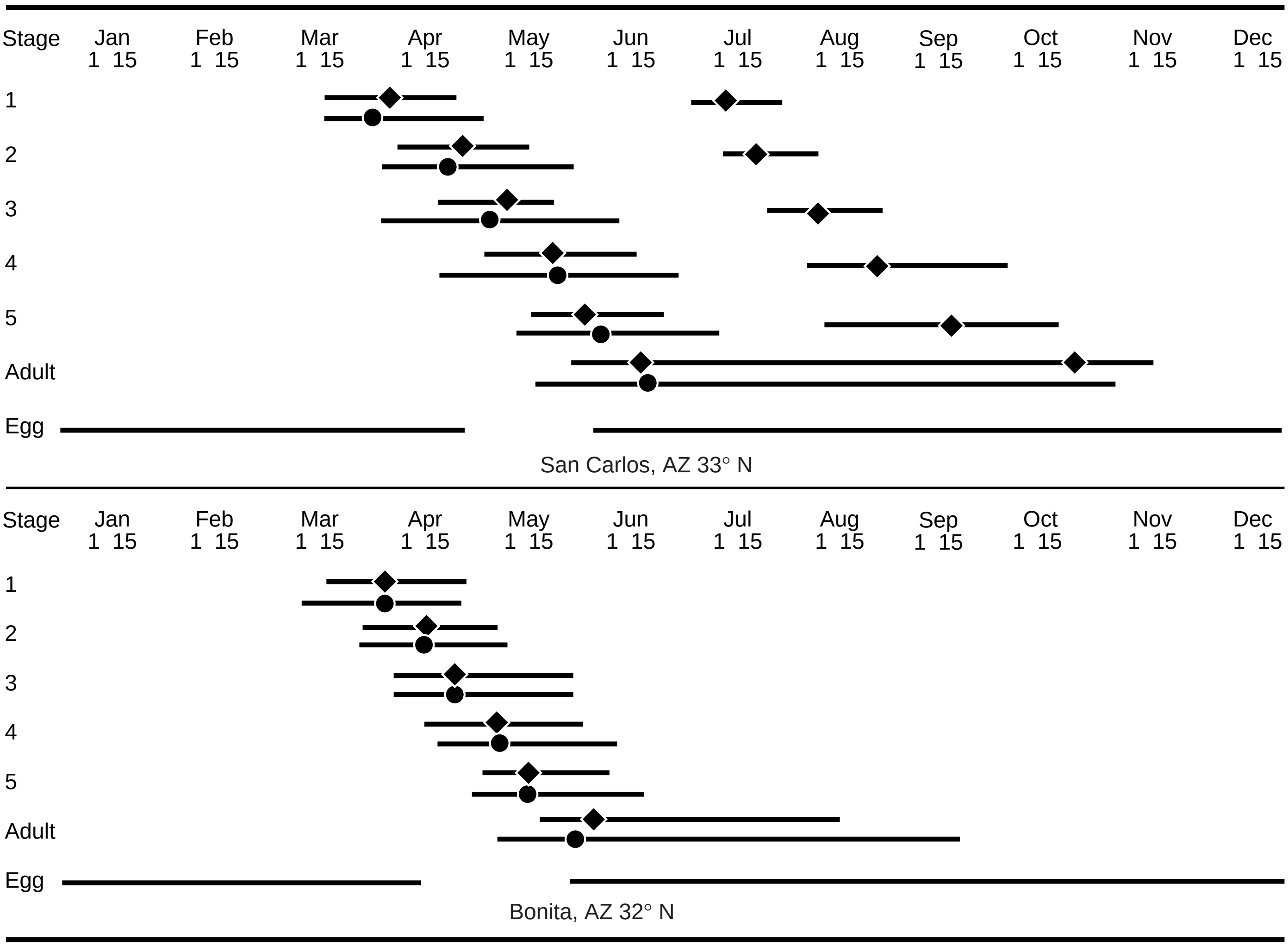
House Rock, AZ 36° N



Paulde, AZ 34° N



Young, AZ 33° N


Stage
Jan 1 15
Feb 1 15
Mar 1 15
Apr 1 15
May 1 15
Jun 1 15
Jul 1 15
Aug 1 15
Sep 1 15
Oct 1 15
Nov 1 15
Dec 1 15
1
2
3
4
5
Adult
Egg
San Carlos, AZ 33° N
Stage
Jan 1 15
Feb 1 15
Mar 1 15
Apr 1 15
May 1 15
Jun 1 15
Jul 1 15
Aug 1 15
Sep 1 15
Oct 1 15
Nov 1 15
Dec 1 15
1
2
3
4
5
Adult
Egg
Bonita, AZ 32° N

shaded areas and by climbing plants to take advantage of wind and cooling effects coming off the plant surfaces. Thus, the rates at which nymphs develop may remain relatively constant despite variable hot and cold weather. But the time when nymphal development starts will always depend on the time when hatch occurs.

In 1993, we observed what appeared to be a possible second generation at two sites, Young and San Carlos, AZ (fig. IV.6–1). The reason for this phenomenon is unknown; it was not observed in 1992. A second generation of *M. sanguinipes* in the southern areas has been mentioned occasionally in the literature (Barnes 1944, Dean 1982, Hebard 1938, Smith 1943). However, this is the first quantitative data provided as evidence of a second generation. We describe this only as a possible second generation because eggs were not collected in the field throughout the season; therefore, we could not document the early (prediapause) stages of embryonic development that would confirm a second generation.

Relationship to Hopper

The grasshopper phenological simulation module of Hopper (see VI.2) is based on thermal unit accumulation starting on January 1. Although historic National Oceanic and Atmospheric Administration weather data bases were not available for all sites, they were available for areas close to Bonita and San Carlos, AZ, and Augusta, MT. Predicted peaks for each nymphal stage were within 10 days of those shown in figure IV.6–1. We feel that Hopper accurately represents *M. sanguinipes* phenology in the field.

References Cited

Barnes, O. L. 1944. Time schedules for grasshopper surveys in Arizona. Journal of Economic Entomology 37: 789–795.

Dean, J. M. 1982. Control of diapause induction by a change in photoperiod in *Melanoplus sanguinipes*. Insect Physiology 28: 1035–1040.

Fisher, J. R. 1993. Location of egg pods of *Aulocara elliotti* (Orthoptera: Acrididae) in a field of crested wheatgrass in Montana. Journal of the Kansas Entomological Society 65: 416–420.

Hardman, J. M; Mukerji, M. K. 1982. A model simulating the population dynamics of the grasshoppers (Acrididae) *Melanoplus sanguinipes* (Fabr.), *Melanoplus packardii* Scudder, and *Camnula pellucida* Scudder. Researches on Population Ecology 24: 276–301.

Hebard, M. 1938. An ecological survey of the Orthoptera of Oklahoma. Tech. Bull. 5. Stillwater, OK: Oklahoma State University and Oklahoma Agricultural Experiment Station. 31 p.

Kemp, W. P. 1986. Thermoregulation in three rangeland grasshopper species. Canadian Entomologist 188: 335–343.

Kemp, W. P.; Sanchez, N. E. 1987. Differences in post-diapause thermal requirements for eggs of two rangeland grasshoppers. Canadian Entomologist 119: 653–661.

Pfadt, R. E. 1994. Field guide to common western grasshoppers. (2d ed.) Wyoming Agric. Exp. Stn. Bull. 912. Laramie, WY: University of Wyoming. 198 p.

Smith, R. C. 1943. Insects in Kansas. Agric. Bull. 62. Manhattan, KS: Kansas State University: 117–414.

IV.7 Nutritional Needs and Control of Feeding

Anthony Joern

The primary concern of range managers is forage loss, not the number of grasshoppers per se. After all, other than causing the loss of forage intended for other uses, grasshoppers do not generally present significant problems. In natural systems, grasshoppers may exhibit many positive attributes unrelated to agriculture (see chapter VII.16). Because forage consumption is the primary issue, understanding the basic nutritional needs and controls on feeding that drive food consumption by grasshoppers is important. From a modeling standpoint (in Hopper, described in chapter VI.2), consumption rates by grasshoppers of different sizes eating food of variable quality become key inputs to estimate forage loss.

Scientists have only a rudimentary understanding of grasshopper nutrition (Simpson and Bernays 1983, Bernays and Simpson 1990). For example, grasshoppers probably require the same 10 essential amino acids as required by mammals to support survival, growth, and reproduction. These include arginine, histidine, isoleucine, leucine, lysine, methionine, phenylalanine, threonine, tryptophan, and valine. However, the exact amino acid requirement for any grasshopper species is unknown. But scientists do know enough to provide a basic framework for understanding grasshopper nutrition. This knowledge is useful for predicting: (1) why grasshopper populations respond as they do, (2) why food consumption rates vary as they do, and (3) why some grasshopper control tactics will be more suited than others, depending on the availability of suitable food. Equally important, cultural management practices developed by range managers must work with naturally occurring constraints on grasshopper food consumption. These new management practices can be successful only if basic underlying nutritional issues are incorporated into the planning process at the beginning.

From the grasshopper's viewpoint, what considerations are important to feeding?

(1) Among insects, grasshoppers exhibit the highest total-nitrogen body content but typically feed on food that is very low in nitrogen. Since high protein content in grasshoppers comes primarily from low soluble-protein content in food plants, grasshoppers must make up this difference in protein concentration by eating and converting sufficient food material.

(2) As with all organisms, an energy source fuels the basic metabolism. Grasshoppers must eat sufficient energy besides protein to prevent the conversion of scarce protein to energy. Allocation of protein to growth and reproductive functions such as cuticle (skin) and muscle formation or egg production optimizes protein use.

(3) The dynamic process of balancing nutritional needs responds to many situations that can cause dramatic changes in feeding behavior. Nutritional needs change as the grasshopper develops and switches from nymphal to adult stages. Reproductively mature adults exhibit striking sex-specific differences in allocating nutritional resources. In addition, depending on the adequacy of the diet for immediate needs, internal physiological and biochemical processes may reallocate internal nutrient budgets to satisfy new requirements. As a result, certain activities, such as egg production or growth, cease if the diet becomes inadequate. These shifts probably happen often in natural environments, given that only poor-quality food is generally available to meet high-quality needs. Consequently, internal reallocation of nutrients may alter feeding behavior. These feedbacks can increase or decrease total consumption or cause switching among available food sources to adjust the intake to meet new nutritional needs.

One can manipulate the following factors to alter the nutritional economy and control of feeding: food acquisition, digestion, assimilation, utilization, and allocation. These factors interact as highly coordinated processes with many feedbacks. Figure IV.7–1 illustrates the principal tissues and organs involved in nutrient acquisition, storage, and metabolism. Such tissues interact to control acquisition and allocation of nutrients. Feedbacks control consumption rates among these components, the quality of the food, and nutrient needs. Because of this interactive system and its feedbacks, insect herbivores achieve remarkable efficiency at extracting required resources from plant material and in compensating for dietary deficiencies.

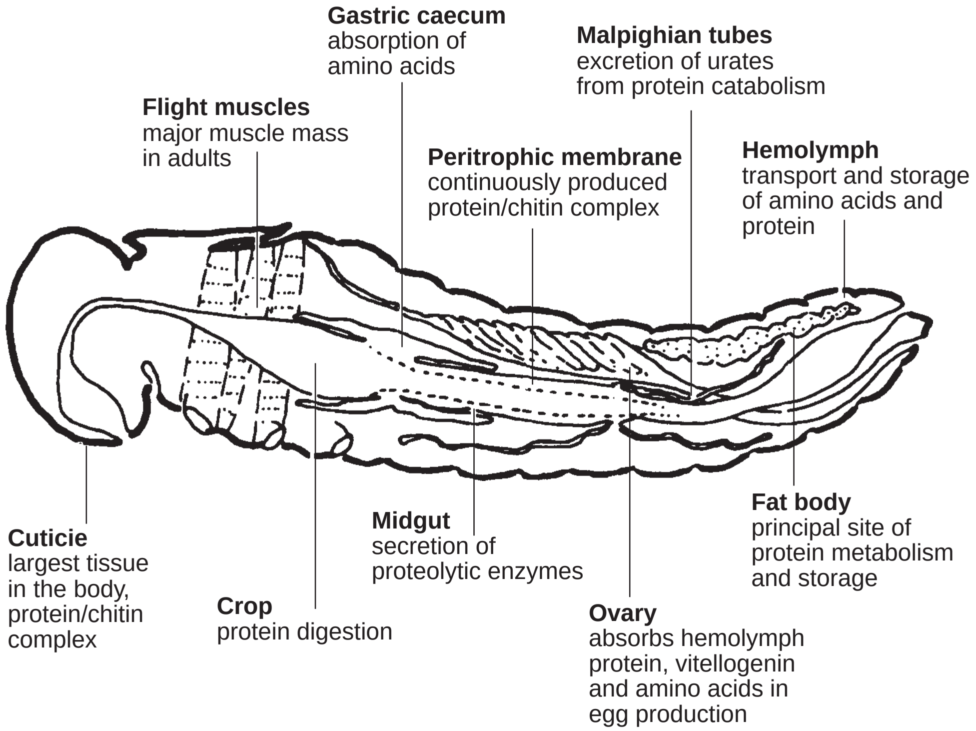


Figure IV.7–1—Multiple organ systems contribute to the acquisition, metabolism, distribution and deposition of proteins in grasshoppers, as depicted (adapted from Hinks et al. 1993).

Internal Needs and Allocation of Nitrogen

Nitrogen Requirements.—An adequate diet requires many components: protein or amino acids, energy-containing substances, water, minerals, and sterols, among many others (Bernays and Simpson 1990). To illustrate the dynamic nature of nutrient use and control, the internal allocation of protein among competing physiological needs provides a good example (fig. IV.7–2); similar relationships can be drawn for other nutrients although the details will differ. I illustrate nitrogen use because of its importance in so many key stages in a grasshoppers life history (McCaffery 1975). As figure IV.7–2 shows, many physiological and biochemical processes require amino acids as building blocks. These processes simultaneously compete for the available amino acid pool (Hinks et al. 1993). An amino acid pool that is insufficient to meet all needs will reduce physiological activities. Protein reallocation to other processes depends on their relative importance to critical life functions.

Why is nitrogen (protein and amino acids) in such demand to an individual grasshopper? Quite simply, proteins not only make up major components of most anatomical structures (such as muscle and cuticle) but are also intricately involved in most physiological and biochemical activity (all enzymes). Two examples from among many illustrate this point (reviewed in Hinks et al. 1993).

(1) Structural components require much protein. Cuticle, which is about half protein, accounts for about 50 percent of the grasshopper total dry mass. Because of cuticle replacement at each molt, both growth and cuticle replacement require massive investments in protein. Upon molting to the adult stage, the cuticle weight almost doubles, and allocation of protein (amino acids) to flight muscle triples.

(2) The hemolymph (body fluid) contains an important amino acid pool most of the time and provides amino acids for use in synthesizing structural, functional,

and storage proteins. Most amino acids come from digested proteins in leaf material. Grasshoppers typically maintain high amino acid concentrations. But some flux occurs, particularly during periods of strong demand for amino acids to drive growth, digestive, and reproductive processes. In addition, many proteins reside in the hemolymph. Fat bodies produce lipophorins that serve as storage proteins that are held in reserve to support future activities. In adults, egg production requires large amounts of the protein vitellogenin. Production and maturation of eggs require the diet-dependent accumulation of vitellogenin. For example, in *Melanoplus sanguinipes*, accumulation of vitellogenin occurs rapidly after wheat consumption but slows following oat consumption (Hinks et al. 1991). Adult males also accumulate various proteins in the hemolymph and accessory reproductive glands with the levels decided by diet.

Nitrogen Allocation.—After acquiring protein or amino acids from food, the strongest sink(s) (processes requiring significant amounts of nitrogen) direct the ultimate fate of these constituents. The sinks change depending on the developmental stage and sex of the grasshopper. For example, nymphal grasshoppers may allocate available protein between growth (soft tissues and cuticle) and digestive enzymes. Adult females exhibit antagonistic protein demands among body growth, digestive enzymes, and ovarian growth (including egg formation) (McCaffery 1975). Under most situations, especially when high-quality food is limited, all activities cannot proceed at maximal rates.

Tissue proteins are quite labile (able to change), so their constituent amino acids are available for transfer to other body functions with greater need. As an example, during starvation, grasshoppers resorb developing ovarioles,

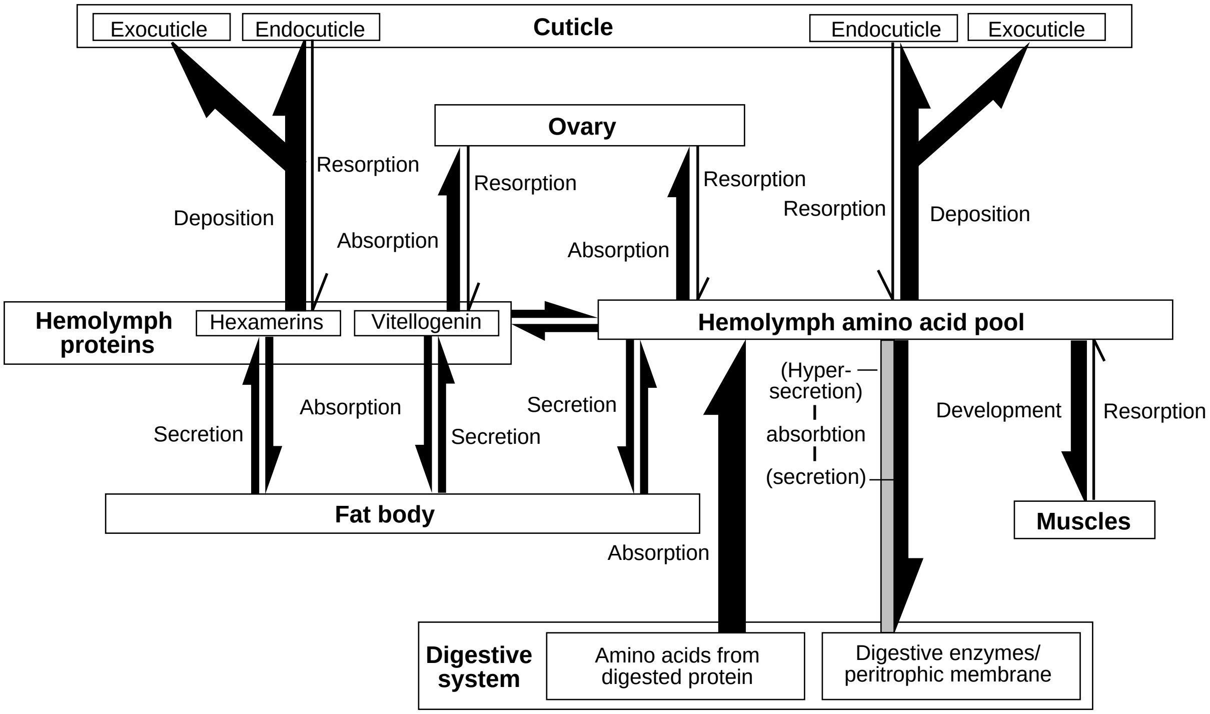


Figure IV.7–2—Diagrammatic representation of protein allocation among cuticle, tissues, and organs of grasshoppers (adapted from Hinks et al. 1993).

muscle, and gut tissue mass, and the fat body mass decreases with a sharp drop in protein reserves. Reassigning the constituents to other processes protects the animal from death (Hinks et al. 1993). When carbohydrate intake is insufficient, grasshoppers may metabolize protein to supplement the depletion of energy reserves. Many of these resorption processes are diet dependent, where different food plants lead to differential resorption rates depending on their nutritional quality.

Dietary Mixing and Compensation

Few grasshopper species eat only a single or even just a few plant species (Chapman 1990). In addition, individuals seldom specialize but readily feed on many plant species and parts. Polyphagous feeding (eating many kinds of food) appears to benefit individuals, and patterns of host plant selection illustrate adaptive behavior. Grasshoppers that feed on mixtures of food plants typically grow at faster rates than when fed single, otherwise suitable, host plants (MacFarlane and Thorsteinson 1980, Lee and Bernays 1988). Such mixing may serve several purposes (Bernays and Bright 1993):

(1) Diet mixing may dilute potentially poisonous plant chemicals that differ significantly among plants.

(2) Diet mixing may provide a better balance of nutrients if grasshoppers cans sense the differences between host plant species and pick plants whose nutritive profiles correct the insect's need. Optimal diets constructed in this fashion would counter incomplete nutrition obtained from single plants.

(3) Because many detoxification systems rely on induced enzymes (enzymes constructed only after the substrate is present), frequent mixing of such plants could maintain broad capabilities to deal with an array of poisons. This variety protects individuals from succumbing to occasional high doses of plant toxins. Evidence supports a variety of additional mechanisms that cause dietary mixing, including learning, chemosensory changes, and arousal with novel feeding cues. Each appears to become important to differing degrees in various grasshopper species.

Dietary imbalance often alters feeding behavior to compensate for suboptimal meals (McGinnis and Kasting 1967, Raubenheimer and Simpson 1990, Raubenheimer 1992, Yang and Joern 1994a–c). A grasshopper that encounters plants low in a critically needed substance (protein, for example) may either reject this plant or choose another. Each meal is unlikely to contain the optimal balance of required nutrients. Also, an insect cannot regulate the intake of one nutritional category without simultaneously altering the intake of all others. Very often, some plant or tissue may exhibit high quality for some nutrients and poor quality for others. By varying the specific intake order of different food plants or tissues, grasshoppers can regulate nutrient balance.

Water Balance

Grasshoppers actively regulate internal water balance. Besides the primary nutrients, water also can sometimes alter patterns of diet selection to maintain internal water balance (Bernays 1990). In very dry years, lack of water may explain grasshopper mortality better than low food availability. Too little information currently exists to tease apart the relative importance of water availability versus other nutritional components, especially under field conditions.

Meal Size and Frequency

Multiple interacting factors in a series of correlated relationships with unclear causal links regulate meal size and number. Persons responsible for developing grasshopper management plans will readily see the use of measuring plant quality to estimate forage losses to grasshoppers. Figures IV.3–3 (on p. IV.3–7) and IV.7–3 *(Melanoplus differentialis* and *Locusta migratoria)* illustrate relationships between host plant quality, temperature, and various components of the feeding responses, including elements of food processing, that enter the equation. In some of these cases, inverse responses (including increased feeding rate and lowered time of digestion in the gut) must hold. How grasshoppers control the process is often unclear (Yang and Joern 1994b, c).

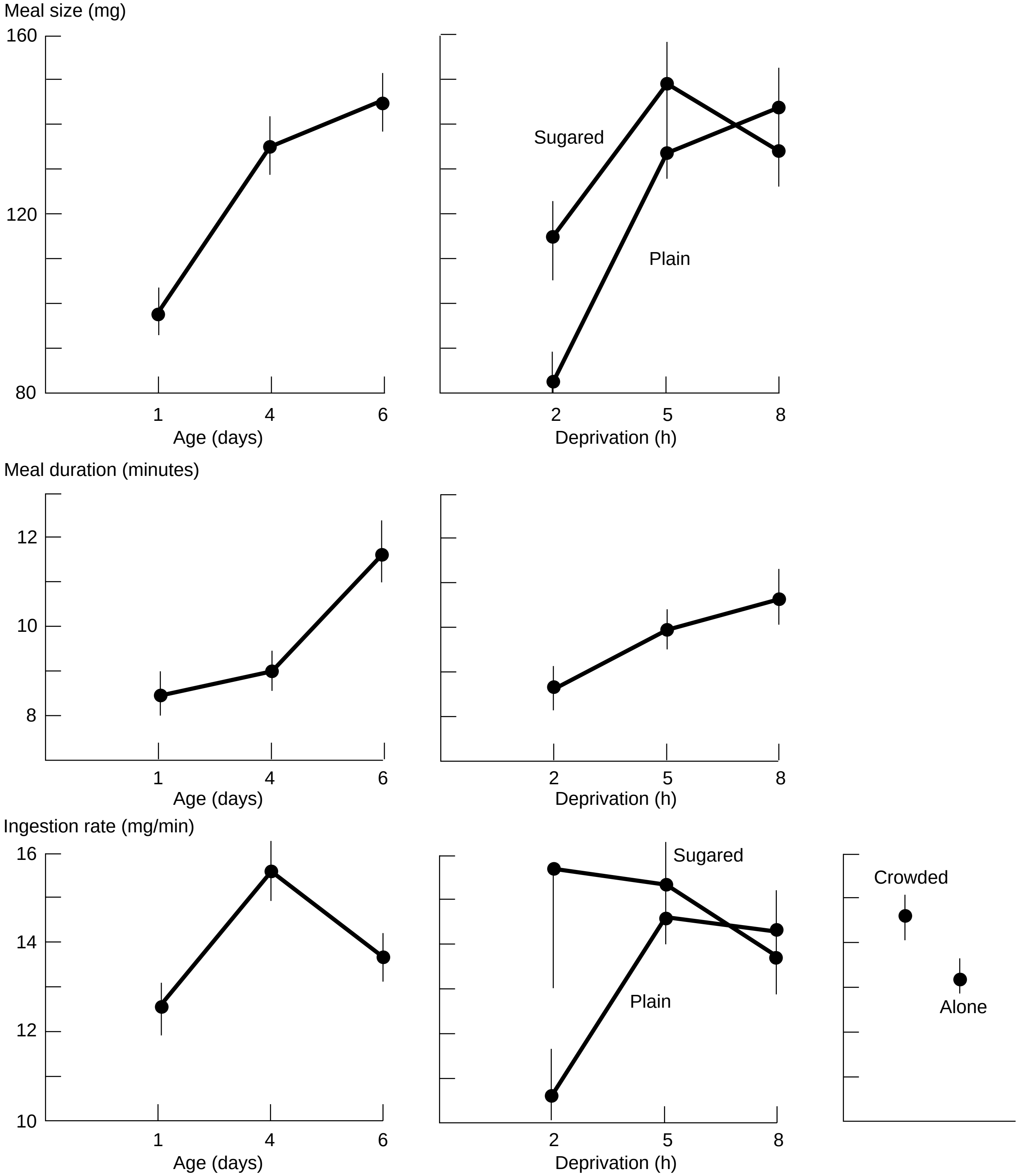


Figure IV.7–3—Effects of food deprivation time, age during the fifth instar, level of phagostimulation, and presence of other individuals on feeding behavior of *Locust migratoria* (adapted from Simpson 1990). Phagostimulation was promoted by dipping wheat seedlings in 1M sugar solution. Crowded conditions represent the presence of two other individuals in the test versus a single grasshopper (alone).

When food is lower in quality, both *M. differentialis* and *L. migratoria* typically eat more often for a longer period. Food residence time (the time that the food remains in the gut for digestion) increases as diet quality increases. As expected, the longer food remains in the gut, the greater is the assimilation rate. In addition, weight gain generally increases as food quality increases, although temperature-dependent metabolic effects can modify this response. Grasshopper metabolic rates increase with temperature, thus requiring faster energy intake to maintain a constant energy balance. At higher temperatures, weight gain may decrease because an increased metabolic rate burns off energy otherwise allocated to growth. Age and prior food deprivation can also exhibit significant impact on feeding responses (fig. IV.7–3). An important interaction between palatability and deprivation also exists as seen for plant material coated with sucrose, a feeding stimulant. After a period of about 5–8 hours, such as that experienced by grasshoppers on cold, cloudy days, food stimulation plays a secondary role to food deprivation.

Grasshopper body size also influences meal size. Large animals can eat more than small ones because of the absolute differences in gut volume (fig. IV.7–4). Grasshoppers also can compensate for poor-quality food by increasing the allocation to the gut. This ability results in a larger gut size, which in turn increases the ability to extract resources from food (Yang and Joern 1994a).

Feeding history can influence grasshopper movement, although few details exist. Grasshoppers exhibit lowered activity levels and move shorter distances after feeding on high-quality food than low-quality food. Such behavior may explain why grasshopper densities increased in grass patches in response to the fertilization level (Heidorn and Joern 1987; see IV.4). From a land manager’s perspective, this relationship means that grasshoppers will seldom be uniformly distributed across rangeland. Land managers may find that for control operations involving baits to be effective, distribution patterns based on food quality are important. Clever land managers may find ways to exploit this relationship in presenting baits for consumption, both by adding eating stimulants and “artificially” increasing concentrations of grasshoppers.

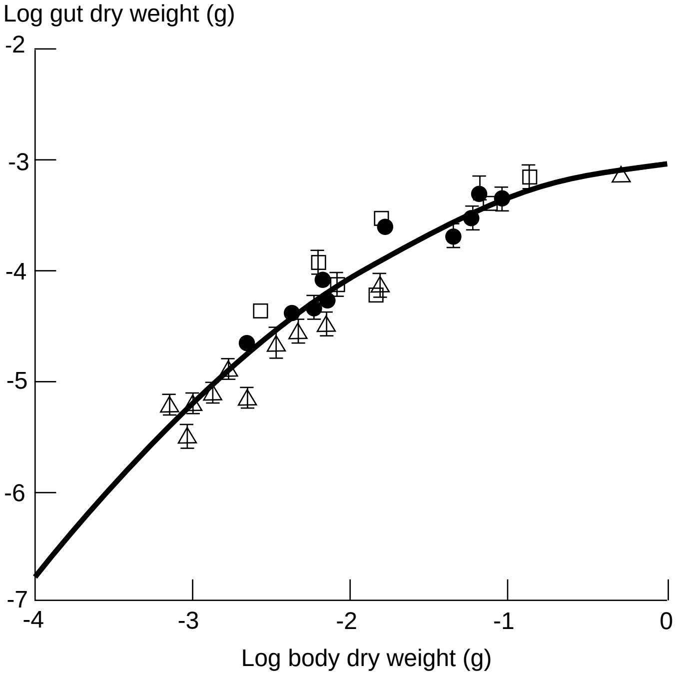


Figure IV.7–4—Regression of log-gut-dry mass to log-body-dry mass of females of 29 species of grasshoppers from a Nebraska sand hills prairie. Vertical bars represent standard errors (adapted from Yang and Joern 1994a).

Regulating Grasshopper Food Consumption

What decides the amount and timing of grasshopper feeding? Not unexpectedly, a variety of internal physiological feedbacks interact to maintain a constant concentration of key nutrients in the hemolymph. For the most part, neither modelers nor land managers will routinely incorporate directly into their planning known physiological responses that regulate feeding. Consequently, this section is short. However, developing some sense of what regulates grasshopper feeding behavior at the physiological level can be useful in trying to understand “motivational responses” that do not act at cross purposes to what the grasshopper does. In addition, clever managers may figure out methods to short-circuit these feedbacks in desirable ways. I feel that even a little insight is helpful.

When physiological needs shift, internal controls must shift accordingly. Thus, feeding-control mechanisms balance nutritional needs at several levels, some of which cannot always be simultaneously satisfied: water, protein, energy, trace minerals, and nutrients (such as sterols and fatty acids, specific free amino acids, and vitamins). Internal physiological feedback mechanisms include neurological control, osmoregulation (maintaining water balance), and responses by chemoreceptors. These mechanisms ultimately interact with environmental features that define the quality of food available and the time available to feed and process food.

In assessing grasshopper damage, food consumption stands at center stage. Regulation of food consumption depends on meal size, meal duration, and ingestion rate (Simpson and Bernays 1983, Simpson 1990). Palatability of food, duration of prior food deprivation, developmental stage, elapsed time within a developmental stage, and presence of other individuals nearby all affect meal size or duration. In addition, internal controls such as fluxes in amino acid concentration in the hemolymph can regulate feeding based on nitrogen needs through a series of physiological feedbacks (Simpson and Simpson 1990). Chemoreceptor sensitivity seems especially reactive to dietary protein levels and hemolymph composition (Abisgold and Simpson 1988).

Substances that promote feeding (phagostimulants) play important roles in grasshopper feeding behavior. Sucrose, a common free-sugar in plants, acts as an important phagostimulant for many grasshopper species. As sucrose levels increase up to 3–4 percent (dry weight), consumption rates increase. Other chemicals, such as specific amino acids, act as phagostimulants as well. During molting, the cuticle is completely rebuilt. Cuticle formation requires large levels of the aromatic amino acid phenylalanine. Phenylalanine in the diet can be limiting to growth, survival, and reproduction. Consequently, grasshoppers choose diets with higher concentrations of this amino acid (Behmer and Joern 1993).

Final Comments

Dynamic relationships that define food consumption require a multidimensional approach, mostly because a change in one variable, food quality, can exhibit so many effects. Because our ultimate goal revolves around reducing forage loss to grasshopper consumption, estimating these losses now and in the future becomes important. Host plant quality and the total number of grasshoppers (weighted by size) drive this relationship. However, most feedbacks that interact with temperature can play havoc with simple regression analyses so that more complex, dynamic models seem desirable in a forecasting sense. Dietary compensation takes place and earns a central position in understanding grasshopper feeding behavior. At present, I feel that these details will obscure relationships at the levels most useful to land managers: too many detailed data are required. However, forecasting modelers should continue to evaluate such notions in the hope that simplified and readily measured variables can increase local forecasting success.

References Cited

Abisgold, J. D.; Simpson, S. J. 1988. The effect of dietary protein levels and hemolymph composition on the sensitivity of the maxillary palp chemoreceptors of locusts. Journal of Experimental Biology 135: 215–229.

Bernays, E. A. 1990. Water regulation. In: Chapman, R. F.; Joern, A., eds. Biology of grasshoppers. New York City: John Wiley and Sons: 129–141.

Bernays, E. A.; Simpson, S. J. 1990. Nutrition. In: Chapman, R. F.; Joern, A., eds. Biology of grasshoppers. New York City: John Wiley and Sons: 105–128.

Bernays, E. A.; Bright, K. L. 1993. Mechanisms of dietary mixing in grasshoppers: a review. Comparative Biochemistry and Physiology 104A: 125–131.

Chapman, R. F. 1990. Food selection. In: Chapman, R. F.: Joern, A., eds. Biology of grasshoppers. New York City: John Wiley and Sons: 39–72.

Heidorn, T. J.; Joern, A. 1987. Feeding preference and spatial distribution by grasshoppers (Acrididae) in response to nitrogen fertilization of *Calamovilfa longifolia*. Functional Ecology 1: 369–375.

Hinks, C. F.; Cheeseman, M. T.; Erlandson, M. A.; Olfert, O.; Westcott, N. D. 1991. The effects of kochia, wheat and oats on digestive proteinases and the protein economy of adult grasshopper, *Melanoplus sanguinipes*. Journal of Insect Physiology 37: 417–430.

Hinks, C. F.; Hupka, D.; Olfert, O. 1993. Nutrition and the protein economy in grasshoppers and locusts. Comparative Biochemistry and Physiology 104A: 133–142.

Lee, J. C.; Bernays, E. A. 1988. Declining acceptability of a food plant for a polyphagous grasshopper, *Schistocerca americana*: the role of food aversion learning. Physiological Entomology 13: 291–301.

MacFarlane, J. H.; Thorsteinson, A. J. 1980. Development and survival of the two-striped grasshopper, *Melanoplus bivittatus* (Say) (Orthoptera: Acrididae) on various single and multiple plant species. Acrida 9: 63–76.

McCaffery, A. R. 1975. Food quality and quantity in relation to egg production in *Locusta migratoria migratorioides*. Journal of Insect Physiology 21: 1551–1558.

McGinnis, A. J.; Kasting, R. 1967. Dietary cellulose: effect on food consumption and growth of a grasshopper. Canadian Journal of Zoology 45: 365–367.

Raubenheimer, D. 1992. Tannic acid, protein and digestible carbohydrate: dietary imbalance and nutritional compensation in locusts. Ecology 73: 1012–1027.

Raubenheimer, D.; Simpson, S. J. 1990. The effects of simultaneous variation in protein, digestible carbohydrate and tannic acid on the feeding behavior of larval *Locusta migratoria* (L.) and *Schistocerca gregaria* (Forskal). I. Short-term studies. Physiological Entomology 15: 219–233.

Simpson, S. J. 1990. The pattern of feeding. In: Chapman, R. F.; Joern, A., eds. Biology of grasshoppers. New York City: John Wiley and Sons: 73–103.

Simpson, S. J.; Bernays, E. A. 1983. The regulation of feeding: locusts and blowflies are not so different from mammals. Appetite 4: 313–346.

Simpson, S. J.; Simpson, C. L. 1990. Mechanisms of nutritional compensation by phytophagous insects. In: Bernays, E. A., ed. Plant insect interactions. Vol. II. Boca Raton, FL: CRC Press: 111–160.

Yang, Y.; Joern, A. 1994a. Gut size changes in relation to variable food quality and body size in grasshoppers. Functional Ecology 8: 36–45.

Yang, Y.; Joern, A. 1994b. Compensatory feeding in response to variable food quality by *Melanoplus differentialis*. Physiological Entomology 19: 75–82.

Yang, Y.; Joern, A. 1994c. Influence of diet, developmental stage and temperature on food residence time in *Melanoplus differentialis*. Physiological Zoology 67: 598–616.

IV.8 Recognizing and Managing Potential Outbreak Conditions

G. E. Belovsky, J. A. Lockwood, and K. Winks

Introduction

An outbreak is defined ecologically as an “explosive increase in the abundance of a particular species that occurs over a relatively short period of time” (Berryman 1987). There is no doubt that certain grasshopper species in Western U.S. rangelands occasionally experience an outbreak and assume pest status, but most species do not exhibit outbreaks. Most species increase only slightly while the pest grasshopper species increase dramatically (Joern and Gaines 1990).

Identifying this rapid and dramatic increase in grasshopper numbers when it occurs is an easy task after the fact by examining regular surveys of grasshopper densities that are part of monitoring programs. However, surveys do not give pest managers the ability to predict the conditions that produce outbreaks. Understanding the ecological processes and events that produce these outbreaks is necessary for pest managers to be able to forecast outbreak events and design better management strategies.

Ecological Explanations for Outbreaks

To date, pest managers have sought simple ecological explanations in attempts to predict when outbreaks will occur in the future based upon past environmental events, such as last year’s temperatures and precipitation. For example, Joern and Gaines (1990) have found research that associates warm, dry springs with grasshopper outbreaks on northern rangelands but cool, wet springs with outbreaks on southern rangelands.

Even when the above weather relationships are observed, they never explain more than 25 percent of the observed variation in grasshopper numbers between years. This explanation is not very powerful scientifically or very useful for management. Nonetheless, these correlations have been widely used to infer that density-independent factors affect mortality (the proportion dying does not vary with the population’s density) because weather is independent of density, and that weather determines grasshopper population outbreaks in Western U.S. rangelands. The existence of an association between weather and grasshopper numbers is undeniable, but the interpretation of this association does not indicate that a straightforward implication of density-independent control of grasshoppers may be part of the association.

A simple analogy will help to illustrate this point. A house’s temperature may be controlled by a thermostat-controlled furnace and air conditioner, but the temperature may still fluctuate with outside temperatures. Does this mean that the house’s temperature is set by weather? No, the average inside temperature is set by the furnace and air conditioner, but fluctuations are created by weather. The thermostat-controlled furnace and air conditioner are equivalent to density-dependent factors operating on a population (the proportion dying or reproduction per individual varies with density) because the furnace and air conditioner adjust to changes in both the inside and outside temperatures.

Likewise, weather could be producing density-independent effects on the population and these could cause the population to increase or decrease, but the average population size could be set by density-dependent factors, such as food abundance and predation (Horn 1968). Another possibility is that the average population size is not constant but varies with weather (the equivalent of raising and lowering the thermostat as the outside temperature gets colder and warmer). For example, weather might influence food abundance, vulnerability to predators and parasitoids, or susceptibility to disease (Capinera 1987, Joern and Gaines 1990), factors that may create density-dependent effects. Therefore, the occurrence of population fluctuations because of weather does not imply that populations are controlled by weather or that density-independent factors are most important. The reliance of managers on the above weather relationships to predict outbreaks and the willingness of scientists to attribute population changes to density-independent mortality have kept our understanding of grasshopper populations in its infancy. Answers to these questions are largely unresolved (see VII.14—Grasshopper Population Regulation) but critical for designing when and how to manage grasshoppers.

Outbreak Patterns

If pest managers do not understand the ecological processes that control grasshopper populations, it becomes difficult to explain why certain populations exhibit outbreaks and how outbreaks develop. With information derived largely from studies of forest and agricultural insect pests, Berryman (1987) categorized insect outbreaks as being eruptive or gradient.

Eruptive Outbreaks.—These outbreaks occur when favorable conditions (such as less stressful weather, abundant food, and lack of predators) at a site permit the population to increase and the additional individuals move out to supplement populations at other sites. These additional individuals create the outbreak at the other sites or enable the populations at these other sites to "escape" the factors, such as predation, that have been keeping densities low. Sites producing surplus individuals are called "sources" or "hot-spots" and sites being supplemented, "sinks" (Pulliam 1988).

Gradient Outbreaks.—These outbreaks are restricted to sites with favorable conditions. Eruptive outbreaks spread over a region and require "hot-spot epicenters" to generate the outbreak, while a widespread outbreak that is gradient in nature requires widespread favorable conditions, such as common weather patterns favorable to a particular insect species.

Resolving whether grasshopper outbreaks are eruptive or gradient requires knowledge about the factors that control grasshopper populations at each site and the dispersal of individuals between populations in the landscape. If pest managers do not understand the factors controlling a single population, they will not be able to answer the issue of gradient versus eruptive, which requires knowledge about several populations. In addition, because the management of grass-hoppers in Western U.S. rangelands involves many species of grasshoppers and a variety of habitats, it is possible that some species and habitats exhibit eruptive outbreaks while others exhibit gradient outbreaks.

Without information on what controls the grasshopper populations that a pest manager is being asked to manage, how can the manager forecast outbreaks, allocate monitoring efforts to populations more prone to outbreak, and design better management strategies to prevent or suppress outbreaks? For example, a manager can prevent eruptive outbreaks by preemptive strikes against hot-spots, but a manager can respond to a gradient outbreak only after it has started. While progress is being made in understanding grasshopper population dynamics (see VII.14), scientists can seldom answer these types of issues with their current knowledge.

Broader Ecological and Economic Considerations

In developing control strategies for grasshoppers, managers must base their decisions on more than the density of grasshoppers. The observed grasshopper density must be considered in a broader ecological and economic context:

- the available forage base provided by plants and the potential reduction of this base by current and future grasshopper densities;
- the economic value of the forage base lost to grasshoppers;
- the economic cost of controlling grasshoppers; and
- the ecological mechanisms that may be controlling grasshopper numbers, and how control efforts might change these mechanisms and future grasshopper densities.

The Grasshopper Integrated Pest Management (GHIPM) Project has demonstrated that reference to a single grasshopper density, such as greater than 13/yd^2 (16/m^2), as constituting outbreak conditions is no longer adequate: density must be assessed in its ecological and economic context. This complexity is being considered in a very simple way by Hopper, the expert system decision-support tool developed by GHIPM. A set of simple examples illustrates this point.

Low Grasshopper Densities.—At densities below 6/yd^2 (8/m^2) grasshoppers can cause considerable damage to the forage base (up to 70 percent loss). High levels of damage occur if the forage base has low potential abundance (low biomass) and/or has low productivity (low regrowth) (Holmes et al. 1979). Such a forage base may be marginal for livestock production and may not be economically practical to protect. In these instances, control may not be warranted from a market perspective (Davis et al. 1992). However, individual ranchers may well call for control if any economic loss makes their ranching operations unprofitable, especially when grasshopper control costs are subsidized by State and Federal agencies.

Pest managers need to consider more than the economic value of lost forage production or the outcry of individual ranchers. Grasshopper control might provide short-term relief but worsen future problems in these environments. From GHIPM findings (see VII.14), it appears that grasshopper populations in these environments have a high potential for being limited by natural enemies. Pesticide applications that reduce grasshopper numbers could also reduce natural enemy numbers directly by outright poisoning of the invertebrate natural enemies, or indirectly by lowering the numbers of vertebrate predators as their invertebrate prey are reduced (Belovsky 1992 unpubl.). Therefore, the ultimate result of control efforts could be an increase in grasshopper numbers for the future, as they are released from the control of natural enemies.

In this kind of environment, grasshopper monitoring and control may not be warranted, except from a political/social mandate. But while these populations may not warrant further attention for management, they may deserve scientific attention. Understanding grasshopper population dynamics under low-density conditions can help explain population dynamics under other conditions where management may be necessary and can aid in the development of management strategies that create populations that do not cause appreciable economic damage. These conditions may represent populations that only outbreak infrequently, when conditions are unusual.

High Grasshopper Densities.—At densities above 13/yd^2, grasshoppers can cause damage to the forage base, even if it is abundant (high biomass) and/or has high productivity (Holmes et al. 1979). This damage may approach 20 percent; however, because of the forage's high abundance and/or productivity, it might still be economically very valuable for livestock production and economically practical to protect despite the low percentage of damage.

Even though in these instances control may be warranted from a market perspective, individual ranchers have some alternatives that may be more cost effective than grasshopper control. These alternatives could include making up for forage losses to grasshoppers by feeding hay to cattle or leasing additional rangeland (Davis et al. 1992). Such alternatives are especially more attractive in scenarios where grasshopper control costs are not subsidized by State or Federal agencies.

From GHIPM findings (VII.14), it appears that grasshopper populations on productive rangelands have a high potential for being limited by food. Control efforts may be frequently warranted in these environments to reduce grasshopper numbers and consumption of forage. Because of the chronic nature of these outbreaks, monitoring efforts may not have to be widespread. These are the circumstances where long-term management strategies that suppress grasshopper populations without repeated application of pesticides (such as habitat manipulation) can be most useful and need to be developed. These conditions can represent populations that serve as hot-spot epicenters from which eruptive outbreaks emerge, and therefore, may deserve special attention for the study of their grasshopper populations.

Intermediate to High Grasshopper Densities.—At densities more than 6/yd^2 but less than 13/yd^2, grasshoppers can cause damage to the forage resource, depending upon its abundance (biomass) and/or productivity. Populations with such densities may demonstrate dynamics that are intermediate to those described above, reflecting natural enemy- or food-limitation in different years (VII.14), and may be the most common circumstance in Western U.S. rangelands.

Given the variability of these populations from year to year, it may not be easy to assess the economic feasibility of control because control may be economically warranted in some outbreak years but not others. When conditions approach those of low densities/low forage, control may be unwarranted; when conditions approach those of high densities/high forage, it may be warranted. Therefore, intermediate populations require very careful monitoring to detect population trends and changes in the forage resource. These situations also demand greater flexibility by managers in developing control strategies that match the varying conditions. Relying on chemical control when populations are food-limited could reduce the numbers of natural enemies and worsen the outbreaks in years when natural enemies would otherwise maintain the grasshoppers at low densities (see above).

From the simple set of scenarios developed above, it is apparent that grasshopper management is neither simple nor straightforward. This job is further complicated when you consider the tradeoff between controlling the negative effects of grasshopper outbreaks versus potential beneficial effects that grasshoppers may produce, such as weed control and nutrient cycling (see VII.16).

Like so many natural resource management issues, the more people begin to understand the dynamics of the ecological processes that they are trying to manipulate, the more difficult the problem becomes to solve. First, we find that traditional perspectives on management are not always appropriate from an ecological and/or economic perspective. Second, we see that new management alternatives that may be more complicated to develop and apply are better suited to help in dealing with the problem. While investigators are still scientifically deciphering grasshopper outbreaks (VII.14), GHIPM's expert system Hopper brings together many of these new findings to aid pest managers in recognizing outbreak conditions, when it may be feasible to control these outbreaks, and how these outbreaks may be most effectively and economically managed.

References Cited

Berryman, A. A. 1987. The theory and classification of outbreaks. In: Barbosa, P.; Schultz, J. C., eds. Insect outbreaks. New York City: Academic Press: 3–30.

Capinera, J. L. 1987. Population ecology of rangeland grasshoppers. In: Capinera, J. L., ed. Integrated pest management on rangeland: a shortgrass prairie perspective. Boulder, CO: Westview Press: 162–182.

Davis, R. M.; Skold, M. D.; Berry, J. S.; Kemp, W. P. 1992. The economic threshold for grasshopper control on public rangelands. Journal of Agricultural Resource Economics 17: 56–65.

Holmes, N. D.; Smith, D. S.; Johnston, A. 1979. Effect of grazing by cattle on the abundance of grasshoppers on fescue grassland. Journal of Range Management 32: 310–311.

Horn, H. S. 1968. Regulation of animal numbers: a model counterexample. Ecology 49: 776–778.

Joern, A.; Gaines, S. B. 1990. Population dynamics and regulation in grasshoppers. In: Chapman, R. F.; Joern, A., eds. Biology of grasshoppers. New York City: John Wiley and Sons: 415–482.

Pulliam, H. R. 1988. Sources, sinks, and population regulation. American Naturalist 132: 652–661.

References Cited—Unpublished

Belovsky, G. E. 1992. Grasshopper control: implications of investigations into population/community ecology. In: Grasshopper Integrated Pest Management Project, 1992 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 57–63.

V. Rangeland Management

V.1 The Importance of Grazing Strategies to Grasshopper Management: An Introduction

Jerome A. Onsager

For some rangeland ecosystems, certain grazing-management strategies appear to offer great potential for reducing periodic grasshopper outbreaks. For most of the prairie grassland ecosystems, grasshopper densities tend to increase with drought and grazing intensity. In several different studies since 1940, grasshoppers have been reported as being most abundant during dry seasons in heavily grazed pastures. The study sites included mixed-grass prairie in Montana and Oklahoma, tall-grass prairie in Kansas, and fescue grassland in Alberta (see Onsager 1987 and Kemp 1992).

In the Montana studies, grasshopper densities generally were inversely proportional to plant height and amount of cover. Therefore, grazing strategies that manipulate the time, rate, and severity of forage harvest can, in turn, affect the time, rate, and degree to which prairie rangeland habitats are improved for grasshoppers.

For some rangeland ecosystems, an almost opposite situation appears to be true. Examples include short-grass prairie in Arizona (Nerney 1958) and Colorado (Capinera and Sechrist 1982) and Intermountain sagebrush-grass range in Idaho (see V.2), where food supply usually limits grasshopper density.

During dry or normal seasons in food-limited habitats, densities generally are low but tend to be highest in ungrazed or lightly grazed pastures. Infestations tend to increase during years with above-normal precipitation and above-normal forage production, but it is not practical to attempt grasshopper suppression through removal of forage with livestock (see V.6). Periodic grasshopper outbreaks, therefore, probably will continue in such habitats regardless of the presence or intensity of livestock grazing.

Hart et al. (1987) discuss some relationships between grazing management and pest management: The primary forage plant species determine to a large degree what pest species will be of most importance, the return from grazing management affects the resources available for pest management, and good grazing practices should maintain vigorous plant communities that resist pest outbreaks and recover from attack.

Hart's team also discusses five "families" of grazing strategies, four of which involve systems for rotation or alternation of periods of grazing versus no grazing. The fifth strategy is continuous or season-long grazing.

Perhaps the primary criticism of continuous grazing is that the plant species most preferred by livestock tend to be grazed and regrazed at the same growth stages year after year. This repetitive selection favors growth of plant species that are less palatable or species with unique competitive advantages and, consequently, favors the same species of grasshoppers year after year.

The boundaries between proper, sustainable, season-long grazing and abusive grazing usually are not clear and can vary from season to season. Management options are largely limited to adjustments in herd size, an option that may or may not stop the abuse. (Reducing the herd size could simply alter the number, area, or distribution of patches where abuse continues unabated.) Because frequent lapses into an abusive scenario can favor undesirable plant species, such lapses can favor undesirable grasshopper populations as well. In fact, the ability to thrive in disturbed habitats is a prominent characteristic of many of the grasshopper species that cause the highest levels of damage. Therefore, the continuous grazing strategy does not seem to offer much opportunity for proactive grasshopper management.

Hart's four "families" of grazing systems include (1) rotationally deferred grazing (grazing is not allowed in selected pastures until after a certain interval, and the deferment is rotated among pastures), (2) rest-rotation grazing (rest periods with no grazing intended to allow seed production and seedling establishment are rotated among pastures), (3) high-intensity, low-frequency grazing (heavy, nonselective grazing is followed by a relatively long period of rest before the next grazing), and (4) high-intensity, short-duration grazing (relatively short periods of intense grazing are interspersed between relatively short periods of rest). Devised in different rangeland ecosystems to meet different goals and objectives, these four grazing systems seem to share some common goals. These include improvement of range condition, maintenance of plant diversity, and avoidance of repetition, all of which are compatible with sound grasshopper management.

Besides providing a food source, plant canopy can affect grasshopper microhabitat in many ways. Thanks to both direct experimentation and modeling studies, we can now predict some of the responses of grasshoppers to grazing. High diversity in canopy structure and plant species composition tends to support high diversity in grasshopper species (Joern 1979, Pfadt 1982). This diversity and composition tend to provide stability and to suppress pest species that exploit disturbance.

Canopy removal increases solar radiation of the soil surface and increases airflow over the ground. Thus, canopy removal increases both soil and air temperatures and decreases relative humidity for grasshoppers. All of this is favorable to pest grasshopper species because sunlight and low humidity discourage important grasshopper pathogens and because higher temperatures accelerate grasshopper egg development, growth, maturation, and egg production. Canopy removal also can affect basking sites, which provide for early morning thermoregulation (to hasten grasshopper warmup); perching sites, which provide for avoidance of high midday temperatures; and availability or frequency of sites favored for egg-laying (some species require patches of bare soil).

The preceding two paragraphs suggest that any range-management practice that significantly opens up the prairie grassland canopy will tend to favor one or more pest grasshopper species. Therefore, the possibility is unlikely that any grazing strategy, season-long or systematic, can negatively affect every pest grasshopper species in every pasture during every season. However, some attributes of grazing systems should provide some benefits in all pastures every year. Both deferment and alternation of grazing can manipulate the time, rate, and degree of defoliation, and these factors affect the timing, rate, and degree of improvement in habitat for discouraging increases in pest grasshoppers. Both strategies also can prevent repetitively favoring the same pest species for consecutive seasons. Even subtle changes in microhabitat can cause significant decreases in grasshopper development rates and survival rates, and reducing these rates can not only increase the interval between periodic outbreaks but also decrease their intensity and duration.

Different grazing systems can rely on different mechanisms to achieve similar goals. For example, in eastern Montana, Banister (1991) essentially uses periodic high-intensity grazing to increase his forage base (he forces utilization of unpalatable forage, which is about as nutritious as palatable forage). He then uses long periods (about 23 months) of rest to allow plant recovery and to generate plant litter and a tall, dense canopy, which discourage grasshoppers.

Meanwhile, in western North Dakota, Manske (see V.7) promotes use of a “twice-over” rotational grazing system that he developed specifically for use in the northern Great Plains. He allows grazing during a critical period of plant growth to induce subsequent increases in total forage production. The system increases cover and encourages the reproduction of preferred forage (the grasses that are preferentially grazed are selectively induced to produce tillers). The heavier canopy created by this rotation of grazing schedules discourages grasshopper populations.

All observations to date indicate that both systems have merit. Infestations on Banister’s lands seem to comprise mostly *Melanoplus sanguinipes* (a very mobile species), and the grasshopper densities seem to decrease with length of the rest period and with distance to adjacent cultivated crop- or rangeland under more traditional management.

Infestations affecting Manske’s land have been shown to suffer from unusually long periods for development of immature grasshoppers and from rather high daily mortality rates of all stages. Neither system supports pest species that need bare soil for egg-laying. The biggest difference seems to be that the former modifies grazing behavior of the animals while the latter increases production of preferred forage plants. Both systems are ingenious, and both represent creative approaches to the solution of complex, interrelated problems. I hope that their examples will inspire similar integrated management packages that will discourage grasshoppers in other rangeland ecosystems.

The chapters in this section provide an overview of grazing management and the role of grasshoppers in healthy range ecosystems. The introduction of nonnative rangeland plants in the rangeland States unquestionably has had an effect on grasshopper populations, and moisture is a key variable in any range management decision. Grasshopper management through controlled removal of vegetative cover appears to have promise in some situations and may prove to be a key approach to integrated grasshopper management in the future.

References Cited

Banister, R. 1991. Eight principles of range management. Rangelands 13: 85–86.

Capinera, J. L.; Sechrist, T. S. 1982. Grasshoppers (Acrididae)—host plant associations: response of grasshopper populations to cattle grazing intensity. Canadian Entomologist 114: 1055–1062.

Hart, R. H.; Samuel, M. J.; Waggoner, J. W., Jr.; Kaltenbach, C. C.; Smith, M. A. 1987. Grazing management systems for the shortgrass prairie. In: Capinera, J. L., ed. Integrated pest management on rangeland: a shortgrass prairie perspective. Boulder, CO: Westview Press: 9–24.

Joern, A. 1979. Resource utilization and community structure in assemblages of arid grassland grasshoppers (Orthoptera: Acrididae). Transactions of American Entomological Society 105: 253–300.

Kemp, W. P. 1992. Temporal variation in rangeland grasshopper (Orthoptera: Acrididae) communities in the steppe region of Montana, USA. Canadian Entomologist 124: 437–450.

Nerney, N. J. 1958. Grasshopper infestations in relation to range condition. Journal of Range Management 11: 247.

Onsager, J. A. 1987. Current tactics for suppression of grasshoppers on range. In: Onsager, J. A., ed. Integrated pest management on rangeland: state of the art in the sagebrush ecosystem. Washington, DC: U.S. Department of Agriculture, Agricultural Research Service; 60–65.

Pfadt, R. E. 1982. Density and diversity of grasshoppers (Orthoptera: Acrididae) in an outbreak on Arizona rangeland. Environmental Entomology 11: 690–694.

V.2 Historical Trends in Grasshopper Populations in Southern Idaho

Dennis J. Fielding and M. A. Brusven

Many people who live in the West want to know when the next grasshopper outbreak will occur. Currently, understanding of grasshopper population dynamics on rangeland is limited. While precise predictions of grasshopper densities cannot be made from place to place and year to year, examining historical records since 1950 reveals trends that may be valuable when managers need to predict when and why the next grasshopper outbreak will occur.

Each year, personnel of the U.S. Department of Agriculture's Animal and Plant Health Inspection Service, Plant Protection and Quarantine (APHIS-PPQ) conduct surveys of adult grasshopper populations. These survey records generally do not give information on species composition and do not represent intensive sampling, but they are useful in documenting large-scale, regional trends in overall grasshopper densities.

These records show that areas of high density (more than 8 grasshoppers/yd^2) occur somewhere in the State of Idaho nearly every year, but usually these areas are small. Most grasshopper problems occur in the southern portion of the State. Major outbreaks, covering large portions of southern Idaho, occurred in each decade since 1950: in the early 1950's, 1963–65, 1971–72, and most recently, in 1985.

Although we cannot detail the relative contribution of all factors influencing grasshopper populations, we believe that weather plays a very important role in grasshopper population fluctuations in southern Idaho. The historical records show that high grasshopper populations are associated with above-average precipitation at most locations in that area.

Figure V.2–1 shows the importance of adequate precipitation for grasshoppers by depicting the relationship between the number of acres sprayed for grasshopper control and the total precipitation of the 2 previous years. Since 1950, APHIS and its predecessors carried out spray programs covering more than 100,000 acres in southern Idaho in 7 years following the 15 wettest 2-year periods. No spray programs of more than 50,000 acres occurred following the 15 driest 2-year periods.

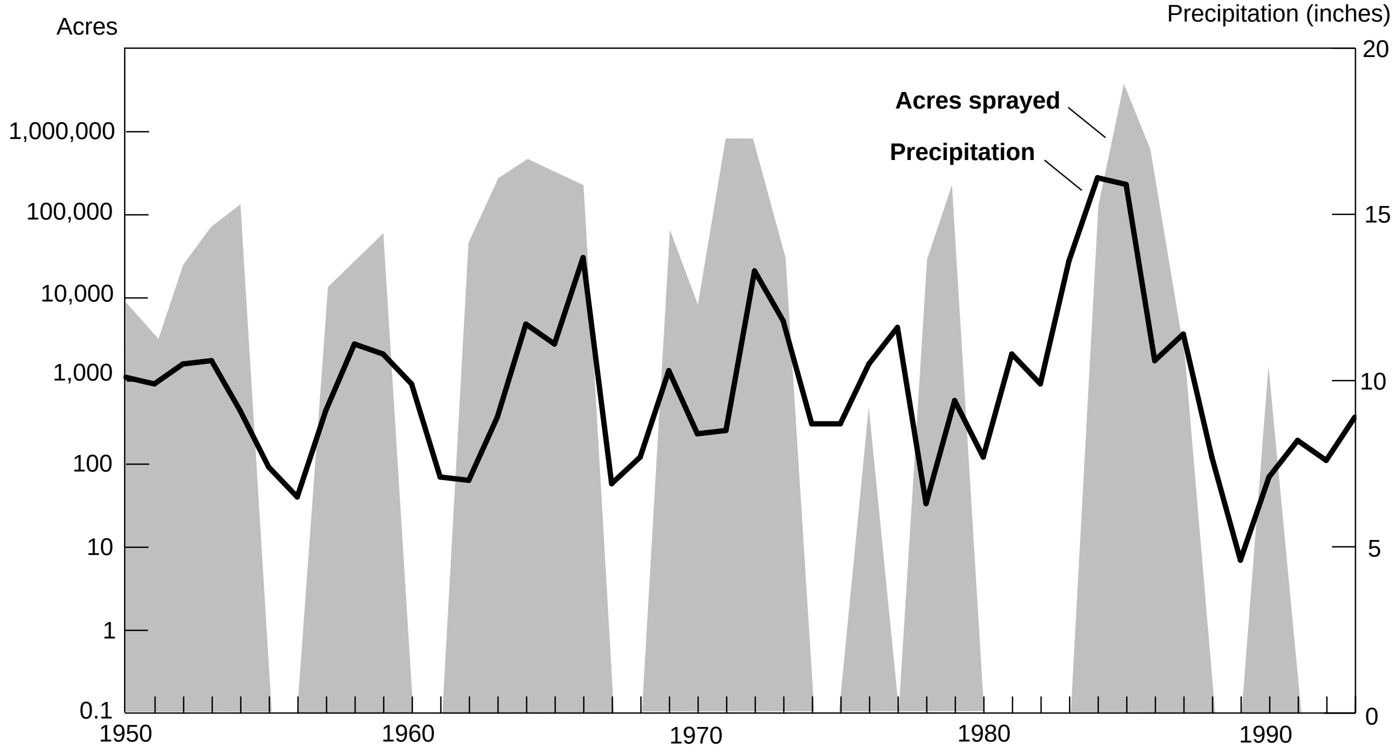


Figure V.2–1—Annual acreage treated for control of grasshoppers in southern Idaho, 1950–92. Precipitation is the average total for the 2 years prior to year of treatment.

Precipitation affects grasshoppers both directly and indirectly. In the Intermountain region, most precipitation occurs between October and April; rainfall in the summer is generally very scant and unpredictable. Grasshopper eggs may be susceptible to drying out during summer drought.

Precipitation also greatly influences the amount and quality of forage available to grasshoppers. As with any herbivore, grasshoppers require abundant energy and protein to achieve maximum reproduction. During drought episodes, rangeland in the Intermountain region provides little green forage in late summer, when many pest grasshopper species reach reproductive stages. Precipitation also may influence the incidence of grasshopper diseases.

Temperature is an important variable. Grasshoppers require a certain amount of heat units to complete development and reproduce. A short growing season at higher elevations may limit grasshopper populations. Cooler, high-elevation areas in southern Idaho usually have lower average grasshopper densities.

Varying 27-year-average densities of adult grasshoppers among 26 locations across southern Idaho reflect the importance of temperature and precipitation. The wetter and warmer locales of southwestern Idaho tend to have the highest average densities, and the cooler, drier areas of eastern Idaho, the lowest.

Biotic (living) factors also help regulate grasshopper populations. Predators, parasites, and pathogens may exert a significant influence on grasshopper population dynamics. Competition for limited resources, especially desired food plants, also may have an impact.

If weather is the primary factor controlling fluctuations in grasshopper populations in southern Idaho, we can do little to prevent occasional outbreaks. Anecdotal evidence from the 1800's suggests that grasshopper outbreaks are a natural feature of southern Idaho rangeland. To date, evidence does not show whether the frequency of outbreaks has increased with the introduction of domesticated livestock or exotic plant communities. Our observations show that maintaining a shrub cover with a perennial grass understory will foster grasshopper populations that are more diverse with more species that are not prone to outbreaks. We therefore suggest that habitat management is the best long-term action to reduce grasshopper problems (see VII.12).

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V.3 Associations Between Grasshoppers and Plant Communities on the Snake River Plains of Idaho

Dennis J. Fielding and M. A. Brusven

A mosaic of vegetation exists across the landscape of the Intermountain region of Idaho (fig. V.3–1). Soils, elevation, and disturbance history strongly influence the mix of plant species growing on a site. Vegetation directly affects watershed functions, suitability of habitat for wildlife, livestock forage, and many recreational uses. Therefore, range managers are very concerned with vegetation management. They try to nurture plant communities that will provide an optimal balance among the multiple demands placed upon America's public rangelands.

The plants growing on a site also provide resources, such as food and shelter, critical to grasshoppers. Because plants define much of a grasshopper's environment, we may expect that different plant communities will harbor different grasshopper species. Our research on the U.S. Department of the Interior, Bureau of Land Management's Shoshone District in south-central Idaho has documented some dominant trends in the associations between grasshoppers and plant communities in the region.

Exotic and Native Plant Communities in Southern Idaho

Compared to some other grassland ecosystems, such as the short-grass prairie of the Great Plains, the sagebrush–grass ecosystem of the Intermountain region is very susceptible to disturbance. Evidence shows that this region did not support heavy concentrations of large, vertebrate herbivores before settlers introduced livestock. (The buffalo [American bison] did not inhabit the Snake River Plains in large numbers.) Grazing, especially during the spring and early summer growing season, easily depletes most of the native perennial grasses in this

Figure V.3–1—Undisturbed Idaho rangeland may contain many native plant species, such as sagebrush and bluebunch wheatgrass. Native plant communities often are host to grasshopper species different from species found in plant communities with introduced grasses. (U.S. Department of the Interior, Bureau of Land Management photo by Mike Pellant.)

region. With the introduction of large numbers of livestock in the 1800's, a substantial decline in the abundance of native perennial grasses occurred over large areas of the region.

Introduced from Eurasia, annual grasses such as cheatgrass *(Bromus tectorum)* and medusahead *(Taeniantherum asperum)* quickly spread through the region. These exotic species are often present in relatively undisturbed plant communities but usually become dominant only on disturbed sites.

Because annual grasses form a continuous, fine fuel that dries out early in the summer fire season, the presence of annual grasses on a site greatly increases the chances of wildfire. Most species of sagebrush are sensitive to fire and with repeated burning are lost from the community. Frequent burning perpetuates the dominance of cheatgrass and maintains these annual grasslands.

This process of shrub loss and conversion to annual grasslands is a key management problem that affects nearly every use of public rangelands on the Snake River Plains. Annual grasses are more susceptible to climatic fluctuations, such as drought, than perennial grasses, so forage production is less predictable on annual grasslands. Cheatgrass matures early in the season, so the grazing season is shorter than on perennial grasslands. The lack of shrub cover makes for poor-quality wildlife habitat, so annual grasslands have diminished plant and animal diversity. Finally, the increased frequency of fire on annual grasslands increases the costs of fire suppression. In the Shoshone District, about 240,000 acres have been converted from perennial to annual grasslands.

Because of the limited resource values of annual grasslands, efforts have been made to reconvert cover in some of these areas to perennial grasses. A primary strategy during the last 40 years has been to plant crested wheatgrass *(Agropyron cristatum),* an introduced perennial bunchgrass that is relatively easy to establish and exhibits competitive abilities against cheatgrass. Crested wheatgrass is often seeded as part of fire-rehabilitation projects or following removal of overabundant sagebrush stands in range-improvement projects. These seedings have typically been established as monocultures, although a new trend involves more diverse seed mixtures that include shrubs and forbs.

A crested wheatgrass monoculture usually has a large percentage of bare ground between the bunchgrasses and fewer annual grasses and weeds than other habitats. Where crested wheatgrass stands fail to become established, because of drought for instance, range-improvement projects can actually promote conversion to highly disturbed annual grassland. As of the mid-1980's, about 20 percent of the Shoshone District below 5,000-ft elevation consisted of crested wheatgrass stands.

Grasshopper Complexes and Principal Species of Southern Idaho

Only about 4 of the 40-plus common species of grasshoppers in southern Idaho attain pest status. The others seldom reach high densities and may be considered harmless or beneficial.

The spurthroated grasshoppers, subfamily Melanoplinae, include some of the most pestiferous species in southern Idaho. Most feed upon a wide range of plants, but some are more specialized. *Melanoplus cinereus,* for instance, feeds mainly on sagebrush and is found only where sagebrush is growing. *Hesperotettix viridus* feeds mainly on rabbitbrush (*Chrysothamnus* spp.) in southern Idaho.

The lesser migratory grasshopper, *M. sanguinipes,* is the number 1 grasshopper pest in southern Idaho. This species occurs in a wide variety of habitats across North America and it feeds upon many forbs and grasses. It has a high reproductive potential, and populations can reach outbreak status within a generation or two when conditions are favorable. This insect will readily migrate to irrigated crops when rangeland vegetation dries during summer droughts.

The valley grasshopper, *Oedaleonotus enigma,* also can reach outbreak densities. It feeds primarily on forbs but will feed extensively on cheatgrass in the spring and on sagebrush during summer droughts. From 50 to 95 percent of a population of this species have short wings and are flightless. Scientists do not know how commonly these grasshoppers migrate from rangeland to cropland. Depending on the proportion of flightless individuals in the population and the distance from cropland, this species is much less significant as a threat to crops than *M. sanguinipes.*

Members of the subfamily Gomphocerinae, the slantfaced grasshoppers, feed almost exclusively on grasses. Except for the bigheaded grasshopper, *Aulocara elliotti,* slantfaced grasshoppers are not major pests in southern Idaho, although *Ageneotettix deorum* and *Amphitornus coloradus* may be common pests elsewhere. *Aulocara elliotti* matures from mid-June to July, about the same time as the perennial grasses on which it feeds. Although it can attain high densities and can be very damaging to rangeland grasses, it does not seem to be a threat to cultivated crops in southern Idaho.

The lower elevations of the Intermountain region have many species in the subfamily Oedipodinae, the bandwinged grasshoppers. About half of the grasshopper species in south-central Idaho are included in this group. Most are large-bodied, generalist feeders, although *Trachyrachys kiowa* is a common, smaller grasshopper that feeds exclusively on grasses. High densities of the clearwinged grasshopper, *Camnula pellucida,* have been recorded at higher elevations in south-central Idaho.

Grasshopper Species Distributions Across Plant Communities in Southern Idaho

We established long-term grasshopper monitoring sites at 30 locations in the Shoshone District, representing annual grasslands, crested wheatgrass seedings, and sagebrush–grass areas. The sagebrush–grass sites covered a variety of vegetation types, with different species and subspecies of sagebrush represented. Dominant understory grasses included cheatgrass or native bunchgrasses, such as bluebunch wheatgrass *(Agropyron spicatum)* or Thurber's needlegrass *(Stipa thurberiana).*

During 5 years of monitoring grasshopper populations on these sites, we have observed differences in grasshopper species composition between exotic and native plant communities.

The annual grasslands had the highest grasshopper densities, along with the highest proportion of pest species, during the 5-year period. The annual grassland sites also had the lowest grasshopper species diversity and were clearly dominated by the Melanoplinae (fig. V.3–2). Other researchers have noted that these species are common in weedy, disturbed habitats.

The grasshopper species commonly found in annual grassland habitats usually are generalist feeders that live in a variety of habitats, characteristics that make them well adapted to exploit unpredictable habitats like the annual grasslands. Two species, *M. sanguinipes* and *O. enigma,* accounted for most of the grasshoppers on the annual grassland sites. The presence of *M. sanguinipes* correlated positively with areas having a high percentage of ground cover of annual vegetation and correlated negatively with areas having sagebrush cover.

The crested wheatgrass seedings had a more even representation of grasshopper species, with the grass-feeding Gomphocerinae being the most abundant group in these habitats (fig. V.3–2). Most slantfaced grasshoppers are closely associated with perennial grasses, such as crested wheatgrass, using them for food and shelter.

The sagebrush–grass sites had an even distribution of grasshopper species across the three subfamilies (fig. V.3–2). Grasshopper assemblages of the sagebrush–grass habitats included a greater proportion of species with specialized habitat requirements. These species tended to be found at fewer sites and to have a more restricted diet.

Implications for Range Managers

We conducted our studies during years of low grasshopper densities. We expect that under outbreak conditions the observed relationships may change. For example, we expect *M. sanguinipes* to be a prominent species in all southern Idaho habitats during an outbreak. We need detailed observations during high-density years. Historical data from the last outbreak (1985) are consistent with our more recent observations in that, although we found high densities in all habitats, the annual grasslands had the highest average densities.

While one may argue that during a major outbreak all habitats will require control operations, we believe that outbreaks will be less frequent and of smaller extent in habitats characterized by sagebrush cover over a perennial grass understory. Moreover, we believe that efforts to prevent further shrub loss and to reconvert annual grasslands to perennial grasses should help restrain future grasshopper outbreaks.

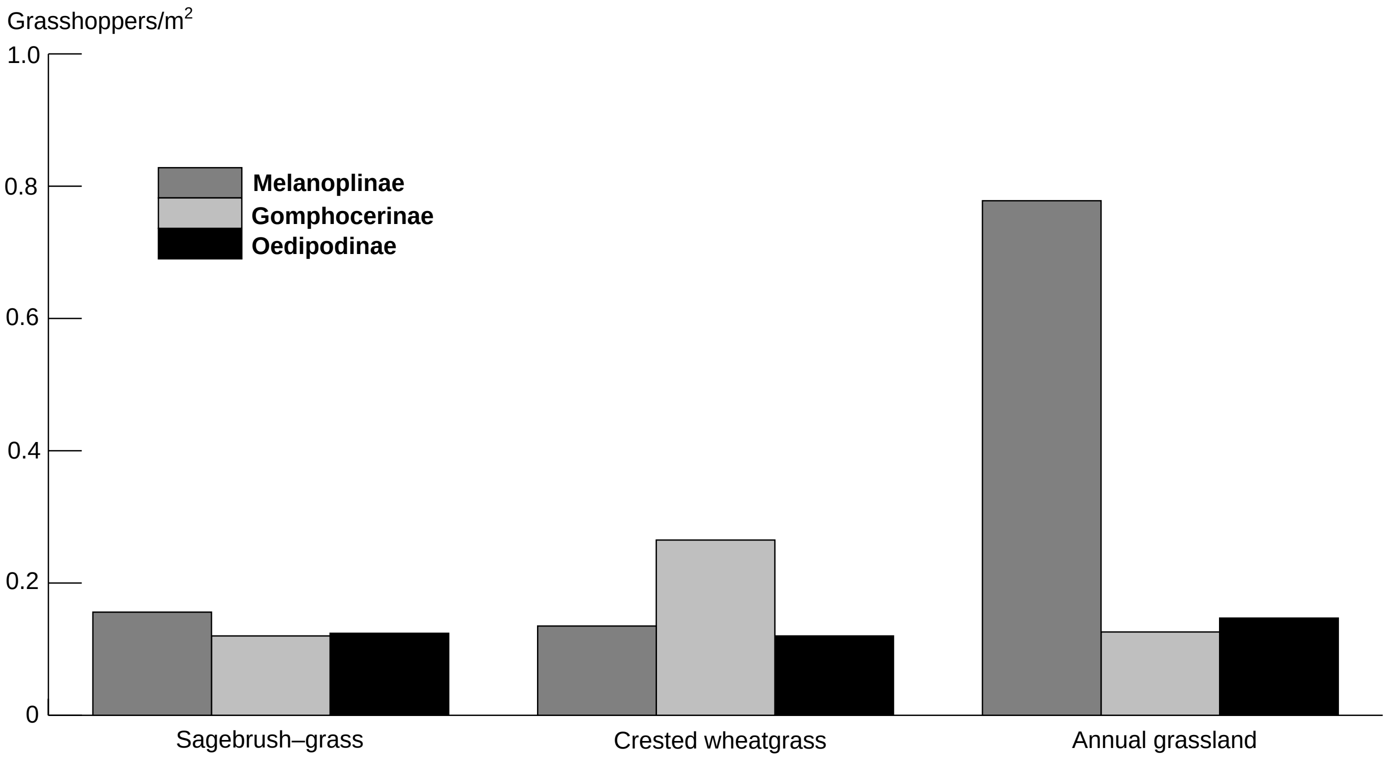

Figure V.3–2—Density distribution of grasshopper subfamilies by grassland sites on the Snake River Plains of southern Idaho.

Although the high cost of rehabilitating annual grasslands may not be justified by reduced grasshopper problems alone, the conversion of annual grasslands to a sagebrush–perennial grass vegetation type is consistent with many other goals of multiple-use management, such as the provision of wildlife habitat, livestock forage, and recreation.

Acknowledgment

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Selected References

Fielding, D. J.; Brusven, M. A. 1993. Spatial analysis of grasshopper density and ecological disturbance in southern Idaho. Agricultural Ecosystem Environment 43: 31–47.

Fielding, D. J.; Brusven, M. A. 1993. Grasshopper (Orthoptera: Acrididae) community composition and ecological disturbance on southern Idaho rangeland. Environmental Entomology 22: 71–81.

Fielding, D. J.; Brusven, M. A. 1994. Grasshopper community responses to shrub loss, annual grasslands, and crested wheatgrass seedings: management implications. In: Monsen, Stephen B.; Kitchen, Stanley G., comps. Proceedings—ecology and management of annual rangelands; 1992 May 18–21; Boise, ID. Gen. Tech. Rep. INT-313. Ogden, UT: U.S. Department of Agriculture, Forest Service, Intermountain Research Station: 162–166.

Tisdale, E. W.; Hironaka, M.; Fosberg, M. A. 1969. The sagebrush region in southern Idaho: a problem in range resource management. Bull. 512. Moscow, ID: University of Idaho, Idaho Agricultural Experiment Station.

Whisenant, S. G. 1990. Changing fire frequencies on Idaho’s Snake River Plains: ecological and management implications. In: McArthur, E. D.; Romney, E. M.; Smith, S. D.; Tueller, P. T., eds. Proceedings of a symposium on cheatgrass invasion, shrub die-off, and other aspects of shrub biology and management. Gen. Tech. Rep. INT-276. Ogden, UT: U.S. Department of Agriculture, Forest Service, Intermountain Forest and Range Experiment Station: 4–10.

V.4 Utilization of Nonnative Rangeland Plants by Grasshoppers on the Snake River Plains of Idaho

Dennis J. Fielding and M. A. Brusven

The Intermountain region of Idaho is highly susceptible to invasions by exotic plant species. At many locations in southern Idaho, exotic plant species comprise 70 to 90 percent of the plant biomass. Cheatgrass *(Bromus tectorum),* medusahead wildrye *(Taeniantherum asper),* knapweeds *(Centaurea* spp.), tumblemustard *(Sisymbrium altissimum),* and Russian-thistle *(Salsola kali)* are widely distributed annual or biennial weeds. Other introduced weeds threatening rangelands in southern Idaho include leafy spurge *(Euphorbia esula)* and rush skeletonweed *(Chondrilla juncea).* The area infested by exotics continues to increase each year. Also, people intentionally have established crested wheatgrass *(Agropyron cristatum),* an exotic perennial bunchgrass, over vast acreages of the Intermountain West.

To learn about the relationship between such exotic plant species and grasshoppers, we investigated the food habits of the most common grasshopper species in southern Idaho. We wanted to gain some insight into the following questions: How palatable are these exotic plant species to native grasshoppers? Do these exotics provide a significant new resource for grasshoppers? Might grasshoppers limit the spread of these new weeds?

We used microscopic analysis of the crops of grasshoppers to learn about their food choices. By examining the contents of a grasshopper's crop under a microscope and comparing the surface characters (hairs, hair structure, arrangement of cells, etc.) of the plant fragments with known reference material, we were able to measure accurately the relative proportion of different plant species and parts of plants (stems, flowers, and leaves) ingested by the grasshopper.

Diffuse Knapweed

Diffuse knapweed *(Centaurea diffusa)* has spread rapidly and widely across Idaho. Knapweeds contain a chemical, cnicin, that is repellent to many herbivores. Concentrations of cnicin vary within the plant: leaves surrounding the flowers have the highest concentrations, and the stem epidermis and flowers have only trace quantities. Because of the unpalatibility of knapweed, infested rangeland has greatly reduced forage value for livestock and wildlife.

We used microscopic analysis to determine the use of diffuse knapweed by the common grasshopper species *Melanoplus sanguinipes.* A spurthroated grasshopper, *M. sanguinipes* is a very opportunistic feeder. Egg hatch in this species often spreads out over a long period, resulting in a highly variable life history. Much of a population of this species typically matures during late summer droughts common in southern Idaho. At such times, most late-maturing plant species that retain some greenness will be a primary food item for *M. sanguinipes.*

Our results showed that *M. sanguinipes* readily consumes knapweed but not in proportion to its availability. The insect prefers other plants, such as cheatgrass and tumblemustard, over knapweed. In late summer, though, when most other plant species are dead, knapweed comprises up to 50 percent of that species' crop contents (table V.4–1). Other plants that are still green then, such as rabbitbrush *(Chrysothamnus* spp.), sagebrush *(Artemesia* spp.), and certain lupine *(Lupinus)* species, also serve as food sources. After autumn rains caused cheatgrass, an exotic annual, to sprout in October, this grass comprised the bulk of *M. sanguinipes'* diet.

Cheatgrass and Crested Wheatgrass

Cheatgrass and another exotic grass species, crested wheatgrass, dominate much of the landscape at lower elevations on the Snake River Plains (figs. V.4–1 and –2). Crested wheatgrass, a perennial bunchgrass, stays green longer in the season than does cheatgrass. We investigated the food habits of *M. sanguinipes* and another common grasshopper species, *Aulocara elliotti*, regarding these two grasses.

A. elliotti, a slantfaced grasshopper, is mostly limited in its diet to grasses but is not selective among grasses. In southern Idaho, populations of *A. elliotti* hatch early and mature at the same time as the grasses on which they feed. In early summer, that species eats crested wheatgrass and cheatgrass equally (table V.4–2). However, as the season progresses and the cheatgrass dries, the diet of *A. elliotti* consists of proportionally greater amounts of crested wheatgrass.

Figure V.4–1—Cheatgrass, an introduced annual grass, can dominate disturbed sites and is widespread across Idaho and in other Pacific Northwest States (Photo by Dennis Fielding, University of Idaho).

Figure V.4–2—Land managers and ranchers often have used crested wheatgrass to reseed areas of Idaho rangeland to enhance forage for livestock and in doing so, sometimes create food sources for pest species of grasshoppers. (U.S. Department of the Interior, Bureau of Land Management photo by Mike Pellant.)

In contrast, *M. sanguinipes* eats mostly cheatgrass in the early summer. As the cheatgrass dries, the insect consumes greater proportions of weedy forbs, such as tumblemustard and Russian-thistle (table V.4–2). Crested wheatgrass did not comprise more than 20 percent of the insect's diet at any time.

Table V.4–1—Crop contents of *M. sanguinipes*, by percentage, on knapweed-infested rangeland east of Jerome, ID, on five different dates in 1989. Grasses were primarily cheatgrass with less than 5 percent western wheatgrass *(Agropyron smithii)*

	June 30	July 20	Aug. 14	Sept. 6	Oct. 13
Diffuse knapweed	18	30	32	55	12
Other forbs	65	48	29	31	1
Grasses	8	13	27	7	86
Litter, detritus	9	9	13	7	1

Note: Percentages may exceed 100 due to rounding.

Table V.4–2—Crop contents of *A. elliotti* and *M. sanguinipes*, by percentage, on a crested wheatgrass seeding north of Bliss, ID, in 1990

	Aulocara elliotti		*Melanoplus sanguinipes*		
	May 18	July 2	May 25	July 9	Aug. 13
Crested wheatgrass	37	75	16	16	19
Cheatgrass	60	17	56	22	17
Forbs	0	7	12	42	50
Litter, detritus	3	2	16	20	14

Note: Percentages may exceed 100 due to rounding.

Conclusions

The manner in which evolutionary history has molded a grasshopper's food habits and other life-history traits decides how a grasshopper will respond to exotic plants. On the Snake River Plains, the most abundant grasshopper species—the ones most likely to achieve outbreak densities—accept a variety of plants and will adapt readily to exotic plant species.

Certain introduced weeds, especially tumblemustard and cheatgrass, may represent a significant new resource for generalist feeders, such as *M. sanguinipes* and *Oedaleonotus enigma.* Rangeland dominated by these plants may provide a more favorable habitat for these grasshoppers, compared to rangeland dominated by native perennial grasses (see section IV, Modeling and Population Dynamics). Less palatable weeds, such as the knapweeds, probably do not provide a significant new resource for native grasshopper populations in southern Idaho; our findings indicate that diffuse knapweed may serve mostly as a survival food during summer droughts.

Our study of grasshopper food habits suggests that land managers should not count on these insects to slow the spread of noxious weeds. While it is conceivable that at high densities grasshoppers may eat large amounts of noxious weeds and reduce seed production, grasshoppers also will eat other plants at the same time, reducing competition to the weeds.

Grasshoppers with specialized feeding habits may offer a better chance of controlling certain weeds. *Hesperotettix viridis*, for example, feeds on broom snakeweed *(Gutierrezia sarothrae).* Such specialist feeders probably would eat mainly native weeds or exotics that are very close relatives of native plants. Specialist feeders would not recognize novel plants as potential food items.

Acknowledgment

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V.5 Local Movement of Grasshoppers Between Public Rangeland and Irrigated Pastures in Southern Idaho

M. A. Brusven, Dennis J. Fielding, Leslie P. Kish, and Russell C. Biggam

On the ground or in flight, grasshoppers have great powers of mobility that allow them to disperse in a way that aids their survival (Dingle 1980, Drake and Farrow 1988, Farrow 1990, Joern 1983, McAnelly and Rankin 1986, Parker et al. 1955). During major outbreak years, ranchers and farmers have noted "clouds" of grasshoppers migrating from one area to another. The exact origin of the migrating grasshopper bands, direction and distance traveled, and the reasons why they disperse are poorly understood for most North American grasshopper species (Riegert et al. 1954, Shotwell 1941). Chapman et al. (1978), Dingle (1972), Southwood (1981), and Uvarov (1977) have given general accounts of insect migration. Laboratory studies have been used to help understand grasshopper flight in confined environments (Riegert 1962).

But the study we conducted is about more localized movement of grasshoppers across the narrow transitions between public rangeland and privately owned, irrigated pastures. A general perception exists that grasshoppers migrate from highly disturbed, overgrazed public rangeland to the more lush, irrigated cropland–pastureland, causing considerable damage to the latter (fig. V.5–1). In southern Idaho, the boundaries between private and public lands, most of which are managed by the U.S. Department of the Interior's Bureau of Land Management (BLM), are long and irregular and usually marked by a fence. Nearly 2 million acres (809,717 ha) make up the BLM Shoshone District. This district is located in the sagebrush–grass ecoregion of southern Idaho. Areas having deeper and more productive soils are largely under private ownership.

The question of whether grasshoppers migrate from public to private land or vice versa and the reasons for localized movements formed the basis for our study. Numerous factors potentially influence the direction and extent of grasshopper migration. Some of these factors include soil moisture; plant composition, height, quality,

Figure V.5–1—An Idaho study provided new information on the belief that grasshoppers migrate from public rangelands to privately owned pastures.

and moisture; vegetative cover; wind velocity; grazing disturbance; predators, inter- and intraspecific competition; grasshopper age and physiological state; and genetically related behavior, such as egg-laying. Our investigations and interpretations were limited to plant cover, composition, moisture content, and height, particularly as they related to grazing of public rangeland and adjacent irrigated pastures.

The Study Area

We studied the lesser migratory grasshopper, *Melanoplus sanguinipes*, and used adults because they display the greatest powers of mobility. The study took place in and adjacent to a 321-acre (130-ha) sprinkler-irrigated pasture bordered on the north, east, and west by BLM rangeland. Studies centered on the west border in 1991 and east border in 1992 to test for directional movements of grasshoppers in response to different rest-rotation grazing regimes, range conditions, prevailing winds, and irrigated pasture conditions.

In 1991, 2 populations of 500 adult *M. sanguinipes* each were differentially marked with fluorescent markers and released in the centers of 2 adjacent 98.4×98.4-ft (30×30-m) plots separated by a fence. The west-side plot was on BLM rangeland that had been rested (not grazed) since the previous year. The east-side plot was on a well-utilized (currently grazed), legume–grass, irrigated pasture. In 1992, 2 populations of 400 grasshoppers each were marked and released in a similar manner, except the plots were on the east side of the irrigated pasture. Again, extensive grazing occurred on the sprinkler-irrigated pasture at the time of the study. Extensive grazing on the BLM pasture during early summer had resulted in a dry, depleted rangeland condition consisting mostly of heavily cropped crested wheatgrass. After releasing marked grasshoppers, we counted them during the night, thus minimizing movement resulting from investigator disturbance. We counted all the marked grasshoppers within the plot borders at 24, 48, and 96 hours after release.

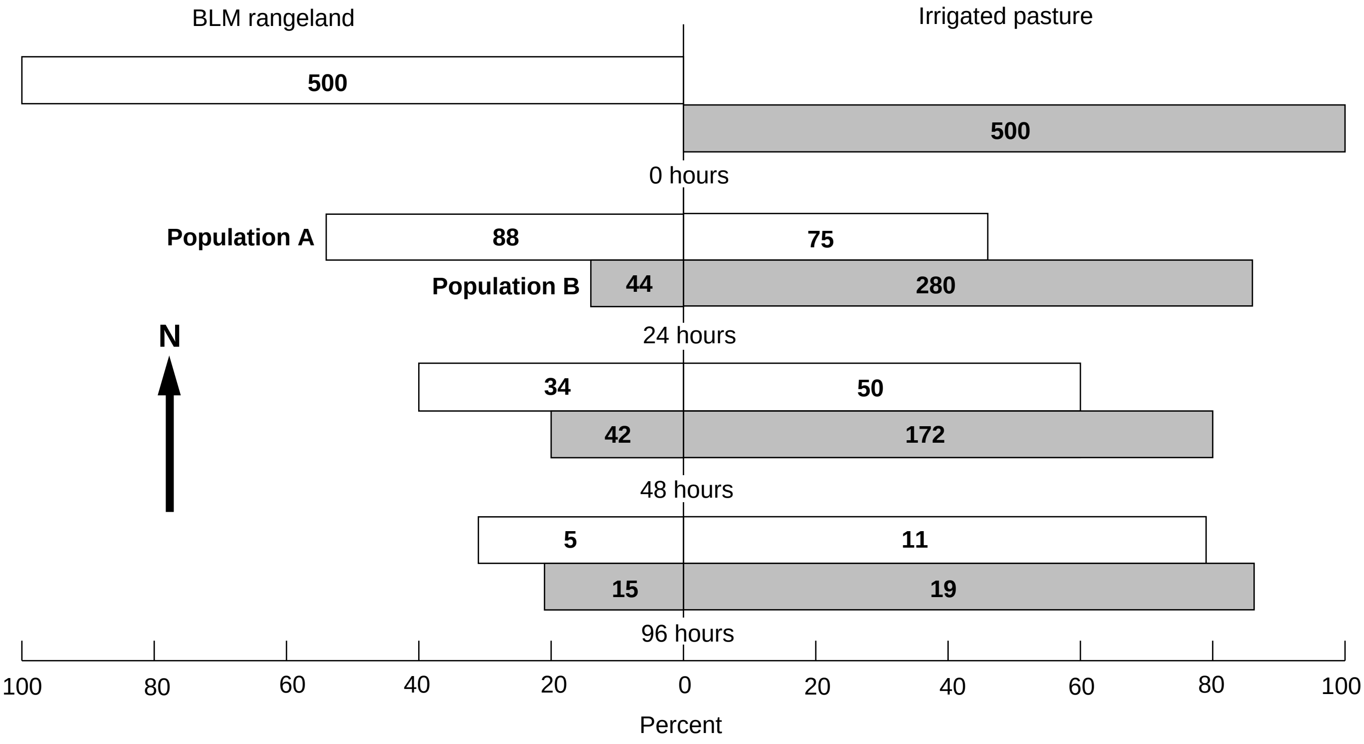

Figure V.5–2—Percent distribution of marked grasshoppers within adjacent rangeland and irrigated pasture plots at 24, 48, and 96 hours after their release, 1991. Numbers inside bars are actual counts.

What We Found

Grasshopper movement between private and public lands differed markedly between the 2 years with regard to “net” directional dispersal. Because the marked populations were not confined to specific plots, the insects’ ultimate movement could be in any direction from the release point and could extend beyond the plot perimeters. For purposes of interpretation, we recorded only marked grasshoppers within adjacent plots. Figures V.5–2 and –3 graph the results on a relative basis (percent of total marked) for each time interval.

In 1991, with prevailing winds from the south to southwest ranging from 6 to 12 miles per hour (mi/hour) (10 to 19 km/hour), net movement of marked populations was easterly from the BLM rangeland to the irrigated pasture (fig. V.5–2). The grasshoppers released in the irrigated pasture showed a much higher affinity for that habitat than grasshoppers released in the BLM plot; however, there was a noticeable presence of grasshoppers from the irrigated pasture in the BLM plot at all times. Only 3 and 5 percent of the marked populations were accounted for in the adjacent plots after 96 hours, indicating a progressive outward dispersal from the release points in all directions.

Because the BLM plot was rested during the spring and summer months preceding the study, the vegetative condition was fair overall, with good plant height and fair cover. The irrigated plot had greater plant cover and moisture content than the BLM plot. Distribution of grasshoppers within the plots correlated significantly with plant height but not with the percent of moisture or cover (bare ground).

In 1992, dispersal patterns were profoundly different from the previous year (fig. V.5–3). Strong, gusty winds from the west and southwest ranged from 14 to 24 mi/hour (23 to 39 km/hour) during the period of study. We recovered only two marked grasshoppers in the heavily grazed BLM plot during the 96-hour test and

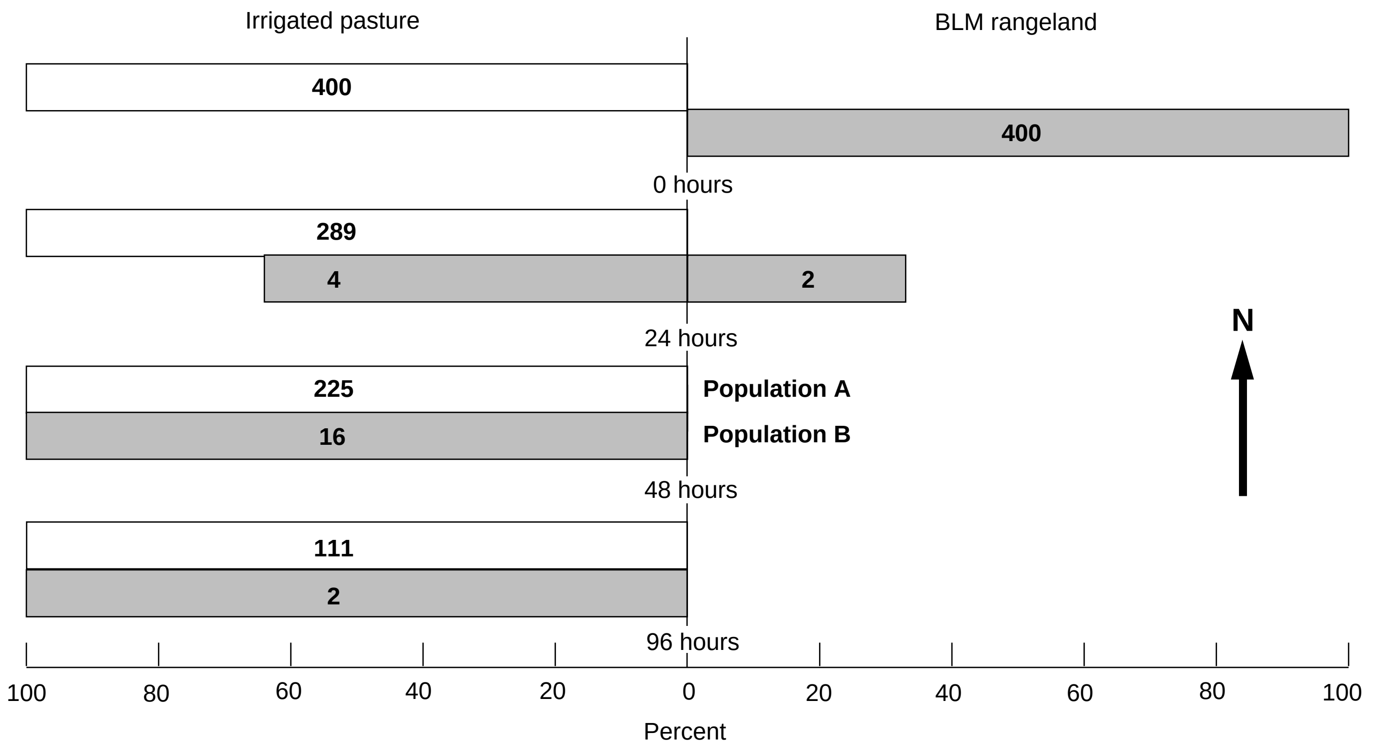

Figure V.5–3—Percent distribution of marked grasshoppers within adjacent BLM rangeland and irrigated pasture plots at 24, 48, and 96 hours after their release, 1992. Numbers inside bars are actual counts.

found 12 of the marked grasshoppers from the BLM plot in the irrigated pasture plot. Conversely, we found no grasshoppers from the irrigated pasture in the BLM plot and noted a very high level of retention of grasshoppers within the irrigated pasture with more than 25 percent still accounted for after 96 hours. The heavily grazed BLM plot was clearly unfavorable to the grasshoppers. Nearly all had moved from the plot within 24 hours or were lost to predation, a factor not readily measurable. The BLM plot was nearly a monoculture of heavily cropped crested wheatgrass. A diversity of weedy forbs was generally absent from the plot, undoubtedly contributing to its objectionable habitat quality for *M. sanguinipes*, which is a mixed feeder preferring forbs.

We believe that strong, westerly to southwesterly, gusty winds aided the dispersal of grasshoppers from the BLM plot in a general downwind direction (northeasterly), even though positive chemical cues were likely coming from the highly diverse, succulent, irrigated pasture to the west. Again, we emphasize the significance of much higher plant height (nearly 3 times greater), plant diversity (mixture of weedy, invasionary plants, grasses, and pasture legumes), and greater vegetative cover (about 2.5 times greater); all are contributing factors to the high retention of grasshoppers in the irrigated pasture compared to the heavily grazed BLM plot, in spite of high, gusty winds.

Conclusions

As to the question of whether grasshoppers migrate from public rangeland to adjacent irrigated pastures, the answer is “not always.” Numerous factors operate individually or together to influence the direction, distance, and magnitude of grasshopper migration. The present study addressed only public rangeland and irrigated pastures. Other types of crops adjoin public rangeland and provide interesting challenges for future studies. A basic axiom of life applies to grasshoppers as with most other mobile organisms on rangeland: When the requirements of survival are limiting (for example, depleted habitat) grasshoppers will migrate, either actively or passively, (wind-aided movement) in search of more favorable habitat conditions.

Acknowledgment

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References Cited

Chapman, R. F.; Cook, A. G.; Mitchell, G. A.; Page, W. W. 1978. Wing dimorphism and flight in *Zonocerus variegatus* (L.) (Orthoptera, Acridoidea). Bulletin of Entomological Research 68: 229–242.

Dingle, H. 1972. Migration strategies of insects. Science 175: 1327–1335.

Dingle, H. 1980. Ecology and evolution of migration. In: Gauthreaux, S. A., ed. Animal migration, orientation and navigation. New York: Academic Press: 1–101.

Drake, V. A.; Farrow, R. A. 1988. The influence of atmospheric structure and motions on insect migration. Annual Review of Entomology 33: 183–210.

Farrow, R. A. 1990. Flight and migration in acridoids. In: Chapman, R. F.; Joern, A., eds. Biology of grasshoppers. New York: John Wiley & Sons: 227–314.

Joern, A. 1983. Small-scale displacements of grasshoppers (Orthoptera, Acrididae) within arid grasslands. Journal of the Kansas Entomological Society 56: 131–139.

McAnelly, M. L.; Rankin, M. A. 1986. Migration in the grasshopper *Melanoplus sanguinipes* (Fab.). I. The capacity for flight in non-swarming populations. Biology Bulletin 170: 368–377.

Parker, J. R.; Newton, R. C.; Shotwell, R. L. 1955. Observations on mass flights and other activities of the migratory grasshopper. Tech. Bull. 110. Washington, DC: U.S. Department of Agriculture.

Riegert, P. W. 1962. Flight of grasshoppers in the laboratory. Nature 194: 1298–1299.

Riegert, P. W.; Fuller, R. A.; Putnam, L. G. 1954. Studies on dispersal of grasshoppers (Acrididae) tagged with phosphorus-32. Canadian Entomologist 5: 223–232.

Shotwell, R. L. 1941. Life histories and habits of some grasshoppers of economic importance on the Great Plains. Tech. Bull. 774. Washington, DC: U.S. Department of Agriculture. 48 p.

Southwood, T.R.E. 1981. Ecological aspects of insect migration. In: Aidly, D. J., ed. Animal migration. London and New York: Cambridge University Press: 196–208.

Uvarov, B. P. 1977. Grasshoppers and locusts, vol. 2. London: Centre for Overseas Pest Research.

V.6 Grazing Effects on Grasshopper Populations in Southern Idaho

Dennis J. Fielding and M. A. Brusven

Many investigators have examined the impact grasshopper populations exert on the availability of forage for livestock. Fewer studies have been done on the reverse relationship: the effects of livestock grazing on grasshoppers. No previous studies have addressed this topic within the Intermountain region of Idaho.

In any discussion of the effects of livestock grazing on grasshoppers, the distinction between long-term and short-term effects of grazing must be maintained. Long-term changes due to grazing may include alterations in the composition of the plant community and changes in soil properties. Short-term changes include reduced forage, altered chemical and physical characteristics of plants, reduced plant height, and possibly a warmer and drier microclimate (see V.1). Only short-term grazing effects will be considered here.

Field Studies

We compared grasshopper densities and species composition between grazed and ungrazed plots from 1990 to 1993. The results have been consistent: we have seen either lower densities on heavily grazed plots or no differences at all. Over the 4-year period, the grazed plots had an average of half as many grasshoppers as the ungrazed plots (fig. V.6–1). One species, *Melanoplus sanguinipes,* accounted for most of the difference in density. The subfamilies Gomphocerinae (slantfaced) and Oedipodinae (bandwinged), as a group, were relatively indifferent to grazing. This does not mean that grazing did not affect certain species within these subfamilies, but densities were too low to evaluate individual species.

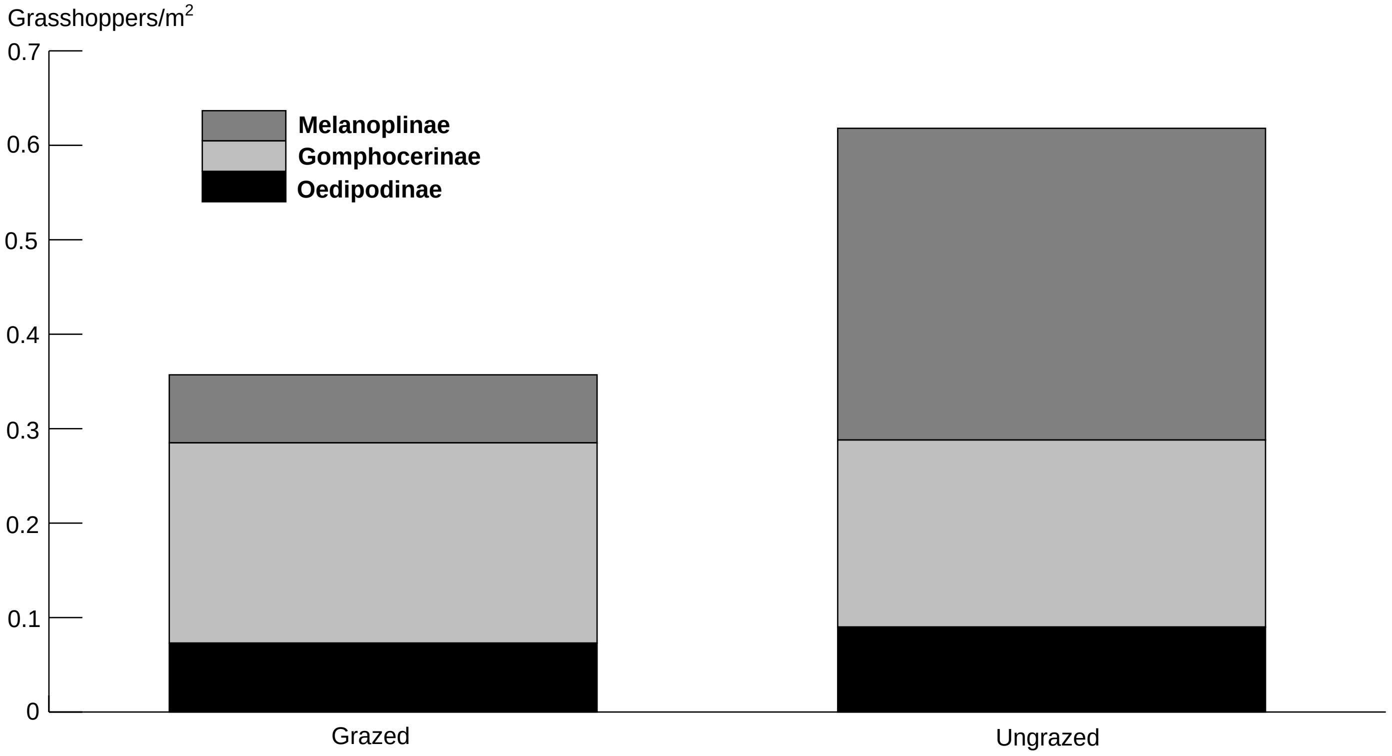

Figure V.6–1—Mean grasshopper densities from five pairs of grazed and ungrazed plots, 1990–93, within the Bureau of Land Management's Shoshone District (N = 3 samples per year × 4 years × 5 plots = 60).

In 1993, a year of above-average precipitation and unusually high rangeland productivity, grazing effects on grasshopper densities were not as pronounced as in other years. These results suggest that by reducing the amount of forage available to grasshoppers, livestock are competing with them and reducing the carrying capacity of the rangeland for grasshoppers. To test this hypothesis under more controlled conditions, we conducted cage studies during 1992 and 1993.

Cage Studies

Cages covering 1 m^2 were set out in an area dominated by cheatgrass *(Bromus tectorum)* and tumblemustard *(Sisymbrium altissimum),* favored habitat of *M. sanguinipes.* We stocked the cages with 10 adult *M. sanguinipes* in July, shortly after adults were first observed in the field. Before we stocked the cages, we clipped half of the aboveground plant biomass (material) and weighed it to the nearest gram in half the cages. We counted grasshoppers within each cage weekly until no grasshoppers survived or until we finished the experiment in October. The remaining plants within the cages were clipped and weighed to the nearest gram after we terminated the experiment, and we sifted the soil to collect any grasshopper egg pods.

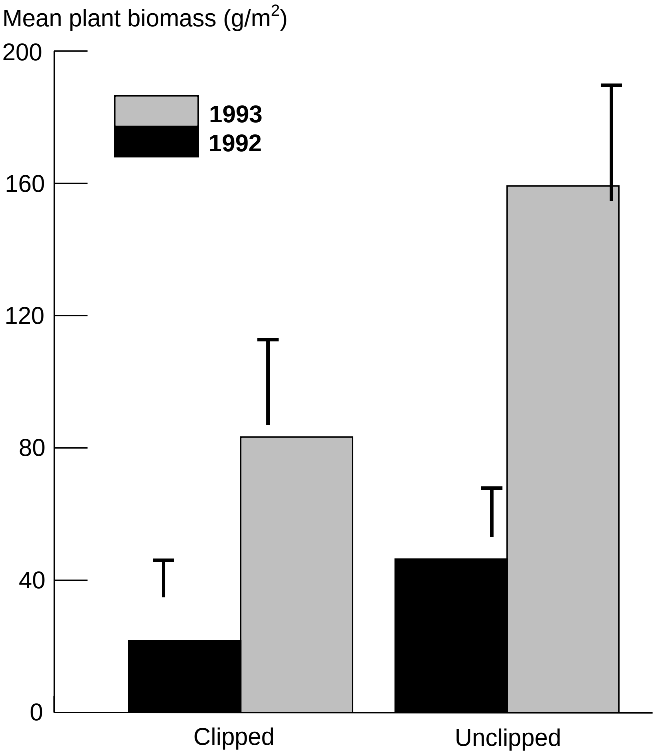

Figure V.6–2—Mean (N = 6 and 9, in 1992 and 1993, respectively) total dry weight of aboveground plant material in clipped and unclipped cages. Error bars indicate 1 standard error of mean. Plants consisted mainly of tumblemustard, cheatgrass, and Sandberg's bluegrass *(Poa sandbergii).*

Abundant precipitation generated much greater plant production in 1993 than the year before (fig. V.6–2). No differences in adult grasshopper survival (measured as total grasshopper-days) occurred between cages of clipped and unclipped plant biomass in either year (fig. V.6–3). However, dramatic differences in fecundity (reproductive capability—measured as eggs per female-day) occurred between the 2 years and between clipped and unclipped cages in 1993 (fig. V.6–4).

These field results suggest that fecundity of *M. sanguinipes* is strongly affected by host plant quality and/or quantity, although adult survival is not. Perhaps maintenance requirements for survival in adults of this species are quite low and can be met by low-quality food, such as dead plant litter. Egg production appears to be much more sensitive to diet.

As the previously cited chapter points out, other factors, besides forage availability, may also play a role in interactions between grazing and grasshoppers. Reduced plant height, increased bare ground, higher temperatures, and lower relative humidity are characteristic of grazed habitats. The behavioral responses of certain grasshopper species to these variables may affect population responses to grazed habitats. For instance, grasshoppers that take refuge in vegetation, such as many slantfaced grasshoppers, may actively seek habitats that provide a greater abundance of refuges, such as ungrazed habitats. Grasshoppers that escape predators by blending in with bare ground, such as many bandwinged species, may be indifferent to grazing-induced habitat changes. These sorts of habitat preferences could explain differing responses to grazing among species.

Concluding Statements

The effects of grazing on rangeland grasshoppers are dependent on so many factors (such as weather and plant community) that generalizations are difficult. Plant responses to grazing depend on the intensity and timing of grazing and the weather. For instance, younger plant tissue is generally more digestible and has higher protein levels than older tissue. In situations where plants can regrow following defoliation, the regrowth may provide higher quality forage for grasshoppers. In dry seasons or climates that do not allow for regrowth, defoliation results in less food, and probably food of lower quality, for grasshoppers. Similarly, the microclimate associated with grazed habitats (warmer and drier) may be beneficial to many grasshopper species during cool, wet spring weather but may be detrimental during summer droughts.

In summary, our observations suggest that livestock grazing often causes a short-term reduction in habitat quality for *M. sanguinipes* in southern Idaho. These observations suggest that grazing could be considered as a management tool for regulating grasshopper populations. However, we are skeptical of the practicality of using livestock grazing as a grasshopper management tool in southern Idaho. Rangeland productivity and the consequent carrying capacity for grasshoppers vary greatly from year to year within the Intermountain region. Livestock numbers are not flexible enough to permit land managers to respond to extreme fluctuations in carrying capacity of rangeland and grasshopper populations. During years of above-normal precipitation and high biomass productivity, grasshopper populations can increase tremendously. Grazing levels would have to be doubled or tripled to inhibit grasshopper reproduction appreciably.

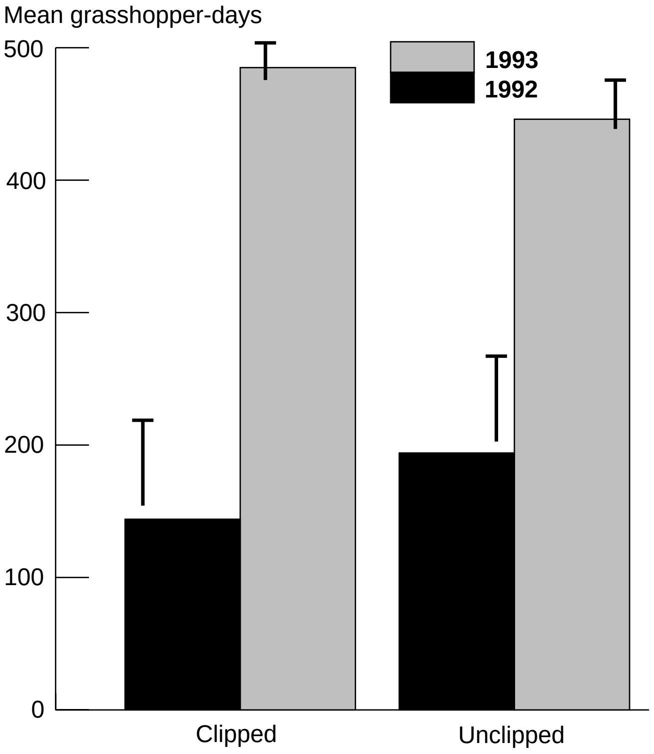

Figure V.6–3—Mean (N = 6 and 9, in 1992 and 1993, respectively) survival of adult grasshoppers *(Melanoplus sanguinipes)* within 1-m^2 cages. Error bars indicate 1 standard error of mean.

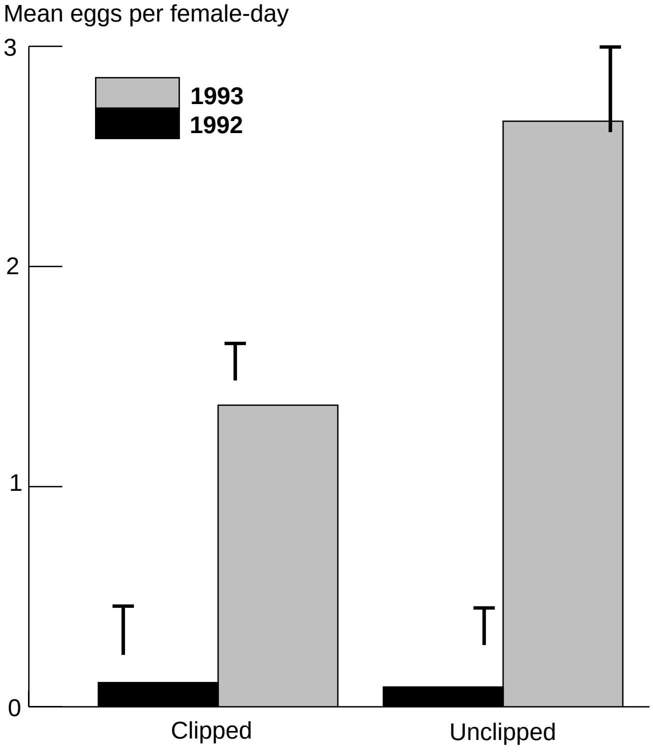

Figure V.6–4—Mean (N = 6 and 9, in 1992 and 1993, respectively) fecundity of female *Melanoplus sanquinipes* within 1-m^2 cages. Error bars indicate 1 standard error of mean.

Assuming that managers could increase livestock grazing to a point where it would reduce grasshopper populations, such levels of grazing could produce negative long-term effects. Chronic, heavy grazing could lead to long-term changes in vegetation toward more of the weedy annuals (fig. V.6–5) that promote high densities of pest grasshopper species (see V.3).

We expect grazing to have the greatest effect on grasshopper populations during drought episodes, when grasshopper populations are already low (see V.2). Under such conditions, grazing potentially could reduce already low grasshopper densities to the point of affecting creatures, such as nesting birds, that depend on grasshoppers for food. (For more information, see chapter I.9, “Birds and Wildlife as Grasshopper Predators.”)

The sustainable level of livestock grazing on public rangelands is an issue that is receiving increased scrutiny. Managers need information regarding ecosystem responses to grazing to manage rangeland resources properly. Presently, knowledge about grazing effects on grasshoppers is fragmentary and incomplete. These issues involve economics, politics, sociology, ecology, and environmental ethics. The full integration and balancing of these considerations leave fertile ground for more holistic studies in the future.

Figure V.6–5—Grazing can produce negative effects on rangeland by removing understory grasses and creating an opportunity for weedy annuals.

V.7 Beneficial Changes of Rangeland Through Proper Grazing

Llewellyn L. Manske

Introduction

Grassland ecosystems are diverse and complex, a fact that makes developing management recommendations difficult. However, increasing knowledge of ecological principles and the intricacies of the numerous mechanisms that function in the grassland ecosystem have allowed for improvements in management strategies.

Several greenhouse and laboratory studies within the last 10 to 12 years have enabled scientists to begin to understand how grassland plants react to being grazed. Grassland plants and herbivores have evolved together for 20 million years. During this long period of coevolution, grassland plants have developed mechanisms to compensate for defoliation from herbivores and fire. These adaptive tolerance mechanisms can be separated into two main categories, but they do not function independently. The first mechanism involves numerous changes in the physiological growth processes within the grassland plant itself, and the second involves numerous changes in the activity levels of the symbiotic (mutually beneficial) soil organisms in the rhizosphere, which is the narrow zone of soil around perennial plant roots.

The physiological responses within the plant caused by defoliation have been reviewed and grouped into nine categories by McNaughton (1983). Physiological responses to defoliation do not occur at all times, and the intensity of the response varies. Grass plants have different physiological responses at various stages of growth. The key to ecological management by defoliation is to match the timing of defoliation events to the appropriate stage of growth that triggers the desired outcome.

All possible combinations of relationships between the physiological responses and the application of the defoliation-management treatment have not yet been quantitatively evaluated with scientific research. One of the main physiological effects of defoliation is the temporary reduction in the production of the blockage hormone auxin in young, developing leaves and within the meristem (the growth point where tissue is formed by cell division).

The reduction of plant auxin in the lead tiller allows either for the synthesis of cytokinin (a growth hormone) in the roots or crown or its utilization in axillary buds, which are growth points with potential to develop into vegetative tillers, resulting in the production of new plants (Murphy and Briske 1992). Partial defoliation of young leaf material reduces the hormonal effects of apical dominance (hormonal suppression of growth of other buds by the lead tiller) and allows secondary tillers to develop from the previous year’s axillary buds. Secondary tillers can develop without defoliation manipulation after the lead tiller has reached the flowering growth stage. Usually, only one secondary tiller develops from the potential of five to eight buds because this secondary tiller also suppresses additional axillary bud development hormonally. When the lead tiller is partially defoliated between the third leaf stage and flowering, several axillary buds can develop subsequently into secondary tillers. No single secondary tiller is apparently capable of developing complete hormonal apical dominance following defoliation of the lead tiller at that time. Some level of hormonal control from the older axillary buds still suppresses development of some of the younger axillary buds. This mechanism is not completely understood, and scientists have not been able to manipulate the hormone levels so that all of the axillary buds develop into secondary tillers.

Besides encouraging grassland plants to tiller, defoliation also stimulates soil organism activity in the rhizosphere. The rhizosphere is that narrow zone of soil around living roots of perennial grassland plants where the exudation (leakage) of materials like sugars, amino acids, glycosides, and other compounds affects micro-organism activity. Bacterial growth in the rhizosphere is stimulated by the presence of carbon from the exuded material (Elliott 1978, Anderson et al. 1981). Protozoa and nematodes graze increasingly on the multiplying bacteria and accelerate the overall nutrient cycling process through the “fast” pathway of substrate decomposition proposed by Coleman et al. (1983). The activity of the microbes in the rhizosphere increases the amount of nitrogen available for plant growth (Ingham et al. 1985, Clarholm 1985). The presence of mycorrhizal fungi (those that live in association with plants) enhances the absorption of ammonia, phosphorus, other mineral nutrients, and water.

Rhizosphere activity can be manipulated by defoliation at early growth stages, when a higher percentage of the total nitrogen of the plant is in the aboveground parts and a higher percentage of the total carbon of the plant is in the belowground parts. At that time, partial defoliation disrupts the plant's relationship of carbon to nitrogen, leaving a relatively high level of carbon in the remaining plant. Some of this carbon is exuded through the roots into the rhizosphere in order to readjust the carbon–nitrogen ratio.

Because of limited access to simple carbon chains, bacteria in the rhizosphere are restricted in growth and activity levels under conditions when defoliation is absent. When defoliation management is used, rhizosphere bacteria increase in activity in response to the increase in exuded carbon. The increases in activity by the bacteria trigger increases in activity levels in the other micro-organisms that make up the nutritional food chain of the rhizosphere. These increases in activity levels ultimately increase available nutrients for the defoliated grass plant. The relationship between grassland plants and organisms in the rhizosphere is truly symbiotic with both entities receiving benefit from their association.

Rhizosphere activity can be stimulated by disrupting the carbon–nitrogen ratio through plant defoliation at early growth stages. During middle and late growth, carbon and nitrogen are distributed fairly evenly throughout the plant, and at these stages defoliation does not remove a disproportionate amount of nitrogen, and very little or no carbon is exuded into the rhizosphere. Also, water levels in the soil generally decrease during the middle and late portions of the grazing season and limit the activity levels of rhizosphere organisms.

The adaptive tolerance mechanisms that pertain to the changes in physiological growth processes within grassland plants, and to the changes in activity levels of the symbiotic organisms in the rhizosphere following defoliation, are the key to understanding the manipulation of beneficial effects from defoliation management under field conditions. Manipulation of these mechanisms by defoliation is also key to the development of ecologically sound recommendations for management of North America's grassland natural resources. Contributions to the development of biological and ecological foundations for grazing management recommendations were major goals of a research project developed to study the ecological effects of defoliation at the Dickinson Research Center in western North Dakota from 1984 to 1992. This study was expanded in 1990 to include sites in McKenzie County, ND.

The objectives of this study were to evaluate changes in plant-exuded material, soil organism activity and biomass, tiller development of grass plants, aboveground and belowground plant biomass, and livestock weight performance among twice-over rotation-grazing treatments, a 4.5-month seasonlong treatment, a 4-month deferred seasonlong treatment, a 6-month seasonlong treatment, and a long-term nongrazed treatment.

The Study Area and Methods

The long-term study site is located 20 miles north of Dickinson in southwestern North Dakota (lat. 47°14' N., long. 102°50' W.) on the Dickinson Research Center operated by North Dakota State University. The McKenzie County sites are located 21 miles west of Watford City (between lat. 47°35' and 47°50' N. and long. 104°00' and 103°45' W.) in the McKenzie County Grazing District of the Little Missouri National Grassland. The National Grassland is administered by the U.S. Department of Agriculture's Forest Service and managed in cooperation with the McKenzie County Grazing Association.

Soils are primarily dark in color and developed under grassland vegetation having cool, continental climate and moderate moisture levels. Average annual precipitation is 14 inches (356 mm) with 80 percent falling as rain between April and September. Temperatures in summer average 66 °F (19 °C) with average daily maximums of 80 °F (27 °C). Winter average daily temperatures are 13 °F (–11 °C) with average daily minimums of 2 °F (–17 °C). The vegetation is the wheatgrass–needlegrass type (Barker and Whitman 1988) of the mixed-grass prairie. The dominant native range species are western wheatgrass *(Agropyron smithii)*, needle-and-thread *(Stipa comata)*, blue grama *(Bouteloua gracilis)*, and threadleaved sedge *(Carex filifolia)*.

The treatments on native range were organized as a paired-plot design with two replications. The twice-over rotation grazing treatments at the Dickinson Research Center had three pastures with each grazed for 15 days between June 1 and July 15 and for 30 days after mid-July and prior to mid-October for a total of 4.5 months. Three seasonlong treatments were used: a 4.5-month seasonlong grazing between mid-June to early November, a 4-month deferred seasonlong grazing between mid-July to mid-November, and a 6-month seasonlong grazing between mid-May and mid-November. The long-term nongrazed treatment areas had not been grazed, mowed, or burned for more than 30 years prior to the start of data collection.

The McKenzie County sites had two grazing treatments. The rotation-grazing treatment had four pastures with each grazed for two periods. The other treatment had a traditional seasonlong grazing method. A long-term nongrazed exclosure was available for nondestructive sampling of control sites. Commercial crossbred cattle were used on all treatments in this trial.

Each of the treatments was stratified on the basis of three range sites (sandy, shallow, and silty). Samples from the grazed treatments were collected on both grazed and ungrazed (protected with cages) quadrats (plots). Aboveground plant biomass was collected on seven sampling dates from May to October. Belowground plant biomass and soil micro-organism data were collected on four sampling periods. Aboveground and belowground net primary productivity (NPP) were determined by methods outlined by Sala et al. (1981) and Bohm (1979), respectively. The major components sampled were live material (by species), standing dead material, and litter.

Plant materials were analyzed for nutrient content using standard procedures (Association of Official Analytical Chemists 1984). Plant species composition was determined between mid-July and mid-August using the 10-pin point frame method as described by Cook and Stubbendieck (1986). Root exudates were determined using procedures outlined by Haller and Stolp (1985). A standard paired-plot t-test (Mosteller and Rourke 1973) was used to analyze differences between means.

Individual animals were weighed on and off each treatment and on each rotation date. Mean weights of cows and calves were adjusted to the 8th and 23d day of each month of the grazing period. Biweekly live-weight performance periods of average daily gain and accumulated weight gain for cows and calves were used to evaluate each treatment. Response surface analysis (Kerlinger and Pedhazur 1973) with a repeated observation design was used to compare animal response curves among treatments and was reported by Manske et al. (1988).

Findings

Percent basal cover of grasses increased 25 percent (from 15 percent to 19 percent basal cover) on the rotation-grazing treatments compared to seasonlong treatments (table V.7–1). Basal cover of sedges and forbs decreased by 4 percent and 36 percent, respectively, on the rotation treatments compared to seasonlong treatments. Plant community relative percent composition changed, with grasses increasing by 14 percent, sedges decreasing by 14 percent, and forbs plus shrubs decreasing by 40 percent, on the rotation treatments compared to seasonlong treatments (table V.7–2).

The amount of herbage that remained standing on September 1 after the rotation treatments was greater than the amount of total current-year’s growth on the long-term nongrazed treatments (table V.7–3). These data do not account for the amount of vegetation removed by livestock on the rotation treatments. During the entire grazing season, an average of 15 percent more herbage biomass was standing after each grazing period on the rotation treatments compared to long-term nongrazed treatments. Seasonlong treatments averaged 8 percent and 29 percent less herbage biomass standing after grazing than on the nongrazed and rotation treatments, respectively. The relatively greater amount of photosynthetic leaf area remaining on the rotation treatments at the end of the grazing season was beneficial for the continued development of the grassland ecosystem at a higher production level. This remaining herbage also provided a benefit as wildlife habitat.

Tiller development of grass plants and the resulting increase in aboveground herbage biomass were greater on the rotation treatments than on the nongrazed and seasonlong treatments. These increases in the vegetation suggest that removal by defoliation of some young leaf material early in the growth cycle has some effect on the

Table V.7–1—Mean percent basal cover, by vegetative growth form categories

	Treatments		
	Season-long	Rotation	Percent difference
Grass	14.7	18.6	+25.2
Sedge	7.7	7.6	–3.8
Forb	3.8	2.4	35.9
Shrub	0.1	0.1	—

Table V.7–2—Mean relative percent composition of plant communities

	Treatments		
	Season-long	Rotation	Percent difference
Grass	55.1	63.2	+14.1
Sedge	30.6	28.0	–13.6
Forb and shrub	14.5	8.7	–39.6

Table V.7–3—Mean monthly aboveground herbage biomass, in pounds per acre, remaining after grazing on three range sites

	Monthly sample periods				
Treatments	1June	1July	1Aug.	1Sept.	1Oct.
Nongrazed	822*a*	1,010*a*	1,144*a*	888*a*	—
Seasonlong	974*a*	1,017*a*	785*b*	717*a*	—
Rotation	990*a*	1,211*b*	1,231*a*	993*b*	987

Means of same column followed by the same letter are not significantly different (P<0.05).

reduction of auxin and the subsequent stimulation of cytokinin, which causes axillary buds to develop into secondary tillers. Thus, defoliation of grass plants at an early growth stage exerts beneficial effects on vegetative tiller development.

Preliminary interpretation of the rhizosphere data collected so far indicates that greater amounts of exuded material were released into the rhizosphere on the rotation treatments than on nongrazed or seasonlong treatments. These data also indicate that the biomass of soil mites was greater on the rotation treatments compared to the nongrazed or seasonlong treatments. This information suggests that removal of some young leaf material by defoliation at early growth stages has some effect on increasing exuded material, which in turn presumably stimulates activity of the bacteria. Greater bacterial activity stimulates activity of subsequent organisms in the nutritional food chain of the rhizosphere. Activity levels were increased in protozoa, nematodes, and mites. Increasing the activity levels of organisms in the rhizosphere increases the amount of nitrogen available for plant growth. Thus, defoliation of grass plants at an early growth stage has beneficial effects on symbiotic rhizosphere organism activity and results in greater amounts of nutrients available for growth by those plants.

The period when defoliation of grass plants showed beneficial effects on the increases in vegetative tillers and symbiotic rhizosphere organism activity occurred between the third leaf stage and the flowering period during this study.

The increase in grass tiller development and symbiotic rhizosphere activity on the twice-over rotation treatments allowed a mean increase in stocking rate of 40 percent greater than on the 4.5-month seasonlong treatments, 96 percent greater than on 6-month seasonlong treatments, and 9 percent greater than the 4-month deferred seasonlong treatments.

Accumulated weight performance of individual cows and calves (table V.7–4), their average daily gain (table V.7–5), and weight gain per acre (table V.7–6), were greater on the rotation treatments compared to the seasonlong and deferred seasonlong treatments. Weight performance of cows and calves on the three grazing treatments was

Table V.7–4—Mean annual accumulated weight gain in pounds for cows and calves

	Treatments		
	Deferred season-long	Season-long	Rotation
	Pounds		
Cows	34	40	107
Calves	204	284	309

Table V.7–5—Mean annual average daily weight gain in pounds for cows and calves

	Treatments		
	Deferred season-long	Season-long	Rotation
	Pounds		
Cows	0.32*a*	0.34*a*	0.62*b*
Calves	1.80*a*	2.09*a*	2.21*b*

Means of same row followed by the same letter are not significantly different (P<0.05).

Table V.7–6—Mean annual weight gain in pounds per acre for cows and calves

	Treatments		
	Deferred season-long	Season-long	Rotation
	Pounds per acre		
Cows	2.6*a*	2.9*a*	8.1*b*
Calves	20.4*a*	20.5*a*	28.5*b*

Means of same row followed by the same letter are not significantly different (P<0.05).

generally not significantly different during the first grazing period of June and July. During the second grazing period, after early August, the animal weight performance on the rotation treatments was significantly greater than on the seasonlong and deferred seasonlong treatments (Manske et al. 1988). Individual animal performance improved on the twice-over rotation-grazing system with an increase in calf average daily gain of 6 percent greater than 4.5-month seasonlong and 23 percent greater than deferred seasonlong grazing treatments. Average daily weight gain of cows improved on the twice-over rotation system by 82 percent greater than 4.5-month seasonlong and 94 percent greater than deferred seasonlong grazing treatments.

The combination of increases in stocking rate and individual animal performance gave the twice-over rotation system a considerable increase in animal weight gain per acre over the other grazing treatments. Calf weight gain per acre on the twice-over rotation system was 39 percent greater than 4.5-month seasonlong and 40 percent greater than deferred seasonlong treatments. Cow weight gain per acre on the twice-over rotation system was 179 percent greater than 4.5-month seasonlong and 212 percent greater than deferred seasonlong grazing treatments.

The improved livestock weight performance during the later portion of the grazing season on the rotation treatments was primarily attributed to the increase in available nutrients from the addition of secondary tillers. These tillers had developed from axillary buds and were at an early growth stage during the second rotation period. Generally, the available herbage on the rotation treatments was 1.5 and 2.5 percentage points greater in crude protein content than the herbage on the seasonlong and deferred seasonlong treatments during the later portion of the grazing season.

The grassland plant community can be changed beneficially when grazing defoliation is properly timed to coincide with the appropriate growth stage of the grass plants (fig. V.7–1). Grass plant density is increased, and total herbage production is increased when defoliation by grazing is timed to occur between the third leaf stage and the flowering stage. A greater amount of vegetation can remain at the end of the grazing season, which causes a noticeable change in the vegetation canopy cover. There

Figure V.7–1—Land managers and ranchers can create beneficial changes on rangeland by using proper and timely grazing systems. Changes in turn can affect the habitat for some grasshopper species, offering another possible tool for long-term grasshopper management.

is a decrease in the amount of bare ground present in the pastures. These changes in plant structure and density should be unfavorable for most troublesome rangeland grasshopper species. Most rangeland pest grasshopper species are favored by open vegetation canopy and bare areas. These open areas in the vegetation structure are used by the grasshoppers to provide access to solar radiation during nymphal development for body temperature regulation and by some species for egg-laying sites.

Grassland areas that have higher percentages of open canopy should have relatively higher grasshopper populations. Grassland areas that have had beneficial changes in the structure and density of the vegetation as a result of the manipulation of the adaptive tolerance mechanisms of the grass plants by the twice-over rotation treatment should show negative effects on grasshopper populations. The changes in vegetation structure and density should lower air and soil temperatures, raise relative humidity, and reduce the level of irradiation within the grasshopper microhabitat. These changes in grasshopper microhabitat should lengthen the time required for nymphal development, exposing the nymphs to numerous causes of death, which would raise the average daily mortality rate and reduce the density of individuals. Lowering the number of nymphs will reduce the number of grasshoppers that develop into adults. This, in turn, will reduce the number of eggs laid. All of these factors should cause an overall reduction in the population of grasshoppers on grassland areas managed with twice-over rotation treatments.

The other characteristic of the twice-over rotation treatment that would negatively affect grasshopper populations is that the sequence of grazing periods on the rotation-system pastures is never the same in consecutive years. This variation should alter the vegetation growth patterns enough so that no single pest grasshopper species would consistently be favored.

Conclusions

Additional research would help quantify exuded material, soil organism activity and biomass, axillary bud development into tillers, and nitrogen, carbon, and phosphorus cyclic flows. These additional findings would allow scientists to understand more completely the adaptive tolerance mechanisms developed by grassland plants to compensate for defoliation. Grassland managers then could manipulate these mechanisms more precisely and be able to use the beneficial defoliation effects on a finer level and further improve the grassland ecosystem. Additional research also needs to document relationships between the changes in vegetation structure and density and the effects on grasshopper population dynamics.

Data collected to date have shown that defoliation of grass plants between the third leaf stage and flowering stage has beneficial effects on the physiological responses within the plant. These effects allow for greater tiller development and beneficial effects on the symbiotic rhizosphere organism activity, which is believed to increase the amount of nitrogen available for plant growth. Deliberate and precise manipulation of these adaptive tolerance mechanisms can increase secondary tiller development and total herbage biomass. The secondary tillers increase the nutrient content of the herbage, and that increase enhances individual animal weight performance during the latter portion of the grazing season.

The increase in herbage biomass permits an increase in stocking rate and leaves a greater amount of herbage after grazing. This increase in residual herbage is beneficial for grassland wildlife habitat. Plant density, canopy cover, and litter cover increase as a result of increased tiller growth, which in turn, reduces the impact of raindrops, reduces and slows runoff, reduces erosion, and increases water infiltration. These improvements in the vegetation density and canopy cover should have negative impacts on grasshopper populations. Grazing management recommendations of systematically rotating 7- to 15-day periods of defoliation between the third leaf stage and flowering growth stage (June 1–July 15 in western North Dakota) on each pasture should maximize beneficial effects on the adaptive tolerance mechanisms of grassland plants.

References Cited

Anderson, R. V.; Coleman, D. C.; Cole, C. V.; Elliott, E. T. 1981. Effect of nematodes *Acrobeloides* sp. and *Mesodiplogaster iheritieri* on substrate utilization and nitrogen and phosphorus mineralization. Ecology 62: 549–555.

Association of Official Analytical Chemists. 1984. Official methods of analysis (14th ed.). Washington, DC: Association of Official Analytical Chemists.

Barker, W. T.; Whitman, W. C. 1988. Vegetation of the northern Great Plains. Rangelands 10: 266–272.

Bohm, W. 1979. Methods of studying root systems. Berlin: Springer-Verlag.

Clarholm, M. 1985. Interactions of bacteria, protozoa, and plants leading to mineralization of soil nitrogen. Soil Biology and Biochemistry 17: 181–187.

Coleman, C. D.; Reid, C.P.P.; Cole. C. V. 1983. Biological strategies of nutrient cycling in soil ecosystems. Advancements in Ecological Research 13: 1–55.

Cook, C. W.; Stubbendieck, J. 1986. Range research: basic problems and techniques. Denver, CO: Society of Range Management. 317 p.

Elliot, E. T. 1978. Carbon, nitrogen, and phosphorus transformations in gnotobiotic soil microcosms. M.S. thesis. Ft. Collins, CO: Colorado State University.

Haller, T.; Stolp, H. 1985. Quantitative estimation of root exudation in maize plants. Plant and Soil 86: 207–216.

Ingham, R. E.; Trofymow, J. A.; Ingham, E. R.; Coleman, D. C. 1985. Interactions of bacteria, fungi, and their nematode grazers: effects on nutrient cycling and plant growth. Ecological Monographs 55: 119–140.

Kerlinger, F. N.; Pedhazur, E. J. 1973. Multiple regression in behavioral research. New York: Holt, Rinehart, and Winston.

Manske, L. L.; Biondini, M. E.; Kirby, D. R.; Nelson, J. L.; Landblom, D. G.; Sjursen, P. J. 1988. Cow and calf performance on seasonlong and twice-over rotation grazing treatments in western North Dakota. In: North Dakota cow/calf conference; [month and dates unknown] 1988; Bismarck, ND. Bismarck, ND: North Dakota Cow/Calf Conference: 5–17.

McNaughton, S. J. 1983. Compensatory plant growth as a response to herbivory. Oikos 40: 329–336.

Mosteller, F.; Rourke, R.E.K. 1973. Sturdy statistics. [City of publication unknown], MA: Addison–Wesley Publishing Co. 395 p.

Murphy, J. S.; Briske, D. D. 1992. Regulation of tillering by apical dominance: chronology, interpretive value, and current perspectives. Journal of Range Management 45: 419–429.

Sala, O. E.; Deregibus, V. A.; Schlichter, T.; Alippe, H. 1981. Productivity dynamics of native temperate grassland in Argentina. Journal of Range Management 34: 41–48.

V.8 Herbage Production, Phenology, and Soil Moisture Dynamics for Plant Communities in Western North Dakota

Daniel W. Uresk and Ardell J. Bjugstad

Increasing demand for intensive management of rangelands requires improved methodologies for classifications, descriptions, and monitoring of plant communities. It is important to document vegetation characteristics of plant communities for a reference point in order to determine how herbivory (the consumption of all or part of a plant by consumers, including cattle, wildlife, insects, etc.) affects vegetation composition and production, insects, and wildlife. An understanding of plant characteristics (production, species composition, canopy cover, phenology, degree of utilization by grazers, and abiotic factors) is important for correlation with grasshopper populations and their dynamics. Knowledge gained from the plant component will be useful in determining grasshopper relationships with vegetation characteristics. Previous vegetation studies describing habitat types and communities in western North Dakota have been limited to subjective evaluations (Hanson and Whitman 1938, Redmann 1975, Lauenroth and Whitman 1977, Hansen et al. 1984, Hansen and Hoffman 1988).

Phenology is the study of the relationship between seasonal climatic changes and plant development. Knowledge of the seasonal timing of flowering events (phenological phases) is useful information for resource managers. This information can be used to determine when to graze livestock on native pastures (Frank and Hofmann 1989), when to burn for enhancement and/or control of plant growth, and when to implement insect control measures (Hewitt 1980, Kemp et al. 1991).

An understanding of soil moisture regimes for native plant communities on the northern Great Plains is basic for improvement of rangeland productivity and development of ecological management practices for each community. On the northern Great Plains, soil moisture is one of the major factors that influence plant growth. Soil types and other factors, including plant composition, plant production, litter, grazing, rocks, and soil nutrients, influence the soil moisture (Rauzi 1960, Smika et al. 1961, Houston 1965, Goetz 1975, Cline et al. 1977, Benkobi et al. 1993). Models have been developed for plant growth at individual or homogeneous (similar) sites as related to soil moisture, precipitation, and temperature (Uresk et al. 1975 and 1979, Wight and Hanks 1981, Wight et al. 1986). However, over large areas, successful attempts to model soil moisture and plant growth relationships have been limited (Rauzi 1960). For additional information, see Branson et al. (1981) for an excellent overview of rangeland hydrology.

The objectives of this study were (1) to classify and describe plant communities quantitively by species using canopy cover, frequency of occurrence, production, and utilization of plants by herbivores in western North Dakota over a 5-year period, (2) to identify the most useful plant species for discriminating, classifying, and monitoring plant communities, (3) to provide information on phenological (growth) development for 10 native plant species, and (4) to determine seasonal trends in soil moisture for native plant communities throughout the study area.

Study Area

The study area was located on the Little Missouri National Grassland and privately owned rangelands in western North Dakota. Climate is semiarid and continental, characterized by long, cold winters and short, warm summers. The coldest month is January with an average low of 10.5 °F (–11.6 °C) and the monthly high for July is 71.6 °F (22 °C). Most of the precipitation falls as rain in early summer. Approximately 75 percent of the precipitation falls during April through September (Hansen et al. 1984, Hansen and Hoffman 1988). Yearly precipitation totals over the 5-year period for four sites within the study area are presented in table V.8–1. Vegetation is dominated by western wheatgrass *(Agropyron smithii)*, blue grama *(Bouteloua gracilis)*, needle-and-thread *(Stipa comata)*, and scarlet globemallow *(Sphaeralcea coccinea)*, with scattered dwarf sagebrush *(Artemisia cana)* and fringed sagebrush *(A. frigida)* (fig. V.8–1).

Study Methods

Plant Communities.—In all, 30 sites were selected throughout the Little Missouri National Grassland. Vegetative characteristics sampled included canopy cover and frequency of occurrence by species (Daubenmire 1959) and plants harvested at peak production. Twenty sites were sampled in 1987–88, and an additional 10 sites were added in 1989. All 30 sites were sampled in 1989–91. Each site had four replicated areas. Three transects were randomly located on each of the four replicates at each site from 1987–89. Sample size estimates for num-

Table V.8–1—Yearly precipitation, in inches, over a 5-year period for four U.S. weather service stations within the western North Dakota study area

Year	Watford City	Trotter's store	Fairfield store	Medora
1987	11	12	13	18
1988	[1]9	[1]6	8	9
1989	14	12	[1]15	[1]13
1990	11	11	12	11
1991	18	18	19	13
Average[2]			16	15

[1] Incomplete or missing data for the year.

[2] Thirty-year station average; similar measurements are not available for Watford City or Trotter's store.

Figure V.8–1—Rangelands support a variety of plant communities. Understanding how plant communities function is important for increasing knowledge about how grasshoppers interact with those communities.

ber of transects and quadrats (plots) were then determined, and for 1990–91, two 98.4-ft (30-m) permanent transects were located at random on each of the replicated sites. Canopy cover and frequency of occurrence by species were estimated at 1-m intervals within 7.9-×19.7-inch (20-×50-cm) frames along each transect (Daubenmire 1959). Data were summarized as means by site for all analyses.

Classification of Communities.—Plant communities were classified and defined by plant canopy cover and frequency of occurrence collected on the 30 sites for 1990 and 1991. Canopy cover times frequency of occurrence (index) of the 10 major plant species were subjected to data reduction (Uresk 1990) and cluster analyses (ISODATA) to determine groupings of similar plant communities (Ball and Hall 1967). Original data reductions to define the 10 major plant species were based on Soil Conservation Service range site classifications. Stepwise discriminant analysis was used to estimate the compactness of the clusters, to identify key variables that accounted for community differences, and to develop Fisher classification coefficients (Uresk 1990). Plant production estimates and utilization were summarized by plant communities.

Plant Production and Utilization.—Plant biomass at time of peak production was determined by harvesting all plants inside cages. The difference between plant biomass harvested inside and outside the cages is expressed as utilization by herbivores. Each site was comprised of four replicated areas. Prior to initiation of spring growth, 10 wire cages measuring 3.3×6.6 ft (1×2 m) were randomly located on each of the 4 replicates for a total of 40 cages/site. Plants were harvested at ground level inside each cage within one 2.69-ft^2 (0.25-m^2) randomly placed circular hoop and sorted by grasses (sedges were included in this category), forbs, and shrubs. Approximately 10–20 ft (3–6 m) from the cages, six 7.9-×19.7-inch (20-×50-cm) quadrats were harvested on each of three transects. In 1990, 5 of the 1.08-ft^2 (0.1-m^2) quadrats were harvested on each of 2 transects/replicate for a total of 10 quadrats. During 1991, a total of 10 2.69-ft^2 (0.25-m^2) circular hoops were harvested along the 2 transects. All plant material was oven dried at 140 °F (60 °C) for 48 hours and weighed to the nearest 0.1 g. Weights were expressed as a mean per site in pounds per acre.

Phenology.—Phenological development was divided into five stages: (1) vegetative, (2) flowering, (3) seed set, (4) seed drop, and (5) dormancy (Sauer and Uresk 1976). Biweekly measurements of 10 plant species were made to determine the timing of developmental stages (phenophases). For each species, 40 plants/site were randomly selected within each of 30 sites in 1989 and were monitored from mid-May through mid-August. In 1990 and 1991, 2 plants of each of the 10 species were located within each of the 4 replicates on each site for a total of 240 plants/species/year. The individual plants were permanently marked with flags in late April on each of 30 sites and were monitored through September. Although some plant species were not found on all sites, a minimum of 192 individual plants was evaluated. New plants were selected each year. Data were summarized for all sites for each of 3 years.

Soil Moisture.—In all, 28 of the study sites were sampled: 18 in 1987–88 and an additional 10 in 1989–91. At each site, four replicates were systematically selected and sampled for soil moisture at a 12-inch (30-cm) depth. On each replicate, three 40-inch soil-moisture access tubes were randomly installed in early June 1987. Neutron soil moisture probes were used and recalibrated each year. Soil samples were collected at the time of installation to determine gravimetric soil moisture. Regression analyses permitted calibration of actual gravimetric soil moisture with estimated soil moisture at each site with value converted to volume percent. Data were summarized as means per site and summarized by plant community.

Results of the Study

Plant Communities.—Cluster analyses on cover and frequency of native plant species separated the 30 sites into 4 native plant community types. Discriminant analyses indicated significant separation (P=0.001) among the plant communities. Five plant species—western wheatgrass, dwarf sagebrush, blue grama, threadleaf sedge *(Carex filifolia)* and needle-and-thread were required to separate the four native communities. The five species accounted for 97 percent of the total variation in three canonical discriminant functions.

These five major plant species were used in the discriminant procedure in SPSS/PC (1990) to develop Fisher classification coefficients to predict the four plant communities (table V.8–2). Needle-and-thread had a greater weighting for community 1, dwarf sagebrush had a greater weighting for communities 2 and 3, and western wheatgrass, a greater weighting for community 4. Based on substitution error rates in SPSS/PC (1990), the four plant communities could be classified with 96-percent accuracy given just these five species.

The four plant communities are (1) needle-and-thread/blue grama/threadleaf sedge, (2) blue grama/western wheatgrass/needle-and-thread, (3) dwarf sagebrush/blue grama/western wheatgrass, and (4) western wheatgrass/blue grama/needle-and-thread. Two additional plant communities with limited sample sizes were defined in this study but not included in the above analyses. These are (5) crested wheatgrass *(Agropyron cristatum)* and (6) dwarf sagebrush/leafy spurge *(Euphorbia esula)*. The sites for each plant community by number and name are listed in table V.8–3.

Table V.8–2—Fisher classification coefficients for plant communities in western North Dakota

	Plant community			
Plant	1	2	3	4
Western wheatgrass	0.00145	0.00306	0.00384	0.00649
Dwarf sagebrush	0.00561	0.01048	0.01443	0.00417
Blue grama	0.00203	0.00649	0.00494	0.00285
Threadleaf sedge	0.00637	–0.00049	–0.00059	0.00006
Needle-and-thread	0.01095	0.00360	0.00234	0.00152
Constant	–17.48374	–17.82723	–14.53323	–13.43716

1. Needle-and-Thread/Blue Grama/Threadleaf Sedge Community.—This plant community is dominated by needle-and-thread (table V.8–4). Canopy cover for this species ranged, over a 5-year period, from 18 to 39 percent. Blue grama is the second most abundant grasslike, with canopy cover that varied from 10 to 22 percent. It was followed by threadleaf sedge, which extended from 7 to 20 percent over the 5-year period. Western wheatgrass is common in this plant community, with an overall average cover of 8 percent. Dwarf sagebrush is present only in trace amounts.

Total plant production estimated inside cages ranged from 584 lb/acre in 1988 to 1,165 lb/acre in 1991 (table V.8–5). Grasses and sedges comprised a major portion of the production in this plant community and ranged from 532 to 1,026 lb/acre. Forb production was variable and extended from 49 to 276 lb/acre. Shrubs were not dominant in this plant community; production varied from 3 to 20 lb/acre.

The difference between plant production estimated inside and outside cages (utilization) over the 5-year period is shown in figure V.8–2. In 1987, no forage utilization was evident. Utilization from 1988 to 1991 averaged 12 percent when sampled at the peak of the growing season in July.

2. Blue Grama/Western Wheatgrass/Needle-and-Thread Community.—This plant community was dominated by blue grama followed by western wheatgrass and needle-and-thread (table V.8–4). Canopy cover for blue grama ranged from 21 to 60 percent over a 5-year period. Canopy cover varied from 7 to 19 percent for western wheatgrass and from 5 to 13 percent for needle-and-thread during this study. Threadleaf sedge averaged 5 percent over the 5-year period. Dwarf sagebrush was present in only trace amounts.

Table V.8–3—List of Grasshopper Integrated Pest Management Project sites and identification number sampled, 1987–91, by plant communities in western North Dakota

1. Needle-and-thread/blue grama/threadleaf sedge

7	101-Exclosure
15	East Twin Butte (natural)
16	Buffalo Gap
19	Dantz Creek
20	Van-Vig Ranch
22	Flat Top Butte
24	Charbonneau Creek
28	Road 881

2. Blue grama/western wheatgrass/needle-and-thread

1	Tobacco Garden
2	Lone Beaver
3	Christ Springs
4	Bear Butte
5	Horse Creek
10	Grassy Butte
11	Devils Pass
18	Kinley Plateau
23	Valley Enclosure
26	French Creek
29	Klandl Springs
30	Bartall Creek

3. Dwarf sagebrush/blue grama/western wheatgrass

8	Prairie Dog Enclosure
9	Little Beicegal
13	Government Creek
17	Tracy Mountain
21	Icebox Canyon

4. Western wheatgrass/blue grama/needle-and-thread

12	Whitetail Creek
25	Bowline Creek
27	Cheney Creek

5. Crested wheatgrass

6	Crested wheatgrass
31	East Twin (crested wheatgrass)

6. Dwarf sagebrush/leafy spurge

14	Wannagan Creek

Table V.8–4—Mean canopy cover (percent) ± standard error for key species, by plant community and year (n=number of sites)

	1987	1988	1989	1990	1991
1. Needle-and-thread/blue grama/threadleaf sedge					
	n=5	n=5	n=8	n=8	n=8
Western wheatgrass	7.6 ± 1.4	6.9 ± 1.8	6.7 ± 1.7	5.6 ± 1.4	17.3 ± 3.3
Blue grama	13.6 ± 3.7	16.5 ± 4.2	10.0 ± 2.4	9.5 ± 2.2	22.2 ± 5.0
Threadleaf sedge	6.7 ± 2.7	11.3 ± 4.6	12.1 ± 4.2	7.3 ± 2.0	19.8 ± 8.0
Needle-and-thread	27.0 ± 8.4	17.5 ± 3.9	19.1 ± 3.6	20.3 ± 5.2	39.2 ± 6.0
Dwarf sagebrush	0.2 ± 0.2	0.1 ± 0.1	0.1 ± 0.1	0.2 ± 0.1	0.4 ± 0.2
2. Blue grama/western wheatgrass/needle-and-thread					
	n=8	n=8	n=12	n=12	n=12
Western wheatgrass	8.4 ± 2.1	6.6 ± 1.4	9.5 ± 1.5	10.1 ± 1.1	19.0 ± 3.5
Blue grama	29.5 ± 2.1	24.8 ± 2.4	21.3 ± 2.2	32.3 ± 2.3	59.9 ± 3.2
Threadleaf sedge	2.1 ± 0.7	2.0 ± 0.7	4.3 ± 1.2	2.8 ± 0.6	5.0 ± 1.4
Needle-and-thread	5.6 ± 1.2	4.9 ± 1.1	4.7 ± 1.0	6.0 ± 1.5	12.5 ± 2.8
Dwarf sagebrush	0.0 ± 0.0	0.0 ± 0.0	0.0 ± 0.0	0.0 ± 0.0	0.0 ± 0.0
3. Dwarf sagebrush/blue grama/western wheatgrass					
	n=4	n=4	n=5	n=5	n=5
Western wheatgrass	19.7 ± 5.8	11.0 ± 2.1	15.4 ± 2.8	13.9 ± 2.0	26.4 ± 3.8
Blue grama	15.4 ± 3.9	20.0 ± 5.0	19.1 ± 3.7	19.8 ± 5.6	38.9 ± 8.3
Threadleaf sedge	0.2 ± 0.1	0.7 ± 0.4	1.1 ± 1.0	0.5 ± 0.3	0.3 ± 0.1
Needle-and-thread	7.1 ± 1.0	5.3 ± 1.8	3.7 ± 1.0	4.1 ± 1.5	7.7 ± 3.6
Dwarf sagebrush	10.0 ± 5.3	6.4 ± 3.6	9.6 ± 3.8	9.2 ± 3.3	13.5 ± 4.9
4. Western wheatgrass/blue grama/needle-and-thread					
	n=1	n=1	n=3	n=3	n=3
Western wheatgrass	14.2	9.3	20.2 ± 3.5	20.2 ± 4.9	41.5 ± 6.5
Blue grama	24.4	37.3	14.7 ± 3.7	10.2 ± 2.8	33.5 ± 8.7
Threadleaf sedge	1.7	0.4	4.4 ± 2.5	1.1 ± 0.5	1.0 ± 0.7
Needle-and-thread	2.3	0.8	5.0 ± 2.4	6.2 ± 2.7	11.3 ± 5.8
Dwarf sagebrush	0.0	0.0	0.9 ± 0.8	0.3 ± 0.2	0.5 ± 0.4

Table V.8–5—Plant production, in total and by grasses, forbs, and shrubs (in lb/acre), over a 5-year period for six plant communities (mean ± standard error)

	1987	1988	1989	1990	1991
1. Needle-and-thread/blue grama/threadleaf sedge					
	n=5	n=5	n=8	n=8	n=8
Total	1,165 ± 96	584 ± 86	1,042 ± 102	1,113 ± 139	1,159 ± 127
Grasses	959 ± 119	532 ± 92	747 ± 75	896 ± 89	1,026 ± 120
Forbs	208 ± 40	49 ± 10	276 ± 84	207 ± 64	118 ± 24
Shrubs	7 ± 4	3 ± 2	20 ± 7	10 ± 5	16 ± 8
2. Blue grama/western wheatgrass/needle-and-thread					
	n=8	n=8	n=12	n=12	n=12
Total	984 ± 60	449 ± 18	889 ± 64	1,021 ± 57	1,144 ± 85
Grasses	733 ± 63	372 ± 19	530 ± 4	826 ± 63	1,019 ± 78
Forbs	249 ± 41	77 ± 15	351 ± 59	194 ± 28	122 ± 22
Shrubs	2 ± 1	<0.1	9 ± 4	1 ± 1	3 ± 1
3. Dwarf sagebrush/blue grama/western wheatgrass					
	n=4	n=4	n=5	n=5	n=5
Total	1,604 ± 244	401 ± 62	1,320 ± 108	1,157 ± 115	1,140 ± 112
Grasses	1,210 ± 195	334 ± 56	853 ± 98	860 ± 80	986 ± 105
Forbs	179 ± 61	30 ± 11	279 ± 96	148 ± 60	72 ± 17
Shrubs	216 ± 107	38 ± 20	289 ± 141	148 ± 73	82 ± 57
4. Western wheatgrass/blue grama/needle-and-thread					
	n=1	n=1	n=3	n=3	n=3
Total	1,271	513	1,332 ± 278	1,167 ± 183	1,308 ± 226
Grasses	878	452	825 ± 148	895 ± 112	1,154 ± 182
Forbs	390	46	459 ± 146	260 ± 96	91 ± 29
Shrubs	3	16	47 ± 45	12 ± 9	63 ± 57
5. Crested wheatgrass					
	n=2	n=2	n=2	n=2	n=2
Total	292 ± 69	391 ± 62	1,170 ± 17	1,167 ± 62	1,366 ± 249
Grasses	1,056 ± 11	377 ± 55	1,120 ± 0	1,091 ± 121	1,316 ± 285
Forbs	101 ± 51	22 ± 1	46 ± 15	72 ± 55	45 ± 30
Shrubs	16 ± 7	5 ± 5	4 ± 3	5 ± 4	6 ± 5
6. Dwarf sagebrush/leafy spurge					
	n=1		n=1	n=1	n=1
Total	2,503		2,089	1,660	2,242
Grasses	197		207	333	182
Forbs	2,055		1,405	127	1,893
Shrubs	251		477	309	168

This community was the least productive of the four major types: total plant production fluctuated from 449 to 1,144 lb/acre over the 5-year period (table V.8–5). Total production of grasses and sedges showed a range of 372 to 1,019 lb/acre. Forbs were less productive and varied from 77 to 351 lb/acre. Shrub production was very limited and averaged 3 lb/acre.

Plant production and herbivore utilization for this plant community is presented in figure V.8–3. Forage used by herbivores during the first 2 years of the study was nominal. However, plant utilization increased the last 3 years from 18 to 28 percent in July.

3. Dwarf Sagebrush/Blue Grama/Western Wheatgrass Community.—Blue grama was the dominant understory grass in this community (table V.8–4). It ranged from a low of 15 percent to a high of 39 percent canopy cover. This was followed by western wheatgrass, which varied from 11 to 26 percent cover. Dwarf sagebrush was the dominant overstory plant with canopy cover values that ranged from 6 to 14 percent over the 5-year period. Needle-and-thread averaged 6 percent canopy cover. Least abundant was threadleaf sedge, which averaged less than 1 percent cover.

Figure V.8–2—Comparison of plant biomass in July over a 5-year period on ungrazed (inside cages) with grazed (outside of cages) habitats.

Figure V.8–3—Comparison of plant biomass in July over a 5-year period on ungrazed (inside cages) with grazed (outside of cages) habitats.

Total production ranged from 401 to 1,604 lb/acre over the 5-year period (table V.8–5). Production of grasses and sedges varied from 334 to 1,210 lb/acre. Grass and sedge production was followed by forbs with 30 to 279 lb/acre, and shrubs, with 38 to 289 lb/acre. Shrub production was greater in this community than in the other three native plant communities.

Plant utilization was nominal in light of total production estimates throughout the 5 years; however, estimates of dwarf sagebrush production were highly variable and masked utilization of grasses (and sedges) and forbs. Herbivore utilization of grasses (and sedges) and forbs is comparable to utilization in other plant communities. Utilization of grasses (and sedges) and forbs was minimal during the first 2 years (fig. V.8–4). Greatest utilization of plants occurred in 1991.

4. Western Wheatgrass/Blue Grama/Needle-and-Thread Community.—Western wheatgrass was the dominant plant species in this community (table V.8–4). Canopy cover ranged from 9 to 42 percent over the 5-year period. Western wheatgrass was followed by blue grama, which ranged from 10 to 37 percent. Needle-and-thread expressed itself less (less than 1 percent cover) during the drier years early in the study; however, when more moisture was available for growth during the last 3 years, canopy cover reached a high of 11 percent. Threadleaf sedge averaged approximately 2 percent cover over the 5-year period, and dwarf sagebrush was present only in trace amounts.

Total plant production on this community ranged from 513 to 1,332 lb/acre over the 5 years (table V.8–5). Grasses and sedges showed similar trends among years, with production varying from 452 to 1,154 lb/acre. Forb production showed a range from 46 to 459 lb/acre over the study period. Shrubs were a minor component and averaged only 28 lb/acre.

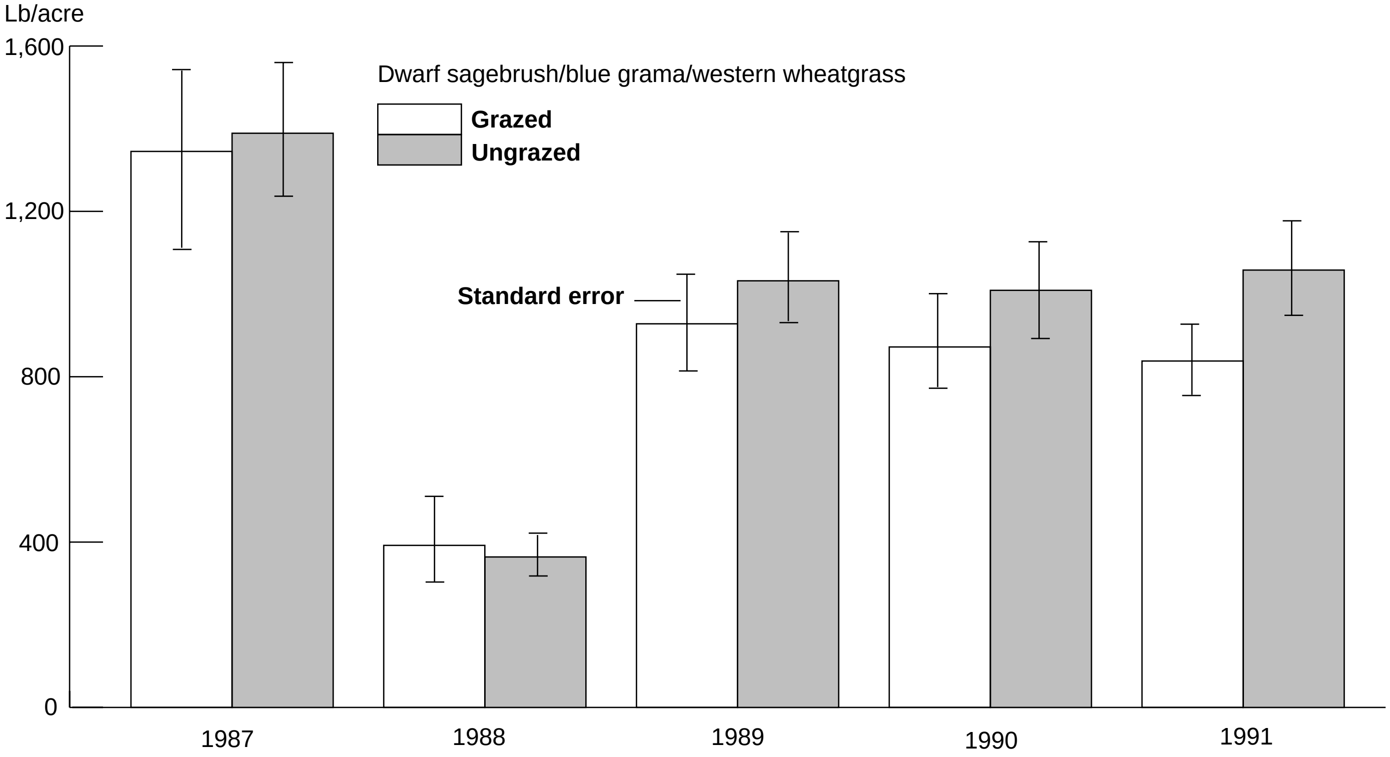

Figure V.8–4—Comparison of plant biomass in July over a 5-year period on ungrazed (inside cages) with grazed (outside of cages) habitats. Shrubs are excluded from this comparison.

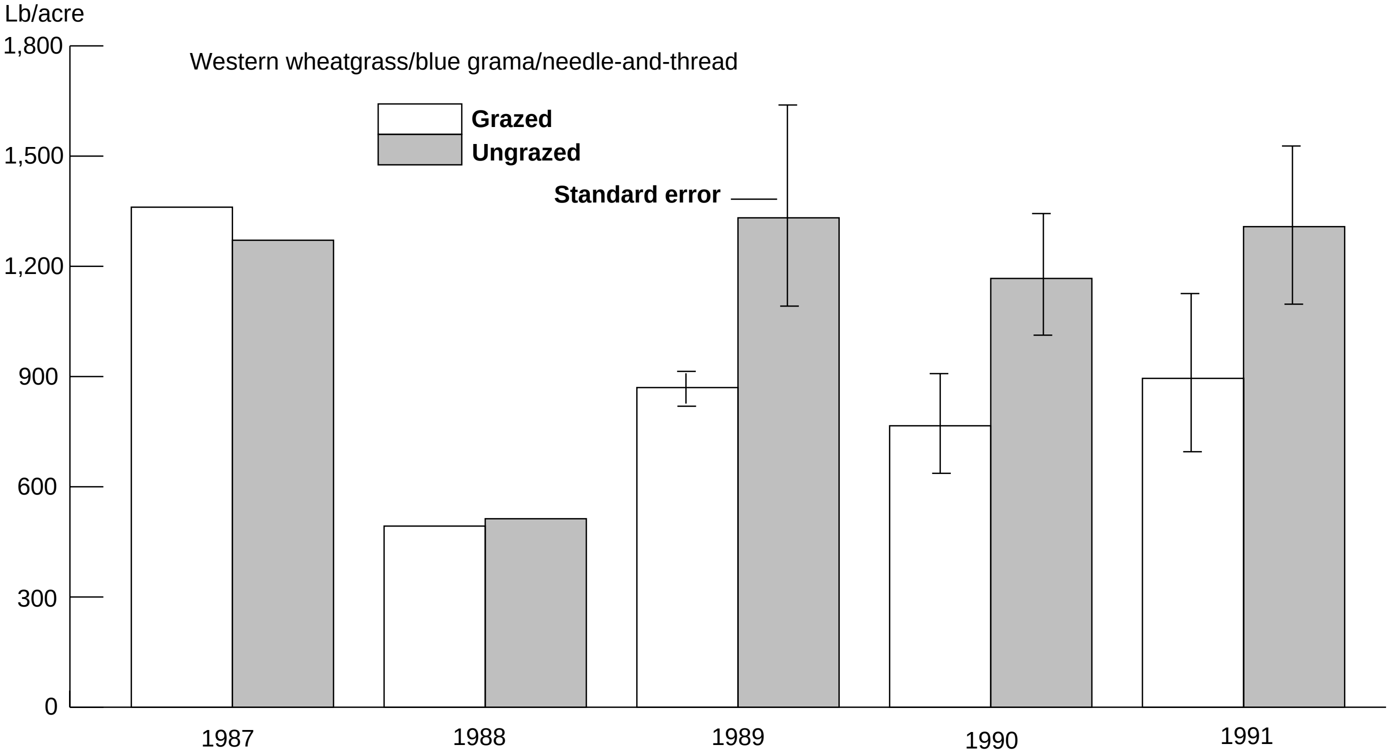

Figure V.8–5—Comparison of plant biomass in July over a 5-year period on ungrazed (inside cages) with grazed (outside of cages) habitats. For the years 1987 and 1988, n=1.

Limited sample size constrained estimates of plant utilization the first 2 years on this community (fig. V.8–5). Nominal utilization occurred in the latter 3 years. Total plant production was low during the first 2 years, but from 1989 to 1991, plant production and utilization were greater. Utilization of forage was similar during the last 3 years, averaging 34 percent. Of the four native plant communities, this one showed the greatest use by herbivores.

5. Crested Wheatgrass Community.**—**This plant community, represented by two sites, was dominated by the non-native crested wheatgrass, whose canopy cover ranged from 33 to 72 percent. Needle-and-thread was the next most dominant grass, ranging from 5 to 11 percent canopy cover.

The community had been seeded to crested wheatgrass, and total plant production was less variable among years (table V.8–5). Total production for this community ranged from 391 to 1,366 lb/acre. Grass and sedge production, primarily crested wheatgrass, varied from 377 to 1,316 lb/acre. Forbs ranged in production from 22 to 101 lb/acre. Shrubs were a minor component in the community at 7 lb/acre.

Utilization of crested wheatgrass was nominal and variable throughout the study (fig. V.8–6). Livestock generally use crested wheatgrass early in the spring before native plants start to grow and then switch to native species as they turn green.

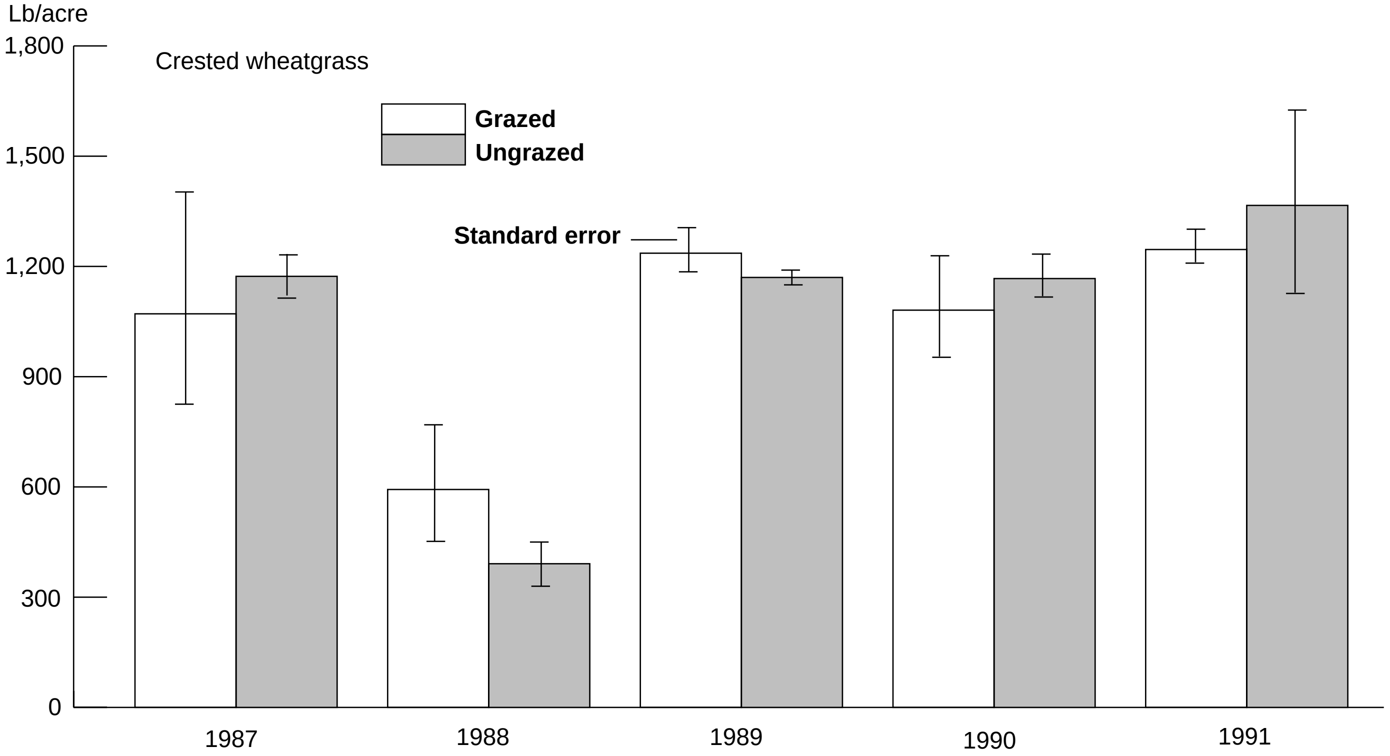

Figure V.8–6—Comparisons of plant biomass in July over a 5-year period on ungrazed (inside cages) with grazed (outside cages) habitats.

***6. Dwarf Sagebrush/Leafy Spurge Community.*—**Only one site dominated by leafy spurge was sampled throughout the 5-year period; its total understory canopy cover ranged from 50 to 77 percent. Dwarf sagebrush was the dominant shrub, and canopy cover varied from 5 to 11 percent. Some western wheatgrass (2 percent) and needle-and-thread (3 percent) was present. Total production over a 4-year period averaged 2,123 lb/acre, with forbs averaging 1,593 lb/acre, shrubs 301 lb/acre, and grasses and sedges 229 lb/acre. Plant utilization was not determined.

Phenology.—Phenological progression through the three seasons for each species is shown in figure V.8–7. These species vary in growth form and include a woody shrub, perennial grasses, *Carex* species, and a forb. The 10 species differed in the timing of their development among years. Western wheatgrass was in a vegetative stage throughout 1991; however, this plant completed all phases of development in 1990. In 1989, when other species were flowering early, fringed sagebrush remained in the vegetative state through the first week in August, when sampling was terminated. Needle-and-thread and green needlegrass *(Stipa viridula)* were similar in phenological development for all 3 years. Blue grama, junegrass *(Koeleria pyramidata),* and Sandberg's bluegrass *(Poa sandbergii)* varied greatly among years in phenophases. Both threadleaf sedge *(Carex filifolia)* and needleleaf sedge *(C. eleocharis)* initiated flowering and seed-set early in the growing season, although length of flowering varied among years. Scarlet globemallow, a forb, flowered longer in 1989 than in the other 2 years; however, other phases of development were similar among all years. In 1991, most species entered the dormancy phase 2–4 weeks later than in 1989–90.

Extensive examination with multivariate analyses, regressions, and correlations of developmental phases through the season for the 10 plant species in our study produced no relationships with degree days, soil moisture, air temperatures, soil temperatures, or precipitation.

Soil Moisture.—Seasonal and yearly amounts of soil moisture are presented in figure V.8–8 and table V.8–6. Overall, soil moisture varied among years. When considering spring moisture available for plant growth, 1988 was the driest year and 1989 the wettest (fig. V.8–9). All years exhibited seasonal variation in soil moisture content among the four native plant communities. Generally, western wheatgrass/blue grama/needle-and-thread community (type 4) was the most moist of the four plant communities (fig. V.8–8). The driest was generally needle-and-thread/blue grama/threadleaf sedge (type 1). Soil moisture trends throughout the growing seasons differed among years. Usually soil moisture decreased on all plant communities as the growing season progressed. Soil moisture and plant production were very low in 1988. Early spring moisture content ranged from 9 to 15 percent among the four plant communities. Years showing greater amounts of soil moisture early in the growing season also showed greater plant production.

Discussion

Plant Community Classification.—The procedures developed in this study to define and classify native plant communities by methods outlined by Uresk (1990) used cover-frequency index for grouping plant communities. Individual plant communities are homogeneous, with minimal variance within each of the communities. Discriminant analyses allowed for identification of groups of variables (species) that collectively were important in separating the major communities.

Five species accounted for most of the variation (97 percent) in separating the four native plant communities in western North Dakota. The plant communities were quantitatively identified with an estimated 96 percent predictability, based on cover-frequency estimates for western wheatgrass, blue grama, threadleaf sedge, needle-and-thread, and dwarf sagebrush. Variation in species composition on a site can be used by resource managers to classify plant communities once canopy cover and frequency-of-occurrence data are collected.

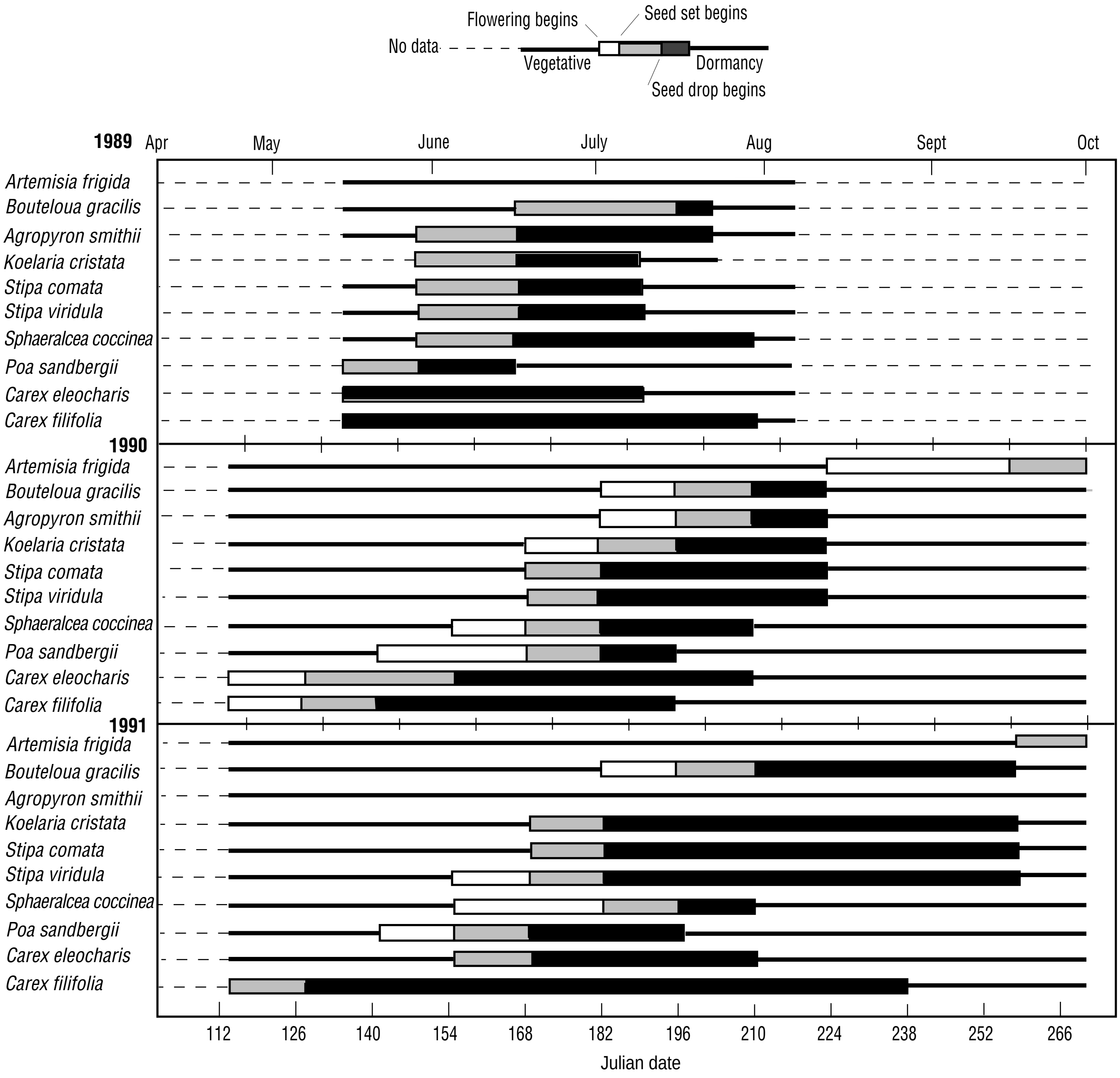

Figure V.8–7—Phenological development for 10 plant species over the 1989–91 growing seasons in western North Dakota.

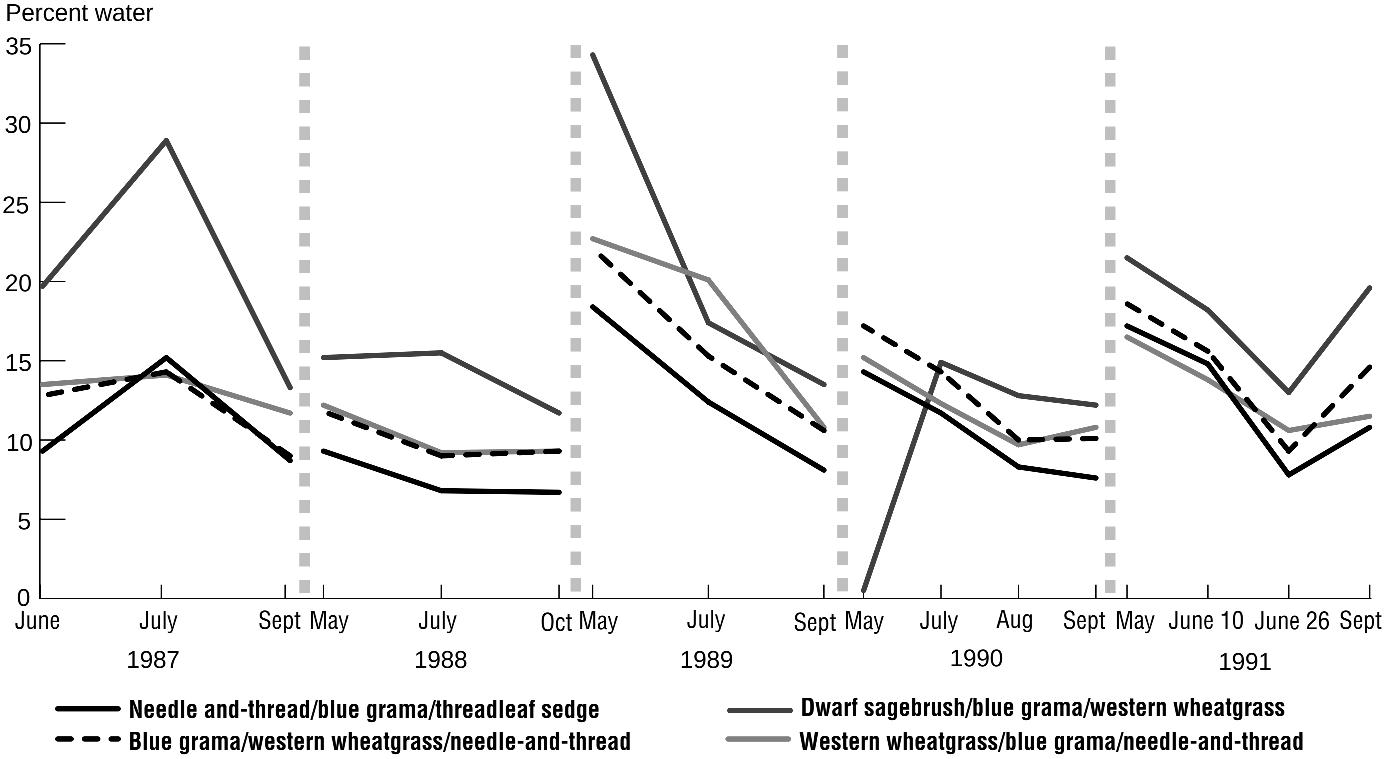

Figure V.8–8—Soil moisture content in percent, at 12-inch depth, by plant community, over a 5-year period.

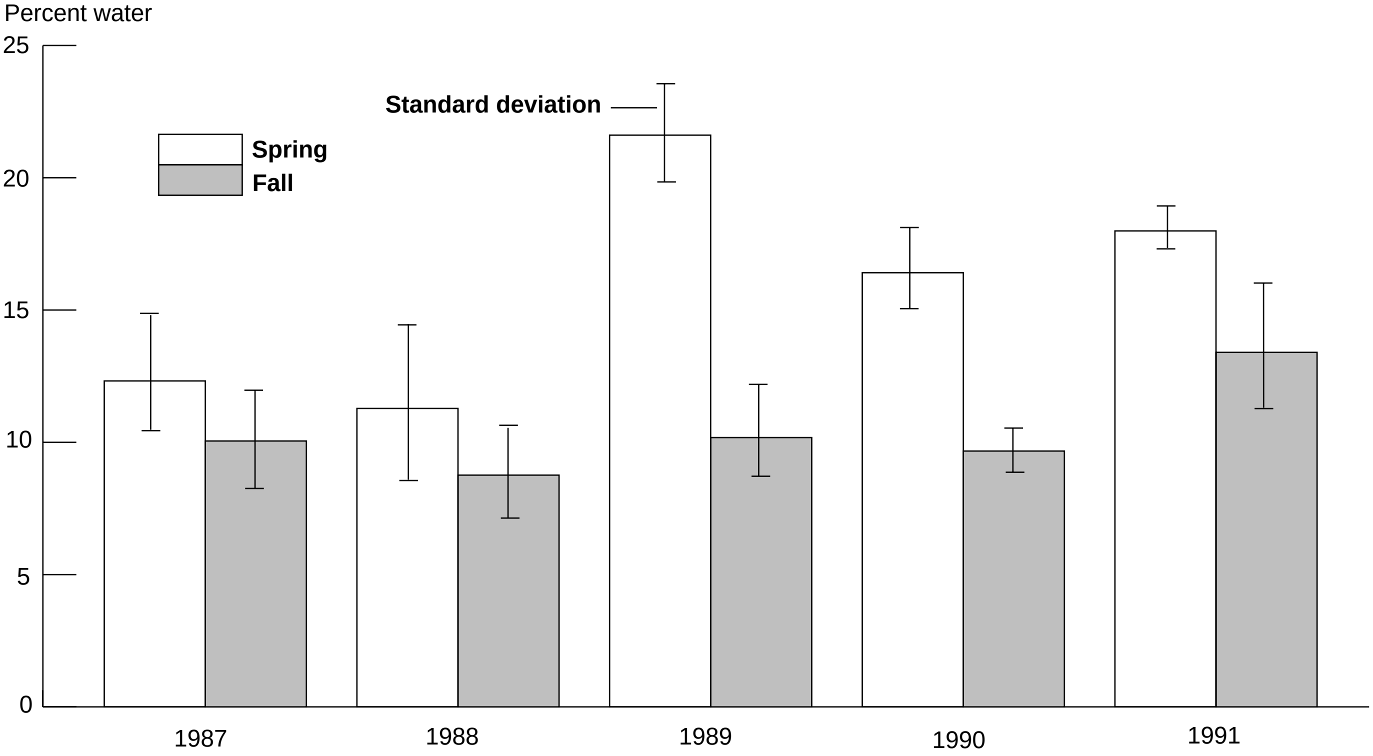

Figure V.8–9—Soil moisture content in percent, at 12-inch depth, across all sites, over a 5-year period.

Table V.8–6—Mean soil moisture (in percent; ± standard error), by plant community, over a 5-year period in western North Dakota

	[1]Type 1	Type 2	Type 3	Type 4
1987	n=5	n=8	n=4	n=1
June	9.3 ± 1.3	12.8 ± 1.9	13.5 ± 1.9	19.7
July	15.2 ± 1.7	14.3 ± 1.6	14.1 ± 2.1	28.9
Sept.	8.7 ± 0.6	9.9 ± 1.5	11.7 ± 1.5	13.3
1988	n=5	n=8	n=4	n=1
June	9.3 ± .7	11.8 ± 1.9	12.2 ± 1.6	15.2
July	6.8 ± 1.0	9.0 ± 1.6	9.2 ± 1.2	15.5
Oct.	6.7 ± .8	9.3 ± 1.7	9.3 ± 1.2	11.7
1989	n=5	n=8	n=4	n=1
1May	18.4 ± 2.7	22.1 ± 2.3	22.7 ± 3.0	34.3
July	12.4 ± 2.0	15.3 ± 2.2	20.1 ± 6.0	17.4 ± 2.4
Sept.	8.1 ± .9	10.6 ± 1.1	10.8 ± 1.2	13.5 ± 0.5
1990	n=8	n=12	n=5	n=3
May	14.3 ± 1.6	17.2 ± 0.9	15.2 ± 1.5	0.5 ± 2.0
July	11.7 ± .9	14.3 ± .9	12.3 ± 1.0	14.9 ± 1.1
Aug.	8.3 ± 1.2	10.0 ± 1.1	9.7 ± 1.1	12.8 ± .4
Sept.	7.6 ± 1.0	10.1 ± 1.1	10.8 ± 1.7	12.2 ± .6
1991	n=8	n=12	n=5	n=3
May 24	17.2 ± 1.4	18.6 ± 2.4	16.5 ± 1.0	21.5 ± 4.7
June 10	14.8 ± .6	15.6 ± 1.2	13.8 ± 1.7	18.2 ± 3.0
June 26	7.8 ± 2.5	9.3 ± 2.2	10.6 ± 2.8	13.0 ± 6.9
Sept. 18	10.8 ± 2.0	14.6 ± 1.5	11.5 ± 2.8	19.6 ± 2.9

[1] Plant community types:
1 = Needle-and-thread/blue grama/threadleaf sedge,
2 = Blue grama/western wheatgrass/needle-and-thread,
3 = Dwarf sagebrush/blue grama/western wheatgrass, and
4 = Western wheatgrass/blue grama/needle-and-thread.

Minimum requirements for data collection to classify plant communities would be to sample on two 98.4-ft (30-m) transects with a minimum of 30 frames (7.9×19.7 inches [20×50 cm]) per transect (Daubenmire 1959) for canopy cover and frequency of occurrence on each site for each of the 5 plant species. Data must be expressed as means for each of the five species. The index is obtained by multiplying canopy cover (percent) and frequency of occurrence (percent) corrected from 30 quadrats to a base of 100.

Once data are obtained for each of the five species, the method to classify a site to a plant community involves multiplying the index for each species with the appropriate Fisher classification coefficients (table V.8–2). All values are summed for each plant community, and the highest score to the positive end indicates the assigned plant community. This method, once developed, provides resource managers with a reliable quantitative tool with replicable results to classify a site to a plant community. With other methods, data sets can be interpreted subjectively to yield different results.

Monitoring Plant Communities.—The five plant species identified in the classification procedures (table V.8–2) can be used to monitor rangelands with respect to herbivory, fire, drought, and disease within these four plant communities. Monitoring can be conducted with canopy-cover and/or frequency-of-occurrence estimates with a minimum of 2 permanent transects and 30 canopy-cover and/or frequency estimates (Daubenmire 1959) per transect on each site. The index (cover × frequency) is the best plant variable to monitor changes (Uresk 1990), but either cover or frequency will do an adequate job for monitoring rangelands. Changes in direction (+/–) from the base data can be used for monitoring purposes with the five species defined for trend. Minor species are too variable for monitoring, and quantitative results are extremely limited. The five species can be easily identified and measured by resource managers in the field. Further refinement for monitoring is discussed by Uresk (1990).

Needle-and-Thread/Blue Grama/Threadleaf Sedge Community.—The eight sites assigned to this community were generally found on upland plateaus and gentle slopes. Soils were primarily sandy. Soil moisture for this community was lowest among the four communities throughout the study. In years of increased precipitation, canopy cover of some species may increase by two- or threefold. When summing canopy cover for individual species, we found that grasses and sedges ranged from 57 percent in a dry year to 125 percent in a wet year. Hansen and Hoffman (1988) reported 90 species in this community. We identified 28 grasses and sedges, 87 forbs, and 9 shrubs in this plant community, for a total of 124 species. Community and soil descriptions are similar to those provided by Hanson and Whitman (1938), Hansen et al. (1984), and Hansen and Hoffman (1988). Under heavy livestock grazing, threadleaf sedge increases and blue grama becomes dominant (Hansen and Hoffman 1988).

Plant production varied considerably from a dry year (1988) to a wet year (1991). Overall this is a very productive community. Eight species of plants make up most of the plant production for this community, with grasses (and sedges) and forbs the major components of production. Forb production showed a tremendous increase in 1989, following the dry year, possibly due to the release of nutrients available for plant growth. Hanson and Whitman (1938), Redmann (1975), and Hansen et al. (1984) described similar trends for canopy cover and production estimates for this community type.

Blue Grama/Western Wheatgrass/Needle-and-Thread Community.—Twelve sites were assigned to this community. Soils for these sites were clayey and silty. This plant community is generally found on drier upland slopes, and the period of optimum moisture for growth is shorter than that of the other communities. We found that blue grama was clearly the dominant vegetation in this plant community, similar to results reported by Hanson and Whitman (1938). Grasses and sedges ranged from 47 to 115 percent canopy cover in this community. In all, 29 grass and sedge species, 89 forbs, and 10 shrub species were identified. Redmann (1975) identified 21 species but sampled only 1 site, which produced 686 lb/acre. Overall plant production on our study ranged from 449 to 1,144 lb/acre. Forbs exhibited a 4.5-fold increase in production following 1988, the dry year. Approximately eight plant species made up the majority of the production.

Dwarf Sagebrush/Blue Grama/Western Wheatgrass Community.—This plant community was made up of five sites dominated by an overstory of dwarf sagebrush and an understory of blue grama and western wheatgrass. The community occupies floodplains and alluvial fans in valleys and is subjected to flooding, erosion, and deposition from storms or minor climatic events (Hanson and Whitman 1938). Soils were silty. Hansen et al. (1984) and Hansen and Hoffman (1988) described this as a dwarf sagebrush/western wheatgrass habitat type. Blue grama becomes the dominant understory plant when heavily grazed, with a reduction in western wheatgrass and needle-and-thread (Hansen et al. 1984). In the present study, grasses and sedges ranged in canopy cover from 44 to 101 percent. The number of grass and sedge species was 30; there were 69 forb species and 9 shrub species. Total production was greatest on this plant community when compared to other native plant communities; however, shrub production was highly variable. Forb production increased following the dry year.

Western Wheatgrass/Blue Grama/Needle-and-Thread Community.—Three sites were assigned to this plant community with western wheatgrass being the dominant plant. Soils were thinbreaks and found on slopes. Throughout the study, soil moisture was greatest for this community. Grasses and sedges ranged from 57 to 120 percent canopy cover. There were 22 grass and sedge species, 67 forb species, and 9 shrub species. Heavy livestock grazing reduces the amount of western wheatgrass and needle-and-thread and increases blue grama and buffalo grass (Uresk 1990). Grasses (and sedges) and forbs were the major component of production. Forb production increased after the dry year, 1988. Hanson and Whitman (1938) described this as a miscellaneous vegetation component in western North Dakota.

Crested Wheatgrass Community and Dwarf Sagebrush/Leafy Spurge Community.—Both of these communities were limited in the number of sites sampled. The crested wheatgrass community had a total of 79 plant species—23 grasses and sedges, 51 forbs, and 5 shrubs. Plant production was primarily from crested wheatgrass. Generally, in 20–30 years crested wheatgrass will decrease and native species become dominant. The dwarf sagebrush/leafy spurge site was dominated by dwarf sagebrush for the overstory plant and had an understory of leafy spurge, which land managers in the West consider a noxious weed. Total number of species in this community consisted of 10 grasses and sedges, 25 forbs, and 4 shrubs.

Phenology

Phenological change has been related to genetics, daily air temperatures, soil moisture, and nutrients (Bassett et al. 1961, Sauer and Uresk 1976, Idso et al. 1978, White 1979, Frank and Hofmann 1989, Callow et al. 1992). Plants on the northern Great Plains are dormant during winter. Seasonal development does not begin until temperatures and daylength exceed dormancy thresholds, adequate moisture is available, and no adverse conditions exist.

Most plants generally initiated flowering earliest in 1989 (fig. V.8–7) with the exception of fringed sagebrush, which remained in a vegetative state through the first week of August. In 1990, most plants were generally later in phenological development. The phases of development in 1991 exhibited a greater range for most plants throughout the season. However, western wheatgrass remained in a vegetative state. Callow et al. (1992) found that flowering events for 97 species varied by year and that temperature seemed more important than precipitation in the flowering dates of spring and early summer plants. They found that midsummer species did not show relationships to climatic effects.

Soil Moisture

Soil moisture could not be used as a variable to model plant growth and development over all 28 sites. Variation among sites was high due to variable precipitation, soil types, grazing, range condition, plant community differences, species composition, litter, and topography. Rauzi (1960) showed that correlations of soil moisture with plant production over several widely spaced sites were lower than for localized sites. Most modeling efforts in western North Dakota with acceptable results have been in homogeneous areas and with individual sites (Rauzi 1960, Wight and Hanks 1981, Wight et al. 1984 and 1986). Tools allowing management decisions to be applied over larger rangeland tracts are needed; unfortunately, it is difficult to model plant growth and development with high reliability over large areas that are highly variable.

As reported by the Agricultural Research Service in Sidney, MT, precipitation was highly variable over the study area. Effective precipitation directly influences soil moisture. Because most summer thunderstorms are localized, some areas may receive precipitation while others remain dry. However, effective thunderstorm events that recharge soil moisture were evident in some seasonal soil-moisture trends in midsummer or fall (fig. V.8–8).

Moisture-holding capacity in soil is a function of particle size. Fine soils generally accumulate and hold greater amounts of moisture; coarse-textured soils, less moisture (Houston 1965). Each plant community in our study was associated with a different soil type—a fact that accounted for some differences in soil moisture. Grazing intensity also influences the amount of moisture. Throughout the 28 sites, grazing, which varied from heavy to light, accounted for some of the variability in soil moisture among the four plant communities. Rangelands in a more productive condition with increased litter absorb greater amounts of moisture as compared to rangelands in poorer condition (Rauzi 1960, Houston 1965, Goetz 1975, Benkobi et al. 1993).

Most soil-moisture changes occur near the surface. Smika et al. (1961) and Cline et al. (1977) found that most variability in soil moisture occurred in the upper 12 inches (30 cm) with little change at the 35-inch (60-cm) depth and below. Soil moisture varied greatly among the 5 years for the four plant communities at the 12-inch depth.

Summary

The 30 sites in our study were classified into six plant communities. Multivariate analyses using the index (cover × frequency) provided a quantitative method to classify four native plant communities with key plant species for separating the communities. These plant species were western wheatgrass, blue grama, threadleaf sedge, needle-and-thread, and dwarf sagebrush. These plants may be used to monitor changes on the rangeland due to management practices, grazing, drought, fire, insects and disease.

Plant communities defined in this study were (1) needle-and-thread/blue grama/threadleaf sedge, (2) blue grama/western wheatgrass/needle-and-thread, (3) dwarf sagebrush/blue grama/western wheatgrass, (4) western wheatgrass/blue grama/needle-and-thread, (5) crested wheatgrass, and (6) dwarf sagebrush/leafy spurge. The latter two communities were limited to just a few sites.

The native grassland communities varied in soils and location. Plant community 2 showed the greatest species richness with 128 species, followed by 124, 108, and 98 for communities 1,3, and 4, respectively. Canopy cover for grasses and sedges ranged from 101 to 125 percent and was greatest on community 1, followed by 4, 2, and 3. Total production on the native communities was similar for all communities with the exception of community 2, which had lower total production. Shrub production in community 3 was highly variable. After a dry year, forb production dramatically increased the following year. Utilization was greatest on plant community 4 and least on community 3. Overall, western wheatgrass and dwarf sagebrush exhibited the greatest variability in phenological development among the 10 plant species over the 3-year period. However, yearly differences in phenological development were evident for all species. Timing for a particular developmental stage (e.g., flowering) varied by 2-4 weeks in some species over the 3 years. The wide range and variability in sites and climatic conditions did not produce definitive models for phenological development.

Soil moisture varied among years, seasons, and plant communities. Seasonal differences were pronounced in most years, with soil moisture decreasing as the growing season progressed. Plant communities dominated by western wheatgrass, blue grama, and needle-and-thread usually showed the greatest soil-moisture content; the needle-and-thread/blue grama/threadleaf sedge community showed the least over the 5-year period.

Acknowledgments

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References Cited

Ball, G. H.; Hall, D. J. 1967. A clustering technique for summarizing multivariate data. Behavioral Science 12: 153–155.

Bassett, I. J.; Holmes, R. M.; MacKay, K. H. 1961. Phenology of several plant species at Ottawa, Ontario, and an examination of the influence of air temperatures. Canadian Journal of Plant Science 41: 643–652.

Benkobi, L.; Trlica, M. J.; Smith, J. L. 1993. Soil loss as affected by different combinations of surface litter and rock. Journal of Environmental Quality 22: 657–661.

Branson, F. A.; Gifford, G. F.; Renard, K. G.; Hadley, R. F. 1981. Rangeland hydrology. Range Sci. Ser. No. 1, (2d ed.). Denver, CO: Society of Range Management. 340 p.

Callow, J. M.; Kantrud, H. A.; Higgins, K. F. 1992. First flowering dates and flowering periods of prairie plants at Woodworth, North Dakota. Prairie Naturalist 24: 57–64.

Cline, J. F.; Uresk, D. W.; Rickard, W. H. 1977. Comparison of soil water used by a sagebrush–bunchgrass and a cheatgrass community. Journal of Range Management 30: 199–201.

Daubenmire, R. 1959. A canopy-coverage method of vegetation analysis. Northwest Scientist 33: 43–64.

Frank A. B; Hofman, L. 1989. Relationship among grazing management, growing degree-days, and morphological development for native grasses on the northern Great Plains. Journal of Range Management 42: 199–202.

Goetz, H. 1975. Availability of nitrogen and other nutrients on four fertilized range sites during the active growing season. Journal of Range Management 28: 305–310.

Hansen, P. L.; Hoffman, G. R. 1988. The vegetation of the Grand River/Cedar River, Sioux, and Ashland Districts of the Custer National Forest: a habitat type classification. Gen. Tech. Rep. RM-157. Ft. Collins, CO: U.S. Department of Agriculture, Forest Service, Rocky Mountain Forest and Range Experiment Station. 68 p.

Hansen, P. L.; Hoffman, G. R.; Bjugstad, A. J. 1984. The vegetation of Theodore Roosevelt National Park, North Dakota: a habitat type classification. Gen. Tech. Rep. RM-113. Ft. Collins, CO: U.S. Department of Agriculture, Forest Service, Rocky Mountain Forest and Range Experiment Station. 35 p.

Hanson, H. C.; Whitman, W. C. 1938. Characteristics of major grassland types in western North Dakota. Ecological Monographs 8: 57–114.

Hewitt, G. B. 1980. Plant phenology as a guide in timing grasshopper control efforts on Montana rangeland. Journal of Range Management 33: 297–299.

Houston, W. R. 1965. Soil moisture response to range improvement in the northern Great Plains. Journal of Range Management 18: 25–30.

Idso, S. B.; Jackson, R. D.; Reginato, R. J. 1978. Extending the “degree day” concept of plant phenological development to include water stress effects. Ecological Society of America 59: 431–433.

Kemp, W. P; Berry, J. S.; Caprio, J. M. 1991. Use of ornamental lilac and honeysuckle phenophases as indicators of rangeland grasshopper development. Journal of Range Management 44: 583–587.

Lauenroth, W. K.; Whitman, W. C. 1977. Dynamics of dry matter production in a mixed-grass prairie in western North Dakota. Oecologia 27: 339–351.

Rauzi F. 1960. Water-intake studies on range soils at three locations in the northern plains. Journal of Range Management 13: 179–184.

Redmann, R. E. 1975. Production ecology of grassland plant communities in western North Dakota. Ecological Monographs 45: 83–106.

Sauer, R. H.; Uresk, D. W. 1976. Phenology of steppe plants in wet and dry years. Northwest Scientist 50: 133–139.

Smika, D. E.; Haas, A. J.; Rogler, G. A.; Lorenz, R. J. 1961. Chemical properties and moisture extraction in rangeland soils as influenced by nitrogen fertilization. Journal of Range Management 14: 213–216.

SPSS, Inc. 1990. SPSS/PC+ advanced statistics 4.0. Chicago: SPSS, Inc. 369 p.

Uresk, D. W. 1990. Using multivariate techniques to quantitatively estimate ecological stages in a mixed grass prairie. Journal of Range Management 43: 282–285.

Uresk, D. W.; Cline, J. F.; Rickard, W. H. 1979. Growth rates of cheat grass community and some associated factors. Journal of Range Management 32: 168–170.

Uresk, D. W.; Sims, P. L.; Jameson, D. A. 1975. Dynamics of blue grama within a shortgrass ecosystem. Journal of Range Management 28: 205–208.

White, L. M. 1979. Relationship between meteorological measurements and flowering of index species to flowering of 53 plant species. Agricultural Meteorology 20: 189–204.

Wight, J. R.; Hanks, R. J. 1981. A water-balance, climate model for range herbage production. Journal of Range Management 34: 307–311.

Wight, J. R.; Fissler, H. G.; Hanson, C. L. 1986. Biology and ecology of sagebrush in Wyoming: IV. Validation of a rangeland production model (ERHYM) for sagebrush sites. In: McArthur, E. D.; Welch, B. L., comps. Proceedings—symposium on the biology of *Artemisia* and *Chrysothamnus;* 1984 July 9–13; Provo, UT. Gen. Tech. Rep. INT-200. Ogden, UT: U.S. Department of Agriculture, Forest Service, Intermountain Forest and Range Experiment Station: 320–325.

Wight, J. R.; Hanson, C. L.; Whitmer, D. 1984. Using weather records with a forage production model to forecast range forage production. Journal of Range Management 37: 3–6.

V.9 Simulation of Near-Surface Soil Temperature on Rangelands

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To effectively control grasshoppers and the damage they cause requires information about when the potential for grasshopper outbreaks exists, the age structure of grasshopper populations, and how grasshopper population densities will change over time. Central to all these objectives is the ability to predict the timing of hatch and the rate of nymphal (immature) development for different species of grasshoppers. Recent Grasshopper Integrated Pest Management (GHIPM) Project results have shown that the growth and development of grasshoppers can be adequately predicted once the time of hatch has been determined (Dennis et al. 1986, Dennis and Kemp 1988). However, predicting the timing of grasshopper hatch is very difficult.

In late summer and fall, most grasshoppers lay eggs that then hatch the following spring. Several weeks after the eggs are laid, they enter what is called an embryonic diapause until the temperature gets very cold later in the fall or winter. Diapause is a state in which the eggs will not develop beyond a certain stage until the right environmental conditions exist. Diapause prevents the eggs from developing and hatching too early during an unfavorable or inappropriate season of the year. After the eggs experience a period of extreme cold, they begin to develop at a rate governed by the amount of heat they receive. Eggs that receive more heat hatch earlier in the year than eggs in cooler locations. Therefore, to predict grasshopper hatch accurately, scientists must first accurately predict soil temperature conditions that exist in the near-surface soil layers, where grasshopper eggs are laid.

Because continuous monitoring of environmental conditions in the soil is time-consuming and costly, computer simulation of soil temperature is the most practical alternative. However, temperature and moisture conditions near the soil surface change quite rapidly and are strongly influenced by small changes in weather patterns and soil types. Vegetation also strongly influences soil water and temperature conditions by controlling how much sunlight reaches the soil surface and how much heat is lost from the soil at night, when the air is cooler. Soil under a shrub receives much less sunlight than bare soil or soil covered by a grass plant immediately adjacent to the shrub. This causes a great deal of variation in how much heat is accumulated at different locations across a landscape. Pierson and Wight (1991) reported that at 1 cm below the surface, soil temperatures varied by as much as 31 °F between soils under a sagebrush plant canopy and a bare soil in the interspace between the shrubs. Their measurements reflect soil temperature conditions in March, when grasshopper eggs are still in the ground and are just beginning rapid development. Near-surface soil temperatures can be equally influenced by grasses or shrubs. In particular, bunch grasses insulate the soil surface like a shrub canopy does and can cause temperature differences of up to 36 °F between locations only a few centimeters apart.

The SHAW Model

The Simultaneous Heat and Water (SHAW) model was modified to estimate near-surface soil temperatures under varying types of rangeland vegetation (Flerchinger and Pierson 1991). The model simulates the movement of water and heat through the vegetation, snow, soil surface residue, and the soil profile. The model includes the influence of soil freezing and thawing, evaporation, transpiration, infiltration, and surface runoff. SHAW provides hourly predictions of soil temperature and water potential at any specified point throughout the plant canopy or soil profile. The model can simultaneously simulate the influence of several plant species as well as dead plant material on soil water and temperature conditions.

The model looks at the plant–soil system as a series of layers starting from the top of the plant canopy and extending down through the soil to a depth of just over 13 ft (4 m). The model requires weather information to tell it how much water and heat are being received into the top layer of the system. Data requirements include hourly estimates of air temperature, precipitation, solar radiation, windspeed, and relative humidity. The model then predicts how much heat and water will move between layers or will be lost out the bottom of the soil profile or back into the atmosphere.

Model Operation

A great deal of descriptive information about the vegetation and soil is needed before the SHAW model can be used to simulate soil water and temperature conditions at a specific site. Supplying this information in terms the model can use is referred to as the model parameterization process. To facilitate this process, there is a user interface that steps the user through each parameter and allows the user either to enter a value or have it estimated by the model. The interface then formats all the information into the proper computer file formats.

The model interface comprises a series of formatted computer screens that a user can select from a menu. Each screen steps through a variety of related parameters and, where applicable, provides helpful information on estimating a proper value. The menu consists of the following screen options, which allow the user to:

FILE: Recall parameter information from a previous simulation or to save the current parameter values,

CONTROL: Input dates of simulation and location of input and output files,

SITE: Input general information for the site (e.g., latitude, slope, aspect and elevation),

VEGETATION: Input data for plant characteristics,

SOILS: Input data for soil characteristics,

SURFACE: Input data for residue, snow, and surface characteristics,

RUN MODEL: Input data to create model input files using current data values and execute SHAW model simulation, and

EXIT: Exit the model interface.

In addition to parameterizing the model, the user must also supply a computer data file of weather information before a model simulation can be conducted. Values of air temperature, precipitation, solar radiation, windspeed, and relative humidity must be supplied on an hourly or daily basis. If weather data are available only on a daily basis and hourly output is desired, the model will estimate hourly weather values based on the daily values provided. Weather data specific to the site provide the most accurate model simulations, but weather data are not always available for all locations. In such situations, weather data can be computer generated using information from nearby weather stations. A climate generator called CLIGEN (Nicks and Gander 1993 and 1994) has been adapted to provide weather data in the proper format needed to run SHAW for many locations throughout the world.

Model Testing

To test how well the model predicts soil water and temperature conditions under different rangeland vegetation and soil conditions, model-predicted values were compared to measured values taken in the field (Pierson et al. 1992). Measurements of soil water and temperature conditions were taken at several depths in the soil within three different rangeland plant communities. One site was a sagebrush *(Artemisia tridentata tridentata)*–grass plant community, where measurements were taken directly under the shrubs and in the bare-soil interspaces between shrubs. The other two sites were shortgrass prairie plant communities dominated by blue grama grass *(Bouteloua gracilis)*, a sod-forming grass, and a stand of seeded crested wheatgrass *(Agropyron cristatum)*, a bunchgrass. The two sites were close to one another but differed in soil characteristics and elevation. Measurements of soil water and temperature were collected directly under the sodgrass and bunchgrass plants and in the bare-soil interspaces between the grass plants.

At the sagebrush site, SHAW predicted hourly soil temperatures at a depth of 1 cm during the spring growth period with average errors of only 4 °F (2.2 °C) for sagebrush locations and 5.8 °F (3.2 °C) for interspace locations. The model performed well throughout the year except for the hot summer months, when it consistently underestimated soil temperatures near the soil surface. SHAW did not simulate soil moisture conditions as well as it did soil temperature. It predicted soil moisture adequately under the sagebrush canopy but predicted dry-down too early in the interspace locations.

On the shortgrass prairie sites, SHAW simulated 1-cm and 2-inch (5-cm) soil temperatures quite well under all conditions. For bare soil conditions, SHAW consistently underestimated soil temperatures during the hot summer months at the 1-cm depth but was much closer at the 5-cm depth. SHAW slightly overestimated soil temperatures during the cooler months, particularly at the 5-cm depth. SHAW predicted periods of wetness very well at both the 1-cm and 5-cm depths but predicted too rapid a dry-down period compared to measured values. Both measured and predicted soil temperature and moisture responses under the sodgrass were similar to those for the bare soil condition.

Under bunchgrass, SHAW simulated 1-cm and 5-cm soil temperatures better than it did under bare-soil conditions. The seasonal problem of underestimating summer soil temperatures exhibited for the bare soil was much less evident. For certain conditions throughout the year, SHAW seemed to overpredict temperatures at both the 1-cm and 5-cm depths, but the errors were generally small. SHAW simulated soil moisture conditions significantly better under the bunchgrass than under bare-soil conditions at both tested depths. Rather than predicting dryness too quickly as SHAW did for the bare soil, the model generally overpredicted the length of the wet periods at both depths.

Testing the SHAW model has shown that it is quite capable of simulating small-scale variations in soil temperature and moisture conditions induced by vegetation. The model performed particularly well under the sagebrush and bunchgrass conditions compared to bare-soil conditions, indicating SHAW's strength at simulating the insulating effect of the plant canopy and the evapotranspiration process.

Model Applications

The ability to simulate the soil water and temperature regimes of the top inch or so of the soil profile will significantly enhance the simulation of grasshopper growth dynamics and the development of management strategies. Simulated soil temperatures can be used to drive other models, such as the grasshopper hatch model developed as part of the GHIPM Project (see IV.2, "Grasshopper Egg Development: the Role of Temperature in Predicting Egg Hatch"). Together these models can be used to develop regional and geographic information systems data bases of the expected time of occurrence of various stages of grasshopper development.

SHAW-simulated soil temperatures were used to drive the grasshopper hatch model and predict grasshopper hatch dynamics at a site near Three Forks, MT. The results were compared against predictions of hatch based on measured soil temperatures and actual field measurements of grasshopper hatch (fig. V.9–1). Early in the season, predictions of grasshopper hatch based on SHAW soil temperatures were very close to those for measured soil temperatures, but both slightly overpredicted the proportion of grasshoppers hatched compared to measured populations. The timing of 50-percent hatch was predicted quite well based on both SHAW-simulated and measured soil temperatures. Later in the season, the hatch model slightly underestimated the proportion of grasshoppers hatched, particularly based on SHAW-simulated soil temperatures. Overall, the grasshopper hatch model performed very well and lost little accuracy when SHAW-simulated soil temperatures were substituted for measured values.

This type of modeling approach can also be used with historical climate information to explore management questions such as how the timing of grasshopper hatch might vary from year to year for different grasshopper species. The SHAW model was used to simulate annual near-surface soil temperatures within a sagebrush–grass plant community for a period of 100 years using simulated climate information. The model output was then used to determine the probability of occurrence of specific temperature conditions that might be associated with the timing of grasshopper hatch. For the purposes of this example, grasshoppers were assumed to hatch when the eggs had accumulated 300 growing degree-days (GDD).

Figure V.9–2 shows the frequency of occurrence of 300 GDD under both sagebrush shrubs and the interspace locations between shrubs. Notice that the distribution of possible hatch times for the entire site covers about 5 weeks (Julian date 124–161) and that there is no overlap of distributions between the two locations. The frequency distribution for the interspace location is only 1 week in length, indicating that there is a very high probability that grasshopper eggs within the interspace locations will hatch every year within 3 days of Julian day 126.

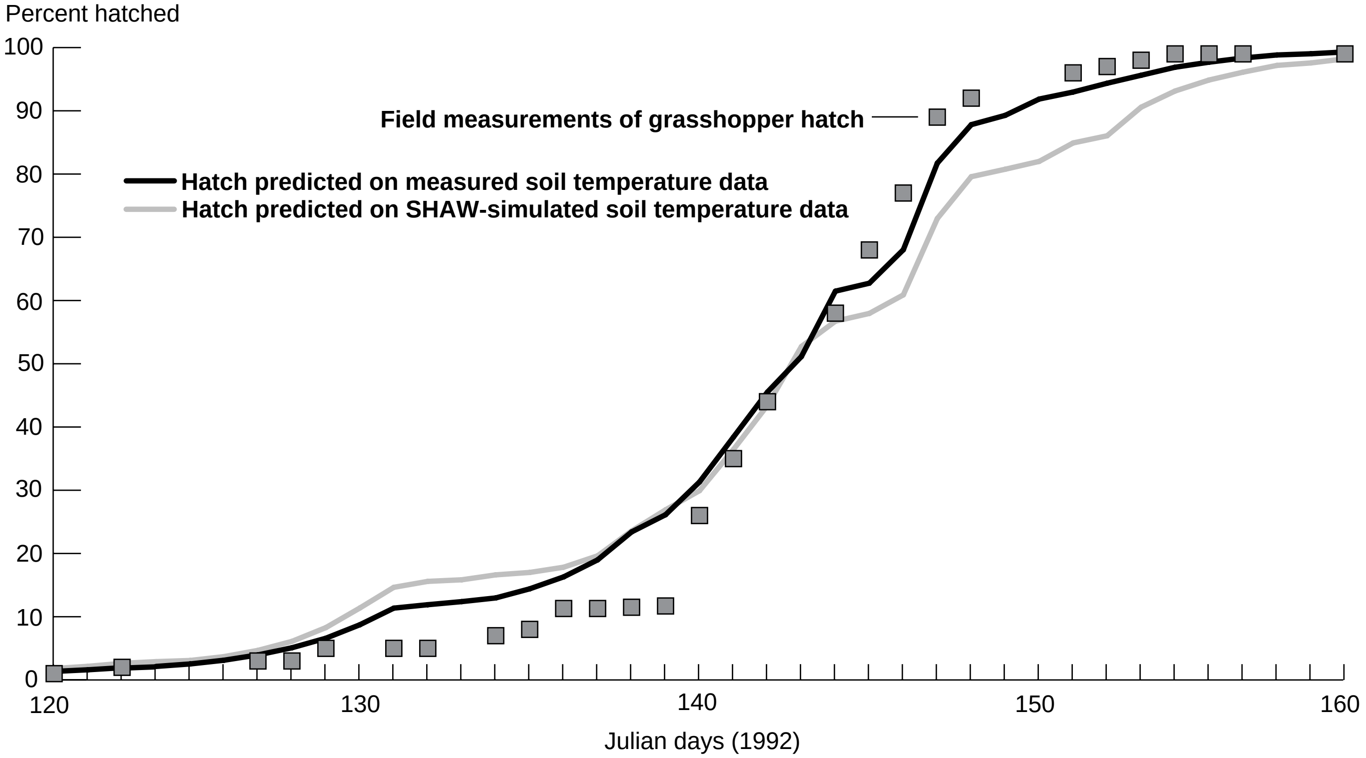


Figure V.9–1—Comparison of measured and predicted proportions of the population of *Aulocara elliotti* grasshoppers hatched for each day during the spring of 1992 near Three Forks, MT.

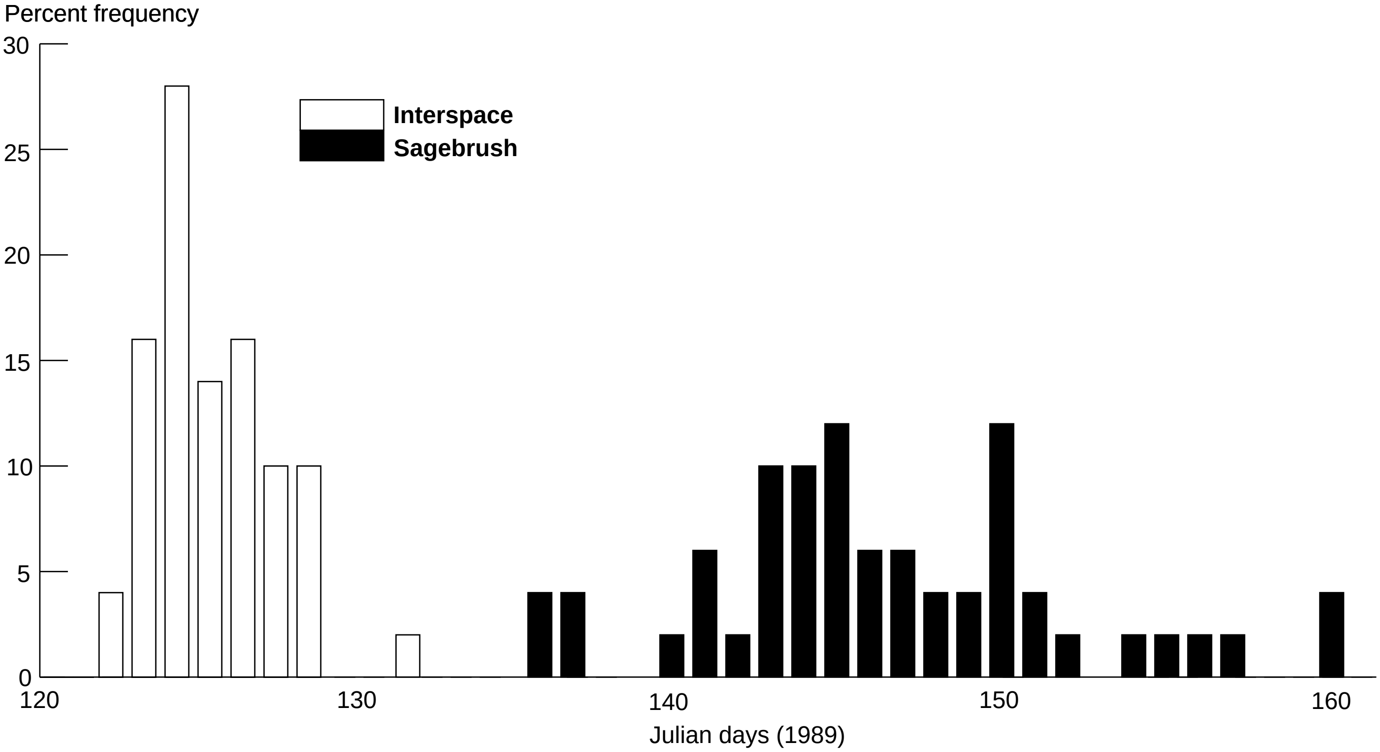


Figure V.9–2—Percent frequency of the timing of the accumulation of 300 degree-days of heat under sagebrush plants and the interspace locations between sagebrush plants at the Quonset site on the Reynolds Creek Experimental Watershed, Reynolds, ID (Wight et al. 1992).

So what does this information mean to grasshopper management? If grasshoppers lay their egg pods at random across the landscape, then the variation in hatch time across the site could be as much as 5 weeks. This variation would result in a very mixed-aged population of grasshoppers. However, research has shown that certain species of grasshoppers do not lay their eggs at random across the landscape but selectively choose specific sites (such as directly under a shrub or in full sunlight between shrubs). Thus, the model results can tell managers when to look for hatch to begin for different grasshopper species. For example, if grasshopper species "X" lays its eggs under shrubs and grasshopper species "Y" lays its eggs in the interspaces, then the entire population of grasshopper X will always hatch before grasshopper Y begins to hatch. This kind of information can be useful for improving resource planning and enhancing the efficiency of grasshopper control applications.

References Cited

Dennis, B.; Kemp, W. P.; Beckwith, R. C. 1986. A stochastic model of insect phenology: estimation and testing. Environmental Entomology 15: 540–546.

Dennis, B.; Kemp, W. P. 1988. Further statistical inference methods for a stochastic model of insect phenology. Environmental Entomology 17: 887–893.

Flerchinger, G. N.; Pierson, F. B. 1991. Modeling plant canopy effects on variability of soil temperature and water. Agriculture and Forest Meteorology 56: 227–246.

Nicks, A. D.; Gander, G. A. 1994. CLIGEN: a weather generator for climate inputs to water resource and other models. In: Watson, D. G.; Zarzueta, F. S.; Harrison, T. V., eds. Proceedings of the fifth international conference on computers in agriculture; 6–9 February 1994; Orlando, FL. [Place of publication unknown]: American Society of Agricultural Engineers: 903–909.

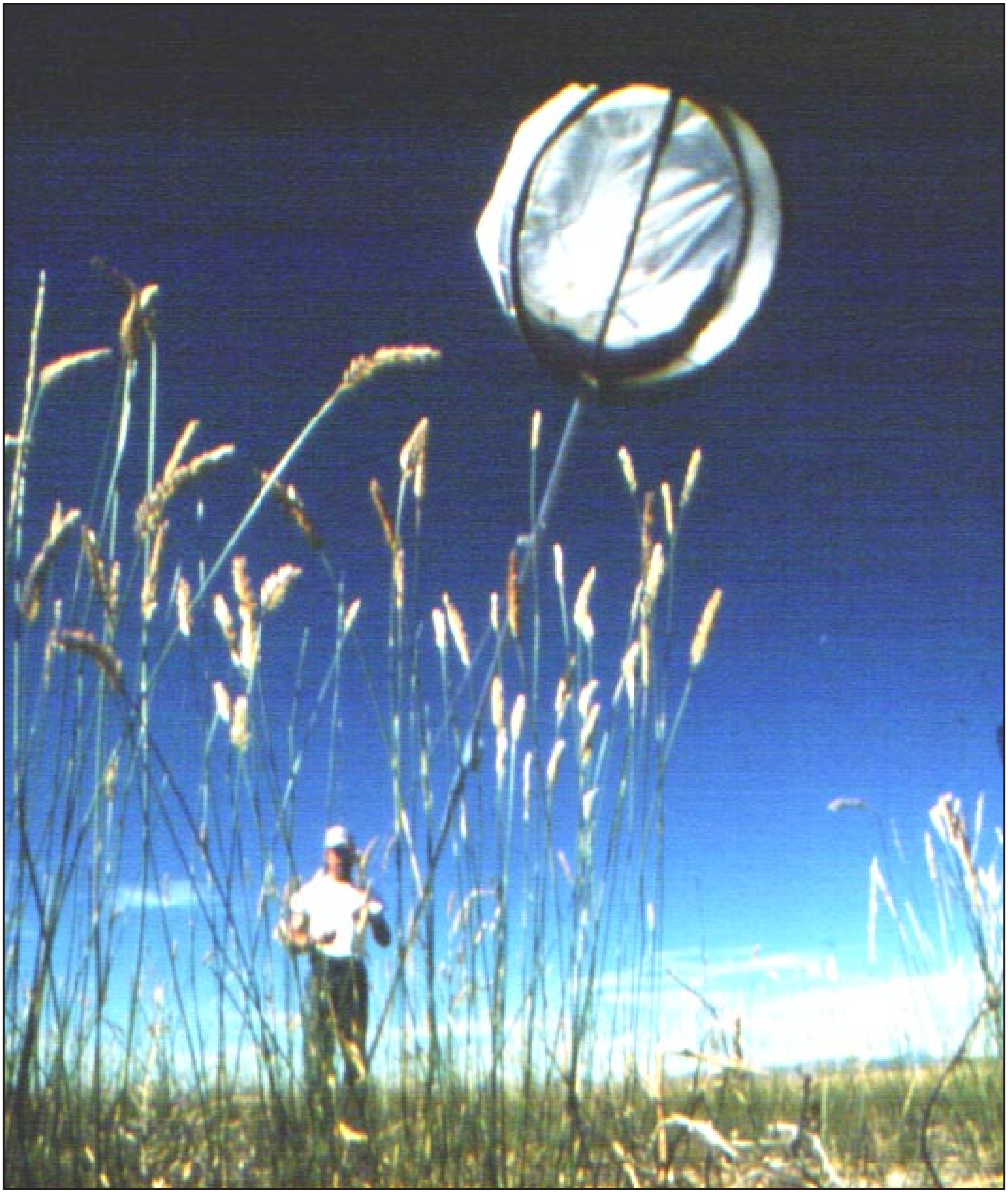
Nicks, A. D.; Gander, G. A. 1993. Using CLIGEN to stochastically generate climate data inputs to WEPP and other water resource models. In: Proceedings of the Federal interagency workshop on hydrologic modeling demands for the 90's. Water Resour. Invest. Rep. 93-4018. Fort Collins, CO: U.S. Geological Survey: 7-14–7-21.

Pierson, F. B.; Flerchinger, G. N.; Wight, J. R. 1992. Simulating near-surface soil temperature and water on sagebrush rangelands: a comparison of models. Transactions of the American Society of Agricultural Engineers 35: 1449–1455.

Pierson, F. B.; Wight, J. R. 1991. Variability of near-surface soil temperature on sagebrush rangeland. Journal of Range Management 44: 491–497.

Wight, J. R.; Pierson, F. B.; Hanson, C. L.; Flerchinger, G. N. 1992. Influence of sagebrush on the soil microclimate. In: Symposium on ecology and management of riparian shrub communities; 29–31 May 1991; Sun Valley, ID. Gen. Tech. Rep. INT-289. Ogden, UT: U.S. Department of Agriculture, Forest Service, Intermountain Forest and Range Experiment Station: 181–185.

VI. Decision Support Tools



The sweep net is a valuable tool for identifying grasshopper species. Knowing the species composition of a grasshopper population is a key element for making correct decisions. (USDA photo.)

VI.1 The Importance of Making Correct Decisions

Jerome A. Onsager

Within the general arena of grasshopper management, it is possible to make decisions that reduce or cancel out expected potential benefits. According to my dictionary, such decisions possibly could qualify as "blunders" (arising from stupidity, ignorance, or carelessness), "mistakes" (arising from misconception or inattention), or "errors" (arising from a violation of standard guidelines). I do not know what to call strict adherence to guidelines based on misconceptions, but that seems to be another possibility for making faulty decisions. Regardless of what we as pest managers call such decisions, an examination of their origins reveals that most are preventable.

Incorrect decisions within grasshopper management can cause us either to take incorrect actions or fail to take correct actions. Examples of the former include treating rangelands too early, treating too late, treating populations of species that are not chronic pests, selecting suboptimal treatments, and treating noneconomical grasshopper infestations. Examples of the latter— failing to take correct actions—include failing to detect infestations in a timely manner, deciding not to treat injurious infestations, and failing to reduce undesirable consequences of treatments. The following chapters on decision support tools are intended to help both novices and experienced personnel gather accurate information about grasshopper populations and thereby increase the probability of making correct management decisions.

W. J. Cushing's chapter (VI.8) on seasonal occurrence of selected grasshopper species is helpful in the proper timing of surveys. Timing of nymphal (immature grasshopper) surveys is critical if managers are to assess accurately the threat of current infestations at a time when all treatment options are available and before irreparable damage occurs. Timing of adult grasshopper surveys must coincide with the adult period of major pest species if managers are to have accuracy in assessing the potential for future infestations. The chapter of J. S. Berry et al. on sampling techniques and sampling intensity (VI.10) provides guidelines that should cover most survey situations.

R. J. Dysart's chapter (VI.6) shows that some of the 400 grasshopper species in the West are serious pests, that the majority of species are fairly innocuous (harmless), and that a few species even have beneficial attributes. Cushing's "Hopper Helper" (VI.7) and R. J. Pfadt's "Field Guide to Common Western Grasshoppers" (VI.5) are useful in deciding if a grasshopper population contains important pest species. Having identification tools and knowing the makeup of a grasshopper population are vital in deciding to control the population.

An example of where timely grasshopper identification averted unnecessary treatment occurred during the first season of the Grasshopper Integrated Pest Management (GHIPM) Project in 1988 in western North Dakota. Potentially threatening grasshopper densities were reported in an area along the Little Missouri River, where nearness to water might have required a complicated integration of chemical spray, carbaryl bait, and *Nosema locustae* bait treatments. However, surveyors determined that the infestation was mostly *Melanoplus keeleri*, a species that feeds abundantly on coarse brushy forbs and that never has been implicated as a major participant in a sustained outbreak. GHIPM Project personnel correctly decided to take no action, and the "outbreak" subsided the following year.

From its inception in 1987, the GHIPM Project placed major emphasis on consolidation of massive sets of information related to biology and control of grasshoppers, on interdisciplinary analysis and interpretation of complex interactions within that body of information, and on organization and presentation of pertinent conclusions in a useful format. The process relied heavily on computer technology to provide solutions to long-standing problems.

Some of the project's products and tools are described in chapters on economic considerations, by M. D. Skold and coworkers (VI.3 and 4); geographic information systems, by W. P. Kemp (VI.9); and the Hopper decision support system, by J. S. Berry (VI.2). These chapters discuss useful but complex analyses that are well beyond the capabilities of many managers who could benefit from those analyses. Fortunately, the authors have contributed to computer software that allows any computer-literate individual to follow the reasoning powers of a panel of experts when trying to make treatment decisions.

The concepts of economic injury levels and economic thresholds are cornerstones in the foundation of IPM. The chapters by Skold and coworkers represent the state of the art in applying economic considerations to grasshopper management. Chapters show very clearly that chemical control is but one of several available management options and is not universally the most economical tactic. Analyses described in the Skold chapters are an integral part of Hopper, which managers can use to estimate public, private, or total benefits versus costs for either public, private, or cooperative rangeland grasshopper control projects.

Clearly, the decision to control or not control rangeland grasshoppers is not simple. Also, the general public rightfully expects a high level of technical competence within the decisionmaking process. This section of the GHIPM User Handbook represents a concerted effort to equip managers with a complete list of definitive questions as well as the means to obtain accurate answers to those questions. Adherence to the suggestions and guidelines in this section will help managers avoid blunders, mistakes, and errors—and will help support rational pest management on public and private rangelands.

Warning

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Hopper, which is described in section VI.2, can be freely copied. All third-party software used in Hopper can be distributed royalty free.

Hopper Disclaimer

Hopper has been tested as much as possible with the available data and experts and has performed satisfactorily. However, the rangeland ecosystem is very complex and unpredictable. In addition, Hopper does not have any control over the data entered by each user. Therefore, the results derived from Hopper cannot be guaranteed. The following disclaimer applies:

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NOTE: Acephate is no longer approved by EPA for rangeland grasshopper control.

VI.2 Hopper, Version 4.0, Users' Guide: Decision Support System for Rangeland Grasshopper Management

James S. Berry, William P. Kemp, and Jerome A. Onsager

Preface

The Users' Guide Is a Teaching Tool.—The goal is to present you with the most critical information and the most likely scenarios you will encounter using Hopper and Hopper Lite. In this way, you can learn the program fast and be free of the documentation soon.

Use the Guide Even If You Can Run Hopper Without It.—Initially, you should follow this Users' Guide, even if you intuitively understand how the programs work. The Users' Guide presents you with the options and situations under which you would use Hopper and Hopper Lite and provides background information to help you understand the data and results.

Hopper and Hopper Lite are simple and intuitive, but the data they require are *not.* Ranching economics and rangeland ecology are complex. Consequently, while the data are easy to enter, they are sometimes hard to collect and understand. The Users' Guide provides useful background information and hints to help you learn and use the system correctly. Used properly, Hopper and Hopper Lite will improve the reliability of your treatment decisions.

Acknowledgments

Hopper was developed for the Grasshopper Integrated Pest Management (GHIPM) Project, a multiyear research and development effort that ended in 1995. Many individuals contributed to Hopper over the life of the Project. We wish to acknowledge the following for their support:

Economics Models: Melvin Skold (Colorado State University, Ft. Collins, CO) and Rob Davis (U.S. Department of the Interior's Bureau of Reclamation, Denver, CO).

Programming Support: Chuck Griffin, Scott Sotka, Larry Emmett (Rangeland Insect Laboratory and Montana State University, Bozeman, MT).

Hopper Implementation Team (Special Advisory Committee):
Gary L. Cunningham, USDA, APHIS, Plant Protection and Quarantine (PPQ)
Robert Averill, USDA, Forest Service
Bob Brittingham, USDA, APHIS, PPQ (retired)
Nelson Foster, USDA, APHIS, PPQ
J. Bruce Helbig, Plant Health Director for South Dakota, USDA, APHIS, PPQ
J. Dan Kail, PPQ Officer for Nevada, USDA, APHIS, PPQ
Jim Krsnak, Division of Regulatory Services, South Dakota (Plant Board Representative)
John Larsen, Plant Health Director for Wyoming, USDA, APHIS, PPQ
David Legg, Department of Plant, Soil and Insect Sciences, University of Wyoming (Extension Service representative)
Melvin Skold, Department of Agricultural and Resource Economics, Colorado State University
B. Barte Smith, Plant Health Director for Nebraska, USDA, APHIS, PPQ
Larry Zaleski, Instructional Design Specialist, USDA, APHIS, Recruitment and Development (R&D)

Original Hopper Users' Guide:
Jim Berry, APHIS, PPQ, Phoenix, AZ
Larry Zaleski, R&D, Frederick, MD

Introduction

There are two versions of the Hopper Decision Support Software, "Hopper" and "Hopper Lite." Hopper Lite is for the infrequent user and could be used without consulting a printed manual. Hopper provides more features and flexibility than Hopper Lite. Consequently, Hopper is more complex and not as simple to use. However, Hopper and Hopper Lite use the same analyses and produce the same results. Each time you start Hopper, you will be asked whether you would like to use Hopper or Hopper Lite.

What Is Hopper?—Hopper and Hopper Lite will allow you to evaluate the validity and cost effectiveness of treating outbreaks of rangeland grasshoppers to protect rangeland in western North America. These analyses are based on the best scientific knowledge currently available. This knowledge represents more than 40 years of research and practical field experience of scientists and field personnel.

Hopper is designed around a menu system that you use to select the options and features you need. On the other hand, Hopper Lite will guide you step-by-step through the treatment selection process. Hopper and Hopper Lite are designed for experienced agriculturalists and resource managers who must make informed treatment decisions. Hopper and Hopper Lite cannot be used to evaluate land enrolled in the Conservation Reserve Program (CRP) or protection of crops adjacent to rangeland. In addition, the economic analysis is based on the value of rangeland forage as a food source for free-roaming cattle. Other values, such as long-term resource protection, wildlife, or social impact, are not considered. Hopper does provide some information that a land manager can use to evaluate some of these factors. For example, forage yield may be useful to big-game managers. The analysis allocates reserve forage to be left after grazing (determined by the proper use factor and the Peak Standing Crop parameter). Grasshoppers consume nonreserve forage according to their need. Any remaining nonreserve forage is available to cattle.

Why Use Hopper?—You should use Hopper or Hopper Lite to assist with and improve the reliability of your treatment decisions. Treating grasshopper outbreaks is costly and complicated; you don’t want to waste time and money treating when treatment is *not* needed.

Treatment decisions are serious business. On the one hand, unneeded treatment wastes money. But failure to treat when treatment is needed may damage the local ranching economy.

Consequently, you want to make the right decision, and you need reliable information to do so. Hopper provides reliability by evaluating your data scientifically.

What Hopper Does.—Hopper and Hopper Lite provide you with a list of treatments and an estimate of cost effectiveness. To provide this information, Hopper asks you for data about your site. Then Hopper analyzes your data using computer models. These models evaluate factors that are critical for making treatment decisions, including many that are otherwise too time consuming for field personnel to consider.

Hopper gives you a benefit–cost ratio (BC) that you can use to help make your decisions. The BC replaces the static treatment thresholds used previously. The BC depends on many factors that change over time and locations (see appendix A, “How Hopper Works and Why”). The BC is based on the benefits and costs incurred during a single year’s operation. The single-year BC does *not* account for multiyear effects, such as the effect of reduced egg deposit on next year’s grasshopper population density. Hopper can calculate a multiyear BC, compounded from the single-year BC. Also, Hopper does not account for environmental costs or benefits, value of beneficial species, and other nonforage-related values.

In summary, Hopper’s economic evaluations include only the value of forage for livestock consumption in a single season. However, there are many other factors that a rancher may consider in addition to possible multiple-year benefits. One factor is maintenance of the brood herd and long-term survivability and profitability of the operation. A 1-year loss may be acceptable over a 10-year cycle of 9 profitable years.

When the BC is 1.0 or more, treatment is economically justified, and you would treat the outbreak to protect forage. But when the BC is less than 1, treatment is economically unjustified, and you would *not* treat the outbreak just to protect the current forage crop. The final decision to treat or not depends on Hopper’s analysis and any other factors important to the ranching community and general public.

Thus, by using Hopper, you can include cost effectiveness in the decisionmaking process.

When To Use Hopper *versus* Hopper Lite.—New users, infrequent users, and managers who need only to evaluate normal treatment scenarios should use Hopper Lite, at least initially. These managers include USDA, APHIS, PPQ personnel. Hopper Lite will direct you, step by step, through Hopper’s essential features to evaluate a potential treatment scenario. The most needed features of Hopper are provided, such as input screens for treatment cost and efficacy and grasshopper information.

After becoming familiar with Hopper Lite, frequent users will probably find Hopper easier to use because of its increased flexibility. Also, Hopper provides the opportunity to determine an economic threshold, change additional economic information, create hard-copy data-entry forms, print graphs, configure attached printers, and run a generalized simulation of rangeland grasshoppers. If you need any of these features, you must choose Hopper. However, remember that the analysis in Hopper Lite is the same as Hopper. There is no reason to use Hopper unless you need its additional features.

Getting Started

System Requirements.—Hopper will run on an IBM™ compatible computer with at least 640 kilobytes (KB) of memory. (A central processing unit 80386, –486, or higher is strongly recommended.) Hopper will probably run with less memory, but the absolute lower limit is not known. Your computer must be running DOS version 3.1 or higher. A VGA monitor is required to view the hazard maps and graphs of the grasshopper and forage simulation results but is not required for other parts of Hopper. To save and print graphs of Hopper's simulations for dot matrix and laser printers, 512–1024 KB of expanded memory (EMS) is required. (See the Installation section of your DOS or MS–Windows™ manual to modify your `config.sys` file with the `emm386.exe` driver.) You can use a mouse to make selections from menus, but the mouse is not required.

A hard drive is required, and there must be at least 3.5 megabytes (MB) free disk space before Hopper is installed. A math coprocessor will speed the simulations in the economic analysis module by almost a factor of 10. However, the math coprocessor is only recommended, not required.

Installation.—There is a simple program (`INSTALL`) supplied with Hopper that will guide you through the installation process and install Hopper on your computer's hard disk. `INSTALL` will also identify the computer's hardware so you can verify system requirements. To install Hopper and Hopper Lite, put the Hopper disk in the floppy disk drive. Then type the letter of the floppy disk drive, a colon, and `INSTALL` (e.g., `A:INSTALL`); do not type any blank spaces; then press the enter key `<ENTER>`. Then follow the directions on the screen. Hopper is supplied in an archived format to save diskette space. `INSTALL` will unarchive the files and copy them to your hard disk. **Note:** Hopper cannot be installed by simply copying the files to your hard disk. You must use the installation program.

If you have previously installed an older version of Hopper in the `\Hopper` directory, you may want to erase the old Hopper files from your hard drive **(Note:** Data files from previous versions and data files (`*.fct` and `*.ec3`) are not compatible with the current version). Removing outdated files will free some disk space for future use. You can keep the old version of Hopper, but you will need to specify a directory other than `\Hopper` when you are prompted by `INSTALL`. If you attempt to install Hopper into a directory where any files exist, `INSTALL` can erase the files for you after prompting you for permission. In this case, all previous information you have saved in that directory will be lost.

If you have at least 2 MB of memory on your computer, you can make some of that memory available to Hopper for creating graphs. To add expanded memory for saving and printing simulation graphics for dot matrix and laser printers, add the following line to your `config.sys` file after the `HIMEM.SYS` line (if present) or on the first line.

```
device=c:\dos\emm386.exe 1024
```

Hopper's default graphic printer (HPGL/2) does not require this line to be added.

Starting Hopper and Hopper Lite.—After `INSTALL` finishes installing Hopper to the hard drive, Hopper is ready for use. Typically, Hopper will be located in a directory called `C:\Hopper`, unless a different drive and directory were specified during installation. Hopper needs to find several of its files while it is running. Therefore, Hopper can be started only from its own directory. To change to the Hopper directory and then to run Hopper, type:

```
cd\hopper <ENTER>
hopper <ENTER>
```

This assumes that Hopper was installed in `C:\Hopper`. If Hopper is started from a menu system, the menu must be programmed to make the Hopper directory the current directory before starting Hopper (similar to the above commands). Each time you start Hopper, you will be asked whether you would like to use Hopper or Hopper Lite.

The User Interface.—In this manual, keystroke commands are in pointed brackets such as <>. The keys are:

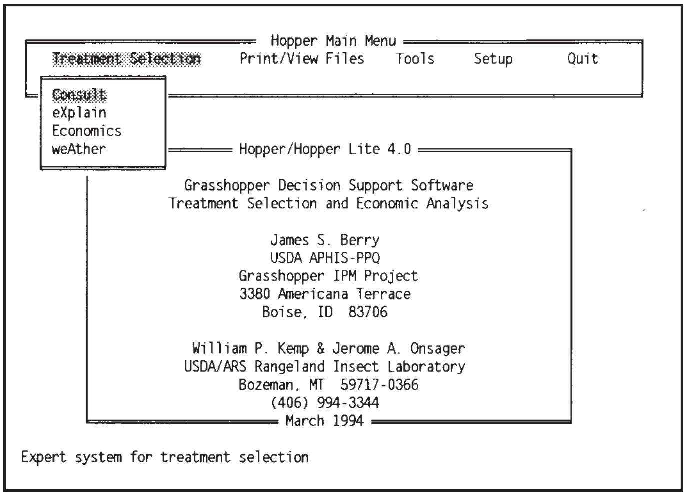
<ENTER>...Enter key
<ESC>...Escape key
<DEL>...Delete key
<INSERT>...Insert key
<PageUp>...Page up key
<PageDown>...Page down key
<Down>...Down arrow
<Up>...Up arrow
<Left>...Left arrow
<Right>...Right arrow
<F1>, <F2>...Function keys.

At times, text or numbers must be entered. These will appear in this Users' Guide without brackets (e.g., 23, some text).

When Hopper or Hopper Lite is started (by typing Hopper <ENTER>, or Hopper MONO <ENTER> if you have a monochrome monitor or monochrome liquid crystal display [LCD] screen), a disclaimer appears and waits for any key to be pressed before continuing. Next, the option to select Hopper or Hopper Lite is presented. If you select Hopper Lite, you will be guided through the treatment selection process. Many of Hopper's and Hopper Lite's features and screens are identical. If you choose Hopper, the main menu screen appears (fig. VI.2–1). This screen contains a title window in the center of the screen. Across the top of the screen is a list of main menu items available. (In this text, main menu items are printed in boldface type.) Use the arrow keys (or mouse) to move to a main menu item and then press <ENTER> (or left mouse button) to select that item. When you select one of these main menu items, a submenu of items appears. (Submenu items are always printed in italics.) You can leave any menu or submenu by pressing <ESC>. In fact, pressing <ESC> will allow you to jump out of most areas in Hopper or back up one step.

Hopper is operated by using menus, so you do not have to remember complicated commands. Instead, look through the menus to find the desired item and press <ESC> to leave the menu if the item is not found. Also, you can press <F1> at any time to get context-sensitive help information (fig. VI.2–2). Therefore, you do not have to remember commands or syntax. This menu-driven architecture increases the ease of operation of Hopper while maintaining flexibility for you. You are always returned to the main menu after exiting from a submenu.

Some information Hopper needs is entered onto onscreen data-entry forms (fig. VI.2–3). At times you will need to type numbers or dates on a form. Use the tab key <TAB>, <ENTER>, or arrow keys to navigate between the fields on a form. Data within a field on a form can be edited using the delete key <DEL> or arrow keys, and by



Grasshopper Decision Support Software
Treatment Selection and Economic Analysis

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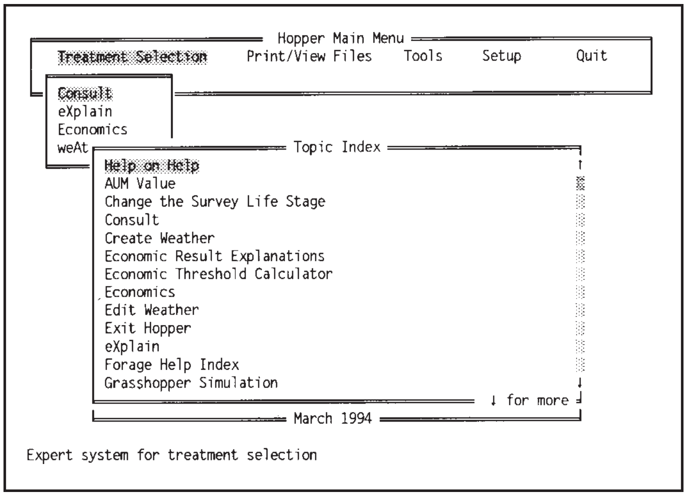
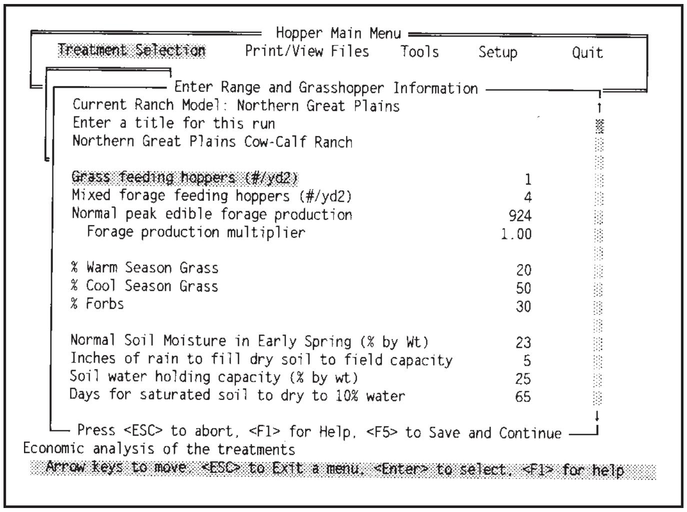
Expert system for treatment selection

Figure VI.2–1—Main screen showing the **Treatment Selection** submenu.



Expert system for treatment selection

Figure VI.2–2—Main screen help after pressing <F1> twice to get the Help index.



```
Enter Range and Grasshopper Information
Current Ranch Model: Northern Great Plains
Enter a title for this run
Northern Great Plains Cow-Calf Ranch

Grass feeding hoppers (#/yd2)                                1
Mixed forage feeding hoppers (#/yd2)                         4
Normal peak edible forage production                       924
  Forage production multiplier                            1.00

% Warm Season Grass                                         20
% Cool Season Grass                                         50
% Forbs                                                     30

Normal Soil Moisture in Early Spring (% by Wt)              23
Inches of rain to fill dry soil to field capacity            5
Soil water holding capacity (% by wt)                       25
Days for saturated soil to dry to 10% water                 65

Press <ESC> to abort, <F1> for Help, <F5> to Save and Continue
Economic analysis of the treatments
Arrow keys to move. <ESC> to Exit a menu. <Enter> to select. <F1> for help
```

Figure VI.2–3—Example of the fields in an onscreen data-entry form.

typing the desired information. Forms are preloaded with default values so that you often do not need to enter much information. Usually you will just change a couple of values on a form.

A good way to learn Hopper is to explore the menu system and try the various features. Hopper filters your input so that you can enter appropriate information only. Hopper is designed to be robust so that you can easily explore its capabilities as you learn how to use it.

Technical Support.—For help in using Hopper or Hopper Lite, contact Jim Berry by telephone at (602) 379–6014 between 8:00 a.m. and 5:00 p.m. (Mountain Standard Time), weekdays. Facsimilies can be sent any time to (602) 379–6005. Send Internet mail to pmdc@xroads.com with "Jim Berry" (minus the quotation marks) in the Subject line.

Hopper Lite Version 4.0 A Simple Decision Support System for Rangeland Grasshopper Management

Hopper Lite is very easy to use. It asks you questions and controls the whole process to the end. A typical scenario should take about 2 to 10 minutes to complete, depending on the speed of your computer.

Installation.—Hopper Lite is installed automatically with Hopper. Hopper Lite is a subset of Hopper and uses the same files as Hopper.

Operation.—Make the Hopper drive and directory current (e.g., `cd\hopper`). Then type `Hopper <ENTER>` to start the program. You will be asked if you would like to use Hopper Lite. Enter `Y` to select Hopper Lite. Then enter the information requested at each prompt. The arrow keys can be used to highlight an answer; then press `<ENTER>` to select that answer. Much of this Users' Guide is contained on the computer and is available by pressing the `<F1>` key. This information will often provide additional explanation or help each step of the way. **Note:** To configure your printer or generate hard-copy data-entry forms, you will need to run Hopper.

For the economic analysis, select a data file that corresponds to your situation (e.g., `NPH_CC.ec3` for northern high plains cow–calf operation, or a generic model [files with "`.gn3`" extension] if no models are available for your area or situation). For more information on economic analysis, see the Economics section.

The economic analysis display at the conclusion of the process shows what treatments were selected and benefit–cost ratio (BC) for each. Remember, these results are the same as those provided by Hopper.

You can change the text printers (default = Hewlett–Packard LaserJet™) or graphics printers (default = HPGL™/2) only in Hopper. In addition, Hopper Lite can only save graphs, not print them. Select **Print/View Files** from the main menu in Hopper to print graphs.

Overview of Hopper

Summary of Features.—There are four items accessible from the main menu. The first is **Treatment Selection**. The submenu provides access to an expert system for selecting appropriate treatments and computer models for economic analyses of those treatments. You can easily try different scenarios to evaluate their economic consequences. The computer simulations for forage production, grasshopper population dynamics, and ranch economic linear programming models in version 4.0 of Hopper and Hopper Lite expand this flexibility for evaluating alternative scenarios.

The second main menu item **(Print/View Files)** will allow you to view on the screen or print any output that

Hopper or Hopper Lite produces. Outputs include reports and data-entry forms. Graphs you save during the economic analysis of treatments can be printed but not viewed.

The third main menu item **(Tools)** has five submenus. There is an interactive *Tutorial* designed to teach a new user how to use Hopper. Next, there is a generalized simulation model of grasshopper population dynamics and treatment effects *(SimHop)*. This is useful for demonstrating the effects of several factors on the overall utility of a control program. *Maps* allows you to select and view rangeland grasshopper hazard maps for several States. These maps are derived, using geostatistical techniques, from surveys of adult grasshoppers in the previous year. Because grasshopper densities are highly correlated with densities 1 year earlier, the maps indicate probable areas of high grasshopper populations. The *Economic Threshold* submenu item will estimate the grasshopper density necessary to produce a benefit–cost ratio you specify. The last submenu item is *Forms,* which will allow you to create hard-copy data-entry forms based on an existing economics data file.

The main menu item **Setup** contains functions to set up printers for text and graphics. Hopper prints graphics indirectly after creating disk files compatible with the graphics printer established in **Setup**. Once you set up both a text and graphic printer, you will not need to set them up again unless you want to use a different printer. The configuration you specify will be used by both Hopper and Hopper Lite.

Strategy for Use.—The main use for Hopper is to select a list of appropriate treatments and then evaluate their economic utility. The *Tutorial* in the **Tools** submenu will demonstrate a typical usage of Hopper. The *Tutorial* will work fine when Hopper is first installed but may not work properly after you have modified some of Hopper's data files. The **Treatment Selection** submenu contains all of the functions for grasshopper control analysis. To develop and evaluate potential treatments, first use the arrow keys to move the highlighted bar to **Treatment Selection** on the main menu and press `<ENTER>`. *Consult* should then be highlighted in the submenu. Press `<ENTER>` to select *Consult* and begin the process to develop a list of appropriate treatments. *Consult* will guide you through this process and ask you for information along the way. In *Consult,* survey and treatment dates are entered. These are used to determine the average grasshopper life stage in *Consult* as a factor for selecting certain treatments. Note that these dates are also used later in the economic analysis to simulate treatment effect on forage availability for livestock. After *Consult* has been used, the treatment list is available to be used for economic analysis. *Economics* is listed below *Consult* in the **Treatment Selection** submenu.

After you select the appropriate economic data file from Hopper's list, Hopper presents onscreen data-entry forms that must be completed. You can accept all the default values except grasshopper density. Typically, of all the data requested by Hopper for the economic analysis, only grasshopper density needs to be entered. More experienced users may change treatment cost and efficacy on the Treatment form. There, scenarios for increasing swath width and the resulting decrease in cost and efficacy can be evaluated.

Once data are correct on an onscreen data-entry form, press `<F5>` to cause Hopper to continue to the next form or function. Most onscreen data-entry forms can just be bypassed by pressing `<F5>` to accept the displayed values when the form appears.

After all data have been entered, the analysis begins. Graphs of the forage and grasshopper simulations can be displayed and/or saved. The economic analysis uses results from the simulations to calculate the benefits and costs of each treatment. The final results can be saved and are also displayed on the screen. Experimenting ("gaming") by changing some values, such as grasshopper density or treatment date, can be very useful and interesting.

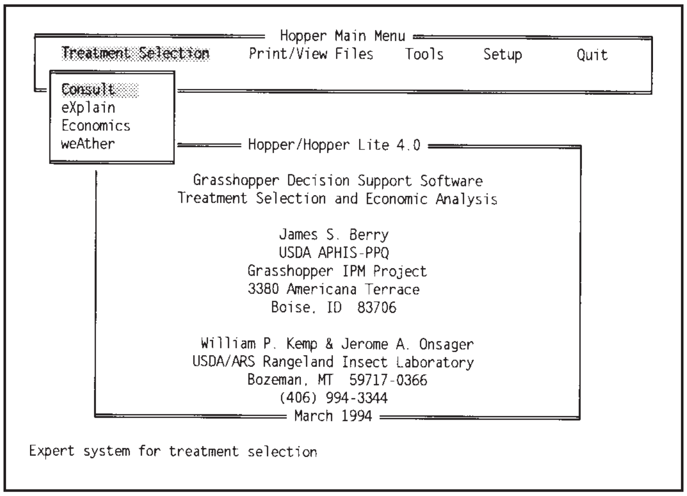
The Modules

Treatment Selection

***Consult.*—**The first item in the **Treatment Selection** submenu is *Consult* (fig. VI.2–4). This is the expert system that selects treatments that are valid for a given situation. Select *Consult* by moving the highlighted bar to *Consult* and pressing `<ENTER>`. The program will ask you relevant questions about the situation and current conditions. Often, *Consult* presents several options on the screen. To select one of the options, use the cursor keys (arrow keys), or you can use a mouse and click once on the left mouse button to move the highlighted bar to the appropriate option. Press <ENTER> or click once on the left mouse button to make your selection and continue with the consultation (fig. VI.2–5). At times you may be asked for data that you will need to type in from the keyboard (e.g., dates). In these situations, you will not use the cursor keys to select an option. Instead, you will type your response (fig. VI.2–6).

First, *Consult* will require you to select weather data for your site unless you have already loaded weather data. The *Weather* submenu will open and present three items. The most common choice is to create a weather file for the site. *Weather* also allows existing files to be used or new files to be created in a spreadsheetlike editor. Once Hopper has weather data for the site, Hopper will present an option to load existing facts into memory. If you choose to load existing facts, Hopper will provide a list of available files from which to select. Hopper will take control and guide the treatment selection process. Just answer any questions that are asked. A second window (Current Value Window) at the bottom of the screen will display the information you have entered.

More explanation or help for a question being asked can be obtained by pressing <F1>. These explanations will help you make sure that your answers are appropriate for the way they will be used in the system (fig. VI.2–7).



```
Hopper/Hopper Lite 4.0

Grasshopper Decision Support Software
Treatment Selection and Economic Analysis

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March 1994

Expert system for treatment selection
```
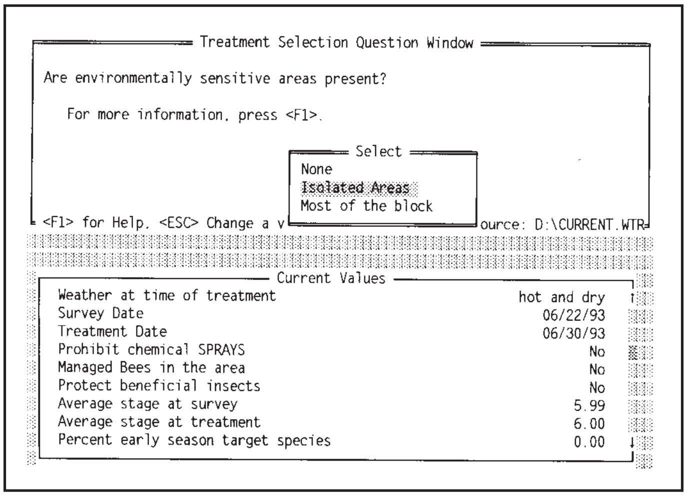
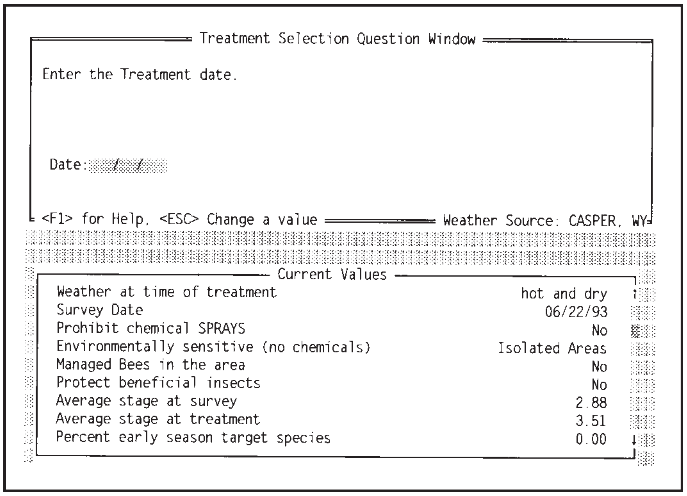

Figure VI.2–4—*Consult* is highlighted and will be selected by pressing <Enter>.



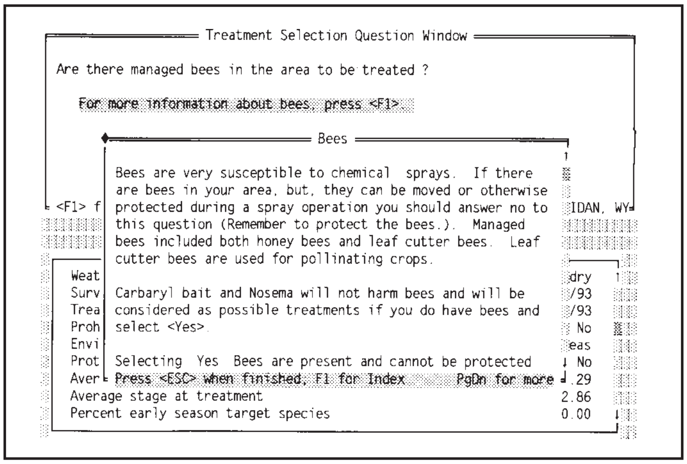
```
Are environmentally sensitive areas present?

   For more information, press <F1>.

<F1> for Help, <ESC> Change a v          ource: D:\CURRENT.WTR

Weather at time of treatment                  hot and dry
Survey Date                                      06/22/93
Treatment Date                                   06/30/93
Prohibit chemical SPRAYS                               No
Managed Bees in the area                               No
Protect beneficial insects                             No
Average stage at survey                              5.99
Average stage at treatment                           6.00
Percent early season target species                  0.00
```

Figure VI.2–5—Typical multiple-choice data entry in *Consult.*



```
Enter the Treatment date.


 Date:  /  /

<F1> for Help, <ESC> Change a value      Weather Source: CASPER, WY

Weather at time of treatment                  hot and dry
Survey Date                                      06/22/93
Prohibit chemical SPRAYS                               No
Environmentally sensitive (no chemicals)   Isolated Areas
Managed Bees in the area                               No
Protect beneficial insects                             No
Average stage at survey                              2.88
Average stage at treatment                           3.51
Percent early season target species                  0.00
```

Figure VI.2–6—Typical numeric entry in *Consult.*



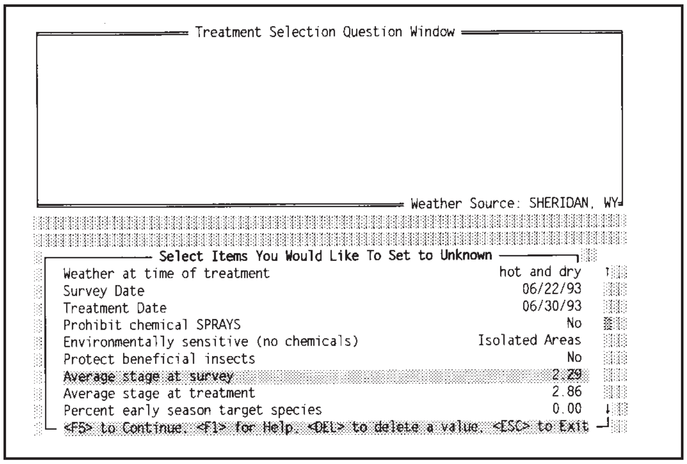
```
Are there managed bees in the area to be treated ?

   For more information about bees, press <F1>.

      Bees are very susceptible to chemical  sprays.  If there
      are bees in your area, but, they can be moved or otherwise
<F1> f protected during a spray operation you should answer no to   IDAN, WY
      this question (Remember to protect the bees.).  Managed
      bees included both honey bees and leaf cutter bees.  Leaf
      cutter bees are used for pollinating crops.
 Weat                                                              dry
 Surv Carbaryl bait and Nosema will not harm bees and will be      /93
 Trea considered as possible treatments if you do have bees and    /93
 Proh select <Yes>.                                                 No
 Envi                                                              eas
 Prot Selecting  Yes  Bees are present and cannot be protected      No
 Aver Press <ESC> when finished, F1 for Index      PgDn for more   .29
 Average stage at treatment                                       2.86
 Percent early season target species                              0.00
```

Figure VI.2–7—While entering information in the *Consult* expert system, help and ancillary information can be displayed.

To change or delete a value (e.g., an incorrectly entered value), you can temporarily exit from the Treatment Selection Question Window by pressing <ESC>. The cursor will be placed in the Current Values Window (fig. VI.2–8). There you can see or delete (highlight the value then press <DEL>) any values you have entered. When you are ready to continue with *Consult* again, press <F5>. Only deleted values and any new information needed by the expert system must be entered during the new or continued run with *Consult.* This feature allows you to build treatment lists rapidly from different scenarios. To quit without selecting a treatment and return to the main menu, press <ESC> while the cursor is in the Current Values Window.

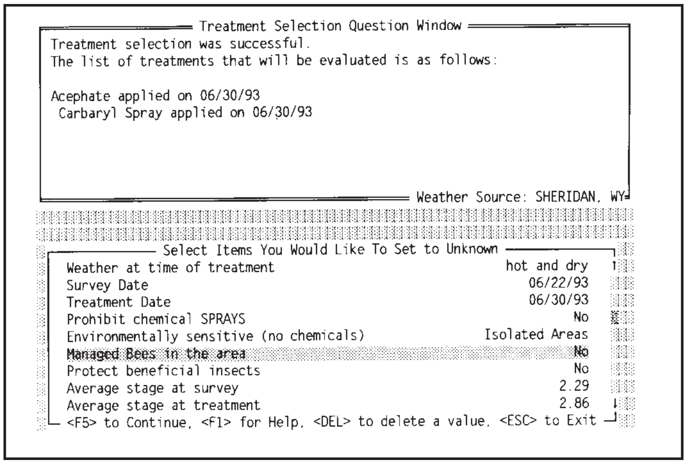
Note: After *Consult* is finished, the information that was entered by you for treatment selection can be saved to a file. This information can later be retrieved when you begin *Consult* again, as previously described. When asked for a file name to save facts, only the filename (eight or fewer characters in length), without an extension, should be entered (e.g., FACT2).

When *Consult* is finished, a list of treatments with corresponding application dates will be displayed (fig. VI.2–9). In some situations, other information will be displayed to show the outcome of the consultation. You could delete some facts and press <F5> to run another scenario. When you press <F5> without deleting any facts, you will be returned to the **Treatment Selection** submenu. Hopper will retain in memory the list of treatments you obtained from *Consult.* This list will be used each time you select the *Economics* module until *Consult* is run with different data.



```
Treatment Selection Question Window

Weather Source: SHERIDAN, WY

Select Items You Would Like To Set to Unknown
Weather at time of treatment                         hot and dry
Survey Date                                             06/22/93
Treatment Date                                          06/30/93
Prohibit chemical SPRAYS                                      No
Environmentally sensitive (no chemicals)          Isolated Areas
Protect beneficial insects                                    No
Average stage at survey                                     2.29
Average stage at treatment                                  2.86
Percent early season target species                         0.00
<F5> to Continue, <F1> for Help, <DEL> to delete a value, <ESC> to Exit
```

Figure VI.2–8—Screen used to delete data from *Consult.*



```
Treatment Selection Question Window
Treatment selection was successful.
The list of treatments that will be evaluated is as follows:

Acephate applied on 06/30/93
 Carbaryl Spray applied on 06/30/93

Weather Source: SHERIDAN, WY

Select Items You Would Like To Set to Unknown
Weather at time of treatment                         hot and dry
Survey Date                                             06/22/93
Treatment Date                                          06/30/93
Prohibit chemical SPRAYS                                      No
Environmentally sensitive (no chemicals)          Isolated Areas
Managed Bees in the area                                      No
Protect beneficial insects                                    No
Average stage at survey                                     2.29
Average stage at treatment                                  2.86
<F5> to Continue, <F1> for Help, <DEL> to delete a value, <ESC> to Exit
```

Figure VI.2–9—*Consult* ends by displaying a list of treatments that can be analyzed in the *Economics* module.

Explain.—The *eXplain* option tells you why Hopper selected or rejected treatments for a given consultation.

You may either:
1. View the explanation onscreen (Read)

Use <PageUp> and <PageDown> to move around the explanation. After reading the explanation, press <ESC> to return to the main menu.

2. Print the explanation (Print)

Follow the onscreen directions to print the explanation. See **Setup** for information on setting up your printer.

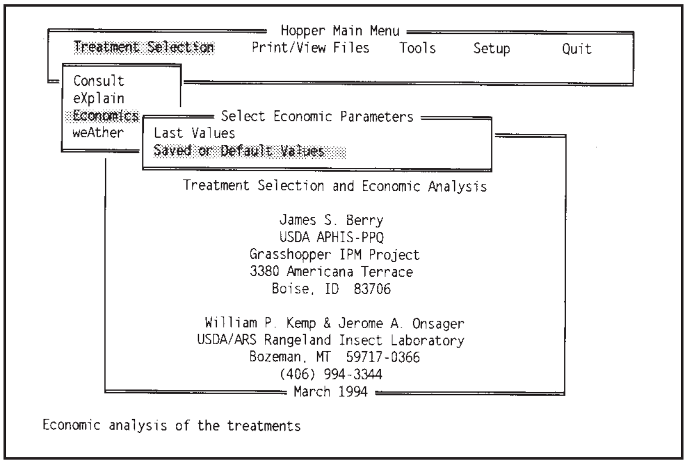
Economics.—*Economics* prompts you to enter economic and environmental data about the infested site. Then, Hopper runs the data through simulation models that provide an economic analysis of the treatments selected by *Consult.* By varying the data, you can evaluate the benefit–cost ratio of treatments for various scenarios. This allows you to determine
- Whether or *not* treatment is cost effective,
- Which treatment is most cost effective, and
- When to use the treatment for maximum effect.

The *Economics* module gives you access to a virtually unlimited number of scenarios for evaluating the economic robustness of the treatments that were selected by the *Consult* module (fig. VI.2–9). This flexibility and power come by using forage and grasshopper simulation models. The *Economics* module manages the models and the details of each simulation. Therefore, it is very easy for you to do the economic analyses. In fact, the only way a user even knows that models are being used is that a display indicates when a simulation is active.

After *Economics* is selected from the submenu, you must select an option to load economic data into Hopper (fig. VI.2–10). Information for the economic analysis is stored in files. The last information used by Hopper can be retrieved by selecting “Last Values.” Information for regional economic models provided with Hopper can be selected by choosing “Saved or Default Values.” In addition, any specific economic information you have saved can also be retrieved this way. Press `<F1>` for descriptions of the economic files. Usually, on the first run for a given area you will select the option for “Saved or Default Values” (existing data file). There are several data files that represent data typical for an area. For example, `NGP_CC.ec3` represents a northern Great Plains cow–calf operation. There is also a generic model available for areas that do not have a specific model. These models use data files that have the extension `.GN3` and can be used anywhere in North America. For a description of the economic models and data files currently available, see appendix B of this Users’ Guide or press `<F1>`.

Six data-entry windows are used to get information from you before the simulations are started. **Help and explanation (press `<F1>`) are available for most parameters.** See figure VI.2–11. These explanations should be read so that you will be able to enter correct information and understand the potential effects of a variable on the economic analyses. To change a value, use the arrow keys or `<TAB>` to move the highlight to the value. Type a new value or use `<DEL>` to edit the value. When you are finished entering information on an onscreen data-entry form, press `<F5>` to move to the next onscreen data-entry form.

The first onscreen data-entry form (Forage and Grasshopper Models) is for information used to simulate forage growth and grasshopper population dynamics (fig. VI.2.–12). Densities of grasshoppers that eat only grass and



```
Treatment Selection and Economic Analysis

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March 1994

Economic analysis of the treatments
```
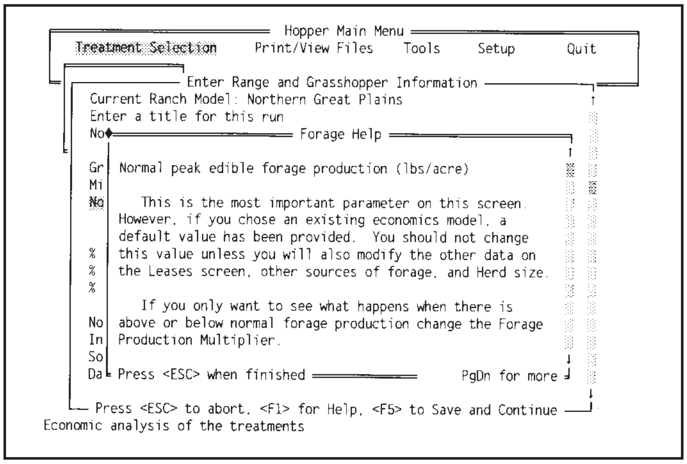
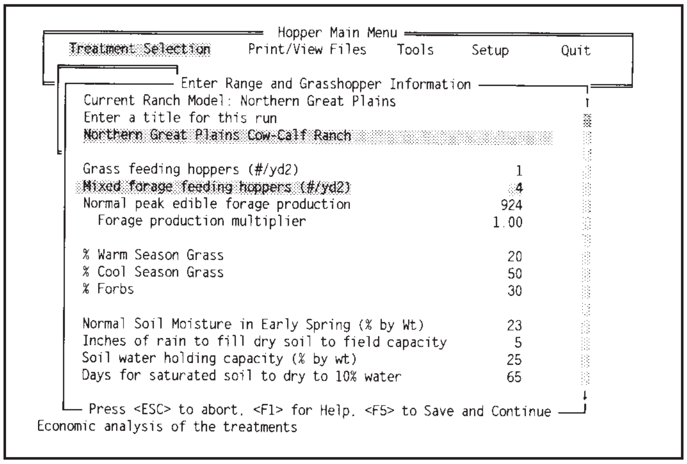

Figure VI.2–10—With *Economics* selected, Hopper then prompts for economic parameters file to use.



```
Current Ranch Model: Northern Great Plains
Enter a title for this run

Normal peak edible forage production (lbs/acre)

   This is the most important parameter on this screen.
However, if you chose an existing economics model, a
default value has been provided.  You should not change
this value unless you will also modify the other data on
the Leases screen, other sources of forage, and Herd size.

   If you only want to see what happens when there is
above or below normal forage production change the Forage
Production Multiplier.

Press <ESC> when finished                    PgDn for more
Press <ESC> to abort, <F1> for Help, <F5> to Save and Continue
Economic analysis of the treatments
```

Figure VI.2–11—Grasshopper and rangeland data entry with help information for peak standing crop displayed.



```
Current Ranch Model: Northern Great Plains
Enter a title for this run
Northern Great Plains Cow-Calf Ranch

Grass feeding hoppers (#/yd2)                          1
Mixed forage feeding hoppers (#/yd2)                   4
Normal peak edible forage production                 924
  Forage production multiplier                      1.00

% Warm Season Grass                                   20
% Cool Season Grass                                   50
% Forbs                                               30

Normal Soil Moisture in Early Spring (% by Wt)        23
Inches of rain to fill dry soil to field capacity      5
Soil water holding capacity (% by wt)                 25
Days for saturated soil to dry to 10% water           65

Press <ESC> to abort, <F1> for Help, <F5> to Save and Continue
Economic analysis of the treatments
```

Figure VI.2–12—Grasshopper and rangeland onscreen data-entry form.

those that eat mixed vegetation and, occasionally, a forage-production multiplier should be entered. Remember to press <F1> for more explanation for each parameter (fig. VI.–11 shows a help screen). The rest of the values are reasonable estimates if you do not have better information. Press <F5> to continue when you are satisfied with the values that are displayed. Percent forbs is calculated by Hopper, based on cool- and warm-season grasses.

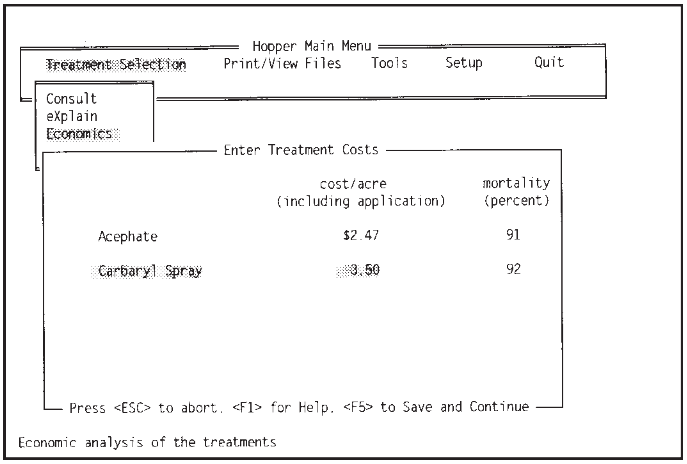
The second onscreen data-entry form (Treatment Cost) displays the list of treatments, with their costs and mortalities, selected by the *Consult* module (fig. VI.2–13). The total cost (material plus application cost) and mortality can be entered (press <ENTER> after typing each value) for each treatment except *Nosema* bait. Only cost can be entered for *Nosema* bait because mortality calculations are too complicated for most users. After all the costs have been entered correctly, press <F5> to accept your entries and continue to the next onscreen data-entry form.

The third onscreen data-entry form allows you to indicate the potential for multiple-year benefit from control. Be sure to read the information on the screen (fig. VI.2–14). Multiple-year benefits are calculated only by compounding single-year benefits over the number of years you enter on this onscreen data-entry form. This is the last screen of data presented when a generic model is used.

The fourth onscreen data-entry form (Hay Information) shows data used in the ranch economic model (fig. VI.2–15). Press <F5> to continue when you are satisfied with the values that are displayed. Only change this information if you have data for a specific ranch or a ranch typical for the treatment block. The values provided by Hopper are for a typical ranch in the area.

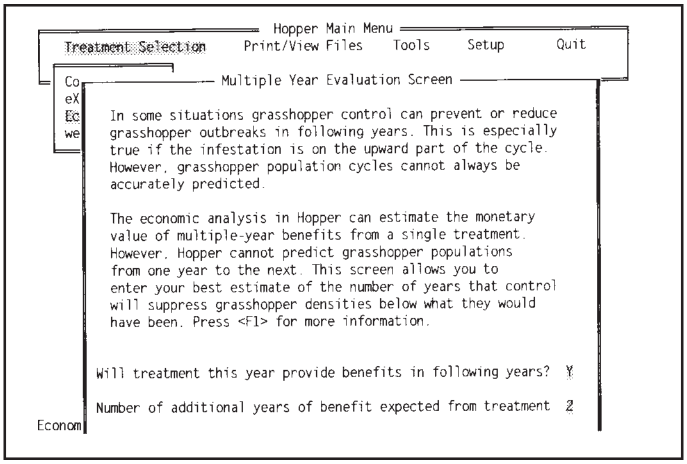
The fifth onscreen data-entry form (Herd Size) shows livestock data used in the ranch economic model (fig. VI.2–16). A land manager may choose to evaluate the effect of reduced herd size versus paying for grasshopper control. Press <F5> to continue when you are satisfied with the values that are displayed.

The sixth onscreen data-entry form (Lease Information) shows lease data used in the ranch economic model (fig. VI.2–17). Press <F5> to continue when you are satisfied with the values that are displayed. Only change this information if you have data for a specific ranch or a



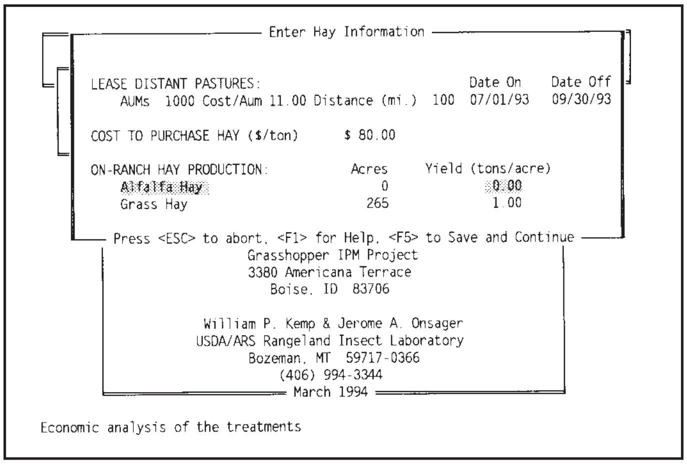
```
Enter Treatment Costs

                              cost/acre            mortality
                       (including application)     (percent)

    Acephate                    $2.47                 91

    Carbaryl Spray               3.50                 92

Press <ESC> to abort. <F1> for Help. <F5> to Save and Continue

Economic analysis of the treatments
```

Figure VI.2–13—Treatment cost and mortality onscreen data-entry form.



```
Multiple Year Evaluation Screen

  In some situations grasshopper control can prevent or reduce
  grasshopper outbreaks in following years. This is especially
  true if the infestation is on the upward part of the cycle.
  However, grasshopper population cycles cannot always be
  accurately predicted.

  The economic analysis in Hopper can estimate the monetary
  value of multiple-year benefits from a single treatment.
  However. Hopper cannot predict grasshopper populations
  from one year to the next. This screen allows you to
  enter your best estimate of the number of years that control
  will suppress grasshopper densities below what they would
  have been. Press <F1> for more information.


Will treatment this year provide benefits in following years?  Y

Number of additional years of benefit expected from treatment  2
```

Figure VI.2–14—Multiple-year onscreen data-entry form.


```
Enter Hay Information

LEASE DISTANT PASTURES:                              Date On    Date Off
   AUMs  1000 Cost/Aum 11.00 Distance (mi.)  100  07/01/93   09/30/93

COST TO PURCHASE HAY ($/ton)      $ 80.00

ON-RANCH HAY PRODUCTION:            Acres     Yield (tons/acre)
   Alfalfa Hay                          0               0.00
   Grass Hay                          265               1.00

Press <ESC> to abort. <F1> for Help. <F5> to Save and Continue
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                           (406) 994-3344
                             March 1994

Economic analysis of the treatments
```
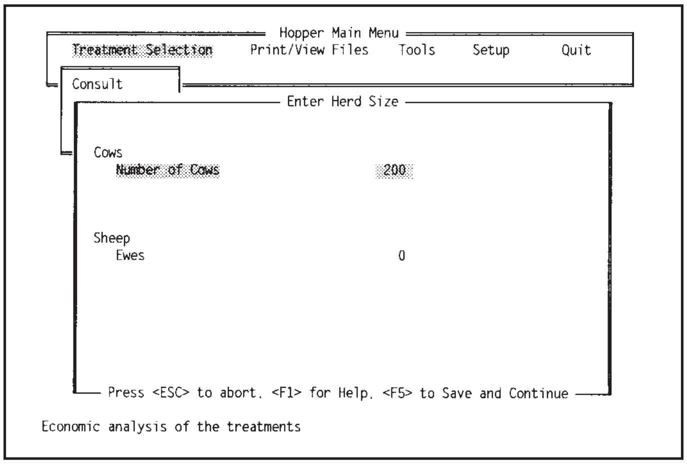

Figure VI.2–15—Hay information onscreen data-entry form and livestock data display.

ranch typical for the treatment block. The values provided by Hopper are for a typical ranch in the area.

Your last entered values for the economic analysis are saved automatically in a file called `Last.ec3`. Before the economic calculations begin, Hopper will ask if you would also like to save in a specific file the information just entered. You could reload and use this information later (fig. VI.2–10).

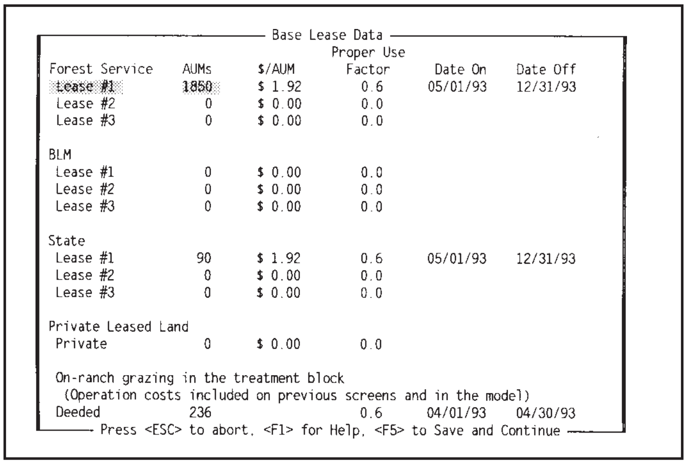
Then you will be asked if you want to continue with the economic analysis. This process can take several minutes on slow computers without math coprocessors. If `N` is entered, the economic analysis will end and the Treatment Selection menu will be displayed. If `Y` is entered, the economic analysis will proceed. The progress of the analysis can be monitored in a window in the center of the screen. You can view graphs after each treatment simulation. Then the ranch economic model will run. There is no user intervention required until the analyses are complete (fig. VI.2–18). (However, you can press `<Ctrl-Break>` to interrupt the economic analysis and return the Hopper menus.) The results are automatically saved in a file called `Results.rpt`. You will be asked if you would like to save this information in a specific file. **Note:** The Primal/Dual Degenerate Problem message should be ignored.

The results of a ranch economic analysis (not generic analysis) are displayed in a window (fig. VI.2–19). The



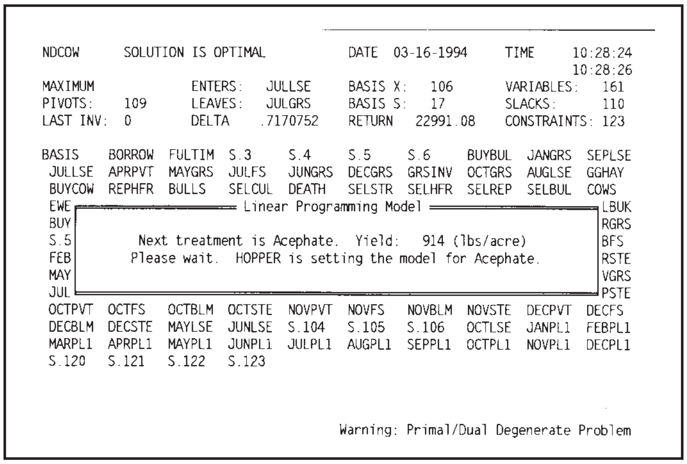
```
Enter Herd Size

Cows
  Number of Cows                    200

Sheep
  Ewes                              0

Press <ESC> to abort. <F1> for Help. <F5> to Save and Continue

Economic analysis of the treatments
```

Figure VI.2–16—Livestock herd-size onscreen data-entry form. Yearlings and sheep are not in the model.



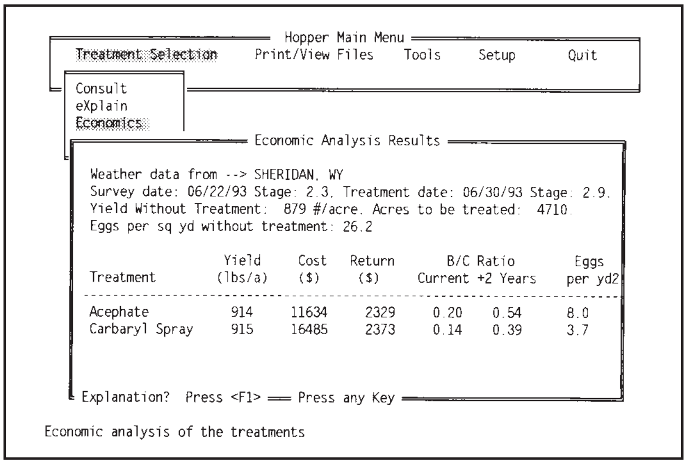
```
                              Base Lease Data
                                         Proper Use
Forest Service     AUMs      $/AUM         Factor       Date On      Date Off
 Lease #1          1850      $ 1.92          0.6        05/01/93     12/31/93
 Lease #2             0      $ 0.00          0.0
 Lease #3             0      $ 0.00          0.0

BLM
 Lease #1             0      $ 0.00          0.0
 Lease #2             0      $ 0.00          0.0
 Lease #3             0      $ 0.00          0.0

State
 Lease #1            90      $ 1.92          0.6        05/01/93     12/31/93
 Lease #2             0      $ 0.00          0.0
 Lease #3             0      $ 0.00          0.0

Private Leased Land
 Private              0      $ 0.00          0.0

 On-ranch grazing in the treatment block
  (Operation costs included on previous screens and in the model)
 Deeded             236                      0.6        04/01/93     04/30/93
        Press <ESC> to abort. <F1> for Help. <F5> to Save and Continue
```

Figure VI.2–17—Range lease onscreen data-entry form.



```
NDCOW     SOLUTION IS OPTIMAL         DATE  03-16-1994     TIME      10:28:24
                                                                     10:28:26
MAXIMUM              ENTERS:   JULLSE    BASIS X:   106     VARIABLES:   161
PIVOTS:    109       LEAVES:   JULGRS    BASIS S:   17      SLACKS:      110
LAST INV:  0         DELTA     .7170752  RETURN   22991.08  CONSTRAINTS: 123

BASIS    BORROW  FULTIM  S.3     S.4     S.5     S.6     BUYBUL  JANGRS  SEPLSE
 JULLSE  APRPVT  MAYGRS  JULFS   JUNGRS  DECGRS  GRSINV  OCTGRS  AUGLSE  GGHAY
 BUYCOW  REPHFR  BULLS   SELCUL  DEATH   SELSTR  SELHFR  SELREP  SELBUL  COWS
 EWE ═══════════════ Linear Programming Model ═════════════════════════ LBUK
 BUY                                                                    RGRS
 S.5       Next treatment is Acephate.  Yield:   914 (lbs/acre)         BFS
 FEB      Please wait.  HOPPER is setting the model for Acephate.       RSTE
 MAY                                                                    VGRS
 JUL                                                                    PSTE
 OCTPVT  OCTFS   OCTBLM  OCTSTE  NOVPVT  NOVFS   NOVBLM  NOVSTE  DECPVT  DECFS
 DECBLM  DECSTE  MAYLSE  JUNLSE  S.104   S.105   S.106   OCTLSE  JANPL1  FEBPL1
 MARPL1  APRPL1  MAYPL1  JUNPL1  JULPL1  AUGPL1  SEPPL1  OCTPL1  NOVPL1  DECPL1
 S.120   S.121   S.122   S.123


                                      Warning: Primal/Dual Degenerate Problem
```

Figure VI.2–18—Working screen for the ranch economic model.



```
                    Economic Analysis Results

 Weather data from --> SHERIDAN, WY
 Survey date: 06/22/93 Stage: 2.3. Treatment date: 06/30/93 Stage: 2.9.
 Yield Without Treatment:  879 #/acre. Acres to be treated:  4710.
 Eggs per sq yd without treatment: 26.2

                     Yield     Cost   Return      B/C Ratio        Eggs
 Treatment          (lbs/a)    ($)     ($)     Current +2 Years   per yd2
-------------------------------------------------------------------------
 Acephate             914     11634    2329     0.20     0.54      8.0
 Carbaryl Spray       915     16485    2373     0.14     0.39      3.7


Explanation?  Press <F1> ══ Press any Key

Economic analysis of the treatments
```

Figure VI.2–19—Final results from the economic analysis.

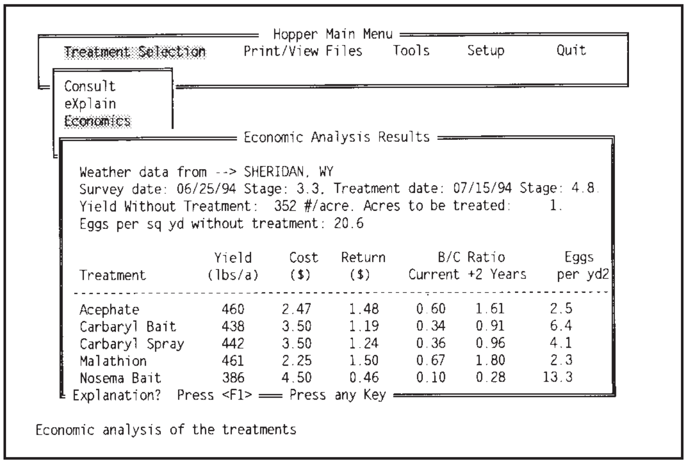
top few lines describe some general results of the analyses. The yields from the simulations are dependent on the scenario you described (Forage and Grasshopper Parameters Window) and on the weather scenario. The yield with grasshoppers accounts only for grasshopper consumption since the survey date or date of average fourth instar, whichever is earlier. The acres to be treated are calculated from the total Animal Unit Months (AUM's) grazed on the ranch, normal production of peak edible forage, and the proper use factor for each lease and total deeded land. [An AUM represents the average amount of forage consumed by one cow and one calf in 1 month—about 800 lb.] Therefore, the acres to be treated represent the total acres grazed by the ranch, except distant pastures on the Hay Information Screen (fig. VI.2–15). The eggs deposited per square yard is an estimate of the density of grasshopper eggs deposited by the end of October. The number of later instar grasshoppers that will be produced next year by these eggs depends on winter survival of the eggs and spring survival of the young instars.

The simulation results from the individual treatments (includes treatment mortality) and their corresponding application dates are listed in tabular form. The dollar return is total return for the ranch and is calculated from the value of an AUM (determined by the ranch economic model or entered by you in the case with the generic economic models), the cost of control, and the AUM's gained from control.

In some situations, the monetary value of forage saved from a treatment does not justify the application of that treatment for short-term economic reasons. However, there may be carryover benefits for the coming year that cannot be quantified economically. For example, the number of eggs deposited may be reduced, possibly preventing continued high densities of grasshoppers during the next growing season. Eggs deposited per square yard are shown for each treatment in the last column. These densities can be compared to the densities simulated for the untreated grasshopper populations (shown at the top of the window). In this way, relative effectiveness of the treatments (and application dates) for reducing next year's potential population can be evaluated. The return is the gain for the ranch if the treatment is applied. Cost is the total cost to treat the ranch (all AUM's on the lease data-entry form, fig. VI.2–17). The benefit–cost ratio (BC) shows if the benefit is greater than the cost (BC > 1.0). Two BC's are displayed. The first is for a single year. The second is combined for a single year plus the number of subsequent years shown. In the example, figure VI.2–20 shows the current year and 2 subsequent years. Although Hopper provides for benefits to be calculated for up to 10 years, 4 or 5 years is more realistic. See Help `<F1>` for additional information about multiple-year benefits. If current BC (single-year) is less than 1.0, the treatment may still be cost effective if you think you will get as much benefit in subsequent years (multiple-year effects BC).

The results from a generic economic analysis are very similar to the results from the ranch models. A difference to note is the acres to be treated. The generic analysis always shows 1.0 acre, whereas the ranch models show the number of treated acres associated with a ranch. The cost and return for the generic model are also for 1.0 acre, not for an entire ranch. In addition, the forage model is not used by the generic model. Therefore, yield is the normal peak edible forage production times the forage production multiplier minus the estimated forage consumption by grasshoppers (from the grasshopper model). In other words, the generic model calculates potential forage consumption by grasshoppers, but the ranch models calculate yield based on the interaction of forage growth with concurrent grasshopper forage consumption.

Hopper's recommendations are derived from the best scientific and field data available (including your own responses). However, remember that there is great



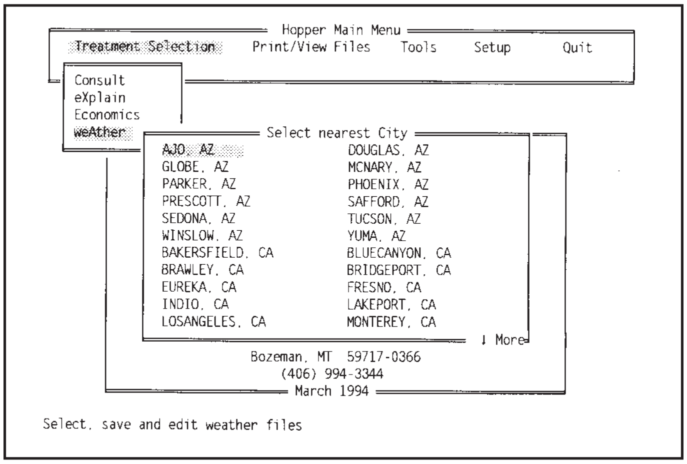
```
Weather data from --> SHERIDAN, WY
Survey date: 06/25/94 Stage: 3.3. Treatment date: 07/15/94 Stage: 4.8.
Yield Without Treatment:  352 #/acre. Acres to be treated:     1.
Eggs per sq yd without treatment: 20.6

                   Yield     Cost   Return       B/C Ratio        Eggs
Treatment         (lbs/a)    ($)     ($)     Current +2 Years   per yd2
-----------------------------------------------------------------------
Acephate            460     2.47    1.48      0.60    1.61       2.5
Carbaryl Bait       438     3.50    1.19      0.34    0.91       6.4
Carbaryl Spray      442     3.50    1.24      0.36    0.96       4.1
Malathion           461     2.25    1.50      0.67    1.80       2.3
Nosema Bait         386     4.50    0.46      0.10    0.28      13.3
```

Figure VI.2–20—Final results from the generic economic analysis.

variability in any biological system. Also, future events, such as drought or changes in the cattle market, cannot be quantified accurately and are not included here. Therefore, you should evaluate the strength of your decision by running Hopper and changing some of the values you enter. For example, decrease the grasshopper density by 20 percent. If BC is greater than 1.0 (assuming it was greater than 1.0 in the first run), then you can have greater confidence in the decision to use the specified treatment. However, if BC drops below 1.0, you should suspect that a decision to use the given treatment is not very robust (not a decision that can be made with much confidence). Gaming with the program in this way can be very informative and is one of the strengths of using computer models.

Weather.—Hopper uses simulation models to predict forage production and grasshopper phenology and oviposition. These models use temperature and precipitation information to make the predictions as accurate as possible. The *Weather* module allows you to retrieve, modify, and save temperature data that are used by Hopper. Currently, precipitation and temperature are generated and stored in a file. Both can be edited or updated for each day of the year in the Temperature Editor provided in the *Weather* module.

You can create average weather (using the weather generator provided with your copy of Hopper, fig. VI.2–21) or provide your own weather files. The files may have any filename but must have `.WTR` as the *file extension* (e.g., `mytemps.wtr`). For example, `mytemps.wtr` might look like this:



```
                    Hopper Main Menu
   Treatment Selection   Print/View Files   Tools   Setup   Quit
   Consult
   eXplain
   Economics
   weAther           Select nearest City
               AJO, AZ                DOUGLAS, AZ
               GLOBE, AZ              MCNARY, AZ
               PARKER, AZ             PHOENIX, AZ
               PRESCOTT, AZ           SAFFORD, AZ
               SEDONA, AZ             TUCSON, AZ
               WINSLOW, AZ            YUMA, AZ
               BAKERSFIELD, CA        BLUECANYON, CA
               BRAWLEY, CA            BRIDGEPORT, CA
               EUREKA, CA             FRESNO, CA
               INDIO, CA              LAKEPORT, CA
               LOSANGELES, CA         MONTEREY, CA
                                                    ↓ More
                      Bozeman, MT  59717-0366
                        (406) 994-3344
                          March 1994

 Select, save and edit weather files
```

Figure VI.2–21—Weather generator submenu to select the weather station nearest your site.

```
    1      14     –5     0.110
    2      13     –3     0.090
    3      17      0     0.000
etc.
```

Column 1 is day of the year (where 1 is January 1, 365 is December 31), column 2 is maximum temperature (°F), column 3 is minimum temperature (°F), and column 4 is precipitation (inches). Incomplete data sets are accepted (whole days can be missing). There must be at least one space between each column. In the Northern States, Hopper uses temperatures from April 1 through October 8. Make sure you have good data for these dates before running the *Consult* or *Economics* modules.

A spreadsheetlike Weather Editor is provided to allow you to edit temperatures and precipitation from several sources (average from sites in your area, created by the weather generator; weather files that you have previously edited or assembled using a text editor; or temperature data that are currently loaded into Hopper). Often you may want to evaluate the effect of generally warmer, cooler, wetter, or dryer conditions. The Weather Editor allows you to increase or decrease temperatures or precipitation for the entire year all at once. When you are finished editing, you may press `<F5>` to update the current temperatures in Hopper and, optionally, to save your changes to a file on the disk. Any file you save may be reloaded later for use by Hopper and/or more editing.

Print/View Files

***Graphs.*—**The *Graphs* option will allow you to print any graphs that were saved during an economic analysis. Note that your graphics printer must have been configured correctly at the time the graphs were created. Your graph will not print correctly if it was created for a printer different than the one you would like to use to print the graph (see **Setup** for more information on graphics printer setup, page VI.2–16).

Reports.—All of the information needed to duplicate a scenario is stored in Hopper's reports. The *Reports* option includes information entered in the *Consult* module and the *Economics* module and identity of the source of weather data. Hopper always saves the last run in the file `results.rpt`, even if you declined to save the results when prompted in the *Economics* module. Print Reports will display any report file on the screen or print it to the current printer (see **Setup** for more information on text printer setup, page VI.2–16).

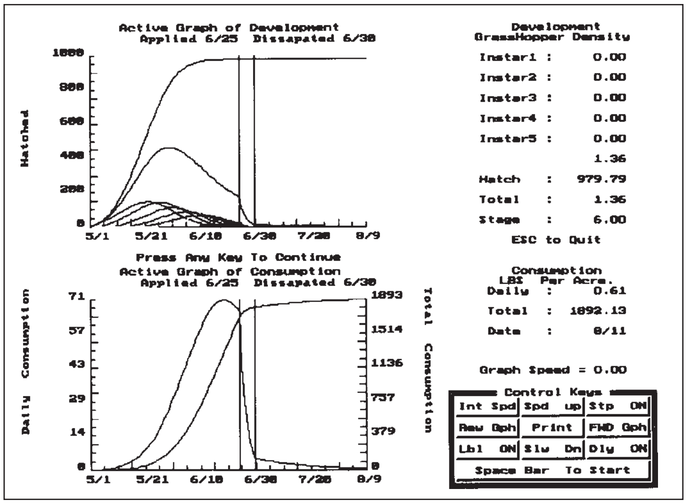
Forms.—You can create hard-copy data-entry forms that you can use to collect input data for Hopper. The forms contain default values from Hopper or values from any data you have saved during an economic analysis. These files have the ending `.ec3`. You can view the forms on the screen or print them. To create a form, see *Forms* in the **Tools** section, page VI.2–16.

Tools

Tutorial.—An automated *Tutorial* will show you a typical run through Hopper. After you modify some of the data files that arrived with Hopper, the tutorial may not run correctly. This problem happens because Hopper may require a different response based on the data that are entered. The tutorial cannot adjust to these changes in advance.

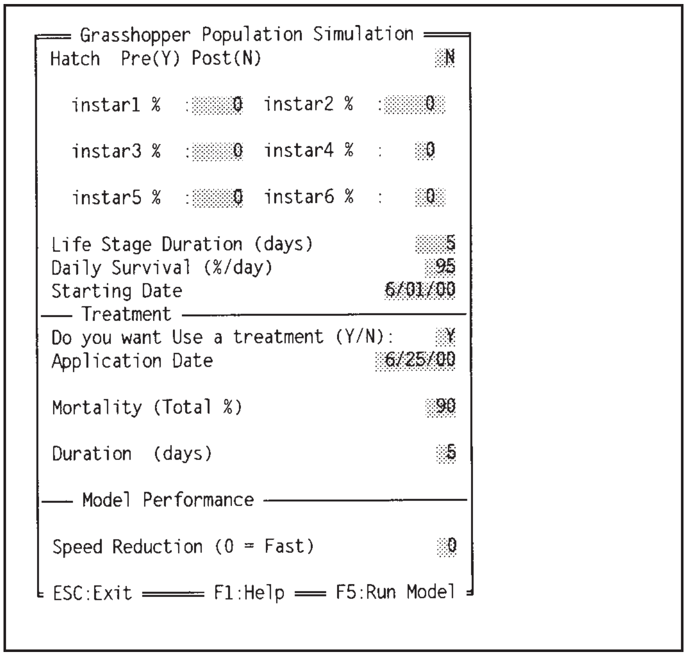
SimHop.—*SimHop* will simulate the general pattern of grasshopper development, forage consumption, and treatment mortality. This is useful for teaching or explaining why it may be too late or too early in the year to treat. The effects of long-lasting (long residual) treatments and timing of treatments can be demonstrated. Text and graphics are used to show the results (fig. VI.–22).

There are two modes of operation. First, a grasshopper population can be simulated from before spring egg hatch (preseason) to the end of season (fig. VI.2–23). You can set the timing, length and size of the hatch period. Second, *SimHop* can begin after egg hatch (midseason). In this case you can specify the density of each instar and starting date of the simulation (fig. VI.2–24). For each type of simulation, you can set the timing, length, and total mortality for a treatment. Therefore, via simulation, you can compare results of a slow-acting biological control applied early to results of a short-residual, fast-acting chemical spray applied later.



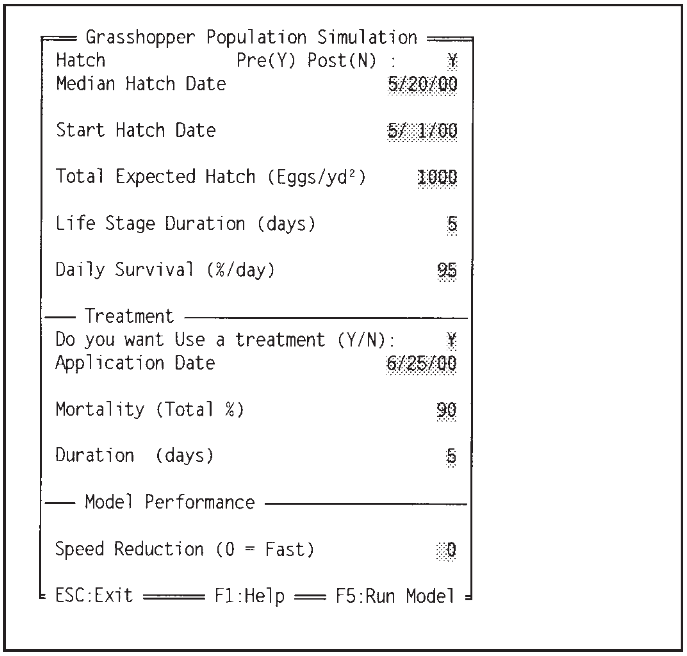
```
     Development
 GrassHopper Density
 Instar1 :      0.00
 Instar2 :      0.00
 Instar3 :      0.00
 Instar4 :      0.00
 Instar5 :      0.00
                1.36
 Hatch   :    979.79
 Total   :      1.36
 Stage   :      6.00
     ESC to Quit

     Consumption
   LBS  Per Acre.
 Daily   :      0.61
 Total   :   1892.13
 Date    :      8/11

 Graph Speed = 0.00
```

Figure VI.2–22—*SimHop* graphics display screen during a simulation beginning before egg hatch.



```
═══ Grasshopper Population Simulation ═══
Hatch  Pre(Y) Post(N)                   N

  instar1 %   :    0  instar2 %   :    0

  instar3 %   :    0  instar4 %   :    0

  instar5 %   :    0  instar6 %   :    0

Life Stage Duration (days)              5
Daily Survival (%/day)                 95
Starting Date                     6/01/00
── Treatment ─────────────────────────────
Do you want Use a treatment (Y/N):      Y
Application Date                  6/25/00

Mortality (Total %)                    90

Duration  (days)                        5

── Model Performance ─────────────────────

Speed Reduction (0 = Fast)              0

ESC:Exit ═════ F1:Help ══ F5:Run Model
```

Figure VI.2–23—Postegg-hatch onscreen data-entry form for *SimHop.*



```
╒══ Grasshopper Population Simulation ══╕
│ Hatch             Pre(Y) Post(N) :     Y
│ Median Hatch Date                5/20/00
│
│ Start Hatch Date                 5/ 1/00
│
│ Total Expected Hatch (Eggs/yd²)     1000
│
│ Life Stage Duration (days)             5
│
│ Daily Survival (%/day)                95
│
├── Treatment ────────────────────────────
│ Do you want Use a treatment (Y/N):     Y
│ Application Date                 6/25/00
│
│ Mortality (Total %)                   90
│
│ Duration  (days)                       5
│
├── Model Performance ────────────────────
│
│ Speed Reduction (0 = Fast)             0
│
╘ ESC:Exit ═════ F1:Help ══ F5:Run Model ╛
```

Figure VI.2–24—Pre-egg-hatch onscreen data-entry form for *SimHop.*

You can change between the prehatch model and the posthatch model by entering `Y` or `N` in the first field. The data-entry screen will switch so you can enter data for the model you chose. The last value on the screen is to slow the simulation so that the graph and data can be viewed in more detail. Press <`F1`> for more information on any current data value (where the cursor is flashing). Change any information on the screen; then press <`F5`> to run the simulation.

While *SimHop* is running, control keys in the lower right corner of the simulation results screen (fig. VI.2–22) can be used to slow, stop, reverse, or increment (step by step) the simulation. The layout of these keys represents the numeric keypad to the right on the computer keyboard. To use the numeric keypad during *Simhop,* turn off the Num-Lock. You could stop a simulation by pressing <`SpaceBar`> and then reverse the simulation by pressing the numeric keypad "4." <`SpaceBar`> will start and stop a simulation.

This tabulation explains the definitions of 1–9 on the numeric keypad:

Int Spd (7) Initial Speed	**Spd Up (8)** Increase speed	**Stp On (9)** Toggles step mode
Rev Gph (4) Reverse graph	**Print (5)** Print current screen to a file	**Fwd Gph (6)** Forward direction for graph
Lbl ON (1) Toggle numeric output to screen	**Slow Dn (2)** Decrease graph speed	**Dly ON (3)** Toggle delay for graph

The consumption rates used in *SimHop* are based on laboratory measurement for *Melanoplus sanguinipes.* Therefore, *SimHop* should be used to help with general understanding of grasshopper population dynamics, not to make precise estimates of forage loss.

Maps.—*Maps* allows you to select and view grasshopper hazard maps for several States. To select a State, move the highlighted bar to the State desired and press <`ENTER`>. When you are finished viewing the map, press <`ENTER`> to continue. To exit the *Maps* module, press <`ESC`> at the State selection submenu.

Economic Threshold.—Hopper can estimate the density of grasshoppers for a specified BC ratio. This estimate is also dependent on grasshopper life stage and species composition and current economic variables. The grasshopper density that corresponds to the BC ratio can be considered an economic threshold. In some situations, you might specify a BC ratio that cannot be achieved—one that is either too high or too low. Hopper will inform you when this situation occurs.

The *Economic Threshold* calculator will first run *Consult* to develop a list of treatments and then allow you to select one of those treatments and enter a BC ratio. Remember that BC ratios greater than 1.0 indicate a treatment profit for the single-year analysis. Next, an economic analysis will begin similar to the analysis in the **Treatment Selection** module. Generic models cannot be used for the *Economic Threshold* calculator. You can enter or modify any of the data to match your situation.

Hopper will run the analysis several times to find the economic threshold. This may take 3 minutes on a 486 computer. However, no intervention is required until the final results are presented. Results can be saved to a report file.

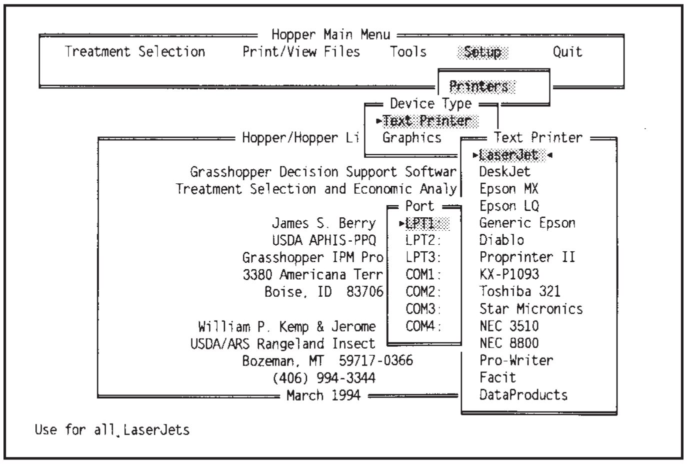
Forms.—You can create and print hard-copy data-entry forms that you can use to collect input data for Hopper. The forms will contain default values from Hopper or values from any data you have saved during an economic analysis. These can be a handy way to prepare to run Hopper because you will have at hand all the information Hopper requests. Select *Forms* and a submenu of items will be presented. You can create, view, or print a form. When you select "Create," a list of economic data files in your Hopper directory is displayed. These files have the ending `.ec3`. Highlight a file and press `<ENTER>` to create the form for those data. Next, you could view the new form on the screen or print it.

Setup

Printers.—Hopper uses the printer type (text and graphics) you select here to format properly the documents it prints. The printer information is stored in a file. Therefore, you need only select a printer once, unless you change printers. Select both a text printer and a graphics printer.

Text Printers.—Hopper will print existing reports and information in *eXplain* to your text printer. With Text Printers highlighted on the menu, press `<ENTER>` and a list of printers will appear (fig. VI.2–25). Use the arrow keys to highlight your text printer (or a similar model). If your printer is not listed, check to see if it is compatible with any other printers listed. The Epson printer is very common, and many printers are compatible with it. If your actual printer-model is not listed in Hopper, try selecting Epson instead. The text printer and graphics printer are often the same. However, you need to set up both types of printers in Hopper if the default printers are not acceptable. The text printer must be connected to a printer port (e.g., LPT). If your printer is connected to a COM port, you can place a Mode command in the file `autoexec.bat` to route the printer data through LPT1. For example, if your printer is attached to the serial port COM1, you should place this line in the `autoexec.bat` file:

```
mode lpt1=com1
```

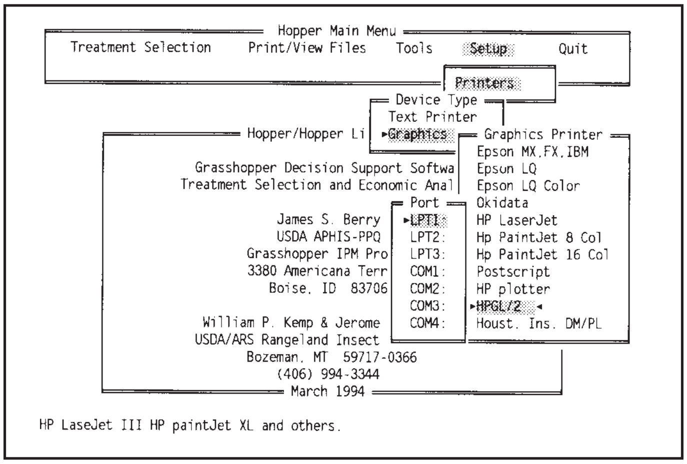


Use for all LaserJets

Figure VI.2–25—Setup submenu selected and Text Printer highlighted.

Graphics Printers.—Hopper does not print graphics information directly to a printer. Instead, graphics are "printed" to a file. The format of the graphics file is determined by the graphics printer you select here. The advantage of having the graphics in a file is that they can be imported into a word processor or graphics software. For example, you can import Hopper's graphics (from the simulation models and *SimHop)* into your word processor document. The Hewlett–Packard Pen Plotter is the default graphics printer. Graphics files for this graphics printer consist of lines and end points (vector graphics), and the format is HPGL/2. Therefore, with this format, graphics can be reproduced at the maximum resolution of the printer device, and most graphics editors (such as Lotus Freelance™ and Harvard Graphics®) can import and edit them. Laser-printer or dot-matrix printer output cannot be imported into these graphics editors. Hopper prints only graphics from the **Print/View Files** submenu (see Graphs, page VI.2–13). Therefore, the graphics printer port is not used if you will only import graphics files into other software without ever printing directly. **Note:** Remember that if you select a dot matrix or laser printer, you will need at least 512 to 1024 KB of expanded memory (EMS). See the Installation section at the front of this manual for instructions for configuring your computer's memory.

To change the graphics printer, highlight “Graphics” on the menu and press <ENTER>; a list of printers will appear (fig. VI.2–26). Use the arrow keys to highlight your printer (or similar model) and press <ENTER>. If your printer is not listed, check to see if it is compatible with any other printers listed. Again, select “Epson” as first try if your printer-model is not listed. Next, a menu of ports for the graphics printer will be displayed. The normal port is LPT1. Select the correct port; then press <ENTER>. The text printer and graphics printer are often the same. However, you need to set up both types of printers in Hopper if the default settings are not acceptable. **Note:** Some graphics printers (dot matrix and some laser printers) will not work in Hopper unless the computer has about 512 KB of EMS (expanded) memory available.



```
HP LaseJet III HP paintJet XL and others.
```

Figure VI.2–26—Setup submenu selected and Graphics selected with the Hewlett–Packard Pen Plotter highlighted (HPGL/2).

Selected References

Berry, J. S.; Kemp, W. P.; Onsager, J. A. 1991. Integration of simulation models and an expert system for management of rangeland grasshoppers. AI Applications 5: 1–14.

Berry, J. S.; Hanson, J. D. 1991. A simple, microcomputer model of rangeland forage growth for management decision support. Journal of Production Agriculture 4: 491–499.

Berry, J. S.; Kemp, W. P.; Onsager, J. A. 1995. Within-year population dynamics and forage destruction model for rangeland grasshoppers (Orthoptera: Acrididae). Environmental Entomology 24: 212–225.

Davis, R. M.; Skold, M. D.; Berry, J. S.; Kemp, W. P. 1992. The economic threshold for grasshopper control on public rangelands. Journal of Agricultural and Resource Economics 17: 56–65.

Foster, R. N.; Reuter, K. C.; Gourd, J. M.; Enis, J.; Wooldridge, A. W. 1983. Field experiments on the toxicity of acephate for control of grasshoppers (Orthoptera: Acrididae) on rangeland. Canadian Entomologist 115: 1163–1168.

Hanson, C. L.; Cumming, K. A.; Woolhiser, D. A.; Richardson, C. W. 1994. Microcomputer program for daily weather simulation in the contiguous United States. Misc. Publ. 114. [Place of publication unknown]: U.S. Department of Agriculture, Agricultural Research Service. 22 p.

Kemp, W. P.; Onsager, J. A. 1986. Rangeland grasshoppers (Orthoptera: Acrididae): modeling phenology of natural population of six species. Environmental Entomology 15: 924–930.

Kemp, W. P.; Onsager, J. A.; Lemmon, H. A. 1988. Rangeland grasshopper treatment selection: application of expert system technology to decision support for resource based management. AI Applications in Natural Resource Management 2: 1–8.

Kemp, W. P.; Kalaris, T. M.; Quimby, W. F. 1989. Rangeland grasshopper (Orthoptera: Acrididae) spatial variability: macroscale population assessment. Journal of Economic Entomology 82: 1270–1276.

Onsager, J. A. 1978. Efficacy of carbaryl applied to different life stages of rangeland grasshoppers. Journal of Economic Entomology 71: 269–273.

Onsager, J. A. 1983. Relationships between survival rate, density, population trends, and forage destruction by instars of grasshoppers (Orthoptera: Acrididae). Environmental Entomology 12: 1099–1102.

Onsager, J. A. 1984. A method for estimating economic injury levels for control of rangeland grasshoppers with malathion and carbaryl. Journal of Range Management 37: 200–203.

Onsager, J. A. 1988. Assessing effectiveness of *Nosema locustae* for grasshopper control. Montana AgResearch. 5(3): 12–16.

Onsager, J. A.; Henry, J. E.; Foster, R. N. 1980. A model for prediction efficacy of carbaryl bait for control of rangeland. Journal of Economic Entomology 73: 726–729.

Software Credits

Borland Pascal 7.0, Borland International, Inc., Scotts Valley, CA.
Pascal Compiler.

Object Professional®, TurboPower Software, Scotts Valley, CA.
User interface and general programmer's software toolbox for Turbo Pascal.

Turbo Analyst™, TurboPower Software, Scotts Valley, CA.
Analytical tools in an integrated development environment, including Pascal formatter, cross referencer, execution timer, execution profiler, program indexer, and program lister.

Tlib Version Control™, Burton Systems Software, Cary, NC.
Source code librarian.

PCX Programmer's Toolkit™, Genus Microprogramming, Houston, TX. Routines to display, save, scale, and print PCX images.

BLP88, Eastern Software Products, Alexandria, VA. Linear programming with bounded variables for the IBM-PC, used for the economic analysis.

INGRAF 6.0™, Integrated Graphics Library for Pascal, SutraSoft, Sugar Land, TX. Routines for scientific plotting and graphs.

INSTALIT™, Helpful Programs, Inc., Huntsville, AL. Hopper installation program.

USCLIMAT.BAS, Weather generator, U.S. Department of Agriculture, Agricultural Research Service, Northwest Watershed Research Center, Boise, ID. (Contact: C. L. Hanson.)

RanchMod, Economics Module, M. Skold and R. Davis, Department of Agricultural and Resource Economics, Colorado State University, Fort Collins, CO.

Appendix A How Hopper Works and Why

By Larry Zaleski

Why You Should Know How Hopper Works.—You should know how Hopper works to help accomplish your treatment responsibilities skillfully and accurately.

Whether you're a rancher or government official, professional and financial considerations demand that you work skillfully and accurately. Applying pesticides when *not* needed may threaten the environment and waste money. Conversely, failure to apply pesticides when conditions warrant may jeopardize native rangeland and potentially threaten the local ranching economy.

Hopper helps you decide objectively whether to treat or *not.* But you must use Hopper correctly for good results. And to use Hopper correctly, you must know how the program works.

What You Should Know.—You should be familiar with the following:

- How Hopper can save time, improve accuracy, and save money
- What the economic research shows
- How Hopper's components work together
- What the expert system (Consult) does
- What the forage model (RangeMod) does
- What the grasshopper model (HopMod) does
- What the economics model (RanchMod) does
- Your role

As you become familiar with Hopper, you will become more knowledgeable about treatment technology, rangeland ecology, and ranching economics.

How Hopper Can Save Time, Improve Accuracy, and Save Money.—Hopper saves time, improves accuracy, and saves money by

- Automating expensive and time-consuming tasks, and
- Using ecological and economic information previously unavailable to decisionmakers.

Automating Expensive and Time-Consuming Tasks.— Hopper automates many tasks that require time, money, and personnel to accomplish. You still collect information about local conditions, but with Hopper, your treatment decisions are greatly *improved* with little additional effort. To understand the value of automation, you should know

- What Hopper does automatically,
- How Hopper automates tasks, and
- How automation improves treatment decisionmaking.

What Hopper Does Automatically.—Hopper automatically

- Estimates the average instar of a grasshopper population (for integration with field data);
- Estimates the effects of precipitation on forage production;
- Estimates forage production and, then, forage loss to grasshoppers;
- Chooses treatments based on local conditions; and
- Determines if treatment is cost effective.

Without automation and computer simulation, many of these tasks are impractical or more likely to be completed with errors.

How Hopper Automates Tasks.—Hopper automates tasks by integrating an expert system with simulation and economic models (Berry et al. 1991, 1992).

Hopper's expert system is rule-based. Rule-based expert systems are computer programs consisting of rules. These rules are the same as those used by human experts, but the expert system uses the computer's ability to apply logic, instead. For example, an expert system program for reacting to a *traffic light* might look like this:

IF THE LIGHT IS RED: Stop and wait for the light to turn green.
IF THE LIGHT IS GREEN: Go on.

IF THE LIGHT IS YELLOW: Slow down, and...
— If the light turns red, stop, wait for it to turn green, then go on.
— If the light doesn't turn red, go on.

The computer runs through the program until it encounters an "if statement" that matches the current condition. Then the program follows the programmed procedure. Hopper's expert system works similarly, but it's designed to select treatments. Hopper asks questions, matches your answers to its rule base, then lists treatments accordingly.

Models, on the other hand, are mathematical formulas that imitate events in the real world. Models allow you to make predictions and estimates about events in the real world. Previously, such models were too time-consuming and complicated for everyday use. Only scientists could use them. But the personal computer has changed that.

How Computer Automation Improves Decisionmaking.—Computer automation improves decisionmaking in two ways. First, automation is comprehensive. That is, Hopper requires that you answer questions needed to make accurate decisions, each time. Critical factors, including those you might *not* ordinarily consider, are routinely considered. Without this prompting, you might ignore some factors to save time or because you don't know how to evaluate them.

Second, automation is consistent. It's consistent because users answer critical questions each time and because Hopper evaluates data the same way each time—something that people seldom do. Consequently, two people independently entering the same data into Hopper achieve the same results each time. Thus, Hopper transforms treatment decisionmaking into a more objective and scientific process.

Simulation, completeness, and consistency result in improved accuracy at roughly the same cost.

Using Ecological and Economic Parameters Previously Unavailable to Decisionmakers.—Hopper achieves improved accuracy because it uses parameters and variables that were previously impractical or unavailable (Davis et al. 1992). Even though these parameters were important, they were often not used because they were too costly and time consuming to obtain or because they could not be analyzed fast enough to help. As a result, treatment decisions were based on partial information.

Recently, however, researchers have shown that many of these unused but critical variables can be simulated mathematically. Other variables have been determined by the Grasshopper Integrated Pest Management Project and cooperators.

Before Hopper, Treatment Decisions Were Based on Less Extensive Information.—Hopper estimates critical variables previously unavailable to decisionmakers. Biologists and economists knew these variables were important, but only well-funded research projects could collect and analyze the data. And the results of their analysis usually came too late to help.

But the economic basis for control of grasshoppers on rangeland depends on several variables, not just grasshopper density (Davis et al. 1992). These critical variables include

- Rangeland productivity,
- Soil moisture,
- Livestock prices,
- Accessibility and cost of alternative forage,
- Effectiveness and timing of treatments, and
- Grasshopper numbers and composition.

These variables, however, are difficult and expensive to measure. Many could not be analyzed quickly. And few scientists, ranchers, or government officers could measure and interpret all of the variables. Consequently, no one could integrate the critical variables into a practical decision support system.

Critical Variables Can Be Estimated Mathematically.—Recently, researchers demonstrated that many critical variables could be estimated mathematically (Berry and Hanson 1991, Berry et al. 1995, Dennis et al. 1986, Kemp and Onsager 1986). Therefore, for some variables, mathematical simulation provides an alternative to sampling and measurement.

When combined with a personal computer, mathematical simulations provide quick, reliable estimates of difficult-to-measure variables. For the first time, critical variables are routinely available to decisionmakers. What's more, estimated variables can be combined with economic calculations to determine if treatment is cost effective.

What the Economic Research Shows.—The economic research reveals three key facts (Davis et al. 1992):

1. Decisionmakers should use an economic threshold as their basis for applying treatment.
2. Economic justification for grasshopper control programs depends on several variables, *not* just grasshopper population density.
3. Economic justification for grasshopper control programs varies from place to place and year to year.

Decisionmakers Should Consider Economic Threshold in Their Decision About Applying Treatment.—Economics is a primary justification for treating grasshopper infestations. So ranchers should treat grasshoppers *not* to reduce their numbers but to improve the profitability of the ranch. Reducing grasshopper numbers is only a tactic for managing the rangeland resource.

From a ranching perspective, even rangeland management—a continuous effort which some use as a justification for grasshopper control—is simply an economic endeavor aimed at preserving rangeland productivity. Preserving productivity preserves profit. To illustrate the profit motive: one way to manage the land and prevent range damage during a grasshopper outbreak is to remove cattle. But this option is unprofitable, so ranchers tend to avoid cattle removal when possible. Generally, ranchers seek more profitable alternatives.

Environmental factors are important, too, and may *prevent* treatment. But in most cases, the basis for your decision to treat or not is economic.

To apply an economic threshold to treatment decisions confidently, you need to understand the concept of the economic threshold and the concept that treatment is an investment.

The Economic Threshold.—The economic threshold is the population density of a pest at which the cost of management intervention equals the resulting benefit from controlling it. The economic threshold varies with the benefits and the cost of treatment (Davis et al. 1992).

When Does Treatment Become Profitable?—The economic threshold is reached when the benefit–cost ratio equals 1 or more.

Hopper determines the economic threshold by calculating the benefits of treatment, then dividing the benefits by the cost. This measure is called the benefit–cost ratio (BC):

$$BC = \frac{\text{Benefits}}{\text{Cost}}$$

When the benefits equal the cost, the ratio is equal to 1 and the economic threshold is achieved. For example:
Benefits of treatment = $5,000
Cost of treatment = $5,000

$$BC = \frac{\text{Benefits}}{\text{Cost}} \begin{matrix} = \\ = \end{matrix} \frac{\$5{,}000}{\$5{,}000} = 1$$

BC's greater than 1 are profitable, but BC's less than 1 are unprofitable. The economic threshold (BC = 1) is the break-even point.

The cost of grasshopper control includes wages and the cost of chemicals, baits, and equipment. The benefit of grasshopper control, on the other hand, is equal to the *value* of the forage saved by treating grasshoppers.

Treatment Is an Investment.—Treatment is an investment in the agricultural economy. You apply treatment to attain or improve profitability.

Typically, you expect a return on your investments. For example, if you invest $100 in a savings account, you expect to collect interest, which is a return. If the account pays 5 percent simple interest, then after a year you would have $105. The BC of your account would be $105 ÷ $100 = 1.05. Because the BC is greater than 1, the account is profitable.

You would *never* knowingly invest in a savings account that loses money (an account whose BC is less than 1). Investing when the BC is less than 1 is unprofitable, and thus, economically unjustified.

Treatment, too, should show a return. Treating when the BC is less than 1 is unprofitable, and thus, economically unjustified.

Variables Affecting Economic Justification of Grasshopper Control Programs.—At least seven variables determine the economic justification for grasshopper control programs on rangeland:

- Rangeland productivity and composition,
- Precipitation and soil moisture,
- Accessibility and cost of alternative forage,
- Effectiveness of treatment,
- Cost of treatment,
- Timing of treatment, and
- Grasshopper population density, life stage, and species composition.

Put simply, these variables determine the *value* of the forage grasshoppers eat (the damage grasshoppers cause) and how much damage can be prevented. The interaction between critical variables is complex.

For example, if rangeland produces too much or too little forage, you cannot economically justify treatment. If *excess* forage is produced, there is enough to feed both grasshoppers and livestock, so you cannot justify treatment. On the other hand, if too little forage is produced, there is no forage to protect, so again, you may not be able to justify treatment purely based on forage value.

Consequently, the effects of the variables below assume that there is forage to protect, but *not* too much or too little. Otherwise, some of the following information would contradict. In practice, Hopper accounts for the effects of forage production automatically.

Rangeland Productivity and Composition.—On *highly productive* rangeland, you can economically justify treatment at lower grasshopper population densities than you can on less productive rangeland (Davis et al. 1992). This is true because treatment saves more forage per acre on highly productive rangeland.

The more forage you save per acre, the lower the cost per unit of forage saved and the greater your benefit for a given per-acre treatment cost. Consequently, on productive rangeland, you can treat *fewer acres* and still get the same per-acre benefit. The fewer acres you treat, the lower the cost.

In addition, some forage species are more valuable than others. Generally, the more valuable the forage, the easier it is to justify treatment.

Precipitation and Soil Moisture.—During *dry years*, you can economically justify treatment at lower grasshopper population densities than in years of normal or high precipitation.

Precipitation is the most important factor affecting rangeland productivity (Berry et al. 1991). Obviously, if it doesn't rain or snow, forage won't grow. When forage is scarce, its value increases because you must supplement it by buying hay or leasing additional land. Remember that, although the value of the forage may increase in dry years, the amount that will be protected by controlling grasshoppers is reduced. Hopper considers both of these factors.

In contrast, during normal and wet years, when forage is plentiful, there is often enough forage to feed both livestock and grasshoppers—even at high grasshopper population densities.

Hopper evaluates the effect of precipitation by calculating soil moisture.

Accessibility and Cost of Alternative Sources of Forage.—When alternative sources of forage are *expensive* or *inaccessible*, you can justify treatment at lower grasshopper population densities than when prices are low and forage accessible. This is true because when alternative sources of forage are expensive, you pay more to supplement or replace your existing forage. Therefore, your existing forage is worth more, and you can justify paying more to protect it.

Effectiveness of Treatments.—Other things being equal, when treatment is highly effective, you can justify treatment at lower grasshopper population densities than when treatment is ineffective. The more effective treatments are, the greater their value, and the higher the benefit–cost ratio.

Cost of Treatment.—When treatment is inexpensive, you can justify treatment at lower grasshopper densities than when treatment is expensive. Several factors influence the cost of treatment, including the price of pesticides, biological control agents, equipment, and personnel. In addition, the cost of treatment varies with demand. In years with lots of spraying, sprayers demand higher fees. Clearly, you need higher grasshopper densities to justify treatment at $4 per acre than you do at $2.25 per acre.

Timing of Treatment.—Timing influences the effectiveness and value of treatment. If you treat too early or too late, you reduce effectiveness. If you treat too early, many grasshopper eggs are still unhatched and will be unaffected. And if you treat too late, the forage is already eaten and next year's eggs are laid. In either case, the benefits are reduced.

Grasshopper Population Density and Composition.—Clearly, you can more readily justify treatment at higher grasshopper population densities than you can at lower grasshopper population densities. The higher their population density, the more forage grasshoppers eat. If the grasshopper density reaches the economic threshold, then grasshoppers literally eat up your profits.

In addition, species composition is important. Some grasshopper species do more harm than others. You can justify treating more-harmful species at lower densities than less-harmful species.

But as you've seen, several factors, in addition to grasshopper population density and composition, determine the economic threshold.

The Economic Justification for Grasshopper Control Varies From Place to Place and Year to Year.—Because the variables affecting the cost effectiveness of treatment vary from place to place and year to year, the economic justification for grasshopper control varies, too.

Conditions vary from place to place. For example, one pasture is more productive than the next, or one county has normal precipitation, while another is dry. Consequently, you may treat grasshoppers profitably at 1 location when densities reach 18 per square yard but not at another location until they reach 25 per square yard.

Similarly, conditions vary from year to year. Over time, a ranch may experience fluctuating precipitation, livestock prices, and lease costs. In 1 year grasshoppers may be worth treating at 30 per square yard; the following year, grasshoppers may be worth treating at 20 per square yard.

Normal variation of ranching conditions demands a flexible response to grasshopper treatment. Hopper provides flexibility by accounting for differences in conditions that vary with location and time.

How Hopper's Programs Work Together.—Hopper uses three kinds of software technology to assist you in making treatment decisions (fig. VI.2–27):

1. An expert system—to select treatments,
2. Simulation models—to estimate difficult-to-measure variables, and
3. An economic model (ranch model)—to determine if treatment is cost effective.

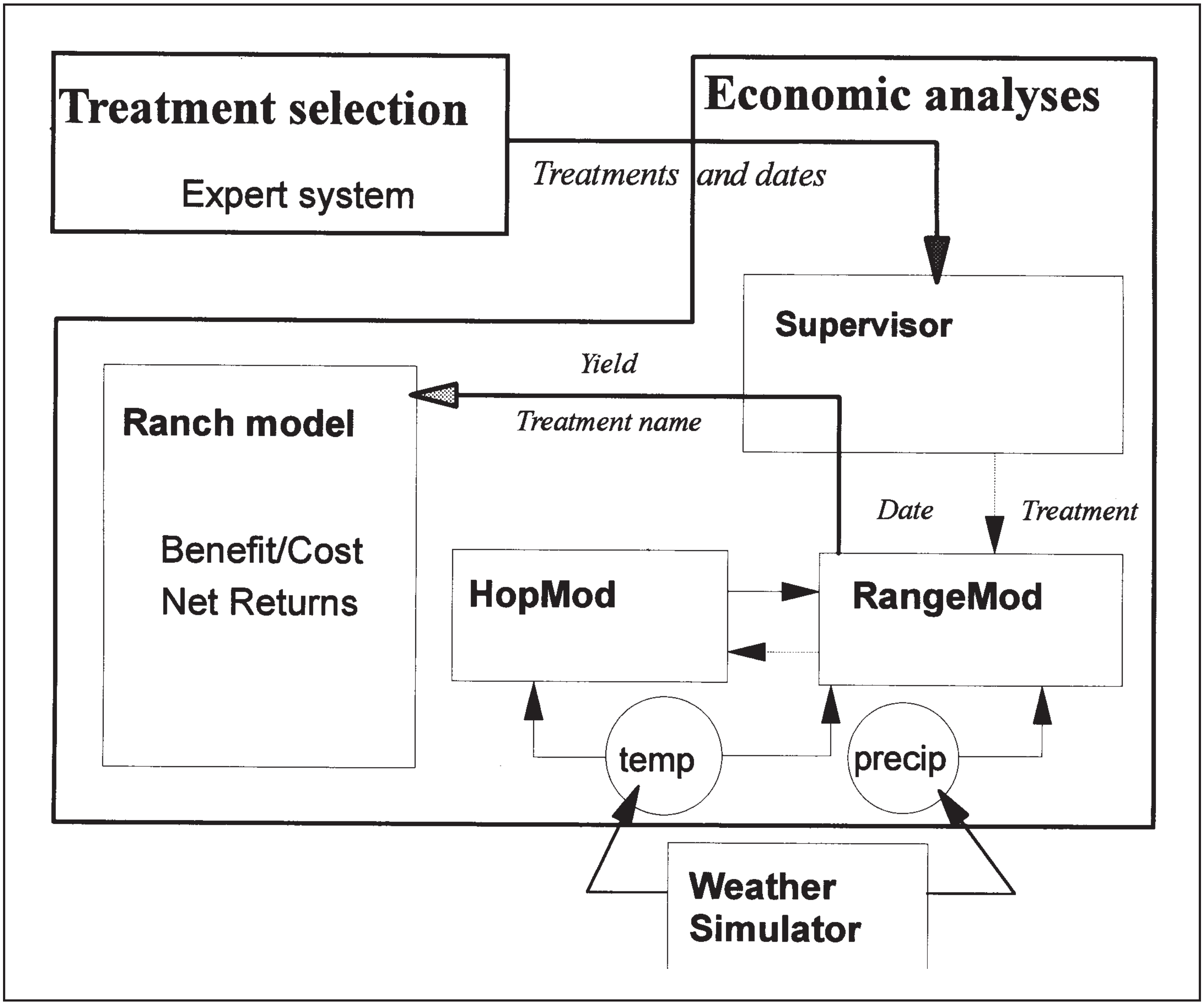


Figure VI.2–27—Overview of Hopper user interface and internal modules.

These technologies work together to provide decision support. Below is an overview of each class of technology. As each technology is introduced, you'll learn how it works with the others.

The Expert System.—The expert system (Consult) helps you choose grasshopper treatments as accurately as an expert. It does this by asking questions about the site, giving some of this information to simulation models to estimate grasshopper life stage, evaluating the data against an internal set of rules, and then providing you with a list of suggested treatments appropriate for the situation (Berry et al. 1991).

The Simulation Models.—The simulation models (HopMod and RangeMod) calculate values for critical variables that would otherwise require additional sampling and analysis.

Hopper uses simulation models to estimate the effects of precipitation, forage production, treatment mortality, grasshopper species composition and life stage (Berry et al. 1995). Information from the simulation models is used by the expert system and economics model.

Simulation models allow Hopper to respond to factors that change over time, like grasshopper life stage and forage production (Berry et al. 1991).

The Economics Model.—The economics model (RanchMod) is a linear programming model that does two things. First it determines if treatment is cost effective. Second, it determines which of the treatments listed by Consult is most cost effective. The economics model uses information from the expert system and simulation models to determine a benefit–cost ratio.

Hopper's models work together to provide reliable decision support. As a result, you can be more confident in your treatment decisions.

What the Expert System Does.—Hopper's expert system (Consult) provides you with a list of treatments appropriate for the conditions you specify. Consult uses internal rules to decide which treatment(s) to list (Berry et al. 1991). In addition, only treatments approved by the Environmental Protection Agency and the Environmental Impact Statement for the Cooperative Rangeland Grasshopper Program are considered.

Where Consult Gets Its Information.—Consult uses information from three sources. First, Consult asks you the following:
- Location?
- Species composition?
- Grasshopper census date?
- Treatment date?
- Presence or absence of managed bees?
- Should treatments harmful to beneficial insects be eliminated from consideration?
- Do conditions prohibit the use of toxic chemicals?
- Vegetation thickness?
- Current weather conditions?
- Percent of the hopper population already hatched (if known)?

Second, Consult uses Hopper's own weather model to enter weather data for the site.

Third, Consult uses the grasshopper model (HopMod) to calculate the average life stage at the time treatment will be applied and number of grasshopper eggs that will be deposited during the current year. This allows Consult to decide if it's too early or too late to treat the infestation economically.

What Consult Does With the Information.—Consult evaluates the information supplied against an internal set of rules. These rules allow Consult to choose treatments appropriate for local conditions.

Consult selects from five treatments approved by the Environmental Protection Agency and the final Environmental Impact Statement for use against grasshoppers on rangeland:
- Acephate spray,
- Carbaryl spray,
- Malathion spray,
- Carbaryl bait, or
- *Nosema locustae* bait (a pathogen of grasshoppers and Mormon crickets).

Depending on the conditions you specify, Consult may recommend none, one, or all of the treatments for economic evaluation. Carbaryl bait, for example, might be recommended when the presence of commercial bees or endangered species prohibit spraying in the area. *Nosema locustae* may be recommended for use near bodies of water, where chemicals are prohibited.

Consult considers species composition and development in making treatment recommendations because:

- Some species *don't* take baits, so you can't use baits.
- Some species *won't* eat the predominant local forage, so you *don't* have to control them.
- Some species develop faster or slower then the bulk of the population, so you should adjust treatment timing.

By accounting for these factors, Consult can alter its treatment list and, ultimately, the decision whether and when to treat.

What the Forage Model (RangeMod) Does.— RangeMod simulates growth of cool- and warm-season grasses and forbs on rangeland during a single growing season (Berry and Hanson 1991). Important features of the model include the following:

- Forage production depends on soil moisture and projected peak standing crop.
- Temperature starts and ends plant growth.
- Forage production occurs logistically (forming an S-shaped curve).

Forage Production Depends on Precipitation and Peak Standing Crop.—RangeMod determines forage production based on daily precipitation and an estimate of peak standing crop. The model uses either known precipitation averages from nearby cities or precipitation data that you supply. Forage consumption by wildlife is *not* estimated or considered by Hopper.

Precipitation directly affects soil moisture, which RangeMod calculates as a function of dry days (consecutive days without precipitation). The model causes soil to dry exponentially (quickly when wet but more slowly as moisture decreases–fig. VI.2–28) down to a minimum of 3 percent by weight. For comparison, the permanent wilting point for plants is reached when soil moisture is 10 percent (Berry and Hanson 1991).

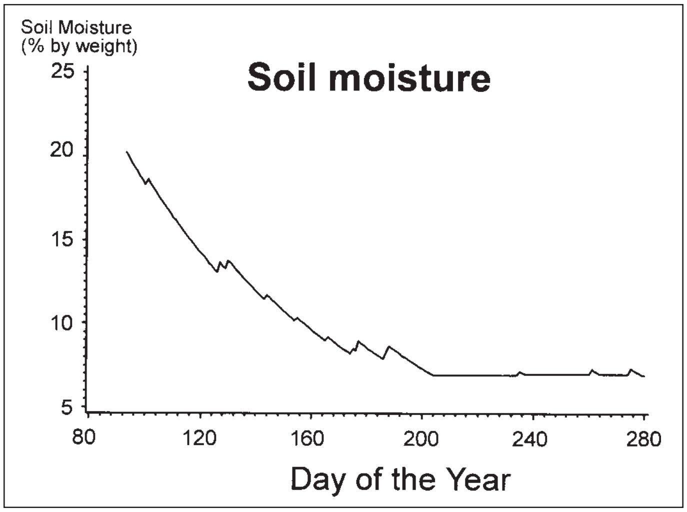


Figure VI.2–28—The effect of drying with occasional precipitation on soil moisture content. This pattern is typical for northern latitudes in the West.

Temperature Starts and Ends Plant Growth.— RangeMod uses a threshold temperature to begin growth in the spring, and to end growth in the fall. The model starts calculating growth when the temperature (the average of the daily high and low) exceeds 32 °F for 5 consecutive days. Growth occurs if daily minimum temperature is above the threshold for the plant type—44.6 °F for forbs and cool-season grasses, and 50 °F for warm-season grasses (Berry and Hanson 1991).

In RangeMod, temperature is *not* a factor in forage production except for its role in starting and ending growth (Berry and Hanson 1991).

Forage Production Occurs Logistically.—When graphed, forage production forms a logistic (S-shaped) curve (fig. VI.2–29). The logistic curve simulates forage production in pounds per acre over time. RangeMod simulates forage production for forbs, cool-season grasses, warm-season grasses, and total production, producing a logistic curve for each.

The *exact* shape of the logistic curve varies with precipitation and forage consumption by grasshoppers. Hopper simulates grasshopper forage consumption in the grasshopper model, HopMod.

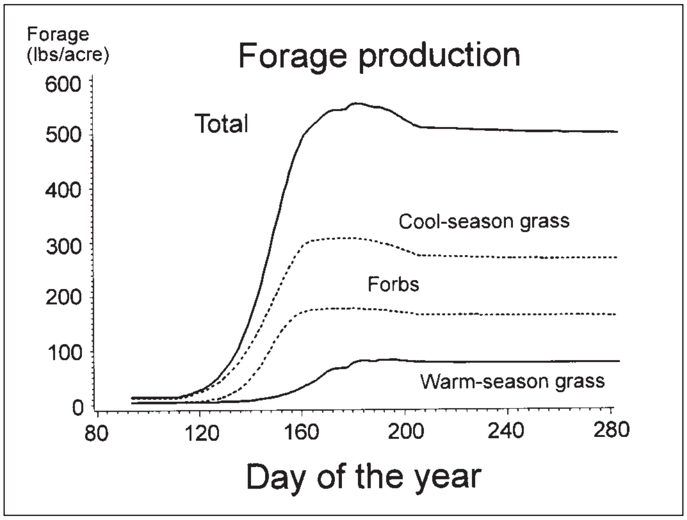


Figure VI.2–29—The logistic growth of forage appears S-shaped.

What the Grasshopper Model (HopMod) Does.—HopMod determines forage loss caused by grasshoppers and determines the loss that you can prevent by applying treatment (Berry et al. 1991).

HopMod simulates grasshopper development through time. Predicting development is important because the amount of forage eaten by grasshoppers per day varies greatly for each life stage. Early instars eat less than later instars. And because the proportion of each instar in the population changes daily, forage consumption changes daily, too.

HopMod's simulation of grasshopper development, in conjunction with the forage and economics models, allows you to decide whether or not to treat at a given time in the grasshopper's growing season.

To understand HopMod, you must understand the following:

- What the grasshopper phenology (growth and development) model does,
- How HopMod determines population size,
- How HopMod calculates forage consumption,
- How HopMod determines oviposition, and
- How accurate HopMod is.

What the Grasshopper Phenology Model Does.**—**Phenology is the study of the relationship between climate and recurring biological events, such as grasshopper life stage. The grasshopper phenology model estimates the *proportion* of the grasshopper population in each life stage on any given day as a function of time and temperature (fig. VI.2–30).

A proportion is a percentage divided by 100. For example, the proportion "0.8" is derived as follows: 0.8 = 80 percent ÷ 100. Most people use proportions frequently for various routine calculations.

How Development Is Calculated.—The model determines grasshopper development based on time and temperature, called development time (Kemp and Onsager 1986). Grasshopper development is controlled primarily by temperature, so development time is measured in degree-days.

Degree-Days Are Accumulated Heat.—A degree-day is a measure of accumulated heat. Degree-days accumulate in HopMod when the air temperature is between 40 °F (4.4 °C) and 100 °F (37.8 °C) (Berry et al. 1995).

For example, when the daily minimum and maximum temperatures are between 40 °F and 100 °F, HopMod calculates degree-days like this: If the air maximum temperature is 70 °F and the minimum is 40 °F, then there are 70–40 = 30 degree-days of development.

HopMod averages degree-days over a day–night cycle. The program adds degree-days when the temperature is

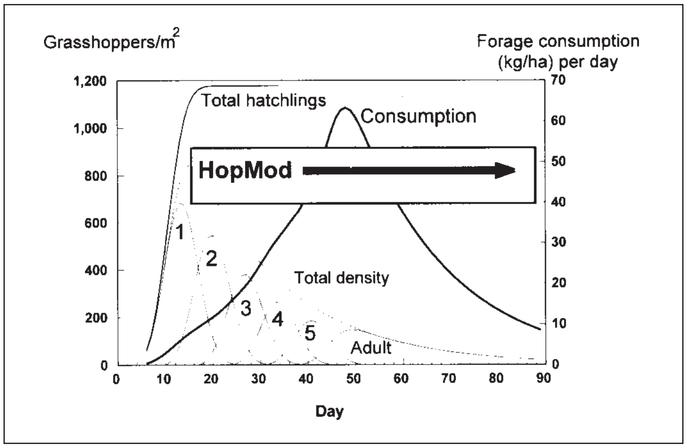


Figure VI.2–30—General progression of a grasshopper population during the spring and summer. HopMod begins when the population has peaked and egg hatch has finished.

within the thresholds. HopMod uses a modified sine-wave formula to adjust and accumulate degree-days as the value changes during the day–night cycle. (A sine-wave formula creates a curve similar to the wave pattern you'd see on an oscilloscope. The wave fluctuates above and below a line. In this case, above the line represents daylight; below the line represents night.) In this way, HopMod calculates the average instar, which is displayed in Consult.

When necessary, you can change Hopper's estimate of the average instar. For example, if you measure an average instar that is different than HopMod's estimate, you can replace Hopper's estimate with your measurement, and HopMod will adjust.

Development Is Based on Accumulated Increments of Development Time.—HopMod assumes that the development rate of a grasshopper depends on accumulated increments of development time (Kemp and Onsager 1986). The process is defined as the amount of development time that a grasshopper has accumulated by a given actual time.

HopMod uses Hopper's weather data base to calculate degree-days. Then HopMod calculates grasshopper development for each calendar day of the growing season. The result is a list of proportions for each life stage for each day. For example, on a given day, you might see the following: instar 1 = 0.1 (10 percent), instar 2 = 0.3 (30 percent), and so on. The proportions must add up to 1.00 (representing 100 percent of the grasshopper population) for the day.

***How HopMod Determines Population Size.*—**HopMod gets the grasshopper population size from you. For example, you count 20 grasshoppers per square yard and type in that number. HopMod, however, adjusts over time for natural grasshopper mortality itself.

HopMod calculates average natural grasshopper mortality using a density-dependent model. The larger the grasshopper population, the faster grasshoppers die.

HopMod, however, does *not* have an egg-hatch model. Consequently, HopMod *cannot* add newly hatched grasshoppers to the population. The program assumes all eggs have hatched by the census date.

***How HopMod Calculates Forage Consumption.*—** HopMod calculates forage consumption in five steps:

1. HopMod determines the *proportion* of grasshoppers in each instar (life stage), each day. For example, instar 1 = 0.1, instar 2 = 0.3, instar 3 = 0.4, instar 4 = 0.15, instar 5 = 0.05. Remember, the total must add up to 1.00, meaning 100 percent of the population. The proportions in each instar change each day but always add up to 1.

2. HopMod determines the *number* of grasshoppers in each instar by multiplying the proportion in each instar by the population density of first grass feeders, then mixed feeders (usually, grass feeders won't eat forbs, so forbs are protected from grass feeders without treatment). You supply the data on population density and composition.

   For example, if the grasshopper population density is 20 per square yard and is 80 percent grass feeders, then—assuming the proportion of instar 2 = 0.4 —the number of grass-feeding grasshoppers in instar 2 is: $20 \times 0.4 \times 0.8 = 6.4$ per square yard.

3. HopMod determines *how much* forage each instar consumes by multiplying the feeding rate of grasshoppers in each instar (supplied by Hopper and based on scientific measurement) by the number of grasshoppers in the instar.

4. HopMod determines *total* forage consumption by adding the consumption of each instar for each day of the growing season. This value is passed to RangeMod and subtracted for each forage type from the amount of forage for the day. If conditions are favorable, forage continues to grow, and forage loss is usually less than the total consumption by grasshoppers.

5. Finally, HopMod *repeats* the process (steps 1–4) after applying simulated treatments. For example, if there are 20 grasshoppers per square yard, and the treatment is 92 percent effective (only 8 percent survive), then *after treatment* the population is $20 \times 0.08 = 1.6$ grasshoppers per square yard.

HopMod calculates forage consumption by grasshoppers on both treated and untreated rangeland to determine the difference in consumption. This difference is the *benefit* to the ranch.

HopMod repeats the process for each treatment selected by Consult. Available forage is used in the economics model (RanchMod) to determine the benefit–cost ratio for each treatment.

How Oviposition Is Determined.—HopMod assumes that grasshoppers lay eggs at a constant rate. The rate is different for grass feeders and mixed feeders. For grass feeders, the rate is 0.6550 eggs/adult female/day; and for mixed grass feeders, the rate is 0.4564 eggs/adult female/day (Berry et al. 1995).

How Accurate Is HopMod.—HopMod has been field validated (Berry et al. 1995). HopMod correctly simulates the general patterns of rangeland grasshopper population dynamics within a given year (Berry et al. 1991).

Comparison of Field Data and the Grasshopper Model.—Figure VI.2–31 shows a comparison between field data and the phenology model's plots. As you can see, the calculated values closely match the field values. In addition, the estimates of forage consumption by the different grasshopper instars are based on scientific measurement. Therefore, you can expect HopMod to produce reasonable approximations of grasshopper forage consumption.

Steps You Can Take To Improve Accuracy.—You can improve accuracy in two ways:

1. Conduct the grasshopper census as close to the treatment date as possible.
2. Enter actual measurements of the average instar instead of accepting calculated values.

Remember, HopMod does *not* have an egg-hatch model. Consequently, HopMod cannot add newly hatched grasshoppers to the population. As a result, the greater the time between field measurement and treatment the greater the error in estimating average instar and density. So for best results, use current data.

Also, observed measurements are the best estimate of reality. Therefore, whenever possible, enter observed measurements instead of relying on Hopper's initial life-stage estimates.

What the Economics Model (RanchMod) Does.—RanchMod determines the value of the forage. With this information, and with information from the other Hopper models, RanchMod can determine if a treatment is cost effective. In addition, RanchMod compares the cost effectiveness of each treatment listed by Consult, so you can decide which treatment is *most* cost effective. The model reports cost-effectiveness as a benefit–cost ratio.

To understand RanchMod, you must know the following:

- How RanchMod determines the benefit–cost ratio,
- What information you may supply, and
- How reliable RanchMod is.

How RanchMod Determines the Benefit–Cost Ratio.—Using the forage and grasshopper models, RanchMod estimates the *value* of forage consumed by grasshoppers when treatment is applied and when treatment is *not* applied. The difference is the damage avoided by treatment, called the benefit. RanchMod assumes that the forage saved (less the forage set aside by the proper use factor) is available to livestock. The proper use factor is the proportion of the forage that will *not* be consumed by livestock, to prevent overgrazing.

The model divides the *value* of the forage saved (benefit) by the *cost* of treatment to determine the benefit–cost ratio.

RanchMod combines information from the forage and grasshopper models within its economic model to determine the value of forage. The value of forage directly affects the benefit–cost ratio.

What Information You May Supply.—The economics model asks you for information on the arrangement, and operation of the local ranch(es). This information includes the following:

- Lease costs,
- Cost and availability of hay,
- Livestock prices, and
- Herd information–size and composition.

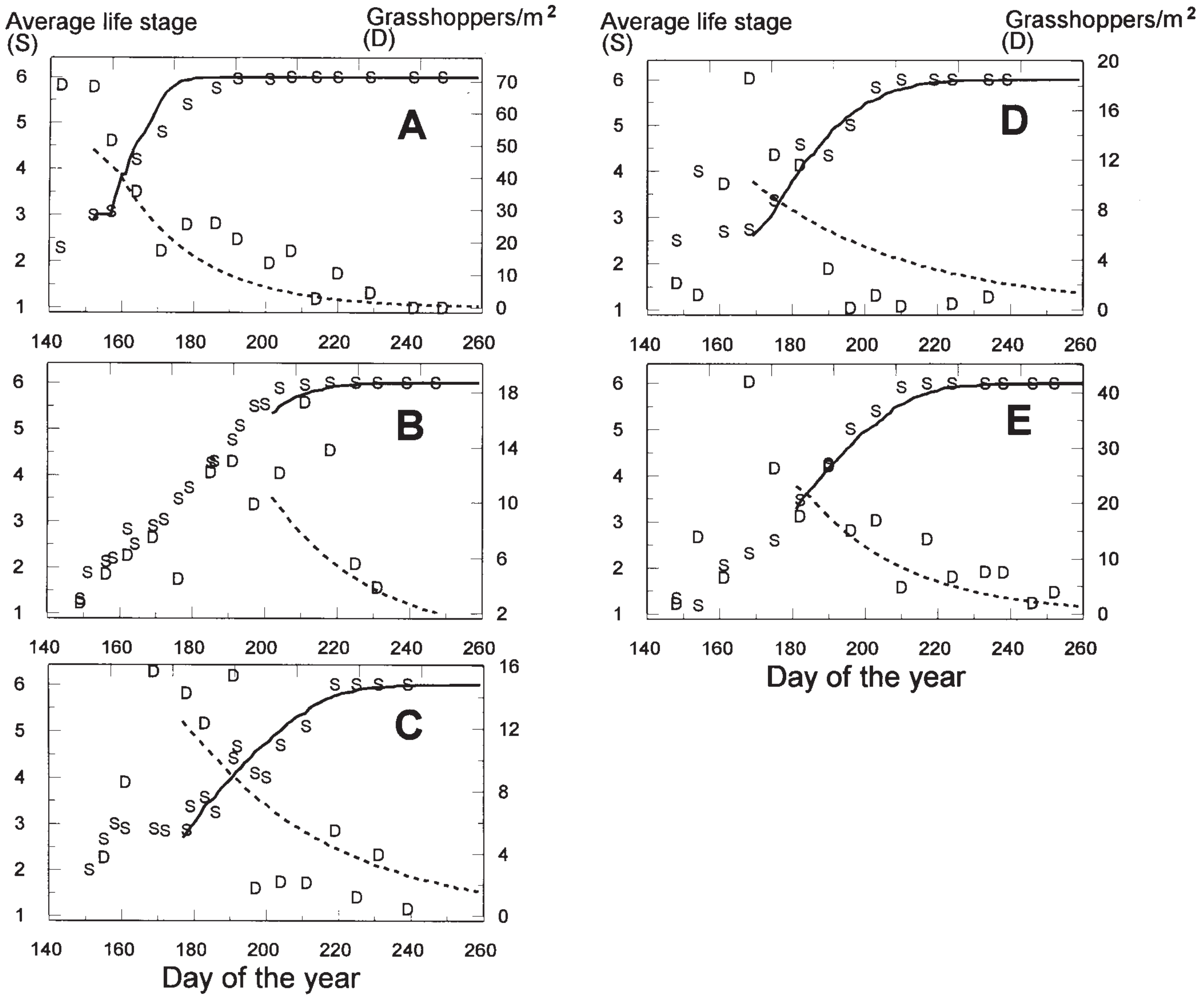


Figure VI.2–31—Validation runs showing average life stage (S, field data; solid line, model) and density (D, field data; dashed line, model) for GHIPM sites in North Dakota.

Hopper provides default values for most of these variables. Default values are averages. When you don't know the actual value, you can use the default value to get a reasonable approximation.

Do not, however, use default values for grasshopper population size and species composition. These values are so variable that your results will be useless. So, for grasshopper density and composition, always use field data. Supply the best information you can for other values as well.

Remember, Hopper is only as accurate as the information you supply. The closer this information matches reality, the more reliable Hopper's recommendation is. Use default values when you must, but supply the best information you can.

***How Reliable RanchMod Is.*—**RanchMod is both reliable and justifiable. RanchMod uses factors previously unavailable to decisionmakers. These factors allow you to account for variation in the ranching environment and to justify your treatment decisions based on economic criteria.

RanchMod's accuracy depends on the accuracy of the data. The closer the data are to reality, the more reliable the benefit–cost ratio. During average years and on the average ranch, the default values will produce good results. But the more conditions stray from average, the more critical that you enter factual data instead of allowing the program to use default values. With accurate data, expect reliable results.

Remember, RanchMod's results are *not* exact. Rather, RanchMod gives you a "ball-park figure," an estimate. RanchMod's estimate, however, is more accurate and more reliable than any you get by other means.

Your Role.—Your role (the role of ranchers, ranching committees, and government officials) in making treatment decisions with Hopper is twofold:

1. To provide accurate data to Hopper.
2. To make the final decision.

Providing Accurate Data to Hopper.—Hopper's recommendation relies on the data you enter. Therefore, to ensure reliability, you must enter the best data available. Collecting this data, however, requires skill, professionalism, and discipline.

Give Hopper the best data you can—it's worth the effort.

Making the Final Decision.—You must make the decision to treat or not. Hopper supplies you with benefit–cost ratios and other useful information. You must decide whether to treat based on the benefit–cost ratio, and other factors *not* accounted for by Hopper, that you judge important. Hopper is a decision support tool, *not* APHIS policy.

Remember, under normal circumstances, treating when the benefit–cost ratio is less than 1 is economically unjustifiable. Failure to treat when the benefit–cost ratio is greater than 1 threatens the ranching economy.

Hopper provides support for your treatment decisions based on scientific and economic research. If you use Hopper's benefit–cost ratio to make your decision, you can claim Hopper's support. But if you use another criteria, you cannot.

References Cited

Berry, J. S.; Hanson, J. D. 1991. A simple, microcomputer model of rangeland forage growth for management decision support. Journal of Production Agriculture 4: 491–499.

Berry, James S.; Kemp, William P.; Onsager, Jerome A. 1992. Hopper decision support system: rangeland grasshopper management for the 1990's. In: Proceedings, 4th international conference: computers in agricultural extension programs; 28–31 January 1992; St. Joseph, MI. St. Joseph, MI: American Society of Agricultural Engineers: 610–615.

Berry, James S.; Kemp, William P.; Onsager, Jerome A. 1991. Integration of simulation models and expert system for management of rangeland grasshoppers. AI Applications in Natural Resource Management 5: 1–14.

Berry, James S.; Kemp, William P.; Onsager, Jerome A. 1995. Within-year population dynamics and forage destruction model for rangeland grasshoppers (Orthoptera: Acrididae). Environmental Entomology 24: 212–225.

Davis, Robert M.; Skold, Melvin D.; Berry, James S.; Kemp, William P. 1992. The economic threshold for grasshopper control on public rangelands. Western Journal of Agricultural and Resource Economics 17: 56–65.

Dennis, Brian; Kemp, William P.; Beckwith, Roy C. 1986. Stochastic model of insect phenology: estimation and testing. Environmental Entomology 15: 540–546.

Kemp, William P.; Onsager, Jerome A. 1986. Rangeland grasshoppers (Orthoptera: Acrididae): modeling phenology of natural populations of six species. Environmental Entomology 15: 924–930.

Appendix B Descriptions of Hopper's Ranch Models

By Melvin Skold, Rob Davis, and James S. Berry

Recent definitions of economic thresholds (ET's) and economic injury levels (EIL's) by economists and entomologists have shown that these concepts are dynamic in nature and must be evaluated for each site under consideration for treatment. Key economic parameters to evaluate include ranch type, rangeland productivity, cost of alternative sources of forage for livestock, and nontreatment options available to the rancher. Biological parameters for evaluating an ET or EIL depend on density of grasshopper species, life stage at time of treatment, mix of economic and noneconomic species, and presence of beneficial insects. Other factors of importance are proximity to waterways and presence of rare or endangered species.

The Grasshopper Integrated Pest Management (GHIPM) Project has provided economic models for eight important range ecosystems in the Western States. Within these range ecosystems, typical ranches are defined which characterize the predominant ranching practices of the area. Between range types, ranches vary considerably with respect to amounts, types, and costs of forages used. Livestock production and management strategies also differ between range types. An evaluation of these typical ranches through Hopper shows how the economic justification for treating rangeland grasshoppers changes between locations and ranching systems.

List of Existing Model Names and Descriptions (Range Types).—The range ecosystems included in Hopper are those identified by APHIS, PPQ personnel as having recurring grasshopper infestations. The selected areas characterize seven range ecosystems and eight typical ranch types. For one area, the Northern Highland Prairie, both beef cow–calf ranches and beef–sheep ranches are common; consequently, two typical ranches were defined to analyze the impacts from grasshoppers infestations more fully.

Generic.—The generic model can be used for any area in the United States or Canada. This model does not use the detailed economic model nor forage production model. Therefore, you will need to use the default value of a replacement AUM ($11.00) or enter a different value (calculated in the other, more detailed models). An AUM (animal unit month) is defined as the amount of forage a cow and calf consume in 1 month (about 800 lb of air-dry forage).

Northern Great Plains.—The rangeland is located within the Northern Great Plains range type, and about 2.2 to 3.3 acres are required to produce 1 AUM. The grazing season is approximately 8 months long; cattle are placed on grazing lands about May 1 and continue to graze until December 31. About half the forage needed on the ranch comes from public land, a quarter from private grazing lands, and the remaining quarter from hay and crop residue.

Located in western North Dakota on the Little Missouri National Grassland, the typical ranch in this model can be used for all of the Little Missouri National Grassland and extrapolated to eastern Montana with changes to rangeland productivity, herd size, leases, weather-generation models, etc. The rangeland is characterized as a northern mixed prairie and is predominantly cool-season grasses, forbs, and shrubs.

Northern Highland Prairie.—About 4 acres are required to produce 1 AUM of forage on this range type. Because elevations in the morthern Highland Prairie are somewhat higher than in the northern Great Plains, the grazing season is shorter. Grazing begins about May 1 and continues through early September.

There are two typical ranches defined for this region. The first is a cow–calf ranch that is supplied 23 percent of forage needs by public grazing lands. Hay stocks are produced for winter feeding needs, and private rangeland supplies the balance of forage AUM’s (56 percent) for the livestock. A calf crop of 85 percent is achieved, with the calving season starting in March.

The second ranch has both a cow–calf enterprise and a range sheep enterprise. This ranch receives 41 percent of forage AUM’s from public rangeland, no hay is produced, and private grazing lands supply the balance of forage needs. Lambing begins about May 15; a lambing crop of 122 percent is the norm. The calving season for this ranch starts in March, with a calving percentage of 80 percent.

Located in Johnson County, WY, this typical operation is a large cow–calf ranch; these model parameters can be used for ranches throughout eastern Wyoming, south-central Montana, and possibly northeastern Colorado (assuming the weather, rangeland productivity, herd size, leases, etc., are changed when data are input). This rangeland, is characterized as Northern Mixed Prairie, is predominantly cool-season grasses, forbs, and shrubs.

Central Great Plains.—This region is characterized by highly productive rangelands of predominantly warm-season grass species. The typical ranch of about 2,200 acres of grazing land is a cow—calf operation with a 6-month summer grazing season. Grazing land can support approximately 1 AUM/acre. Hay is fed in the winter to supplement crop-residue grazing and supplies 14 percent of the total AUM's of forage. Public grazing land is available to only a portion of these ranches. Livestock graze on rangeland owned by the rancher and rangeland leased from other landowners.

This typical ranch is located in western Nebraska, and its parameters can be extrapolated to ranches located in southeastern Wyoming, north-central Colorado, and the Nebraska panhandle.

Southern Great Plains.—The typical southern Great Plains ranch has both cow–calf and sheep enterprises. There is an 8-month grazing season, with 34 percent of the total AUM's of forage coming from public rangelands. The typical ranch includes about 15,600 acres. Privately owned rangelands supply 26 percent of total needed AUM's, and raised hay stocks supply the remaining 40 percent of forage needs.

Almost 53 acres are required to produce 1 AUM of forage. The grass and forb species in this area are predominantly warm season, and most vegetative growth occurs in July, when the monsoon rains come.

The typical ranch in this model is located in eastern New Mexico.

Mexican Highland Scrub.—The typical ranch for this region is a cow–calf operation. Total forage comes from public grazing land (34 percent), from privately owned grazing land (13 percent), private rangeland (10 percent), and from raised hay stocks (43 percent). The elevation is low, and summers are very hot. Most vegetative growth occurs in late summer, when monsoon rains occur. Almost all plant species present are warm season. The grazing season is 9 months long, and hay is fed to supplement the grazing.

Located in southeastern Arizona, this typical operation is a smaller cow–calf ranch operating in the "hot desert" environment. About 64 acres are required to produce 1 AUM of forage. Results from the Mexican Highland Scrub typical ranch profile can be extrapolated to ranches in southwestern New Mexico.

Gila Mountains.—Grazing needs are satisfied for this cow–calf ranch with a year-round grazing season. About 6.5 acres are required to produce 1 AUM of forage. There are no hay stocks produced. The split between public and private grazing lands is about 50–50. The grass species in this region have high percentages of both warm- and cool-season grasses. Most vegetative growth occurs in late July with the onset of summer monsoon rains.

Located in central Arizona in the Chino Valley near Prescott, this typical ranch is a very large cow–calf operation in a transition zone next to a hot desert zone.

Eastern Intermountain Basin.—The typical ranch for the southeastern Great Basin region is a cow–calf ranch that receives about 7 percent of its total forage supplies from public rangelands, 32 percent from leased private rangelands, 41 percent from owned rangeland, and 20 percent from hay produced on the ranch. About 12 acres are required to produce 1 AUM of forage. The grazing season is year-round, with hay stocks supplementing the rangeland forage supplies during the winter. Public rangelands are used during the spring months.

This typical ranch is located in western Utah, and results from this ranch profile can be extrapolated to ranches in southern Idaho and eastern Nevada.

Northern Intermountain Basin.—A cow–calf ranch was defined for this region. The grazing season starts in mid-April and runs until early November. About 9 to 10 acres are required to produce 1 AUM of forage. Public rangelands supply 44 percent of the total forage needs of the cow herd. Raised hay stocks supply 22 percent of the forage and are used in the winter months. Privately owned rangelands and leased private rangelands supply the remainder of forage needs (34 percent).

Located in Harney County, OR, this typical ranch is a cow–calf operation in the Great Basin Desert, which is dominated by big sagebrush. Results from this ranch profile can be extrapolated to operations in southern Idaho and northern Nevada.

Model Names in Hopper

(CC = cow–calf, CS = cow–sheep enterprise)

NGP = Northern Great Plains (western North Dakota)
NHP = Northern Highland Prairie (north-central Wyoming)
CGP = Central Great Plains (southeastern Wyoming, north-central Colorado, Nebraska panhandle)
SGP = Southern Great Plains (eastern New Mexico)
MHS = Mexican Highland Scrub (southeastern Arizona, southwestern New Mexico)
GM = Gila Mountain (central Arizona)
EIB = Eastern Intermountain Basin (western Utah, southern Idaho, eastern Nevada)
NIB = Northern Intermountain Basin (eastern Oregon, western Idaho)
Generic = Any area in the United States or Canada. This model does not use the detailed economic model nor forage model. Therefore, you will need to enter the value of replacement AUM’s (calculated in the other, more detailed models). These files will have the extension `*.gn3` (e.g., `generic.gn3`).

VI.3 Applying Economics to Grasshopper Management

Melvin D. Skold and Robert M. Davis

Economic considerations are a major part of grasshopper management. Rangeland grasshopper control programs, as well as other pest management strategies, use the concepts of economic threshold (ET) and economic injury level (EIL). The ET is defined as the pest population (density) that produces incremental damage which is just equal to the incremental cost of control (Headley 1972). Pedigo and Higley (1992) advance an identical definition. Viewed from this perspective, the damage caused by the pest must be at least as great as the cost of treatment before the ET is reached. The EIL and ET are related concepts. For some pests, observations of earlier life stages can define an ET for an EIL density of a subsequent life stage. For grasshoppers, however, density surveys are completed and ET evaluations are made based on those surveys.

For many years, grasshopper control programs followed an administrative guideline intervention level of 8 grasshoppers/yd^2 as suggested by Parker in 1939. However, the Grasshopper Integrated Pest Management (GHIPM) Project found the ET to vary, depending on a number of conditions in the range forage, grasshopper, and ranch system. Because the ET for rangeland grasshoppers varies with conditions, the GHIPM Project developed a microcomputer-based decision-support system (Hopper) to help those responsible for grasshopper control programs make realistic estimates of the ET. This chapter discusses the physical, biological, and economic rationale that determines the ET.

Evaluating Benefits

There is a long history of public support for control of rangeland grasshoppers. Individual efforts cannot control widespread grasshopper outbreaks. However, there also is a public benefit from protecting rangelands from serious outbreaks of grasshoppers. Public rangeland has many uses. Ranchers lease rangeland for domestic livestock grazing, the traditional economic use. Rangeland also supports a diverse population of wildlife, provides recreation and open space, protects soil from erosion, and contributes to the watershed for rivers and streams. Rangeland grasshoppers eat and destroy forage that livestock and range-consuming wildlife could use. When grasshopper infestations occur on rangelands, ranchers relying on those lands for livestock grazing incur economic losses. Reducing the density of grasshoppers reduces losses to ranchers. The difference in ranch net returns with and without grasshopper treatments is the basis for the benefits calculation. If grasshoppers exceed the ET and land managers or agencies apply treatments, those treatments can limit the reduction in the ranchers' net returns.

The GHIPM Project's decision-support system, Hopper, includes an economics component that evaluates damage reduction (limiting the decrease in net returns for ranchers) for each of the approved grasshopper treatment alternatives. The damages abated are the benefits resulting from the treatment program. The estimate of damages abated likely is unique for a typical ranch and makes use of the type of range being considered for grasshopper control programs.

Typical Ranches

Because it would be very costly to estimate the damage caused by grasshoppers for each ranch using a grasshopper-infested rangeland, we estimated benefits from grasshopper treatments for "typical ranches" on the major range types for which a version of Hopper is available.

Typical ranches reflect the characteristics of ranches in an area. They are typical with respect to rangeland productivity, livestock on the ranch, grazing management practices, and livestock management practices. To define typical ranches, we interviewed ranchers in an area to identify the common practices. The typical ranch became the barometer to evaluate benefits of grasshopper treatment programs for a given range type. Consequently, typical ranches could be indicators of the extent of the economic impact of grasshoppers on the net incomes of ranchers using that range-type.

Suppose that, as a land manager, you are responsible for making the decision about whether or not to conduct a grasshopper control program in a given area. You know the typical ranch in your area is a cow–calf operation that uses public grazing land along with intermingled deeded rangeland. An economic decision model for the typical ranch is available to show the options you can choose among for dealing with an infestation of grasshoppers.

Here are some management strategies you may consider.

- Have a reserve of hay to supplement grazed forage, which may vary with climate or grasshoppers;
- Find additional grazing land to lease;
- Use crop residues to replace forage lost to grasshoppers;
- Change livestock management practices to reduce forage requirements (such as shift from a cow–yearling to a cow–calf marketing strategy, purchase rather than raise herd replacements, or reduce the size of the cow herd through culling);
- Purchase hay; and/or
- Initiate grasshopper control programs.

The economic decision model lets you consider simultaneously which of these options will result in the least reduction in the expected net returns from the ranch. You choose the option least costly to the ranch, based on your current expectations about prices and costs.

The economic decision model for the nine typical ranches is incorporated into Hopper. In Hopper, the decision model for the typical ranch works with two other components that consider the physical and biological systems present on the ranch. One component estimates the growth of rangeland forage, given soil type(s), temperature, precipitation, and related climatic variables. A second component estimates grasshopper population dynamics and the amount of forage that grasshoppers eat and destroy on the ranch.

The grasshopper population dynamics component of Hopper works with the rangeland forage growth model to predict how much forage will be available for grazing animals. Because some types of wildlife also use rangeland forage, the amount of grazable forage available to livestock depends on how much forage grew and how much remains after grasshoppers and wildlife have eaten.

The grasshopper population dynamics component of Hopper also lets you consider each of the approved treatment options available. Treatment options are determined by physical and biological conditions as well as by the cost effectiveness of the options. Each option comes at a different cost and behaves differently in its timing and effectiveness on grasshoppers. The economic decision model for the typical ranch uses these other two components of Hopper to evaluate the nontreatment adjustments available to the rancher along with the cost and effectiveness of alternative treatments.

To evaluate the benefits, Hopper compares the ranch net returns with no treatment to the ranch net returns for a given treatment at various grasshopper densities. Treatment benefits are the difference in ranch net returns between a treatment option at a given grasshopper density and ranch net returns with the no-treatment option. At low grasshopper densities, ranchers may adjust their grazing or livestock herd management to the loss of forage from grasshoppers. As grasshopper densities increase, losses in net returns also increase. At some point, the density of grasshoppers approaches the ET, and the use of treatments becomes economically justified (fig. VI.3–1).

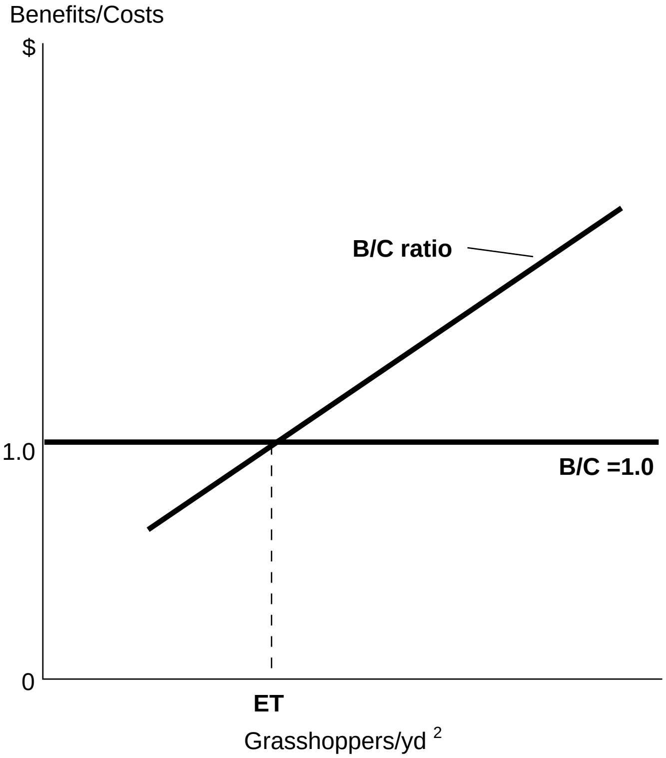


Figure VI.3–1—Determining the benefit–cost (B/C) ratio and the economic threshold (ET), based on grasshopper density per square yard and the cost of treatments.

Cost of Treatments

Hopper determines the costs in addition to the benefits for each treatment at varied densities of grasshoppers. Costs include materials and application expenses per acre, based on recent experience. The costs to apply a given treatment on the typical ranch in your area vary directly with number of acres in the ranch. If you expect the per-acre costs for the treatment(s) considered to differ from those specified in Hopper, you can change the costs to your current best estimate.

Hopper includes expected mortality (grasshopper kill) from each treatment. If dosage, treatment strategy, plant cover, or terrain is likely to change treatment effectiveness, the effective cost of treatment also will change. The benefits (damages abated) will not be as great from a treatment that is less effective (kills fewer grasshoppers) than a treatment that kills more grasshoppers.

The treatment costs reflected in Hopper are the total cost of treatments regardless of who pays. Through its Animal and Plant Health Inspection Service (APHIS), Plant Protection and Quarantine (PPQ) staff, the U.S. Department of Agriculture pays treatment costs for controlling grasshoppers on Federal lands. The Department also pays a portion of the cost of treating intermingled and adjacent private lands. Some States also cost-share in the treatment programs. States may pay a portion of the cost of treating leased State land and a portion of the cost of treating private land. While the cost share may affect the out-of-pocket costs that a given rancher must pay, cost-sharing is not a part of the benefit and cost calculations of Hopper. Rather, in Hopper, benefits are directly compared to total costs, regardless of who pays.

Benefit–Cost Ratios

The ET is defined by a ratio of the per-acre benefits (B) and costs (C), or B/C (B ÷ C). When B/C = 1.0, the ET is reached (fig. VI.3–1). The B/C = 1.0 when the benefits line crosses the treatment cost line. At that grasshopper density, the ET is reached. At grasshopper densities less than where B/C = 1.0, damages (net return reductions) are occurring but are less than the cost of treatment. At densities greater than where B/C = 1.0, benefits (damages abated) are greater than treatment costs, and economic losses occur in the absence of treatments.

The B/C calculations in Hopper initially compare the costs of treatments to the benefits that result in the year of treatment. Many ranchers believe the benefits from effective treatments can last for several years. Consequently, with Hopper you can specify the expected duration (number of years) of control. If that number is >1, Hopper automatically takes it into account when calculating the B/C ratio.

Analysis with Hopper under varied conditions shows that the long-applied intervention level of 8 grasshoppers/yd^2 is not appropriate. Rather than a fixed ET, the ET in Hopper varies depending on rangeland productivity, the cost of replacing forage lost to grasshoppers, treatment costs, and treatment efficacy. Other physical, biological, and economic factors can affect the ET, too. By running Hopper, you can determine the grasshopper densities necessary to reach the ET on parcels like yours and the sensitivity of the ET to various conditions.

By using Hopper to define the ET, the ET is dynamic and may change from year to year at a given location. Further, the ET is different from location to location in any given year. The ET is determined by running Hopper for a typical ranch such as exists on a major range type. The typical ranch reflects the most common practices for the range type.

To characterize the ranches incorporated into Hopper, a ranch of a given size is described. Size is measured by the number of livestock as well as the amount of land available. The amount of grazing land is determined and for the deeded land, the amount that is owned and the amount that is leased are both specified. Public grazing land is divided by management agency between Federal and State. Grazing practices are also reflected in the economics component of Hopper. The use specifies the length of the grazing season, the time during which the different grazing land types are used, and the time when other sources of feed are fed. If some grazed forage is obtained from crop residue, that fact is reflected in Hopper. If harvested forage is fed, the time of its feeding and its source are also important.

The livestock management systems practiced and viable alternative livestock systems also are built into Hopper. Thus, the herd culling practices, typical calf crop, and disposition of steer and heifer calves must be accurately represented in Hopper.

As Hopper is used to evaluate a treatment decision and to determine the grasshopper density at which the ET is reached, several nontreatment management adjustments are automatically considered. The options available to each typical ranch are built into Hopper. Thus, if a grasshopper invasion occurs, the relevant changes in forage management and livestock herd management are considered simultaneously with the authorized treatment options. If leasing grazing land to replace grasshopper damaged grazing land is an option and leasing is less costly than any treatment, leasing other grazing land will occur before any treatment is applied. The availability of alternative forage and livestock management options affects the position of the benefits line and the grasshopper density at which the ET is reached.

Upon running Hopper, you can determine a separate benefits line for each approved treatment. Because treatments vary as to their cost and efficacy, Hopper calculates different ET's for each treatment. Of course, some treatments may not be possible because of environmental and biological circumstances present. In such cases, Hopper determines the ET only for the treatment options consistent with the conditions that prevail. Changes in treatment costs and efficacy also are important to the position of the B/C line. If treatments can be obtained at a reduced cost, the line shifts left and the ET is reached at lower grasshopper densities than for higher treatment costs.

Applying economic analysis to estimate the ET's for grasshopper treatments provides information-based decisions. Hopper defines typical ranches for important range ecosystems in which recurring grasshopper problems occur. We discuss these ranches in chapter VI.4.

References Cited

Headley, J. C. 1972. Defining the economic threshold. In: Pest control strategies for the future. Washington, DC: National Academy of Sciences: 100–108.

Parker, J. R. 1930. Some effects of temperature and moisture upon *Melanoplus mexicanus* Sanssure and *Camnula pellucida* (Orthoptera). Bull. 233. Bozeman, MT: Montana Agricultural Experiment Station. 132 p.

Pedigo, L. P.; Higley, L. G. 1992. The economic injury level concept and environmental quality: a new perspective. American Entomologist 38: 12–21.

VI.4 Regional Economic Thresholds in Grasshopper Management

Robert M. Davis and Melvin D. Skold

Rangeland grasshopper treatment programs traditionally have started when an economic threshold (ET) was reached. In 1939, Parker defined 8 grasshoppers/yd^2 as the density of grasshoppers at which economic damage to the rangeland begins. Therefore, this density became a “trigger” for beginning consideration of a treatment program. Until recently, the 8 grasshoppers/yd^2 intervention level was used for evaluating grasshopper treatment programs on public rangelands throughout the Western United States.

Recent definitions of ET’s and economic injury levels (EIL’s) by economists and entomologists have shown that these concepts are dynamic in nature and must be evaluated for each site under consideration for treatment. Key economic parameters to evaluate include ranch type, rangeland productivity, cost of alternative sources of forage for livestock, and nontreatment options available to the rancher. Biological parameters for evaluating an ET and/or EIL depend on density of grasshopper species, life stage at time of treatment, mix of economic/noneconomic species, and presence of beneficial insects. Other factors of importance are closeness to waterways and presence of rare and endangered species.

The Grasshopper Integrated Pest Management Project has provided estimates of ET’s for eight important range-type regions in the Western States. Within these range-types, typical ranches are defined—ranches that characterize the predominant ranching practices of the area, as discussed in chapter VI.3. Between range-types, ranches vary considerably with respect to amounts, types, and costs of forage used. Livestock production and management strategies also differ between range ecoregions. An evaluation of these typical ranches through Hopper shows how the economic justification for treating rangeland grasshoppers changes between locations and ranching systems.

Range-Type Regions

The range-type regions included in Hopper are those identified by U.S. Department of Agriculture, Animal and Plant Health Inspection Service (APHIS), Plant Protection and Quarantine (PPQ) personnel as having recurring grasshopper infestations. Nine typical ranches are defined for the eight generalized range-type regions. While county lines were used to designate the range-type regions, the regions should be considered to represent a general area. Similarly, local variation may cause some ranches within the defined area to be different from the typical ranches used to characterize ranching in the eight areas. See figure VI.4–1 for details.

Northern Great Plains.—Rangelands within the Northern Great Plains range-type vary between 2.2 and 3.3 acres per animal unit month (AUM). The grazing season is approximately 8 months long; cattle are placed on grazing lands about May 1 and continue to graze until December 31. On the typical ranch, half the forage comes public land, a quarter from private grazing lands, and the remaining quarter from hay and crop residue.

Ranchers are typically cow–calf operators. Calving begins in March. Most ranchers raise their own herd replacements. On average, about 86 percent of the cows bear a calf each spring.

Northern Highland Prairie.—Here grazing lands average about 4 acres per AUM. Since elevations in the Northern Highland Prairie are somewhat higher than in the Northern Great Plains, the grazing season is shorter. Grazing begins about May 1 and continues through early September.

There are two typical ranches defined for this range-type. One is a cow–calf ranch that gets 23 percent of needed forage from public grazing lands. Hay stocks are produced for winter feeding needs, and private rangeland supplies the balance of forage AUM’s (56 percent) for the livestock. A calf crop of 85 percent is achieved, with the calving season starting in March.

Another typical ranch has both a cow–calf enterprise and a range sheep enterprise. This ranch receives 41 percent of forage AUM’s from public rangeland, no hay is produced, and private grazing lands supply the balance of forage needs. Lambing begins about May 15; a lambing crop of 122 percent is the norm. The calving season for this ranch starts in March, with a calving percentage of 80 percent.

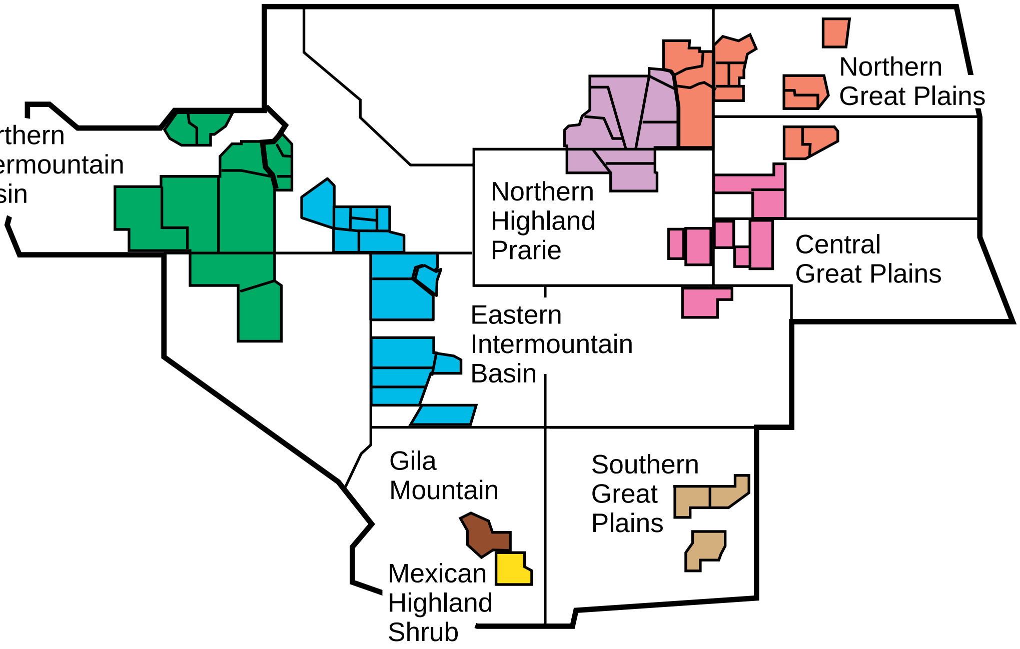


Figure VI.4–1—Map of the Western United States showing the eight generalized range-type regions.

Central Great Plains.—This region is characterized by highly productive rangelands of predominantly warm-season grass species. The typical ranch of about 2,200 acres of grazing land is a cow–calf operation with a 6-month summer grazing season. Grazing land can support approximately 1 animal unit (AU) per acre. Ranchers feed hay (supplying 14 percent of the total AUM's of forage) in the winter to supplement crop residue grazing. Public grazing land is available to only a portion of the ranches. Livestock graze on rangeland owned by the rancher and rangeland leased from other landowners.

Southern Great Plains.—The Southern Great Plains ranch has both cow–calf and sheep enterprises. There is an 8-month grazing season, with 34 percent of the total AUM's of forage coming from public rangelands. The typical ranch includes about 15,600 acres. Privately owned rangelands supply 26 percent of needed forage, and raised hay stocks supply the remaining 40 percent. The rangeland has a productivity rating of about 12 AU's per section (640 acres). The grass and forb species in this area are predominantly warm season, and most vegetative growth occurs in July, when the monsoon rains come.

Mexican Highland Shrub.—The typical ranch for this region is a cow–calf operation. Of total forage needed, 34 percent comes from public grazing land and 13 percent from privately owned grazing land. Another 10 percent is supplied by leasing private rangeland from other landowners. Raised hay stocks furnish the remaining 43 percent of forage. The elevation is low, and summers are very hot. Vegetative growth occurs when the monsoon rains come in late summer. Almost all plant species present are warm season. The grazing season is 9 months long.

Gila Mountains.—Grazing needs are satisfied for this cow–calf ranch with a year-round grazing season. Grazing land provides enough grazable forage to support an

AU for each 6.5 acres. There are no hay stocks produced. The split between public and private grazing lands is about 50–50. The grass cover in this region has high percentages of both warm- and cool-season grasses. Most vegetative growth occurs in late July with the onset of summer monsoon rains.

Eastern Intermountain Basin.—The typical ranch for the Eastern Intermountain Basin region is a cow–calf ranch that receives about 7 percent of its total forage supplies from public rangelands, 32 percent from leased private rangelands, 41 percent from owned rangeland, and 20 percent from hay produced on the ranch. Rangelands carry about 1 AUM/12 acres. The grazing season is year-round, with hay stocks supplementing the rangeland forage supplies during the winter. Public rangelands are used during the spring months.

Northern Intermountain Basin.—A cow–calf ranch was defined for this region. The grazing season starts in mid-April and runs until early November. Rangelands carry 1 AUM/9–10 acres. Public rangelands supply 44 percent of the total forage needs of the cow herd. Raised hay stocks supply 22 percent of the forage and are used in the winter months. Privately owned rangelands and leased private rangeland supply the remainder of forage needs (34 percent).

Results

The ET is the point at which the incremental damage caused by rangeland grasshoppers becomes equal to the incremental cost of applying treatment programs (see chapter VI.3). The ET varies from year to year at a given site; during a given year, it varies between sites. Benefits are measured in terms of the prevention of grasshopper-caused reductions in net returns from rangeland (forage production). Costs are the dollars required to conduct a grasshopper treatment program.

In figure VI.4–2, how the ET is determined is illustrated by ET_0. The ET is reached when the ratio of benefits (B) to costs (C) is equal to 1; B/C = 1.0. At grasshopper densities that are less than where B/C = 1.0, damages are occurring but the cost of applying a treatment exceeds the amount of damage experienced. Only when the ratio of B to C reaches 1.0 or higher does treatment become economically justified.

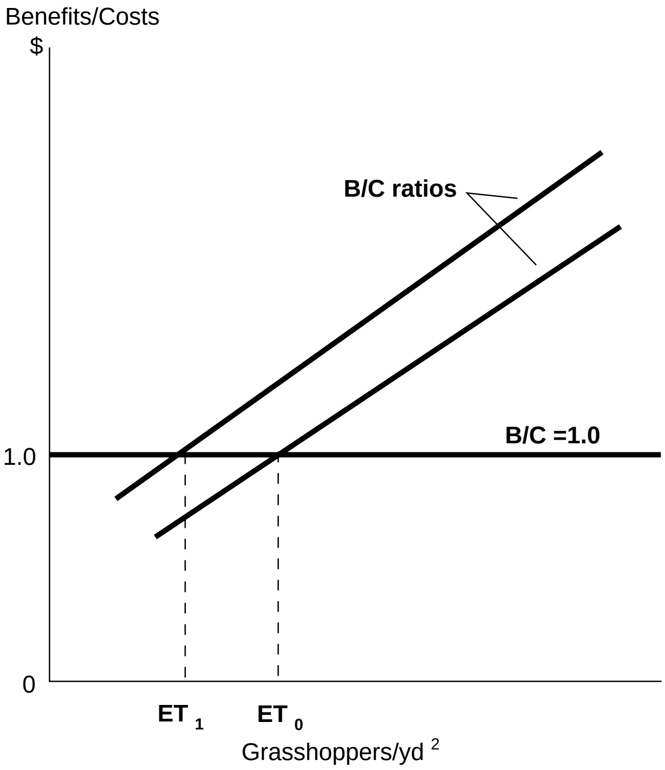


Figure VI.4–2—The relationship of benefit–cost ratios to economic thresholds.

Several factors may cause the ET to vary between years on any of the range-types shown on the map. A drought year will make grazable and harvested forage more valuable; the B/C line shown in figure VI.4–2 will shift to the left, indicating that the ET is reached at a lower grasshopper density (ET_1) than would occur during a year with normal precipitation. The cost and sources of forage to replace that destroyed by grasshoppers will also cause the ET to vary from year to year. If the cost of hay or leased grazing land decreases, the ET at which the B/C = 1.0 will shift to the right or to greater grasshopper densities.

Within a given year, variation in the productivity of rangeland results in a different ET for each range-type. The mix of cool- and warm-season forages and the emergence and maturing of grasshoppers relative to the growth of grasses also causes variation between sites. Further, the species mix of grasshoppers between grass feeders and mixed-forage feeders results in between-site variation in the grasshopper density at which the ET is reached. The ET is quite sensitive to the species composition of grasshoppers so it becomes very important to identify the species of grasshoppers present in the nymphal survey (fig. VI.4–3).

The ET is a dynamic number which changes from year to year and place to place. The conditions may be such that a given grasshopper density is sufficient to reach the ET one year; conditions may have changed by the next year to where that density of grasshoppers does not meet the ET. The ET also can be expected to be different among each of the range types represented in Hopper.

Figure VI.4–3—Monitoring and identifying grasshopper populations while the insects are in the nymph (young) stage allows pest managers to make timely decisions. Knowing species composition is important for calculating the economic threshold. (APHIS photo by Mike Sampson.)

VI.5 Field Guide to Common Western Grasshoppers

R. Nelson Foster and Mike W. Sampson

For many years, personnel who deal with survey and control of grasshoppers have voiced the need for a practical and comprehensive grasshopper identification and informational field guide. Numerous taxonomic keys exist, but most generally are designed only for adult grasshopper species, are for a single State, and are designed for laboratory use.

A wealth of information on certain grasshopper species can be found in the literature; however, information on many other species is scarce. When information does exist, it is scattered throughout numerous scientific journals, State and Federal publications, and textbooks.

When the Grasshopper Integrated Pest Management (GHIPM) Project began in 1987, one of the first needs identified by survey and control personnel was a field guide to the grasshopper species most commonly encountered on rangeland. The project asked Robert E. Pfadt, professor emeritus of entomology at the University of Wyoming, to prepare the field guide. Pfadt's grasshopper experience spans more than 50 years and includes more than 50 publications and several books. The general format of the guide was developed by Pfadt and the U.S. Department of Agriculture, Animal and Plant Health Inspection Service's (USDA, APHIS) Phoenix, AZ, Methods Development unit, and GHIPM Project personnel working collaboratively.

The guide was designed around a four-page factsheet on each selected grasshopper species. A shrink-wrapped collection of all the factsheets, grouped under the title "Field Guide to Common Western Grasshoppers," follows this chapter.

Color photographs of grasshoppers in the immature stages, the adult male and female, and the eggs and eggpod of each species are shown on the inner pages of each factsheet. Here also appear the diagnostic characteristics used to distinguish the identity of the species. The layout is organized so readers can examine all photos and read the diagnostic descriptions without turning any more pages.

Each factsheet contains other important information, such as distribution and habitat, economic importance, food habits, dispersal and migration, hatching, nymphal development, adults and reproduction, population ecology, and daily activities. The information is a collection of existing published information and Pfadt's own personal observances.

Pfadt has color coded the factsheets to educate the user subtly in the taxonomic grouping of the grasshopper species to the subfamily level. The common name, distribution map, and subheadings are green for the slantfaced species (Gomphocerinae), tan for the spurthroated species (Melanoplinae), and blue for the bandwinged species (Oedipodinae). The Mormon cricket, which is really a longhorned grasshopper (Tettigoniidae), is color coded lavender.

Each factsheet is designed as a stand-alone publication so users in different States and regions may organize these field guides in an order most useful for individual needs. The factsheets, following a 41-page introductory publication, presently are arranged alphabetically for easy location of species.

Originally intended to take only 2 years, Pfadt's project eventually expanded to 4 years so he could complete descriptions and photographs of some 40 grasshopper species. Under a cooperative agreement between the GHIPM Project and the University of Wyoming, Pfadt produced his field guide, released as Bulletin 912, in September 1988 with the first four-page species factsheets in color.

Each year since 1988, Pfadt has added additional factsheets to his field guide. Finding all of the instars of some species has meant working in remote locations and being at the mercy of the up's and down's of grasshopper populations. To produce the required photographs of common grasshopper species has been a time-consuming, and sometimes frustrating, endeavor.

In April 1995, Pfadt and the University of Wyoming issued a second edition of Bulletin 912, with more detailed information about grasshopper identification and new and better photographs. The April 1995 revision contains factsheets describing 39 grasshopper species, and Pfadt continues to work on additional factsheets.

During the GHIPM Project, the field guide has become a valuable asset for land managers charged with grasshopper identification. Field guide users now include not only APHIS personnel but also Federal, State, and private land managers, pest control specialists, and scientists.

The knowledge of the most commonly encountered species in each State contained in Pfadt's "Field Guide" will promote a better understanding of grasshopper populations. In turn, that understanding will provide the foundation for making good management and pest-treatment decisions involving rangeland grasshoppers.

Additional free copies of Pfadt's revised "Field Guide to Common Western Grasshoppers" are available on a first-come, first-served basis from USDA, APHIS, Plant Protection and Quarantine; Operational Support Staff; 4700 River Road, Unit 134; Riverdale, MD 20737-1236. You may request a copy by telephone as well (301 734-8247). Once APHIS' supply is exhausted, you may write to the University of Wyoming Bulletin Room, P.O. Box 3313, Laramie, WY 82071-3313 for information on buying the factsheets.

VI.6 Relative Importance of Rangeland Grasshoppers in Western North America: A Numerical Ranking From the Literature

Richard J. Dysart

Introduction

There are about 400 species of grasshoppers found in the 17 Western States (Pfadt 1988). However, only a small percentage of these species ever become abundant enough to cause economic concern. The problem for any rangeland entomologist is how to arrange these species into meaningful groups for purposes of making management decisions. The assessment of the economic status of a particular grasshopper species is difficult because of variations in food availability and host selectivity. Mulkern et al. (1964) reported that the degree of selectivity is inherent in the grasshopper species but the expression of selectivity is determined by the habitat. To add to the complexity, grasshopper preferences may change with plant maturity during the growing season (Fielding and Brusven 1992). Because of their known food habits and capacity for survival, about two dozen grasshopper species generally are considered as pests, and a few other species have been called beneficials (Watts et al. 1989).

Between these extremes are more than 350 grasshopper species that are of little or no economic concern. However, while most species alone never cause serious economic loss, together an assemblage of minor species can inflict serious damage to rangeland. Through the years, the pest grasshoppers have received the greatest attention. Grasshoppers of the family Acrididae surpass all other arthropods in their destructiveness to rangeland (Watts et al. 1982). Although few in number, the pest grasshoppers cause losses to western rangeland estimated at $393 million per year, based on 1977 dollars (Hewitt and Onsager 1983).

Reviewing the Literature

Several authors have made estimates of the relative importance of the major pest grasshoppers on western rangeland, but the work by Hewitt (1977) is probably the most thorough and the most cited. To my knowledge, however, no estimates have been made on the relative importance of the minor, occasional, and nonpest grasshoppers. The purpose of this chapter is to score and rank the western grasshopper species, in terms of relative economic importance, on the basis of remarks made by many grasshopper experts in their reports and publications. It is important to point out that these estimates represent merely the opinions of those involved, not conclusive proof. By including a large number of articles and authors that cover most of the literature on the subject, I hope that the resulting compilation will be a consensus from the literature, without introduction of bias on my part.

This review is restricted to grasshoppers found in 17 Western United States (Arizona, California, Colorado, Idaho, Kansas, Montana, Nebraska, Nevada, New Mexico, North Dakota, Oklahoma, Oregon, South Dakota, Texas, Utah, Washington, and Wyoming) plus the 4 western provinces of Canada (Alberta, British Columbia, Manitoba, and Saskatchewan). Furthermore, only grasshoppers belonging to the family Acrididae are included here, even though many research papers reviewed mentioned species from other families of Orthoptera.

In my evaluation, I have emphasized the impact of grasshoppers on rangeland rather than on cropland. In cases where authors made comparative remarks, such as “this species is of major importance to crops, but only of minor importance to range ...,” I used only the rangeland remarks to assign a pest-status category to that species.

For my review of the North American grasshopper literature, I selected only articles in which the authors had grouped or characterized a number of grasshopper species according to their importance. Because of this limitation, several important taxonomic analyses (Brooks 1958, Handford 1946, and Otte 1981 and 1984) could not be used for my purposes.

Pest-Status Categories

Grasshoppers are important herbivores, and any pest classification is based on whether they compete with or benefit human activities. Many articles I reviewed contained proof that a species actually caused measurable injury to rangeland, but many did not. Also, most articles which claimed that certain grasshopper species were beneficial presented no data to support the claim.

In my review, I have used the authors’ remarks regardless of the evidence presented. In most instances, it was not difficult to assign species to one of my pest-status categories because the authors had made clear statements concerning the relative importance of grasshopper species in a study. However, it was sometimes necessary to make an interpretation from somewhat vague statements, such as "... occasionally common on rangeland." After reviewing each article, I translated the authors' remarks on a particular grasshopper species into one of five categories:

Serious pest species (S) Authors usually made clear statements about grasshoppers in this category, such as "... frequently causes major damage to rangeland," or "... one of the 10 most destructive species in our study."

Minor pest species (M) Authors categorized such species with phrases like "... this species occasionally causes injury to forage grasses," or "... populations may require control treatment in specific areas."

Innocuous species (I) Authors' remarks often contained phrases like "... of no economic importance," or "... this species was rarely encountered in the study area." Also, if an author categorized 10 grasshopper species as "serious pests" and another 10 species as "minor pests" but then discussed 10 additional species without mention of economic importance, I classified the latter species as "innocuous."

Possibly beneficial species (b?) In this and the next category I included grasshoppers that feed to some extent on undesirable rangeland plants, such as the perennial snakeweeds *(Gutierrezia* spp.). I also assigned species to the "possibly beneficial" category when the authors' remarks were either uncertain or intentionally ambiguous, for example "... possibly beneficial since it feeds on noxious forbs."

Beneficial species (B) In these cases the author's remarks were clear and unequivocal: "... this grasshopper is a beneficial insect."

Scientific Names

In this chapter, grasshopper names follow the usage recognized by the following sources, by subfamily:
Acridinae—Otte (1981)
Cyrtacanthacridinae—Arnett (1985), Helfer (1987)
Gomphocerinae—Otte (1981)
Melanoplinae—Arnett (1985), Helfer (1987)
Oedipodinae—Otte (1984)
Also the scientific names of all grasshoppers discussed were checked for proper usage by Dan Otte (Academy of Natural Sciences, Philadelphia) while this chapter was still in manuscript form. However, I am responsible for the accuracy of all names as printed here. In general, I have tried not to use names of subspecies, but in several instances that was unavoidable.

My Findings

My review of the literature yielded 69 articles (table VI.6–1) in which the authors provided opinions of the relative pest status for the grasshopper species in their studies. In the articles selected, a total of 377 different grasshopper species were discussed by 77 different authors and coauthors over a period of 70 years (1924–93). When these authors' opinions were translated into my five pest-status categories, there were a total of 2,731 rankings on the 377 species. The 2,731 rankings broke down into the five categories as follows:

	Percent
Serious pest species	17.4
Minor pest species	15.7
Innocuous species	65.7
Possibly beneficial species	0.5
Beneficial species	0.7

The 377 grasshoppers (table VI.6–2) included species in the following five acridid subfamilies: Acridinae (1), Cyrtacanthacridinae (8), Gomphocerinae (63), Melanoplinae (185), and Oedipodinae (120). Also listed in table VI.6–2 are the status category tally counts for each species. In order to make calculations, I assigned points for each status category, as follows:
Serious = +2, Minor = +1, Innocuous = 0, Possibly beneficial = –1, and Beneficial = –2.

The total score for each grasshopper species was calculated by multiplying the category tally count times the respective point values for each pest-status category. The rank number was determined by the magnitude of the total score for each species. In cases of tie scores, the species with the highest frequency of mentions as a "serious" and "minor" pest was given the higher rank.

Table VI.6–1—Summary of pest-status rankings of 377 western rangeland grasshoppers from 69 articles

Literature citation	Geographic region	Number of grasshopper species in each status[1]					Total species
		“S”	“M”	“I”	“b?”	“B”	
Arnett (1985)	17 Western States	10	1	59	0	0	70
Ball (1936)	Arizona	0	0	10	0	13	23
Ball et al. (1942)	Arizona	13	27	99	1	1	141
Banfill and Brusven (1973)	Idaho	3	4	19	0	0	26
Bird (1961)	Western Canada	3	2	0	0	0	5
Brusven (1967)	Kansas	1	6	15	0	0	22
Brusven (1972)	Idaho	4	9	2	1	0	16
Brusven and Lambley (1971)	Idaho	2	13	13	0	0	28
Buckell (1936a)	Western Canada	5	1	0	0	0	6
Buckell (1936b)	Western Canada	6	0	0	0	0	6
Capinera (1987)	17 Western States	25	0	0	0	0	25
Capinera and Sechrist (1982)	Colorado	16	11	99	3	0	129
Capinera and Thompson (1987)	Colorado	2	4	3	0	0	9
Coppock (1962)	Oklahoma	10	5	97	1	0	113
Ewen and Mukerji (1984)	Western Canada	4	0	0	0	0	4
Fielding and Brusven (1990)	Idaho	3	4	0	0	0	7
Gibson (1938)	Western Canada	7	6	0	0	0	13
Hagen (1970)	Nebraska	4	8	62	0	0	74
Harper (1952)	California	4	19	1	0	0	24
Hauke (1953)	Nebraska	8	8	97	0	0	113
Hebard (1936)	North Dakota	6	3	59	0	0	68
Hebard (1938)	Oklahoma	10	15	36	0	0	61
Helfer (1987)	17 Western States	19	16	234	0	0	269
Henderson (1924)	Utah	4	8	26	0	0	38
Henderson (1931)	Utah	6	5	1	0	0	12
Hewitt (1977)	17 Western States	26	0	0	0	0	26
Hewitt and Barr (1967)	Idaho	1	5	30	0	0	36
Hewitt et al. (1974)	17 Western States	26	0	0	0	0	26
Isely (1938)	Texas	2	0	36	0	0	38
Kemp and Dennis (1991)	Montana	6	0	0	0	0	6
Kemp and Onsager (1986)	Montana	6	0	0	0	0	6
Kevan (1979)	Western Canada	5	0	1	0	0	6
Knowlton and Janes (1932)	Utah	6	21	0	0	0	27
La Rivers (1948)	Nevada	4	9	63	0	0	76
Middlekauff (1958)	California	2	2	0	0	0	4
Mitchener (1953)	Manitoba	3	2	0	0	0	5
Mulkern (1980)	North Dakota	2	10	25	0	0	37
Mulkern et al. (1962)	North Dakota	7	0	19	0	0	26
Mulkern et al. (1969)	17 Western States	7	11	40	3	0	61
Nerney (1960)	Arizona	3	1	0	0	0	4

Table VI.6–1—Summary of pest-status rankings of 377 western rangeland grasshoppers from 69 articles (Continued)

Literature citation	Geographic region	Number of grasshopper species in each status[1]					Total species
		"S"	"M"	"I"	"b?"	"B"	
Nerney (1961)	Arizona	2	3	0	0	0	5
Nerney and Hamilton (1969)	Arizona	2	6	0	0	0	8
Newton et al. (1954)	Montana and Wyoming	12	0	52	0	0	64
Parker (1952)	17 Western States	19	3	0	0	0	22
Parker (1957)	17 Western States	3	9	2	0	0	14
Parker and Connin (1964)	17 Western States	3	9	1	0	0	13
Pfadt (1949)	17 Western States	8	2	0	4	0	14
Pfadt (1977)	17 Western States	4	8	15	0	0	27
Pfadt (1982)	Arizona	2	1	14	0	0	17
Pfadt (1984)	Colorado	1	12	11	0	0	24
Pfadt (1988)	17 Western States	13	17	5	0	1	36
Pfadt and Hardy (1987)	17 Western States	13	0	0	0	0	13
Putnam (1962)	British Columbia	2	1	0	0	0	3
Richman et al. (1993)	New Mexico	19	23	122	0	1	165
Scoggan and Brusven (1972)	Idaho	4	12	21	0	0	37
Scoggan and Brusven (1973)	Idaho	1	9	38	0	0	48
Shewchuk and Kerr (1993)	Alberta	3	0	0	0	0	3
Shotwell (1938a)	Northern Great Plains	5	0	4	0	0	9
Shotwell (1938b)	17 Western States	10	16	13	0	0	39
Shotwell (1941)	17 Western States	2	10	0	0	0	12
Strohecker et al. (1968)	California	11	9	146	1	0	167
Turnock (1977)	Western Canada	3	0	0	0	0	3
Van Horn (1972)	Colorado	5	10	19	0	0	34
Vickery and Scudder (1987)	Western Canada	7	3	91	0	0	101
Wakeland (1951)	17 Western States	5	11	0	0	0	16
Watts et al. (1989)	17 Western States	25	0	0	0	2	27
White and Rock (1945)	Alberta	5	5	66	0	0	76
Wilbur and Fritz (1940)	Kansas	4	8	18	0	0	30
Woodruff (1937)	Kansas	0	7	11	0	0	18
Totals		474	430	1,795	14	18	2,731
Percent of total rankings		17.4	15.7	65.7	0.5	0.7	100.0

[1] S = serious, M = minor, I = innocuous, b? = possibly beneficial, B = beneficial.

Table VI.6–2—Alphabetical list of 377 western rangeland grasshoppers with pest-status scores and ranks

Grasshopper species	Sub-family[2]	Number of rankings[1] “S”	“M”	“I”	“b?”	“B”	Total	Score	Rank
Acantherus piperatus Scudder & Cockerell	G	0	0	4	0	0	4	0	163
Achurum sumichrasti (Saussure)	G	0	0	5	0	0	5	0	148
Acrolophitus hirtipes (Say)	G	0	0	16	0	0	16	0	113
Acrolophitus maculipennis (Scudder)	G	0	0	5	0	0	5	0	149
Acrolophitus nevadensis (Thomas)	G	0	0	7	0	0	7	0	133
Aeoloplides chenopodii (Bruner)	M	0	0	3	0	1	4	–2	374
Aeoloplides elegans (Scudder)	M	0	0	1	0	0	1	0	264
Aeoloplides fratercula (Hebard)	M	0	0	1	0	0	1	0	265
Aeoloplides fuscipes (Scudder)	M	0	0	1	0	0	1	0	266
Aeoloplides minor (Bruner)	M	0	0	2	0	0	2	0	214
Aeoloplides rotundipennis Wallace	M	0	0	1	0	0	1	0	267
Aeoloplides turnbulli (Caudell)	M	0	3	9	1	0	13	2	65
Aeoloplus californicus Scudder	M	0	0	1	0	0	1	0	268
Aeoloplus tenuipennis (Scudder)	M	0	0	7	0	1	8	–2	368
Aeropedellus clavatus (Thomas)	G	6	2	13	0	0	21	14	32
Ageneotettix brevipennis (Bruner)	G	0	0	1	0	0	1	0	269
Ageneotettix deorum (Scudder)	G	27	7	11	0	0	45	61	5
Ageneotettix salutator (Rehn)	G	0	0	2	0	0	2	0	215
Agnostokasia sublima Gurney & Rentz	M	0	0	2	0	0	2	0	216
Agroecotettix modestus Bruner	M	0	0	2	0	0	2	0	217
Agymnastus ingens (Scudder)	O	0	0	3	0	0	3	0	183
Aidemona azteca Saussure	M	0	0	3	0	0	3	0	184
Amblytropidia mysteca (Saussure)	G	0	0	5	0	0	5	0	150
Amphitornus coloradus (Thomas)	G	18	12	12	0	0	42	48	8
Anconia hebardi Rehn	O	0	0	2	0	0	2	0	218
Anconia integra Scudder	O	0	0	5	0	1	6	–2	370
Argiacris militaris (Scudder)	M	0	0	1	0	0	1	0	270
Argiacris rehni Hebard	M	0	0	2	0	0	2	0	219
Arphia behrensi Saussure	O	0	0	3	0	0	3	0	185
Arphia conspersa Scudder	O	0	2	22	0	0	24	2	66
Arphia pseudonietana (Thomas)	O	1	8	20	0	0	29	10	36
Arphia ramona Rehn	O	0	0	1	0	0	1	0	271
Arphia saussureana Bruner	O	0	0	1	0	0	1	0	272
Arphia simplex Scudder	O	0	0	8	0	0	8	0	128
Arphia sulphurea (Fabricius)	O	0	0	5	0	0	5	0	151
Arphia xanthoptera (Burmeister)	O	0	0	8	0	0	8	0	129
Asemoplus hispidus (Bruner)	M	0	0	1	0	0	1	0	273
Asemoplus montanus (Bruner)	M	0	1	3	0	0	4	1	105
Asemoplus sierranus Hebard	M	0	0	1	0	0	1	0	274
Aulocara elliotti (Thomas)	G	39	7	3	0	0	49	85	2

Table VI.6–2—Alphabetical list of 377 western rangeland grasshoppers with pest-status scores and ranks (Continued)

Grasshopper species	Sub-family[2]	Number of rankings[1] “S”	“M”	“I”	“b?”	“B”	Total	Score	Rank
Aulocara femoratum (Scudder)	G	12	8	6	0	0	26	32	12
Aztecacris gloriosus (Hebard)	M	0	0	4	0	0	4	0	164
Barytettix cochisei Gurney	M	0	0	1	0	0	1	0	275
Barytettix humphreysii (Thomas)	M	0	0	3	0	0	3	0	186
Booneacris glacialis (Scudder)	M	0	0	2	0	0	2	0	220
Boopedon auriventris McNeill	G	0	0	6	0	0	6	0	142
Boopedon flaviventris (Bruner)	G	2	1	0	0	0	3	5	54
Boopedon gracile Rehn	G	0	0	5	0	0	5	0	152
Boopedon nubilum (Say)	G	4	6	11	0	0	21	14	31
Bootettix argentatus Bruner	G	0	0	6	0	1	7	–2	369
Bradynotes obesa (Thomas)	M	0	1	8	0	0	9	1	91
Buckellacris chilcotinae (Hebard)	M	0	0	1	0	0	1	0	276
Buckellacris hispida (Bruner)	M	0	0	1	0	0	1	0	277
Buckellacris nuda (Walker)	M	0	0	3	0	0	3	0	187
Camnula pellucida (Scudder)	O	35	7	5	0	0	47	77	3
Campylacantha olivacea (Scudder)	M	1	0	9	0	0	10	2	80
Chimarocephala elongata Rentz	O	0	0	1	0	0	1	0	278
Chimarocephala pacifica (Thomas)	O	0	0	3	0	0	3	0	188
Chloealtis abdominalis (Thomas)	G	0	0	9	0	0	9	0	125
Chloealtis aspasma (Rehn & Hebard)	G	0	0	1	0	0	1	0	279
Chloealtis conspersa (Harris)	G	0	0	14	0	0	14	0	116
Chloealtis dianae (Gur., Stro. & Helf.)	G	0	0	2	0	0	2	0	221
Chloealtis gracilis (McNeill)	G	0	0	2	0	0	2	0	222
Chloroplus cactocaetes Hebard	M	0	0	2	0	0	2	0	223
Chorthippus curtipennis (Harris)	G	6	7	15	0	0	28	19	19
Chortophaga mendocino Rentz	O	0	0	1	0	0	1	0	280
Chortophaga viridifasciata (DeGeer)	O	0	3	17	0	0	20	3	58
Chrysochraon petraea (Gur., Stro. & Helf.)	G	0	0	2	0	0	2	0	224
Cibolacris parviceps (Walker)	G	0	0	8	0	0	8	0	130
Cibolacris samalayucae Tinkham	G	0	0	1	0	0	1	0	281
Circotettix carlinianus (Thomas)	O	0	1	13	0	0	14	1	84
Circotettix crotalum Rehn	O	0	0	2	0	0	2	0	225
Circotettix maculatus Scudder	O	0	0	3	0	0	3	0	189
Circotettix rabula Rehn & Hebard	O	0	0	14	0	0	14	0	117
Circotettix shastanus Bruner	O	0	0	2	0	0	2	0	226
Circotettix stenometopus (Stro. & Buxt.)	O	0	0	2	0	0	2	0	227
Circotettix undulatus (Thomas)	O	0	2	9	0	0	11	2	72
Clematodes larreae Scudder	M	0	0	4	0	0	4	0	165
Conalcea huachucana Rehn	M	0	0	3	0	0	3	0	190
Conozoa carinata Rehn	O	0	1	2	0	0	3	1	109

Table VI.6–2—Alphabetical list of 377 western rangeland grasshoppers with pest-status scores and ranks (Continued)

Grasshopper species	Sub-family[2]	Number of rankings[1]					Total	Score	Rank
		“S”	“M”	“I”	“b?”	“B”			
Conozoa hyalina (McNeill)	O	0	0	1	0	0	1	0	282
Conozoa rebellis (Saussure)	O	0	0	4	0	0	4	0	166
Conozoa sulcifrons (Scudder)	O	0	6	10	0	0	16	6	46
Conozoa texana (Bruner)	O	0	0	12	0	0	12	0	121
Cordillacris crenulata (Bruner)	G	4	7	11	0	0	22	15	29
Cordillacris occipitalis (Thomas)	G	13	4	14	0	0	31	30	15
Cratypedes lateritius (Saussure)	O	0	0	6	0	0	6	0	143
Cratypedes neglectus (Thomas)	O	0	5	12	0	0	17	5	51
Dactylotum bicolor pictum (Thomas)	M	0	1	12	0	0	13	1	86
Dactylotum bicolor variegatum (Scudder)	M	0	0	3	0	0	3	0	191
Dendrotettix hesperus (Hebard)	M	0	0	2	0	0	2	0	228
Derotmema delicatulum Scudder	O	0	0	4	0	0	4	0	167
Derotmema haydeni (Thomas)	O	0	1	20	0	0	21	1	83
Derotmema laticinctum Scudder	O	0	0	3	0	0	3	0	192
Derotmema saussureanum Scudder	O	0	0	2	0	0	2	0	229
Dichromorpha elegans (Morse)	G	0	0	1	0	0	1	0	283
Dichromorpha viridis (Scudder)	G	0	0	7	0	0	7	0	134
Dissosteira carolina (Linnaeus)	O	3	11	18	0	0	32	17	24
Dissosteira longipennis (Thomas)	O	8	2	3	0	0	13	18	23
Dissosteira pictipennis Bruner	O	0	2	2	0	0	4	2	75
Dissosteira spurcata Saussure	O	3	8	6	0	0	17	14	30
Encoptolophus californicus (Bruner)	O	0	0	1	0	0	1	0	284
Encoptolophus costalis (Scudder)	O	5	3	7	0	0	15	13	34
Encoptolophus pallidus Bruner	O	0	0	3	0	0	3	0	193
Encoptolophus robustus Rehn & Hebard	O	0	0	1	0	0	1	0	285
Encoptolophus sordidus (Burmeister)	O	2	3	6	0	0	11	7	43
Encoptolophus subgracilis Caudell	O	0	3	6	0	0	9	3	60
Eritettix abortivus (Bruner)	G	0	0	2	0	0	2	0	230
Eritettix simplex (Scudder)	G	7	3	15	0	0	25	17	26
Esselenia vanduzeei Hebard	G	0	1	3	0	0	4	1	106
Eupnigodes megacephala (McNeill)	G	0	1	2	0	0	3	1	110
Eupnigodes sierranus Rehn & Hebard	G	0	0	2	0	0	2	0	231
Hadrotettix magnificus (Rehn)	O	0	0	5	0	0	5	0	153
Hadrotettix trifasciatus (Say)	O	0	3	22	0	0	25	3	57
Hebardacris albida (Hebard)	M	0	0	3	0	0	3	0	194
Hebardacris excelsa (Rehn)	M	0	0	2	0	0	2	0	232
Hebardacris mono Rehn	M	0	0	2	0	0	2	0	233
Heliastus benjamini Caudell	O	0	0	4	0	0	4	0	168
Heliaula rufa (Scudder)	G	0	0	11	0	0	11	0	124
Hesperotettix curtipennis Scudder	M	0	0	1	0	1	2	–2	375

Table VI.6–2—Alphabetical list of 377 western rangeland grasshoppers with pest-status scores and ranks (Continued)

Grasshopper species	Sub-family[2]	Number of rankings[1] “S”	“M”	“I”	“b?”	“B”	Total	Score	Rank
Hesperotettix nevadensis Morse	M	0	0	1	0	0	1	0	286
Hesperotettix pacificus Scudder	M	0	0	1	0	0	1	0	287
Hesperotettix speciosus (Scudder)	M	1	0	7	2	0	10	0	112
Hesperotettix viridis (Thomas)	M	0	2	17	5	5	29	–13	377
Hippiscus ocelote (Saussure)	O	0	2	12	0	0	14	2	69
Hippopedon capito (Stal)	O	0	0	3	0	0	3	0	195
Hippopedon gracilipes (Caudell)	O	0	0	3	0	0	3	0	196
Horesidotes cinereus Scudder	G	0	0	5	0	0	5	0	154
Hypochlora alba (Dodge)	M	0	0	13	2	1	16	–4	376
Hypsalonia merga Gurney & Buxton	M	0	0	1	0	0	1	0	288
Hypsalonia miwoki Gurney & Eades	M	0	0	1	0	0	1	0	289
Hypsalonia petasata Gurney & Eades	M	0	0	1	0	0	1	0	290
Hypsalonia rentzi Gurney & Eades	M	0	0	1	0	0	1	0	291
Hypsalonia satur (Scudder)	M	0	0	1	0	0	1	0	292
Hypsalonia tioga Gurney & Eades	M	0	0	1	0	0	1	0	293
Karokia blanci (Rehn)	M	0	0	1	0	0	1	0	294
Lactista aztecus (Saussure)	O	0	2	2	0	0	4	2	76
Lactista gibbosus Saussure	O	0	0	3	0	0	3	0	197
Leprus intermedius Saussure	O	0	0	9	0	0	9	0	126
Leprus wheeleri (Thomas)	O	0	1	6	0	0	7	1	97
Leptysma marginicollis (Serville)	M	0	0	6	0	0	6	0	144
Leuronotina ritensis (Rehn)	O	0	0	3	0	0	3	0	198
Ligurotettix coquilletti McNeill	G	0	0	4	0	1	5	–2	372
Ligurotettix planum (Bruner)	G	0	0	2	0	0	2	0	234
Melanoplus ablutus Scudder	M	0	0	1	0	0	1	0	295
Melanoplus alpinus Scudder	M	0	1	7	0	0	8	1	95
Melanoplus angustipennis (Dodge)	M	4	4	12	0	0	20	12	35
Melanoplus aridus (Scudder)	M	0	2	4	0	0	6	2	73
Melanoplus arizonae Scudder	M	0	3	4	0	0	7	3	61
Melanoplus artemesiae (Bruner)	M	0	0	1	0	0	1	0	296
Melanoplus ascensus Scudder	M	0	0	1	0	0	1	0	297
Melanoplus aspasmus Hebard	M	0	0	1	0	0	1	0	298
Melanoplus beameri Hebard	M	0	0	1	0	0	1	0	299
Melanoplus bernardinae Hebard	M	0	0	1	0	0	1	0	300
Melanoplus bispinosus Scudder	M	0	2	3	0	0	5	2	74
Melanoplus bivittatus (Say)	M	2	14	6	0	0	47	68	4
Melanoplus bohemani (Stal)	M	0	0	1	0	0	1	0	301
Melanoplus borealis (Fieber)	M	2	1	8	0	0	11	5	53
Melanoplus bowditchi Scudder	M	1	0	13	1	0	15	1	82
Melanoplus bruneri Scudder	M	3	1	6	0	0	10	7	44

Table VI.6–2—Alphabetical list of 377 western rangeland grasshoppers with pest-status scores and ranks (Continued)

Grasshopper species	Sub-family[2]	Number of rankings[1]					Total	Score	Rank
		“S”	“M”	“I”	“b?”	“B”			
Melanoplus buxtoni Strohecker	M	0	0	1	0	0	1	0	302
Melanoplus caroli Gurney & Helfer	M	0	0	2	0	0	2	0	235
Melanoplus chimariki Gurney & Buxton	M	0	0	1	0	0	1	0	303
Melanoplus chiricahuae Hebard	M	0	0	1	0	0	1	0	304
Melanoplus cinereus Scudder	M	0	1	5	0	0	6	1	99
Melanoplus complanatipes Scudder	M	0	1	5	0	0	6	1	100
Melanoplus confusus Scudder	M	1	4	17	0	0	22	6	48
Melanoplus daemon Strohecker	M	0	0	1	0	0	1	0	305
Melanoplus dawsoni (Scudder)	M	2	5	11	0	0	18	9	39
Melanoplus desultorius Rehn	M	1	0	1	1	1	4	–1	366
Melanoplus devastator Scudder	M	9	1	0	0	0	10	19	20
Melanoplus differentialis (Thomas)	M	13	11	4	0	0	28	37	10
Melanoplus discolor (Scudder)	M	0	0	7	0	0	7	0	135
Melanoplus dodgei (Thomas)	M	0	0	3	0	0	3	0	199
Melanoplus elaphrus Strohecker	M	0	0	1	0	0	1	0	306
Melanoplus elater Strohecker	M	0	0	1	0	0	1	0	307
Melanoplus eremitus Strohecker	M	0	0	1	0	0	1	0	308
Melanoplus fasciatus (Walker)	M	0	0	7	0	0	7	0	136
Melanoplus femurnigrum Scudder	M	0	0	2	0	0	2	0	236
Melanoplus femurrubrum (DeGeer)	M	18	19	3	0	0	40	55	7
Melanoplus flabellatus Scudder	M	0	0	1	0	0	1	0	309
Melanoplus flavidus Scudder	M	0	2	11	0	0	13	2	70
Melanoplus foedus Scudder	M	2	9	13	0	0	24	13	33
Melanoplus franciscanus Scudder	M	0	0	2	0	0	2	0	237
Melanoplus fricki Strohecker	M	0	0	1	0	0	1	0	310
Melanoplus frigidus (Boheman)	M	0	0	1	0	0	1	0	311
Melanoplus fultoni Hebard	M	0	0	1	0	0	1	0	312
Melanoplus gladstoni Scudder	M	8	3	11	1	0	23	18	21
Melanoplus glaucipes (Scudder)	M	0	1	4	0	0	5	1	102
Melanoplus gracilipes Scudder	M	0	0	1	0	0	1	0	313
Melanoplus gracilis (Bruner)	M	0	0	3	0	0	3	0	200
Melanoplus harperi Gurney & Buxton	M	0	0	1	0	0	1	0	314
Melanoplus herbaceus Bruner	M	0	0	5	0	1	6	–2	371
Melanoplus hesperus Hebard	M	0	0	1	0	0	1	0	315
Melanoplus hupah Strohecker & Helfer	M	0	0	1	0	0	1	0	316
Melanoplus huporeus Hebard	M	0	0	1	0	0	1	0	317
Melanoplus huroni Blatchley	M	0	0	5	0	0	5	0	155
Melanoplus immunis Scudder	M	0	0	1	0	0	1	0	318
Melanoplus impudicus Scudder	M	0	0	2	0	0	2	0	238
Melanoplus inconspicuous Caudell	M	0	0	2	0	0	2	0	239

Table VI.6–2—Alphabetical list of 377 western rangeland grasshoppers with pest-status scores and ranks (Continued)

Grasshopper species	Sub-family[2]	Number of rankings[1] “S”	“M”	“I”	“b?”	“B”	Total	Score	Rank
Melanoplus indigens Scudder	M	0	1	3	0	0	4	1	107
Melanoplus infantilis Scudder	M	12	7	11	0	0	30	31	13
Melanoplus islandicus Blatchley	M	0	0	1	0	0	1	0	319
Melanoplus keeleri (Thomas)	M	0	2	14	0	0	16	2	67
Melanoplus keiferi Gurney & Buxton	M	0	0	1	0	0	1	0	320
Melanoplus kennicotti Scudder	M	0	0	5	0	0	5	0	156
Melanoplus lakinus (Scudder)	M	0	1	7	0	0	8	1	96
Melanoplus lemhiensis Hebard	M	0	0	1	0	0	1	0	321
Melanoplus lepidus Scudder	M	0	0	2	0	0	2	0	240
Melanoplus ligneolus Scudder	M	0	0	1	0	0	1	0	322
Melanoplus lithophilus Gurney & Buxton	M	0	0	1	0	0	1	0	323
Melanoplus magdalenae Hebard	M	0	0	2	0	0	2	0	241
Melanoplus marginatus (Scudder)	M	1	3	0	0	0	4	5	52
Melanoplus microtatus Hebard	M	0	0	1	0	0	1	0	324
Melanoplus montanus (Thomas)	M	0	0	3	0	0	3	0	201
Melanoplus muricolor Strohecker	M	0	0	1	0	0	1	0	325
Melanoplus nanus Scudder	M	0	0	1	0	0	1	0	326
Melanoplus occidentalis (Thomas)	M	7	7	10	0	1	25	19	18
Melanoplus oklahomae Hebard	M	0	0	2	0	0	2	0	242
Melanoplus olamentke Hebard	M	0	0	1	0	0	1	0	327
Melanoplus oregonensis (Thomas)	M	0	0	4	0	0	4	0	169
Melanoplus pacificus (Scudder)	M	0	0	1	0	0	1	0	328
Melanoplus packardii Scudder	M	23	12	5	0	0	40	58	6
Melanoplus payettei Hebard	M	0	0	1	0	0	1	0	329
Melanoplus pictus Scudder	M	0	2	1	0	0	3	2	78
Melanoplus pinaleno Hebard	M	0	0	1	0	0	1	0	330
Melanoplus platycercus Hebard	M	0	0	1	0	0	1	0	331
Melanoplus plebejus (Stal)	M	0	0	3	0	0	3	0	202
Melanoplus ponderosus Scudder	M	0	0	7	0	0	7	0	137
Melanoplus punctulatus (Scudder)	M	0	0	3	0	0	3	0	203
Melanoplus regalis (Dodge)	M	0	0	7	0	0	7	0	138
Melanoplus rileyanus Scudder	M	0	0	2	0	0	2	0	243
Melanoplus rugglesi Gurney	M	5	0	3	0	0	8	10	38
Melanoplus rusticus (Stal)	M	0	0	1	0	0	1	0	332
Melanoplus saltator Scudder	M	0	0	1	0	0	1	0	333
Melanoplus sanguinipes (Fabricius)	M	53	7	1	1	0	62	112	1
Melanoplus scudderi (Uhler)	M	0	0	7	0	0	7	0	139
Melanoplus siskiyou Strohecker	M	0	0	1	0	0	1	0	334
Melanoplus snowii (Scudder)	M	0	0	2	0	0	2	0	244
Melanoplus sonomaensis Caudell	M	0	0	2	0	0	2	0	245

Table VI.6–2—Alphabetical list of 377 western rangeland grasshoppers with pest-status scores and ranks (Continued)

Grasshopper species	Sub-family[2]	Number of rankings[1] “S”	“M”	“I”	“b?”	“B”	Total	Score	Rank
Melanoplus splendidus Hebard	M	0	0	4	0	0	4	0	170
Melanoplus stonei Rehn	M	0	0	1	0	0	1	0	335
Melanoplus texanus (Scudder)	M	0	0	4	0	0	4	0	171
Melanoplus thomasi Scudder	M	0	2	1	0	0	3	2	79
Melanoplus tristis Bruner	M	0	0	2	0	0	2	0	246
Melanoplus truncatus Scudder	M	0	0	1	0	0	1	0	336
Melanoplus tuberculatus Morse	M	0	0	1	0	0	1	0	337
Melanoplus tunicae Hebard	M	0	0	1	0	0	1	0	338
Melanoplus viridipes Scudder	M	0	0	1	0	0	1	0	339
Melanoplus walshii Scudder	M	0	0	1	0	0	1	0	340
Melanoplus warneri Little	M	0	0	1	0	0	1	0	341
Melanoplus washingtonius (Bruner)	M	0	0	1	0	0	1	0	342
Melanoplus wilsoni Gurney	M	0	0	1	0	0	1	0	343
Melanoplus wintunus Strohecker & Helfer	M	0	0	1	0	0	1	0	344
Melanoplus yarrowii (Thomas)	M	0	4	1	0	0	5	4	55
Mermiria bivittata (Serville)	G	6	12	10	0	0	28	24	17
Mermiria picta (Walker)	G	0	1	8	0	0	9	1	92
Mermiria texana Bruner	G	0	0	6	0	0	6	0	145
Mestobregma impexum Rehn	O	0	0	4	0	0	4	0	172
Mestobregma plattei (Thomas)	O	0	1	8	0	0	9	1	93
Mestobregma terricolor Rehn	O	0	0	3	0	0	3	0	204
Metaleptea brevicornis (Johannson)	A	0	0	2	0	0	2	0	247
Metator nevadensis (Bruner)	O	0	0	5	0	0	5	0	157
Metator pardalinus (Saussure)	O	4	9	15	0	0	28	17	25
Microtes helferi (Strohecker)	O	0	0	3	0	0	3	0	205
Microtes occidentalis (Bruner)	O	0	0	3	0	0	3	0	206
Microtes pogonata (Strohecker)	O	0	0	1	0	0	1	0	345
Netrosoma nigropleura Scudder	M	0	0	1	0	0	1	0	346
Nisquallia olympica Rehn	M	0	0	2	0	0	2	0	248
Oedaleonotus borckii (Stal)	M	0	1	4	0	0	5	1	103
Oedaleonotus enigma (Scudder)	M	7	4	4	0	0	15	18	22
Oedaleonotus orientis Hebard	M	0	0	1	0	0	1	0	347
Oedaleonotus pacificus (Scudder)	M	0	0	1	0	0	1	0	348
Oedaleonotus phryneicus Hebard	M	0	0	1	0	0	1	0	349
Oedaleonotus pictus (Scudder)	M	0	0	1	0	0	1	0	350
Oedaleonotus tenuipennis (Scudder)	M	0	0	1	0	0	1	0	351
Oedomerus corallipes Bruner	M	0	0	1	0	0	1	0	352
Opeia atascosa Hebard	G	0	0	2	0	0	2	0	249
Opeia obscura (Thomas)	G	13	5	11	0	0	29	31	14
Orphulella pelidna (Burmeister)	G	0	3	10	0	0	13	3	59

Table VI.6–2—Alphabetical list of 377 western rangeland grasshoppers with pest-status scores and ranks (Continued)

Grasshopper species	Sub-family[2]	Number of rankings[1] “S”	“M”	“I”	“b?”	“B”	Total	Score	Rank
Orphulella speciosa (Scudder)	G	3	3	14	0	0	20	9	40
Paraidemona mimica (Scudder)	M	0	0	1	0	0	1	0	353
Paraidemona punctata (Stal)	M	0	0	1	0	0	1	0	354
Paratylotropidia brunneri Scudder	M	0	0	4	0	0	4	0	173
Paratylotropidia morsei Rehn & Rehn	M	0	0	2	0	0	2	0	250
Pardalophora apiculata (Harris)	O	0	0	12	0	0	12	0	122
Pardalophora haldemani (Scudder)	O	0	1	13	0	0	14	1	85
Pardalophora phoenicoptera (Burmeister)	O	0	0	3	0	0	3	0	207
Pardalophora saussurei (Scudder)	O	0	0	5	0	0	5	0	158
Paropomala pallida Bruner	G	0	0	7	0	0	7	0	140
Paropomala virgata (Scudder)	G	0	0	4	0	0	4	0	174
Paropomala wyomingensis (Thomas)	G	1	1	13	0	0	15	3	62
Paroxya atlantica Scudder	M	0	0	2	0	0	2	0	251
Paroxya clavuliger (Serville)	M	0	0	1	0	0	1	0	355
Phaedrotettix dumicola palmeri (Scudder)	M	0	0	1	0	0	1	0	356
Phaulotettix compressus Scudder	M	0	0	1	0	0	1	0	357
Phaulotettix eurycercus Hebard	M	0	0	1	0	0	1	0	358
Phlibostroma quadrimaculatum (Thomas)	G	13	11	6	0	0	30	37	9
Phoetaliotes nebrascensis (Thomas)	M	8	11	10	0	0	29	27	16
Poecilotettix longipennis (Townsend)	M	0	0	1	0	0	1	0	359
Poecilotettix pantherinus (Walker)	M	0	0	4	0	1	5	–2	373
Poecilotettix sanguineus Scudder	M	0	0	4	0	0	4	0	175
Prorocorypha snowi Rehn	M	0	0	3	0	0	3	0	208
Prumnacris rainierensis (Caudell)	M	0	0	2	0	0	2	0	252
Pseudopomala brachyptera (Scudder)	G	0	0	15	0	0	15	0	114
Psinidia amplicornis Caudell	O	0	0	1	0	0	1	0	360
Psinidia fenestralis (Serville)	O	0	0	3	0	0	3	0	209
Psoloessa delicatula (Scudder)	G	1	4	20	0	0	25	6	47
Psoloessa texana Scudder	G	1	1	8	0	0	10	3	63
Rhammatocerus viatorius (Saussure)	G	0	0	3	0	0	3	0	210
Schistocerca alutacea albolineata (Thomas)	C	0	1	4	0	0	5	1	104
Schistocerca alutacea rubiginosa (Harris)	C	0	0	1	0	0	1	0	361
Schistocerca alutacea shoshone (Thomas)	C	2	6	3	0	0	11	10	37
Schistocerca americana (Drury)	C	2	2	5	0	0	9	6	50
Schistocerca damnifica (Saussure)	C	0	0	2	0	0	2	0	253
Schistocerca emarginata Scudder	C	1	4	11	0	0	16	6	49
Schistocerca nitens (Thunberg)	C	2	4	1	0	1	8	6	45
Schistocerca obscura (Fabricius)	C	0	0	5	0	0	5	0	159
Shotwellia isleta Gurney	O	0	0	3	0	0	3	0	211
Spharagemon bolli Scudder	O	0	0	7	0	0	7	0	141

Table VI.6–2—Alphabetical list of 377 western rangeland grasshoppers with pest-status scores and ranks (Continued)

Grasshopper species	Sub-family[2]	Number of rankings[1]					Total	Score	Rank
		“S”	“M”	“I”	“b?”	“B”			
Spharagemon campestris (McNeill)	O	0	0	15	0	0	15	0	115
Spharagemon collare (Scudder)	O	1	5	20	0	0	26	7	42
Spharagemon cristatum (Scudder)	O	0	0	2	0	0	2	0	254
Spharagemon equale (Say)	O	0	7	17	0	0	24	7	41
Spharagemon superbum Hebard	O	0	0	2	0	0	2	0	255
Stenobothrus brunneus Thomas	G	1	1	7	0	0	9	3	64
Stenobothrus shastanus (Scudder)	G	0	2	2	0	0	4	2	77
Stethophyma gracile (Scudder)	G	0	0	6	0	0	6	0	146
Stethophyma lineata (Scudder)	G	0	0	4	0	0	4	0	176
Sticthippus californicus (Scudder)	O	1	2	2	0	0	5	4	56
Syrbula admirabilis (Uhler)	G	0	1	10	0	0	11	1	88
Syrbula montezuma (Saussure)	G	0	1	3	0	0	4	1	108
Tomonotus ferruginosus Bruner	O	0	0	4	0	0	4	0	177
Trachyrhachys aspera Scudder	O	0	1	5	0	0	6	1	101
Trachyrhachys coronata Scudder	O	0	0	4	0	0	4	0	178
Trachyrhachys kiowa (Thomas)	O	13	10	13	0	0	36	36	11
Trepidulus hyalinus (Scudder)	O	0	0	2	0	0	2	0	256
Trepidulus rosaceus (Scudder)	O	0	0	5	0	0	5	0	160
Trimerotropis agrestis McNeill	O	0	0	13	0	0	13	0	118
Trimerotropis albescens McNeill	O	0	0	3	0	0	3	0	212
Trimerotropis arenacea Rehn	O	0	0	6	0	0	6	0	147
Trimerotropis arizonensis Tinkham	O	0	0	2	0	0	2	0	257
Trimerotropis barnumi Tinkham	O	0	0	2	0	0	2	0	258
Trimerotropis bifaciata Bruner	O	0	1	1	0	0	2	1	111
Trimerotropis californica Bruner	O	0	1	10	0	0	11	1	89
Trimerotropis cincta (Thomas)	O	0	0	9	0	0	9	0	127
Trimerotropis cyaneipennis Bruner	O	0	1	9	0	0	10	1	90
Trimerotropis diversellus Hebard	O	0	0	1	0	0	1	0	362
Trimerotropis fontana Thomas	O	0	2	10	0	0	12	2	71
Trimerotropis fratercula McNeill	O	0	0	4	0	0	4	0	179
Trimerotropis gracilis (Thomas)	O	0	1	11	0	0	12	1	87
Trimerotropis inconspicua Bruner	O	0	0	8	0	0	8	0	131
Trimerotropis koebelei (Bruner)	O	0	0	3	0	0	3	0	213
Trimerotropis latifasciata Scudder	O	0	2	13	0	0	15	2	68
Trimerotropis maritima (Harris)	O	0	1	8	0	0	9	1	94
Trimerotropis melanoptera McNeill	O	0	0	4	0	0	4	0	180
Trimerotropis modesta Bruner	O	0	0	4	0	0	4	0	181
Trimerotropis occidentalis (Bruner)	O	0	0	2	0	0	2	0	259
Trimerotropis pacifica Bruner	O	0	0	2	0	0	2	0	260
Trimerotropis pallidipennis (Burmeister)	O	1	13	9	0	0	23	15	27

Table VI.6–2—Alphabetical list of 377 western rangeland grasshoppers with pest-status scores and ranks (Continued)

Grasshopper species	Sub-family[2]	Number of rankings[1] “S”	“M”	“I”	“b?”	“B”	Total	Score	Rank
Trimerotropis pistrinaria Saussure	O	0	0	13	0	0	13	0	119
Trimerotropis pseudofasciata Scudder	O	0	1	6	0	0	7	1	98
Trimerotropis salina McNeill	O	0	0	4	0	0	4	0	182
Trimerotropis saxatilis McNeill	O	0	0	2	0	0	2	0	261
Trimerotropis sparsa (Thomas)	O	0	0	13	0	0	13	0	120
Trimerotropis thalassica Bruner	O	0	0	2	0	0	2	0	262
Trimerotropis titusi Caudell	O	0	0	1	0	0	1	0	363
Trimerotropis tolteca (Saussure)	O	0	0	1	0	0	1	0	364
Trimerotropis verruculata (Kirby)	O	1	0	5	0	0	6	2	81
Trimerotropis verruculata suffusa Scudder	O	0	0	12	0	0	12	0	123
Tropidolophus formosus (Say)	O	0	0	9	0	1	10	–2	367
Xanthippus aquilonius Otte	O	0	0	1	0	0	1	0	365
Xanthippus corallipes (Haldeman)	O	3	9	17	0	0	29	15	28
Xanthippus montanus (Thomas)	O	0	0	8	0	0	8	0	132
Xanthippus olancha (Caudell)	O	0	0	2	0	0	2	0	263
Xeracris minimus (Scudder)	G	0	0	5	0	0	5	0	161
Xeracris snowi (Caudell)	G	0	0	5	0	0	5	0	162

[1] S = serious, M = minor, I = innocuous, b? = possibly beneficial, B = beneficial.
[2] A = Acridinae, C = Cyrtacanthacridinae, G = Gomphocerinae, M = Melanoplinae, O = Oedipodinae.

Each of the 377 species is represented (in order of overall score and rank) in the bar graph shown in figure VI.6–1. From left to right, it displays 111 grasshopper species with scores above zero (“pests”), 254 species with a score of zero (“innocuous”), and 12 species with scores below zero (“possibly beneficial” or “beneficial”).

Pest Species.—A total of 114 different grasshoppers were categorized as either a serious or a minor pest in at least one paper, but only 111 species had total scores above zero. In table VI.6–3, I have listed 38 of the highest ranked “pest” species, those with scores of 10 and above. As expected, the migratory grasshopper *(Melanoplus sanguinipes)* was ranked as the number 1 pest, with the highest total score (112 points) of the 377 grasshopper species.

Innocuous Species.—There were 254 grasshopper species with a total score of zero. Within this group, higher rank numbers were assigned to species having the highest frequency of mention. Several species, including *Acrolophitus hirtipes, Pseudopomala brachyptera,* and *Spharagemon campestris,* were mentioned frequently but were never described as either a pest or a beneficial. For innocuous species with only a single ranking, the rank number has no significance; it was assigned due to the alphabetical arrangement of scientific names.

Beneficial Species.—Overall, 19 different grasshoppers were categorized by at least one author as either beneficial or possibly beneficial, but only 12 species had total scores below zero. The highest ranked “beneficial” grasshoppers are listed in table VI.6–4. Although 12 spe-

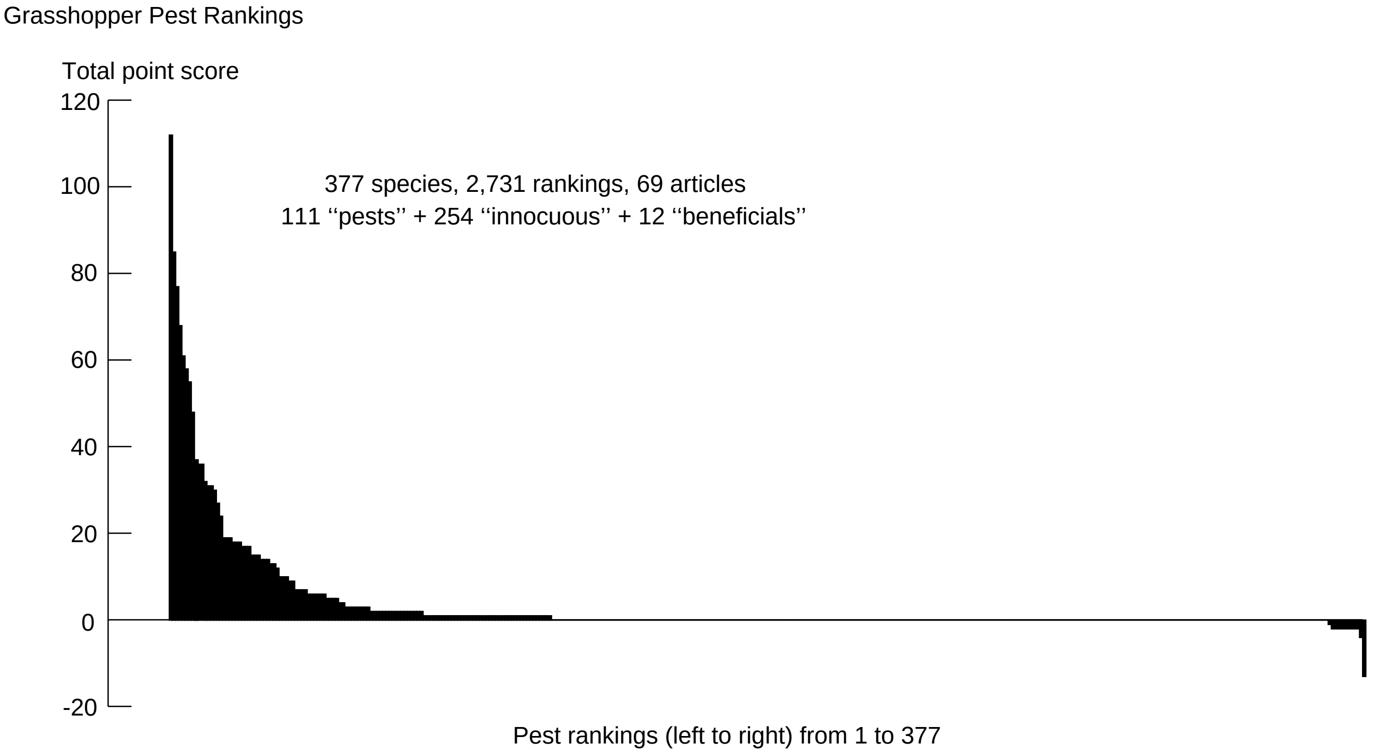


Figure VI.6–1—Graphic display of total scores of 377 western range grasshoppers arranged (left to right) by pest-status rank number. Graph is plotted from data shown in table VI.6–2.

cies were scored as “beneficial,” only 2 were mentioned as such with any frequency: *Hesperotettix viridis* Thomas, a grasshopper commonly associated with snakeweed *(Gutierrezia* spp.), and *Hypochlora alba* Dodge, which prefers to feed on sagebrush *(Artemisia* spp.).

Conclusions

In his 1977 review, Hewitt divided the western rangelands into three different regions: Great Plains, Intermountain, and Pacific Coastal. The literature I reviewed covered a cross section of these same regions, but the reader should be aware that not all of the 377 grasshoppers listed here are common to all regions. Indeed, one limitation of my scoring scheme is that widespread species are cited more frequently and thus accumulate higher total scores than species with a more restricted distribution. A serious pest that occurs in a small geographic area would not be such a pest in the big picture. Three such species, listed in table VI.6–3, are *Dissosteira longipennis, Melanoplus devastator,* and *Oedaleonotus enigma.*

The graph in figure VI.6–1 offers a view of the whole spectrum of western grasshoppers and should provide some perspective when evaluating their relative importance as pests and as beneficials. From the graph it seems clear that nearly one-third (111) of the western grasshopper species are at least occasionally classified as pests. Again I must stress that damage to rangeland is rarely caused by only a single pest species but usually by an assemblage of several grasshopper species.

About two-thirds (254) of the western grasshoppers are thought to be of no economic importance, and only 12 species are considered to be of possible benefit to the

Table VI.6–3 —List of the 38 most serious “pest” grasshoppers on western rangeland (those listed have scores of 10 and above)

Grasshopper species	Sub-family[2]	Number of rankings[1]					Total	Score	Rank
		“S”	“M”	“I”	“b?”	“B”			
Melanoplus sanguinipes (Fabricius)	M	53	7	1	1	0	62	112	1
Aulocara elliotti (Thomas)	G	39	7	3	0	0	49	85	2
Camnula pellucida (Scudder)	O	35	7	5	0	0	47	77	3
Melanoplus bivittatus (Say)	M	27	14	6	0	0	47	68	4
Ageneotettix deorum (Scudder)	G	27	7	11	0	0	45	61	5
Melanoplus packardii Scudder	M	23	12	5	0	0	40	58	6
Melanoplus femurrubrum (DeGeer)	M	18	19	3	0	0	40	55	7
Amphitornus coloradus (Thomas)	G	18	12	12	0	0	42	48	8
Phlibostroma quadrimaculatum (Thomas)	G	13	11	6	0	0	30	37	9
Melanoplus differentialis (Thomas)	M	13	11	4	0	0	28	37	10
Trachyrhachys kiowa (Thomas)	O	13	10	13	0	0	36	36	11
Aulocara femoratum (Scudder)	G	12	8	6	0	0	26	32	12
Melanoplus infantilis Scudder	M	12	7	11	0	0	30	31	13
Opeia obscura (Thomas)	G	13	5	11	0	0	29	31	14
Cordillacris occipitalis (Thomas)	G	13	4	14	0	0	31	30	15
Phoetaliotes nebrascensis (Thomas)	M	8	11	10	0	0	29	27	16
Mermiria bivittata (Serville)	G	6	12	10	0	0	28	24	17
Melanoplus occidentalis (Thomas)	M	7	7	10	0	1	25	19	18
Chorthippus curtipennis (Harris)	G	6	7	15	0	0	28	19	19
Melanoplus devastator Scudder	M	9	1	0	0	0	10	19	20
Melanoplus gladstoni Scudder	M	8	3	11	1	0	23	18	21
Oedaleonotus enigma (Scudder)	M	7	4	4	0	0	15	18	22
Dissosteira longipennis (Thomas)	O	8	2	3	0	0	13	18	23
Dissosteira carolina (Linnaeus)	O	3	11	18	0	0	32	17	24
Metator pardalinus (Saussure)	O	4	9	15	0	0	28	17	25
Eritettix simplex (Scudder)	G	7	3	15	0	0	25	17	26
Trimerotropis pallidipennis (Burmeister)	O	1	13	9	0	0	23	15	27
Xanthippus corallipes (Haldeman)	O	3	9	17	0	0	29	15	28
Cordillacris crenulata (Bruner)	G	4	7	11	0	0	22	15	29
Dissosteira spurcata Saussure	O	3	8	6	0	0	17	14	30
Boopedon nubilum (Say)	G	4	6	11	0	0	21	14	31
Aeropedellus clavatus (Thomas)	G	6	2	13	0	0	21	14	32
Melanoplus foedus Scudder	M	2	9	13	0	0	24	13	33
Encoptolophus costalis (Scudder)	O	5	3	7	0	0	15	13	34
Melanoplus angustipennis (Dodge)	M	4	4	12	0	0	20	12	35
Arphia pseudonietana (Thomas)	O	1	8	20	0	0	29	10	36
Schistocerca alutacea shoshone (Thomas)	C	2	6	3	0	0	11	10	37
Melanoplus rugglesi Gurney	M	5	0	3	0	0	8	10	38

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[1] S = serious, M = minor, I = innocuous, b? = possibly beneficial, B = beneficial.

[2] G = Gomphocerinae, M = Melanoplinae, O = Oedipodinae.

Table VI.6–4 —List of the 12 highest ranked “beneficial” grasshoppers on western rangeland (those listed all have scores below zero)

Grasshopper species	Sub-family[2]	Number of rankings[1]					Total	Score	Rank
		“S”	“M”	“I”	“b?”	“B”			
Hesperotettix viridis (Thomas)	M	0	2	17	5	5	29	–13	377
Hypochlora alba (Dodge)	M	0	0	13	2	1	16	–4	376
Hesperotettix curtipennis Scudder	M	0	0	1	0	1	2	–2	375
Aeoloplides chenopodii (Bruner)	M	0	0	3	0	1	4	–2	374
Poecilotettix pantherinus (Walker)	M	0	0	4	0	1	5	–2	373
Ligurotettix coquilletti McNeill	G	0	0	4	0	1	5	–2	372
Melanoplus herbaceus Bruner	M	0	0	5	0	1	6	–2	371
Anconia integra Scudder	O	0	0	5	0	1	6	–2	370
Bootettix argentatus Bruner	G	0	0	6	0	1	7	–2	369
Aeoloplus tenuipennis (Scudder)	M	0	0	7	0	1	8	–2	368
Tropidolophus formosus (Say)	O	0	0	9	0	1	10	–2	367
Melanoplus desultorius Rehn	M	1	0	1	1	1	4	–1	366

[1] S = serious, M = minor, I = innocuous, b? = possibly beneficial, B = beneficial.
[2] G = Gomphocerinae, M = Melanoplinae, O = Oedipodinae.

rangeland. This small number of “beneficial” grasshoppers, amounts to only 3 percent of the 377 species involved in this review, which is several orders of magnitude less than the recent estimate of 10 percent claimed by Lockwood (1993). The grasshopper most frequently called a beneficial is *Hesperotettix viridis*. Although often seen feeding on snakeweed, it also feeds on more than 30 other rangeland plants (Pfadt 1988). Another grasshopper, *Hypochlora alba,* is highly ranked as a beneficial because of its preference for sagebrush. But the value of sagebrush on rangeland is widely debated. As a strong competitor with desirable forage plants for domestic livestock, it is considered by some as an undesirable weed. Others consider sagebrush a beneficial plant because it comprises an important portion of the diet of mule deer, antelope, and the sage grouse (Watts et al. 1982).

Concerning the relative importance of the major pest grasshoppers, I believe that the rankings shown in table VI.6–3 represent a good concensus of opinions from the North American literature. Although experts differ over the ranking of individual species, most agree that there are about 2 dozen western grasshoppers that should be categorized as pests. I believe that a statement by Watts et al. (1989) summarized the pest issue quite well: “About a dozen species frequently occur in high densities, and . . . an additional 12 species occasionally occur in high densities.” Readers are free to compare their own opinions with the species listed and the pest-status rankings shown.

References Cited

Arnett, R. H. 1985. Orthoptera (grasshoppers, crickets, and katydids), Order 11. In: American insects, a handbook of the insects of America north of Mexico. New York: Van Nostrand Reinhold: 116–138.

Ball, E. D. 1936. Food plants of some Arizona grasshoppers. Journal of Economic Entomology 29: 679–684.

Ball, E. D.; Tinkham, E. R.; Flock, R.; Vorhies, C. T. 1942. The grasshoppers and other Orthoptera of Arizona. Tech. Bull. 93. Tucson, AZ: Arizona Agricultural Experiment Station. 373 p.

Banfill, J. C.; Brusven, M. A. 1973. Food habits and ecology of grasshoppers in the Seven Devils Mountains and Salmon River breaks of Idaho. Melanderia 12: 1–21.

Bird, R. D. 1961. Ecology of the aspen parkland of western Canada in relation to land use. Res. Branch Publ. 1066. Ottawa, ON: Canada Department of Agriculture: 89–92.

Brooks, A. R. 1958. Acridoidea of southern Alberta, Saskatchewan, and Manitoba (Orthoptera). Canadian Entomologist 90, Suppl. 9: 1–92.

Brusven, M. A. 1967. Differentiation, ecology and distribution of immature slant-faced grasshoppers (Acridinae) in Kansas. Tech. Bull. 1490. Manhattan, KS: Kansas Agricultural Experiment Station: 1–59.

Brusven, M. A. 1972. Differentiation and ecology of common Catantopinae and Cyrtacanthacridinae nymphs (Orthoptera: Acrididae) of Idaho and adjacent areas. Melanderia 9: 1–31.

Brusven, M. A.; Lambley, J. D. 1971. Part I. The food habits and ecology of grasshoppers from southern Idaho rangeland. Coop. Proj. Rep. 12-14-100-9726 (33). Moscow, ID: University of Idaho and U.S. Department of Agriculture. 85 p., 46 tables.

Buckell, E. R. 1936a. Part 1. The influence of man on the distribution of grasshoppers in Canada. In: Proceedings of the 4th international locust conference, anti-locust research; March, 1936; Cairo, Egypt. [Place of publication and publisher unknown.] Appendix 12: 1–7.

Buckell, E. R. 1936b. Part 3. Summary of losses and expenditures due to grasshoppers in Canada 1925–1934. In: Proceedings of the 4th international locust conference, anti-locust research; March, 1936; Cairo, Egypt. [Place of publication and publisher unknown.] Appendix 1: 1–13.

Capinera, J. L. 1987. Population ecology of rangeland grasshoppers. In: Capinera, J. L., ed. Integrated pest management on rangeland: a shortgrass prairie perspective. Boulder, CO: Westview Press: 162–182.

Capinera, J. L.; Sechrist, T. S. 1982. Grasshoppers (Acrididae) of Colorado: identification, biology and management. Bull. 584S. Fort Collins, CO: Colorado State University and Colorado Agricultural Experiment Station. 161 p.

Capinera, J. L.; Thompson, D. C. 1987. Dynamics and structure of grasshopper assemblages in shortgrass prairie. Canadian Entomologist 119: 567–575.

Coppock, S. 1962. The grasshoppers of Oklahoma (Orthoptera: Acrididae). Processed Ser. P-399. Stillwater, OK: Oklahoma Agricultural Experiment Station. 143 p.

Ewen, A. B., Mukerji, M. K. 1984. *Melanoplus* spp., *Camnula pellucida* (Scudder), and other grasshoppers (Orthoptera: Acrididae). In: Kelleher, J. S.; Hulme, M. A., eds. Biological control programs against insects and weeds in Canada 1969–1980. Slough, UK: Commonwealth Agricultural Bureaux: 61–62.

Fielding, D. J.; Brusven, M. A. 1990. Historical analysis of grasshopper (Orthoptera: Acrididae) population responses to climate in southern Idaho. Environmental Entomology 19: 1786–1791.

Fielding, D. J.; Brusven, M. A. 1992. Food and habitat preferences of *Melanoplus sanguinipes* and *Aulocara elliotti* (Orthoptera: Acrididae) disturbed rangeland in southern Idaho. Journal of Economic Entomology 85: 783–788.

Gibson, A. 1938. Report on the grasshopper situation and organization for grasshopper control in Canada, Rep. 4. In: Proceedings of the 5th international locust conference, anti-locust research; [dates of meeting unknown]; Brussels, Belgium. [Place of publication and publisher unknown]: 10–107.

Hagen, A. F. 1970. An annotated list of grasshoppers (Orthoptera, Acrididae) from the eleven panhandle counties of Nebraska. Res. Bull. 238. Lincoln, NE: Nebraska Agricultural Experiment Station. 60 p.

Handford, R. H. 1946. The identification of nymphs of the genus *Melanoplus* of Manitoba and adjacent areas. Scientific Agriculture 26(4): 147–180.

Harper, R. W. 1952. Grasshoppers of economic importance in California. Sacramento, CA: California Department of Agriculture Bulletin 41(3): 153–175.

Hauke, H. A. 1953. An annotated list of the Orthoptera of Nebraska, part II, the Tettigidae and Acrididae. Lincoln, NE: Bulletin of the University of Nebraska State Museum 3(9): 1–79.

Hebard, M. 1936. Orthoptera of North Dakota. Tech. Bull. 284. Fargo, ND: North Dakota Agricultural Experiment Station. 69 p.

Hebard, M. 1938. An ecological survey of the Orthoptera of Oklahoma. Tech. Bull. 5. Stillwater, OK: Oklahoma Agricultural Experiment Station. 31 p.

Helfer, J. R. 1987. How to know the grasshoppers, crickets, cockroaches and their allies, 2d edition. New York: Dover. 363 p.

Henderson, W. W. 1924. A taxonomic and ecological study of the species of the subfamily Oedipodinae (Orthoptera—Acrididae) in Utah. Bull. 191. Logan, UT: Utah Agricultural Experiment Station. 150 p.

Henderson, W. W. 1931. Crickets and grasshoppers in Utah. Circ. 96. Logan, UT: Utah Agricultural Experiment Station. 38 p.

Hewitt, G. B. 1977. Review of forage losses caused by rangeland grasshoppers. Misc. Publ. 1348. Washington, DC: U.S. Department of Agriculture, Agricultural Research Service. 24 p.

Hewitt, G. B.; Barr, W. F. 1967. The banded-wing grasshoppers of Idaho (Orthoptera: Oedipodinae). Res. Bull. 72. Moscow, ID: University of Idaho and Idaho Agricultural Experiment Station. 64 p.

Hewitt, G. B.; Onsager, J. A. 1983. Control of grasshoppers on rangeland in the United States—a perspective. Journal of Range Management 36: 202–207.

Hewitt, G. B.; Huddleston, E. W.; Lavigne, R. J.; Ueckert, D. N.; Watts. J. G. 1974. Rangeland entomology. Range Sci. Ser. 2. Denver, CO: Society for Range Management. 99 p.

Isely, F. B. 1938. The relations of Texas Acrididae to plants and soils. Ecolological Monographs 8(4): 551–604.

Kemp, W. P.; Dennis, B. 1991. Toward a general model of rangeland grasshopper (Orthoptera: Acrididae) phenology in the steppe region of Montana. Environmental Entomology 20: 1504–1515.

Kemp, W. P.; Onsager, J. A. 1986. Rangeland grasshoppers (Orthoptera: Acrididae): modeling phenology of natural populations of six species. Environmental Entomology 15: 924–930.

Kevan, D.K.McE. 1979. Orthoptera (s. str.). In: Danks, H. V., ed. Canada and its insect fauna. Memoirs Entomol. Soc. Can. 108. Ottawa, ON: Entomological Society of Canada: 321–323.

Knowlton, G. F.; Janes, M. F. 1932. The 1931 grasshopper outbreak in Utah. Proceedings of the Utah Academy of Science 9: 105–108.

La Rivers, Ira. 1948. A synopsis of Nevada Orthoptera. American Midland Naturalist 39(3): 652–720.

Lockwood, J. A. 1993. Environmental issues involved in biological control of rangeland grasshoppers (Orthoptera: Acrididae) with exotic agents. Environmental Entomology 22: 503–518.

Middlekauff, W. W. 1958. Biology and ecology of several species of California rangeland grasshoppers (Orthoptera: Acrididae). Pan-Pacific Entomologist 34: 1–11.

Mitchener, A. V. 1953. A history of grasshopper outbreaks and their control in Manitoba, 1799–1953. Ann. Rep. 84. Ottawa, ON: Entomological Society of Ontario: 27–35.

Mulkern, G. B. 1980. Population fluctuations and competitive relationships grasshopper species (Orthoptera: Acrididae). Transactions of the American Entomological Society 106: 1–41.

Mulkern, G. B; Anderson, J. F.; Brusven, M. A. 1962. Biology and ecology of North Dakota grasshoppers. I. Food habits and preferences of grasshoppers associated with alfalfa fields. Res. Rep. 7. Fargo, ND: North Dakota Agricultural Experiment Station. 26 p.

Mulkern, G. B.; Toczek, D. R.; Brusven, M. A. 1964. Biology and ecology of North Dakota grasshoppers. II. Food habits and preferences of grasshoppers associated with the sandhills prairie. Res. Rep. 11. Fargo, ND: North Dakota Agricultural Experiment Station. 59 p.

Mulkern, G. B.; Pruess, K. P.; Knutson, H.; Hagen, A. F.; Campbell, J. B.; Lambley, J. D. 1969. Food habits and preferences of grassland grasshoppers of the North Central Great Plains. Bull. 481. Fargo, ND: North Dakota Agricultural Experiment Station. 32 p.

Nerney, N. J. 1960. Grasshopper damage on short-grass rangeland of the San Carlos Apache Indian Reservation, Arizona. Journal of Economic Entomology 53: 640–646.

Nerney, N. J. 1961. Effects of seasonal rainfall on range condition and grasshopper population, San Carlos Apache Indian Reservation, Arizona. Journal of Economic Entomology 54: 382–385.

Nerney, N. J.; Hamilton, A. G. 1969. Effects of rainfall on range forage and populations of grasshoppers, San Carlos Apache Indian Reservation, Arizona. Journal of Economic Entomology 62: 329–333.

Newton, R. C.; Esselbaugh, C. O.; York, G. T.; Prescott, H. W. 1954. Seasonal development of range grasshoppers as related to control. Rep. E-873. Billings, MT: U.S. Department of Agriculture, Agricultural Research Service, Bureau of Entomology and Plant Quarantine. 18 p.

Otte, D. 1981. The North American grasshoppers. Vol. I. Acrididae: Gomphocerinae and Acridinae. Cambridge, MA: Harvard University Press. 275 p.

Otte, D. 1984. The North American grasshoppers. Vol. II. Acrididae: Oedipodinae. Cambridge, MA: Harvard University Press. 366 p.

Parker, J. R. 1952. Grasshoppers. In: Insects, the yearbook of agriculture 1952. Washington, DC: U.S. Department of Agriculture: 595–605.

Parker, J. R. 1957. Grasshoppers, a new look at an ancient enemy. Farm. Bull. 2064. Washington, DC: U.S. Department of Agriculture. 40 p.

Parker, J. R.; Connin, R. V. 1964. Grasshoppers, their habits and damage. Agric. Inf. Bull. 287. Washington, DC: U.S. Department of Agriculture. 28 p.

Pfadt, R. E. 1949. Range grasshoppers as an economic factor in the production of livestock. Wyoming Range Management 7: 1–7.

Pfadt, R. E. 1977. Some aspects of the ecology of grasshopper populations inhabiting the shortgrass plains. In: Kulman, H. M.; Chiang, H. C., eds. Insect ecology. Tech. Bull. 310. St. Paul, MN: University of Minnesota and Minnesota Agricultural Experiment Station: 77–79.

Pfadt, R. E. 1982. Density and diversity of grasshoppers (Orthoptera: Acrididae) in an outbreak on Arizona rangeland. Environmental Entomology 11: 690–694.

Pfadt, R. E. 1984. Species richness, density, and diversity of grasshoppers (Orthoptera: Acrididae) in a habitat of the mixed grass prairie. Canadian Entomologist 116: 703–709.

Pfadt, R. E. 1988. Field guide to common western grasshoppers. Bull. 912. Laramie, WY: University of Wyoming and Wyoming Agricultural Experiment Station. [Species factsheets nos. 1–37, dated through Sept. 1993.]

Pfadt, R. E.; Hardy, D. M. 1987. A historical look at rangeland grasshoppers and the value of grasshopper control programs. In: Capinera, J. L., ed. Integrated pest management on rangeland: a shortgrass prairie perspective. Boulder, CO: Westview Press: 162–182.

Putnam, L. G. 1962. The damage potential of some grasshoppers (Orthoptera: Acrididae) of the native grasslands of British Columbia. Canadian Journal of Plant Science 42: 596–601.

Richman, D. B.; Lightfoot, D. C.; Sutherland, C. A.; Ferguson, D. J. 1993. A manual of the grasshoppers of New Mexico, Orthoptera: Acrididae and Romaleidae. Handbk. 7. Las Cruces, NM: New Mexico State University and New Mexico Cooperative Extension Service. 112 p.

Scoggan, A. C.; Brusven, M. A. 1972. Differentiation and ecology of common immature Gomphocerinae and Oedipodinae (Orthoptera: Acrididae) of Idaho and adjacent areas. Melanderia 8: 1–76.

Scoggan, A. C.; Brusven; M. A. 1973. Grasshopper—plant community associations in Idaho in relation to the natural and altered environment. Melanderia 12: 22–33.

Shewchuk, B. A.; Kerr, W. A. 1993. Returns to grasshopper control on rangelands in southern Alberta. Journal of Range Management 46(5): 458–462.

Shotwell, R. L. 1938a. Some problems of the annual grasshopper survey. Journal of Economic Entomology 31: 523–533.

Shotwell, R. L. 1938b. Species and distribution of grasshoppers responsible for recent outbreaks. Journal of Economic Entomology 31: 602–610.

Shotwell, R. L. 1941. Life histories and habits of some grasshoppers of economic importance on the Great Plains. Tech. Bull. 774. Washington, DC: U.S. Department of Agriculture. 47 p.

Strohecker, H. F.; Middlekauff, W. W.; Rentz, D. C. 1968. The grasshoppers of California (Orthoptera: Acridoidea). Bull. 10. Berkeley, CA: California Insect Survey. 177 p.

Turnock, W. J. 1977. Adaptability and stability of insect pest populations in prairie agricultural ecosystems. In: Kulman, H. M.; Chiang, H. C., eds. Insect ecology. Tech. Bull. 310. St. Paul, MN: University of Minnesota and Minnesota Agricultural Experiment Station: 89–101.

Van Horn, D. H. 1972. Grasshopper population numbers and biomass dynamics on the Pawnee site from fall of 1968 through 1970. Tech. Rep. 148. Colorado Springs, CO: U.S. International Biological Program, Grassland Biome. 70 p.

Vickery, V. R.; Scudder, G.G.E. 1987. The Canadian orthopteroid insects summarized and updated, including a tabular check-list and ecological notes. Proceedings of the Entomological Society of Ontario 118: 25–45.

Wakeland, C. 1951. Changing problems and procedures in grasshopper and Mormon cricket control. Journal of Economic Entomology 44: 76–82.

Watts, J. G.; Hewitt, G. B.; Huddleston, E. W.; Kinzer, H. G.; Lavigne, R. J.; Ueckert, D. N. 1989. Rangeland entomology, 2d edition. Range Sci. Ser. 2. Denver, CO: Society for Range Management. 388 p.

Watts, J. G.; Huddleston, E. W.; Owens, J. C. 1982. Rangeland entomology. Annual Review of Entomology 27: 283–311.

White, R. M.; Rock, P.J.G. 1945. A contribution to the knowledge of the Acrididae of Alberta. Scientific Agriculture 25: 577–596.

Wilbur, D. A.; Fritz, R. F. 1940. Grasshopper populations (Orthoptera, Acrididae) of typical pastures in the bluestem region of Kansas. Journal of the Kansas Entomological Society 13: 86–100.

Woodruff, L. C. 1937. A grasshopper survey for eastern Kansas, 1936. Journal of the Kansas Entomological Society 10: 75–83.

VI.7 Hopper Helper

By Wendal Cushing

Preface

This reference was developed as a resource for personnel after years of observing them struggle to identify the life stages and species of grasshoppers while in the field. Although many resource tools are available, they often are too technical or too bulky to be used in survey operations.

Data for this reference were based on studies done in the Grasshopper Integrated Pest Management (GHIPM) Project demonstration area in McKenzie County, ND. Pocket Hopper Helper, which fits in a shirt pocket, provides necessary information about grasshoppers that will aid the user in identifying different species found in southwestern North Dakota and on western rangelands.

Acknowledgments

The production of Pocket Hopper Helper and Hopper Helper has entailed the efforts and expertise of many coworkers. I wish to acknowledge their valued contributions which made this publication possible.

In particular, I wish to thank three employees of the Animal and Plant Health Inspection Service's (APHIS) Plant Protection and Quarantine (PPQ) Phoenix Methods Development Center: Nelson Foster, for facilitating the production of this aid to be used in conjunction with factsheets for field identification of common grasshoppers; K. Chris Reuter, who provided assistance with identification characters of immature and adult grasshoppers and review of the manuscript; and Lonnie Black, who prepared final drawings from my originals and representative specimens of individual species.

Introduction

Hopper Helper provides field personnel with an easy-to-use guide for survey operations. Data gained through direct observation in field operations in southwestern North Dakota provided the basis for this guide. Please observe the following seven additional facts in applying this field guide:

1. The data in the Seasonal Life History Chart (see next chapter) are based on each instar stage, which lasts about 7 days. In other words, it takes about 35 days, from the day it hatches, for the average grasshopper to become an adult. Changing weather conditions can lengthen or shorten this process.

2. When applying the Seasonal Life History Chart to your operation, for every 100 miles south of latitude 47°46'N (Watford City, ND), instar stages will be ahead of schedule by about 7 days (one instar stage).

3. To improve readability, words and symbols used to represent approximate size are defined as:
   Small = approximately 11 mm.
   Average = approximately 22 mm.
   Large = approximately 33 mm.
   Robust = approximately 44 mm.

   G, M, and F indicate preferred food sources for grasshoppers. A “G” appearing next to a grasshopper's name indicates the species' preferred food is grass. “M” stands for mixed food sources (grass and forbs). “F” stands for forbs.

   * = the particular characteristic mentioned is the primary identification characteristic of the grasshopper species.

4. For quick reference, all grasshopper species are numbered 1–44.

5. To make the most effective use of this guide, become familiar with the external morphological structures (physical characteristics) most often used in identification.

6. To make full use of the color description in this outline, use fresh specimens when possible.

7. Have available a copy of Robert Pfadt's “Field Guide to Common Western Grasshoppers.”

Physical Characteristics Used To Identify Grasshoppers

The following drawings are useful in pinpointing physical characteristics (morphology) of nymphal and adult grasshoppers. Learning the morphology of grasshoppers will speed identification in the field.

Figure A—Lateral view of an adult female.

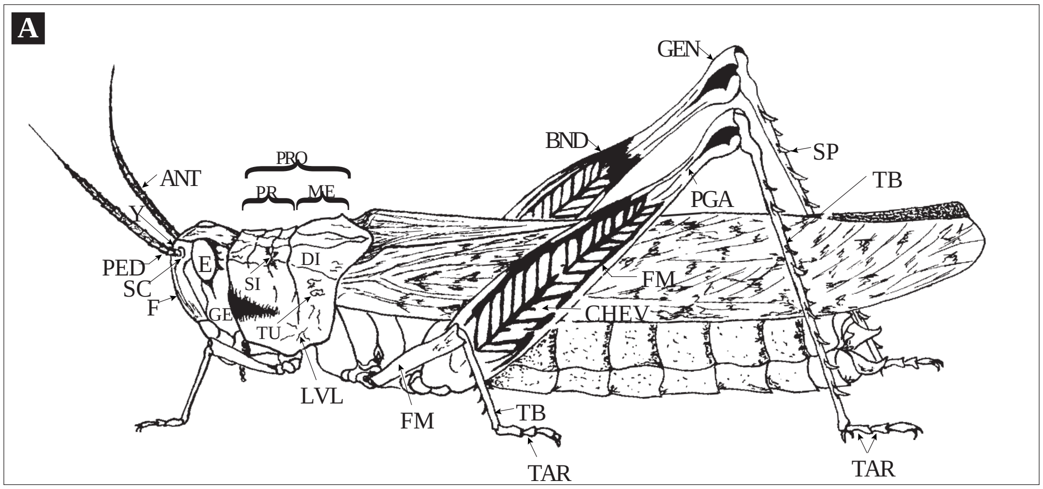


Figure B—Anterior view of head of adult female.

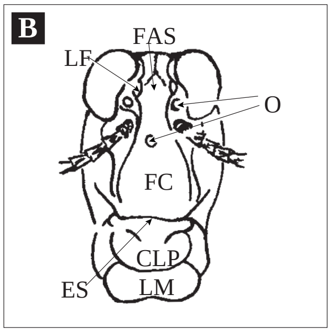
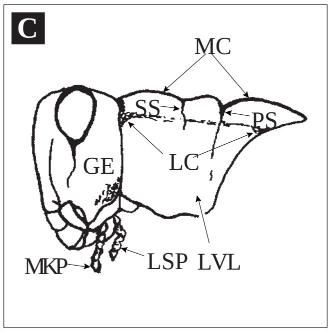


Figure C—Lateral view of head and pronotum of adult female.

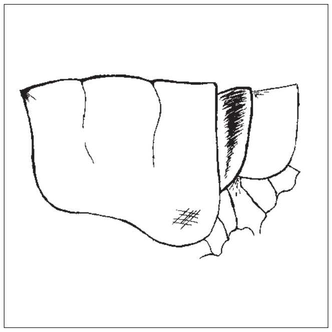


*ANT	Antenna
*BND	Band
CHEV	Chevrons
CLP	Clypeus
DI	Disk of pronotum
*E	Compound eye
ES	Epistomal suture
*F	Frons
FAS	Fastigium
*FC	Frontal costa
*FM	Femur
*GE	Gena
GEN	Genicular area
LSP	Labial palpus
LM	Labrum
*LC	Lateral carina
*LF	Lateral foveolae
*LVL	Lateral ventral lobe of pronotum
*MC	Median carina
*ME	Metazona
MKP	Maxillary palpus
O	Ocelli
PED	Pedicel
PGA	Pregenicular area
*PR	Prozona
*PRO	Pronotum
*PS	Primary sulcus
SC	Scape
SCU	Scutellum
Si	Sinus
SP	Spines
SS	Secondary sulcus
TAR	Tarsus
*TB	Tibia
*TU	Tubercule
V	Vertex

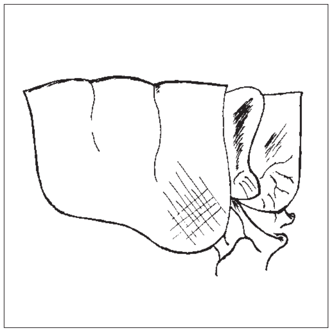
* = characteristics most used in identification.

Key to Normal Nymphal Instars

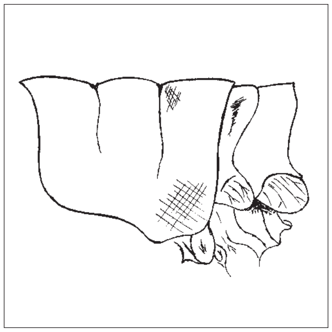
(From Handford 1946)



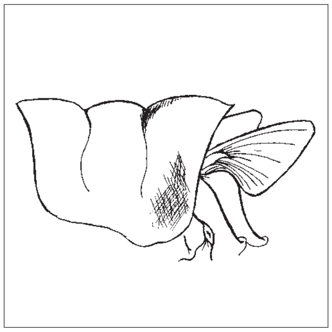
Wing pads rounded with no visible bulge at apex ..... first instar



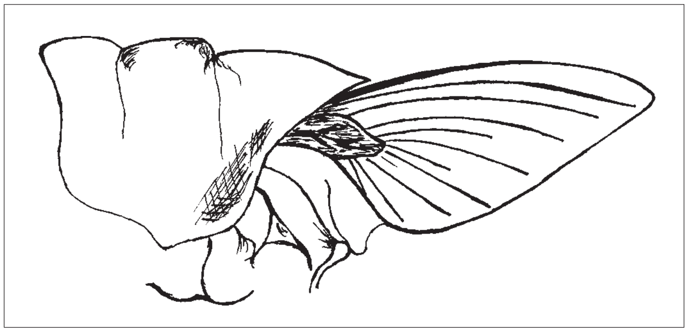
Wing pads rounded with visible bulge at apex ..... second instar



Wing pads more sharply triangular and showing slight venation ..... third instar



Wing pads short, not extending beyond first abdominal segment, more truncated ..... fourth instar



Wing pads elongated, extending beyond the second but hardly beyond the third abdominal segment, more pointed at the apex ..... fifth instar

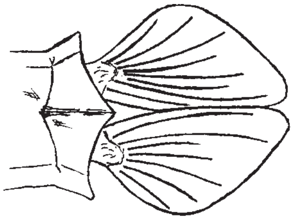
Several of the adult grasshoppers possess wings that are not of the typical form and are sometimes confused with the wing pads of immatures. Examples of some short-winged species are shown below.

Figure 1—Immature wing pads.

Figure 2—
Hypochlora alba
Melanoplus dawsoni
Phoetaliotes nebrascensis
Both sexes

Figure 3—*Aeropedellus clavatus*
Females only

1



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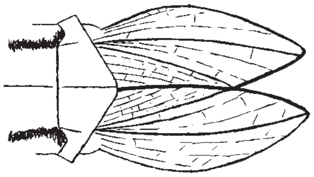

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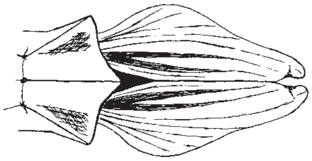
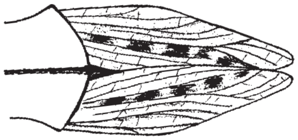


Figure 4—*Boopedon nubilum*
Females only

Figure 5—*Pseudopomala brachyptera*
Both sexes

Figure 6—*Chorthippus curtipennis*
Females only

4



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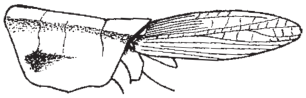

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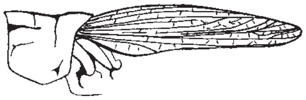
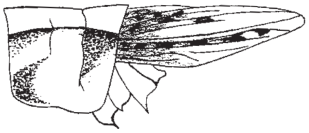


Figure 7—*Chloealtis conspersa*
Females only

Figure 8—*Oedaleonotus enigma*
Both short- and long-winged forms are common in both sexes.

7

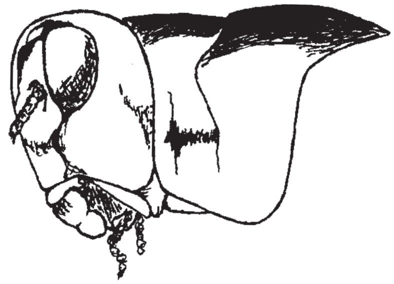


8

Overwintering Species

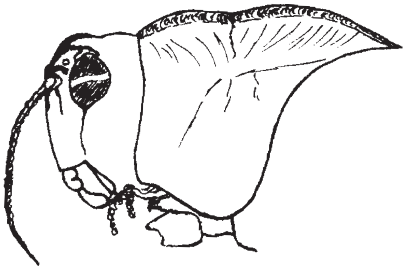
(To be adults at spring greenup.)

Arphia conspersa *1–G*



Adult: A large brown grasshopper with red or yellow wings. Lower abdomen and hind tibia yellowish. This species often will flush before you get close enough to catch them in a net.
Immature: Usually dark brown and having many of the adult morphological characteristics, *two light bands on inner face of femur.

Chortophaga viridifasciata *2–G*



Adult: A large grasshopper with smoke-colored wings, greenish-yellow at base. Color usually green, antennae red with the pronotum slightly arched. *A visible band through the compound eye.
Immature: Body color may range from green to brown speckled with white, but the median carina is always high and sharp. First instars usually appear near mid-July.

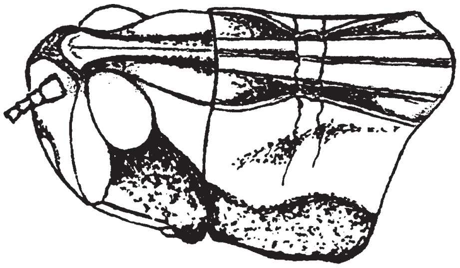
Pardalophora haldemanii *3–G*

Adult: A large, robust grasshopper with one sulcus cutting the pronotum. *Inner surface of the hind femora usually a greenish yellow. Dark spots on forewing, rough pronotum.
Immature: Later instars are large with one sulcus cutting the pronotum. Very similar to *Xanthippus*, can have two sulci on pronotum.

Xanthippus corallipes *4–G*

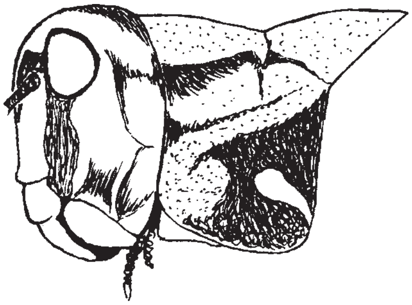
Adult: A large, robust grasshopper with *two sulci cutting the pronotum. Inner surface of the hind femora and tibiae a bright reddish pink. Dark spots on forewing, rough pronotum.
Immature: Overwinter in the later instar stages. *Usually dark blue on inner femur in first four instars, becoming more reddish pink instars five and six. A slight “X” is sometimes visible on the dorsal area of the pronotum. First instars appear in early July.

Eritettix simplex *5–G*

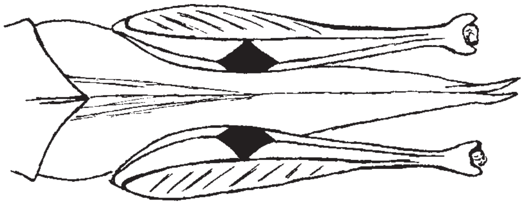


Adult: An average-sized grasshopper. Colors range from a bright green to a light tan. Adults normally begin to appear in early May. *Adults and immatures share tricarinate feature on head and pronotum.
Immature: Apparently overwinter in the fourth and fifth instar stage and can be found from fall to early spring. First instars usually appear around the first week of July.

Psoloessa delicatula *6–G*



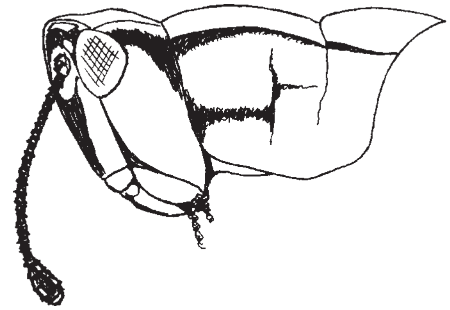
Adult: A small, drab grasshopper with a *diamond visible on the hind femora. Posterior dorsal area of pronotum very flat. Lateral carinae strongly constricted in the middle for immatures and adults.



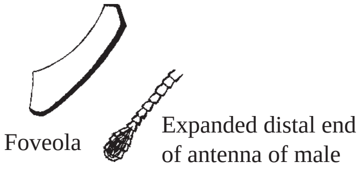
Immature: Color somewhat darker than *Eritettix* sp. with an evident white mark on the pronotum. Face not as slanted as *Eritettix* sp. First instars usually appear around the first week of July. Diamond on hind femora often visible in immatures.

Aeropedellus clavatus *7–G*

Adult: Females have short wings, white cheeks, and a line ahead of the eye. The drawing shows an early summer adult. The lateral carinae constrict near the middle.



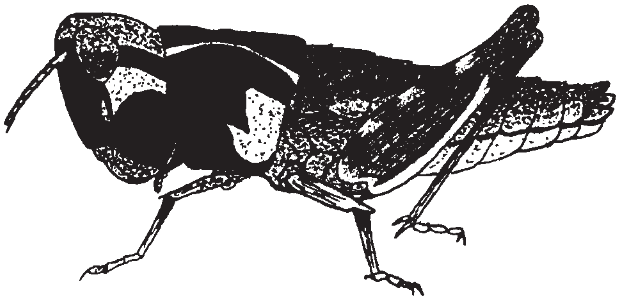
Immature: Lateral foveolae evident in all instars. First instars usually appear by the first week of June.



Ageneotettix deorum *8–G*

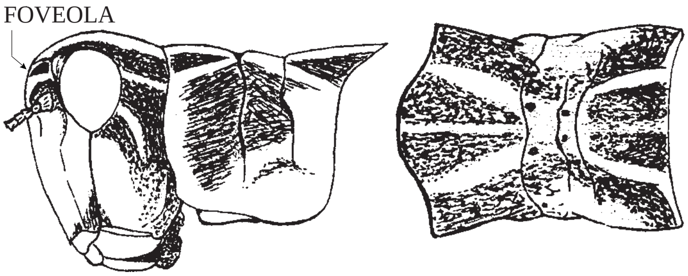
Adult: *Face usually dark, body color speckled, knee black with an orange tibia. Dorsal pronotum with an hourglass shape. *Whitish antennae while grasshopper is alive. Foveolae appear almost square. Inner hind tarsal claw unusually long.

Immature: Face usually dark with lateral foveolae evident. First instars usually appear by mid-May.

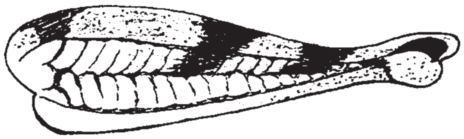


Aulocara elliotti *9–G*

Adult: *Banding of the inner surface of hind femora and “X” mark on the top of the pronotum. Lateral foveolae usually teardrop shaped or triangular.



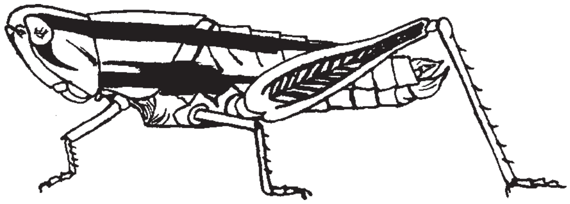
Immature: Banding of the inner surface femora. Lateral foveolae evident. First instars usually appear by the second week in May.



Amphitornus coloradus *10–G*

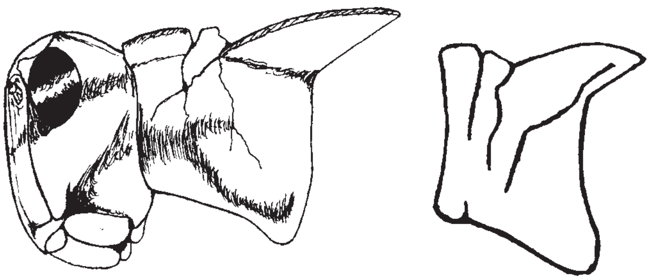
Adult: *Pair of brown stripes running from the head to the end of the pronotum. Hind femora with very visible bands on the outer surface and having a blue tibia.

Immature: A small version of the adult. First instars normally appear by mid-May.



Trachyrhachys kiowa *11–G*

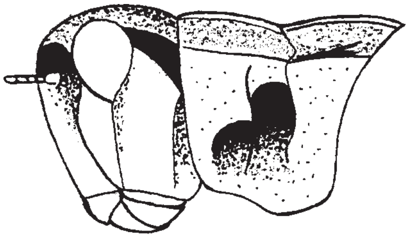
Adult: *A small- to medium-sized grasshopper with bands on the forewing. Banding on the inner surface of femora and having a blue tibia. *Rough pronotum with a lateral ventral flange.



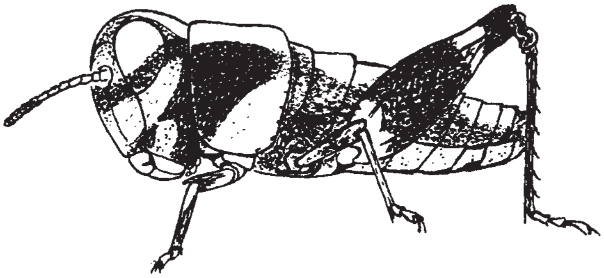
Immature: Body size small and stout. Pronotum rough and the lower hind femora is hirsute (hairy). First instars normally appear by late May.

Camnula pellucida *12-G*

Adult: Both sexes a straw yellow. Lateral carina continuous to posterior end of the pronotum. Spotted forewing and clear hindwings. *Population usually found in hatching beds, hay yards, etc. *Continuous lateral carina.



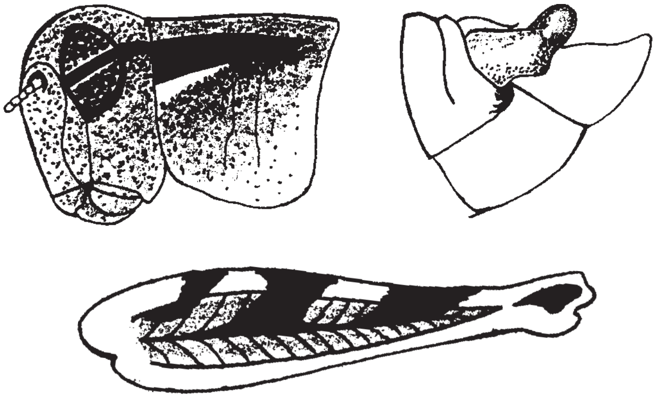
Immature: First instars distinctive with a tan saddle. All later instars have a tan color. First instars normally appear by mid-May.



Problem *Melanoplus* Species

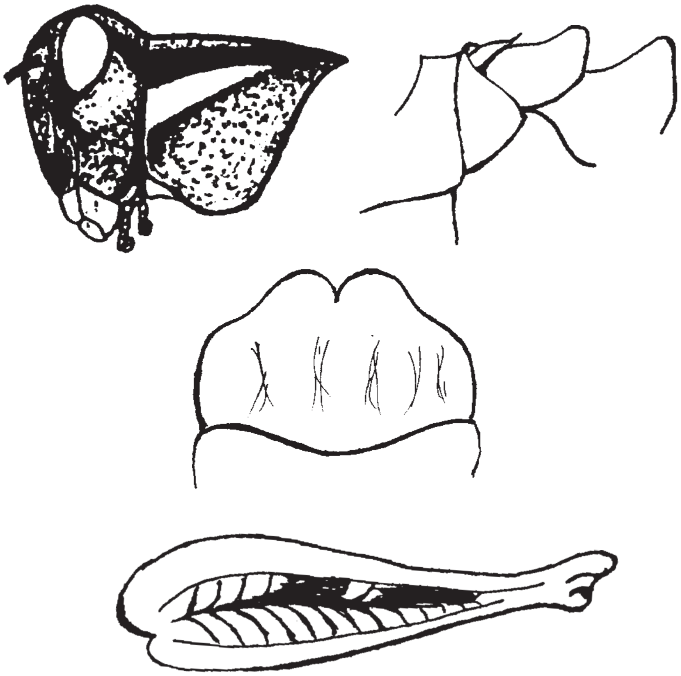
Melanoplus confusus *13-G*

Adult: *Side of pronotum with a patent leather shine and a definite line through the eye.
Immature: *Diagonal dark stripe bordered by narrow light lines through the eye. Cercus evident in later instars. First instars usually appear by early May.



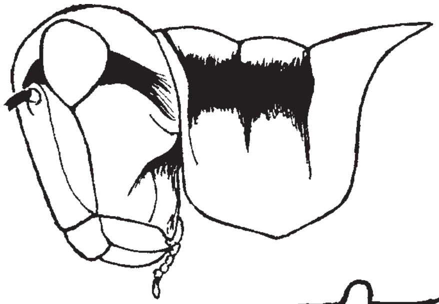
Melanoplus sanguinipes *14-F*

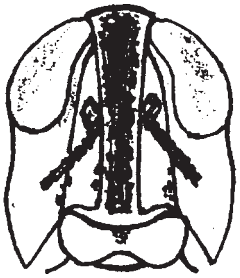
Adult: *Distinctive hump between the second pair of legs in males. The male subgenital plate distinctive.
Immature: First instars usually appear in late May, about 2 weeks later than *M. confusus.* *Early instars have speckled appearance.

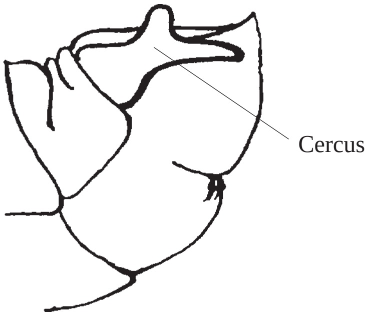


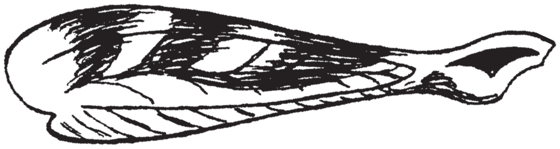
Melanoplus infantilis *15–G*

Adult: *Size small with a beelike striping on the abdomen. *Frontal costa dark, sometimes with spots along the margins. The cheek area is usually cream-colored. Most are adults by the end of June. Cercus boot shaped.
Immature: First instars usually appear by mid-May.









Melanoplus gladstoni *16–M*

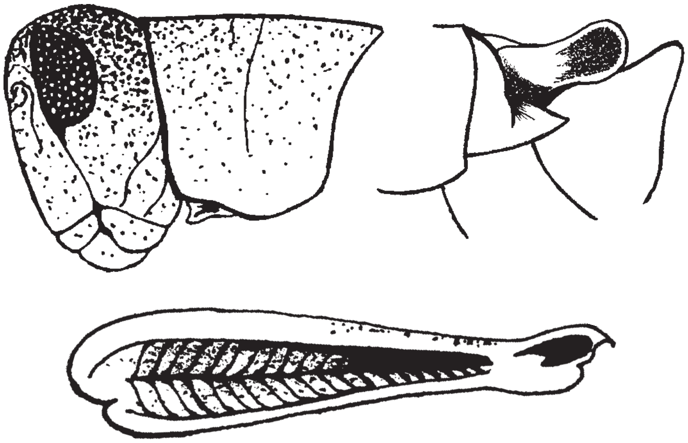
Adult: *Hind femora banding. *Hind femora flattened below base.
Immature: Look much like *M. infantilis* except *gladstoni* are usually adults by the end of June. This species lacks the frontal costal spots but has a very “dark” clypeus.

Dorsal view

Outer face

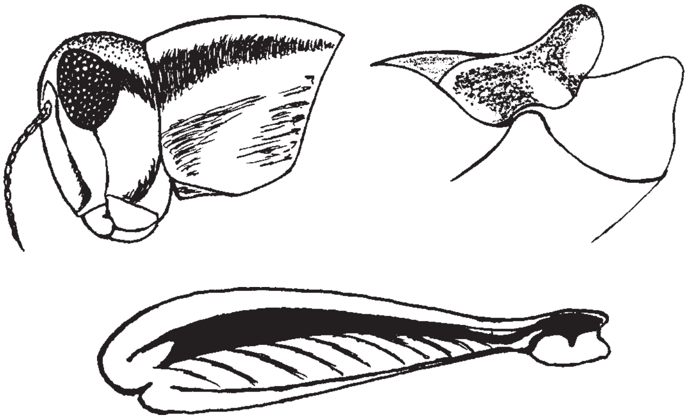
Melanoplus packardii *17–M*

Adult: Most resemble *M. bivittatus* but are smaller. *Two light stripes down the pronotum.
Immature: *Generally tan or green and covered with brown spots over the whole body.



Melanoplus bivittatus *18–M*

Adult: *Compound eye uniformly spotted. *Two clear yellow stripes from the head to the wing tips. Size large. Color usually an olive green with yellow.
Immature: *Bright green or tan is the general body color. The definite black band on the femur and large size usually aid in this species' identification. First instars usually appear by mid-May.



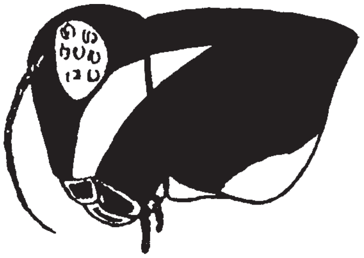
Melanoplus femurrubrum *19–M*

Adult: *Black band on outer face of femur. A pronounced crest and usually a large cream-colored cheek. Strongly contrasting black and white color is similar to *M. dawsoni*. *Underside of abdomen and inner surface of femur bright yellow with red tibia. Tip of male abdomen swollen.
Immature: First instars usually appear by early June.

Melanoplus dawsoni *20–M*

Adult: *General body color a shiny patent leather look.

Compound eye with up to 10 white spots. *Both sexes usually have reduced wings. See fig. 2 on p. 4, description of wings. Underside bright yellow.
Immature: First instars usually appear by early July.



Melanoplus keeleri *21–G*

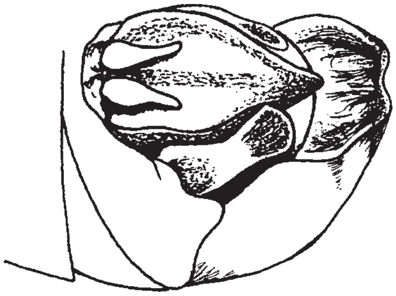
Adult: Hind femora yellow below. Hind tibia red with a black spot or band at its base.
Immature: *Two distinct white lines running parallel through the compound eye. *Large cream-colored area covers the cheek and extends to cover the whole side of the pronotum (pattern may vary). First instars usually appear by mid-June.

Intermediate-Hatching Species

Melanoplus angustipennis *22-G*

Adult: Markings inconspicuous. It may look much like the *M. sanguinipes* male except for the cercus and furcula. *This species is associated with sandy or “blow out” (windswept) land. No noticeable femoral markings. Cercus spoon shaped.

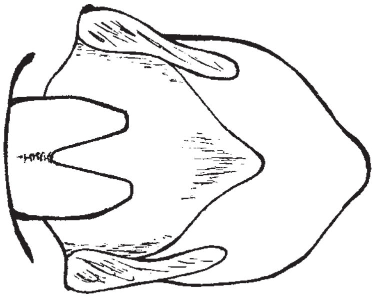
Immature: Tan or green with fine brown spots over most of the body. No banding evident on the outer femur.



Melanoplus bowditchi *23–F*

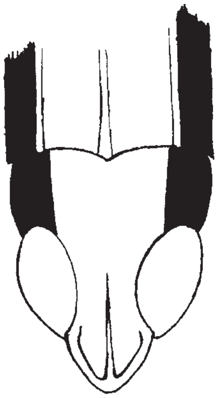
Adult: Markings inconspicuous. Body color usually a brownish olive with a spattering of brown. *Associated with sagebrush or near the base of steep eroded banks. *No noticeable femoral markings.

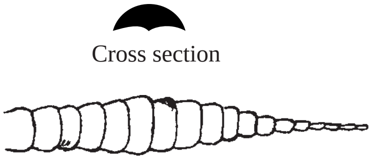
Immature: Pale gray with dark markings and generally a speckled appearance.



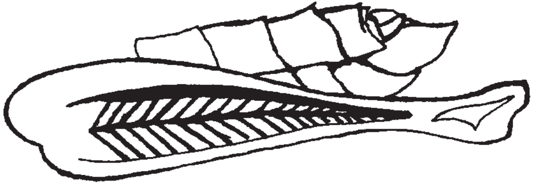
Opeia obscura *24–G*

Adult: Females larger than males. Size small to average. Parallel lateral carina evident. Forewing usually with some green. Forewing with a dark longitudinal stripe. Below the stripe there is a white line in the marginal field. Antennae triangular in cross section, swordshaped (ensiform).





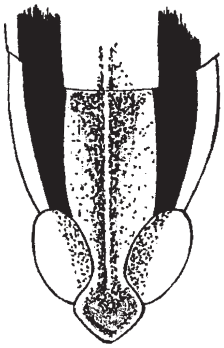
Immature: Resembles *Amphitornus* sp. except without external bands on the hind femora, and does not have brown stripes above eyes. *Hind femora long.

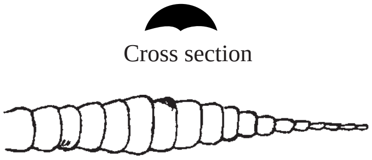


Mermiria bivittata *25–G*

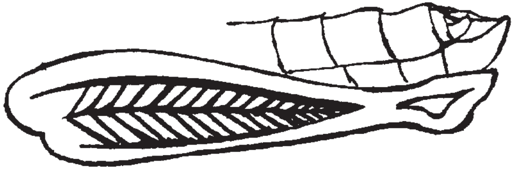
Adult: *Body yellow to greenish. Yellow underneath.

Size large. *No lateral carina evident. Brown stripes behind eye and onto the pronotum. Strongly slanted face. *Depression of vertex without a median carina. Associated with tall, coarse grass.



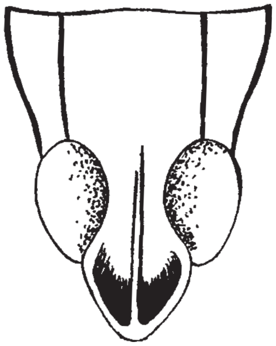


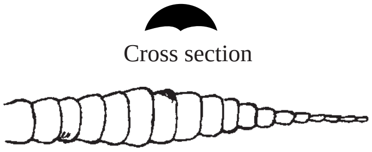
Immature: Quite large and generally green or tan. Fine brown spots cover the body. Antennae triangular in cross section, swordshaped.



Pseudopomala brachyptera *26–G*

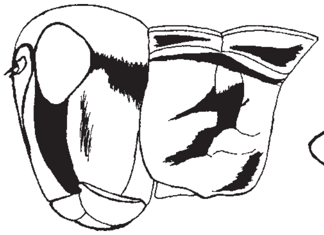
Adult: *Abdomen extending beyond the hind femora in adults. Size large. Lateral carina well developed. Body color light brown. Fastigium divided by a median carina. Both sexes short winged. Antennae triangular in cross section, swordshaped.

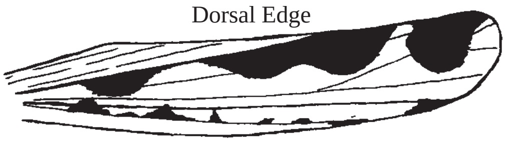




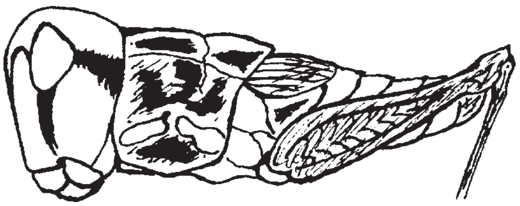
Phlibostroma quadrimaculatum *27–G*

Adult: *Forewing with four spots. Tibia reddish orange. Color brownish olive with some green. Size: Females large, males small. Distinct constricted lateral carinae, vertical white stripe below eye.



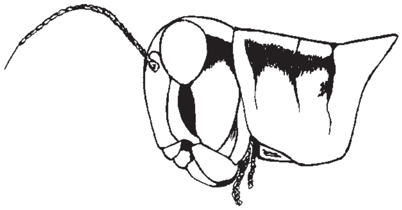


Immature: *Usually a lateral carina and some green color. Hind femora a light brown. No noticeable banding. *Two white areas are usually visible on the lower pronotum.

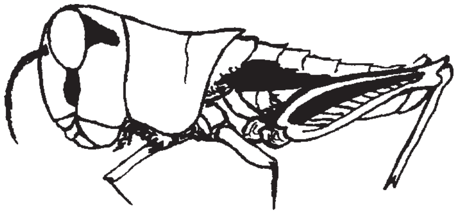


Phoetaliotes nebrascensis *28–M*

Adult: *Both sexes usually with reduced forewing. (See fig. 2, description of wings.) *Head larger than pronotum. Black teardrop below compound eye.

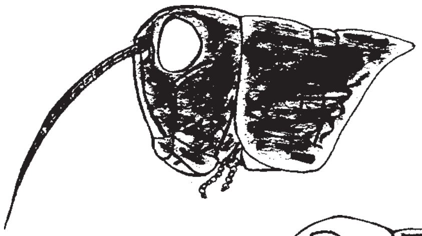


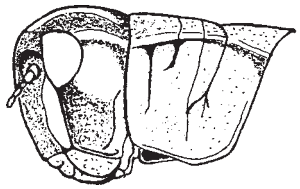
Immature: *No visible lateral carina. *The hind femora with noticeable band on the upper half. Immatures appear to be soft and delicate.



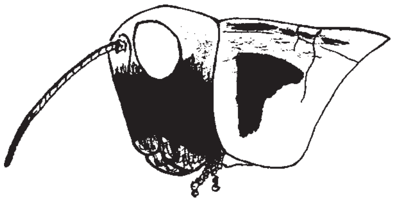
Boopedon nubilum *29–G*

Adult: Males are jet black and with fully developed wings. Females are large and have an olive green and brown color and short wings.



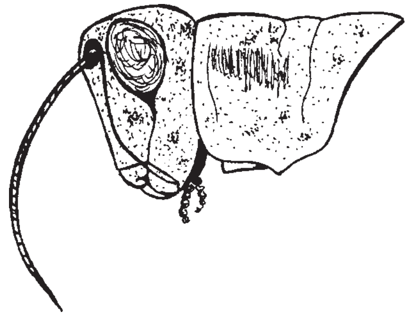


Immature: Pronotum is very distinctive with a dark saddle area.



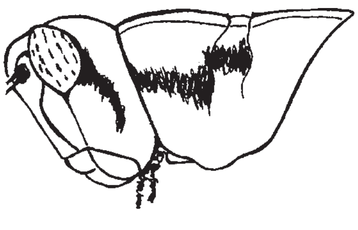
Hypochlora alba *30–F*

Adult: *Both sexes with pointed, reduced forewing. (See fig. 2, description of wings.) *Color a sage-gray green that resembles the host plant (Mulkern et al. 1969). *The entire body is covered with small rust-colored dots.
Immature: A small version of the adult.



Hesperotettix viridis *31–F*

Adult: Pronotum green with a pale white middorsal stripe. *A reddish orange band around the femur near the knee. Compound eye with vertical rows of spots.
Immature: Compound eyes with light spots. Antennae dark with light colored rings. A light-colored line running from the head to the posterior tip of the pronotum. In later instars, hind femoral chevrons are dark.

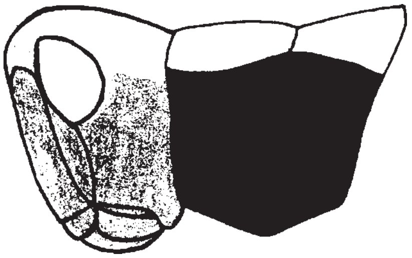


Late-Hatching Species

(To be adults by late summer.)

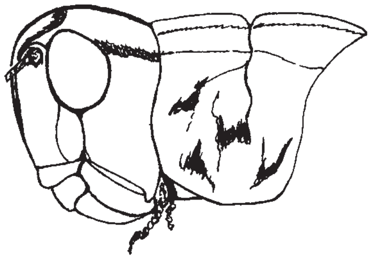
Chloealtis conspersa *32–G*

Adult: Lateral pronotal area of male entirely black. Female with reduced wings. (See fig. 7, p. 4, description of wings.) Sides of female pronotum lighter colored. Black knee in both sexes.

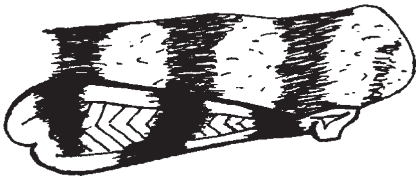


Encoptolophus costalis *33–M*

Adult: Corresponding bands on forewing and femur. A small late bandwing. Inner surface of hind femora dark bluish-black on the basal half and with a dark band toward the apex.

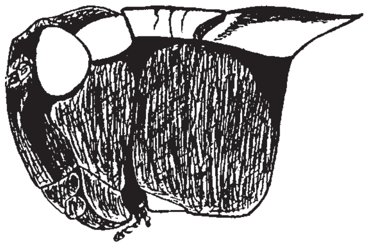


Immature: Similar to *Chortophaga* sp. in color and morphology, but this species is in an advanced instar stage when *Chortophaga* hatches.



Arphia pseudonietana *34–M*

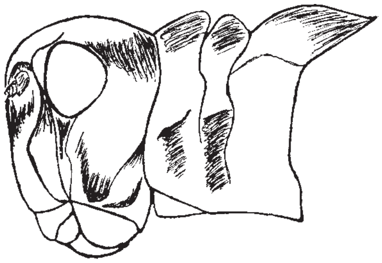
Adult: A late-season adult bandwing. Color bronze, almost black. Color varies from grayish-brown to black, mottled appearance. Usually a red wing disk with a black band.



Immature: This species is usually at least two instars ahead of *Arphia conspersa* near the middle of July.

Metator pardalinus *35–M*

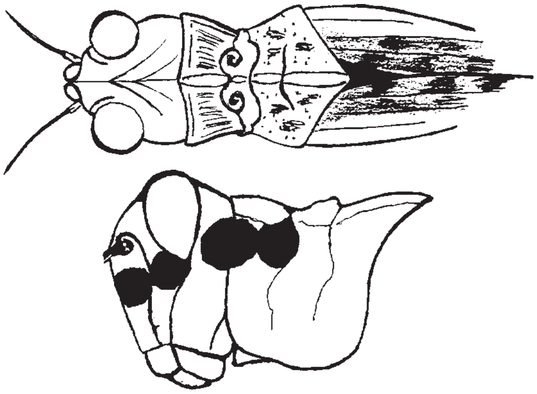
Adult: A large bandwing grasshopper. Females are almost robust. Males are smaller and have dark blue abdomen, tibia, and inner femur. Dark spots on forewing.
Immature: Early instars resemble *Trachyrhachys*, but this species does not have any dense hair on the femora.



Derotmema haydeni *36–M*

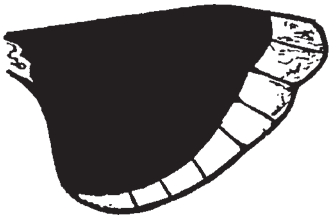
Adult: A small- to medium-sized bandwing grasshopper with large, bulbous eyes and a very wrinkled pronotum and speckled spots quite evident in the forewing.

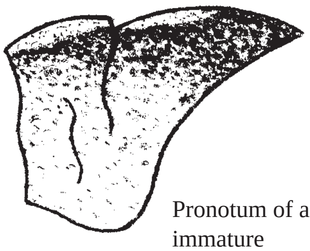
Immature: Early instars have four shiny black spots on the front of the head and two on the pronotum. All instars have two rust spots on each ventral abdominal segment.



Dissosteira carolina *37–M*

Adult: Adults are known as “road dusters.” The hind wing is black with a pale yellow border. This species has the largest wingspan of our grasshoppers. Mimics local soil coloration.
Immature: Early instars possess a morphology much like *Arphia* sp. except the body color is like wet beach sand. Later *Dissosteira* instars are much larger, and the pronotum is shaped like a buffalo’s hump.

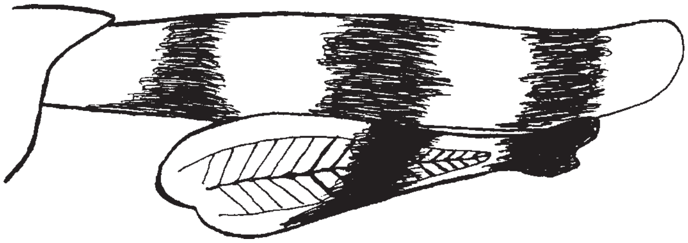




Pronotum of an immature

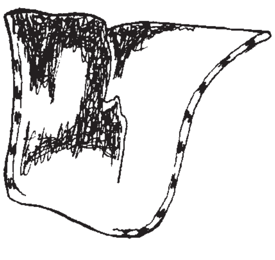
Hadrotettix trifasciatus *38–M*

Adult: *Forewing reddish-brown with conspicuous dark crossbands; apex clear. *Inner surface of hind femora a deep blue color on the basal two-thirds followed by a white band and a dark apex. Hind tibia orange.
Immature: All later instars exhibit the above femoral coloration. Stout appearance.



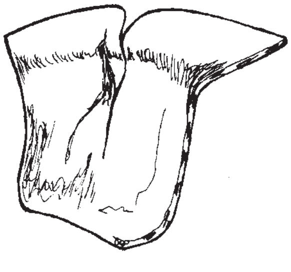
Spharagemon equale *39–M*

Adult: *General body color is a speckled, sandy look with a bright orange inner femora and tibia. *Pronotum with the median carina slightly elevated, usually cut once. Forewing banded.
Immature: *All later instars exhibit the basic adult coloration. On first instars, hind tibia dark.



Spharagemon collare *40–G*

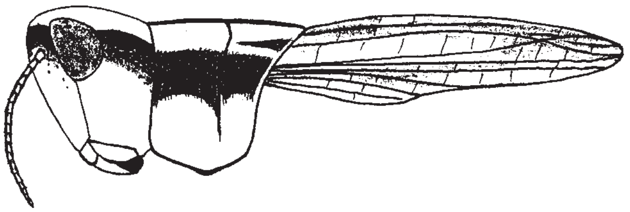
Adult: *General body color is a speckled, sandy look with a yellowish femora and orange tibia. *Pronotum with the median carina raised into a high crest and cut deeply by one sulcus. Forewing not noticeably banded.
Immature: *All later instars exhibit the basic adult coloration. On first instars, hind tibia dark.



Chorthippus curtipennis *41–G*

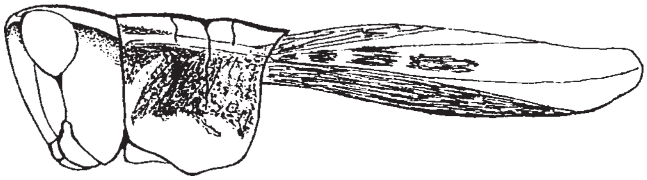
Adult: Body color usually varies from green to a yellowish brown with the ventral portion yellowish. The hind femora of the males have a black knee and are longer

than the abdomen. The lateral foveolae are visible from above. Female wings short. Male wings reach end of abdomen.
Immature: Quite variable in body striping and color. First and second instars have distinct brown stripe from eye well onto the abdomen.



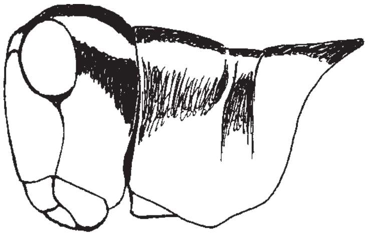
Orphulella speciosa *42–G*

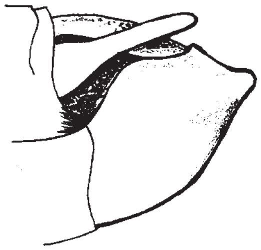
Adult: *Body color variable, greens and browns with a dark band extending from behind the compound eye to the pronotum. *A dark triangular area inside the rear portion of the lateral carina. Hind femora a brownish tan in color and longer than the abdomen in the males. *A visible depression on the point of the head. Lateral carinae of pronotum cut by one sulcus.
Immature: Possess many of the adult morphological characters.



Aeoloplides turnbulli *43–F*

Adult: *Stout body with a greenish yellow color. Body widest at the posterior end of the pronotum. Hind tibia blue. Male subgenital plate with a subapical tubercle. Distinctive stripe on head and pronotum. Outer femur distinctively marked with dark chevrons.





Oedaleonotus enigma *44–M*

Adult: Not found in North Dakota. *Found in California, Nevada, Utah, Idaho, Washington, and Oregon. The anterior edge of the pronotum has a conspicuous cream-colored band giving the appearance of wearing a clergyman's collar. The lower portion of the femora has a thin orange line. The cercus is drumstick shaped. An early hatching species in Idaho.
Immatures: Robust appearance. Distinctive white stripe on middle of pronotum, extends onto abdomen.

Selected References

Brooks, A. R. 1958. Acridoidea of southern Alberta, Saskatchewan and Manitoba (Orthoptera). Suppl. 9. Canadian Entomologist 90: 1–92.

Capinera, J. L., ed. 1987. Integrated pest management on rangeland, a shortgrass prairie perspective. Boulder, CO: Westview Press.

Handford. R. H. 1946. The identification of nymphs of the genus *Melanoplus* of Manitoba and adjacent areas. Scientific Agriculture 26: 147–180 and 12 plates.

Hewitt, G. B.; Barr, W. F. 1967. The banded-wing grasshoppers of Idaho. (Orthoptera: Oedipodinae). Sta. Bull. 72. Moscow, ID: Idaho Agricultural Experiment Station.

Mulkern, G. B.; Pruess, K. P.; Knutson, H.; Hagen, A. F.; Campbell, J. B.; Lanbley, J. D. 1969. Food habits and preferences of grassland grasshoppers of the north central Great Plains. Sta. Bull. 481. Fargo, ND: North Dakota Agricultural Experiment Station.

Newton, R. C.; Esselbaugh, C. O.; York, G. T.; Prescott, H. W. 1954. Seasonal development of range grasshoppers as related to control. Bull. E-873. Division of Cereal and Forest Insect Investigations: U.S. Department of Agriculture, Agricultural Research Service, Bureau of Entomology and Plant Quarantine. 18 p.

Pfadt, R. E. 1988. Field guide to common western grasshoppers. Sta. Bull. 912. Laramie, WY: U.S. Department of Agriculture, Animal and Plant Health Inspection Service, Wyoming Agricultural Experiment Station. 25 p.

Selected References—Unpublished

Cushing, W. J. 1970. Characteristics of the immature stages of North Dakota bandwinged grasshoppers with a key for their identification. M.S. thesis. Fargo, ND: North Dakota State University.

Turley, D. M. 1964. Acridinae nymphs of North Dakota. National Science Foundation final report.

VI.8 Seasonal Occurrence of Common Western North Dakota Grasshoppers

By W. J. Cushing, R. N. Foster, K. C. Reuter, and Dave Hirsch

Several authors have compiled excellent taxonomic keys for identifying various grasshopper groups in North America: slantfaced and bandwinged adults by Otte (1981), spurthroated adults by Brooks (1958), and the identification of nymphs of the genus *Melanoplus* by Hanford (1946). Others have used hatching dates and developmental charts to aid in grasshopper identification. For Wyoming and Montana, excellent examples are the charts developed by Newton (1954) and the charts modified for use in Colorado by Capinera (1981).

Many of the identification aids are not commonly available and are technical and difficult to use in a field situation because of bulk and terminology. Also, the field person attempting to use such identification aids usually is a temporary summer employee with little or no background in entomology.

Although scientists have computer mapping technology and sophisticated methods of conducting grasshopper surveys, grasshoppers still need to be identified at each survey stop. A small, easy-to-use reference such as this one will help in the identification process.

Used in combination, the seasonal occurrence chart (table 1) and the Pocket Hopper Helper can help a field person identify grasshopper species in the field. In a year with average grasshopper populations, a field person using the two aids in combination can identify an unknown grasshopper of known life stage (instar) in western North Dakota.

In 1987, the U.S. Department of Agriculture's (USDA) Animal and Plant Health Inspection Service (APHIS) funded a multiyear Grasshopper Integrated Pest Management (GHIPM) Project to investigate ways to control rangeland grasshoppers in the West. The GHIPM Project set up a study area in McKenzie County, ND, with a demonstration area and several study sites. At each treatment location, there were 10 or more treatment-evaluation sites. Approximately one-half mile outside the treatment areas, 10 untreated sites were also monitored.

Field personnel collected data on pretreatment and posttreatment grasshopper densities, species composition, and age structure at permanent sampling sites on treated and untreated plots. To determine density, each site had a circular transect of 40 0.1-m^2 rings placed 5 m apart (Onsager and Henry 1977). Rings were in place for the duration of the season.

To sample, field personnel took 400 sweeps, 200 high and fast and 200 low and slow, with standard sweep nets during the grasshopper season. Samples were sacked, frozen, and later identified in the laboratory by species and age class for each site and sampling date.

During a 7-year period from 1987 to 1993, the GHIPM Project studied 25 separate demonstration areas. Laboratory personnel examined and recorded data on approximately 250,000 individual grasshoppers comprising 57 species (table 2).

Of the 57 species, no more than 38 are typical in western North Dakota rangeland samples. Of the 50 species listed in the seasonal history chart, surveyors in western North Dakota commonly find the 15 noted in table 3.

The seasonal history chart is divided into four developmental time periods of 4 months each. These four time periods are subdivided into approximately three 10-day periods. The numbers 1 through 5 represent a grasshopper's instar stage, and the letter "A" stands for adulthood. The placement of the numbers and letter A's in the chart represents the time a certain species has reached a stage of development. These data come from 7 years of observing and recording thousands of individual grasshoppers.

Several species listed on the seasonal chart have almost no early dates of occurrence indicated. This void results from a lack of basic identification tools available on important bandwinged and *Melanoplus* species and from the small number of these species examined.

The arrangement of grasshopper-hatch time periods in order by type of species are (1) overwintering,
(2) early-hatching, (3) intermediate-hatching, and
(4) late-hatching.

Overwintering Species

North Dakota has six species that commonly pass the winter in various instar stages, and others occasionally overwinter as adults. Ranchers and survey personnel usually find these species early in the season. Although damage caused by most of the six species is below the threshold of economic significance, their appearance can cause concern because many lay persons are unaware of this group and may think the season's hatch of genuinely threatening species has begun.

Early-Hatching Species

This group of grasshoppers, whose eggs hatch from about late May to mid-June, probably is the most important. Many of the species that cause economically unacceptable levels of damage begin to develop at this time. Most agencies and Cooperative Extension Service personnel advise ranchers and farmers to check their fields and rangeland for possible infestations at this time. Late spring is the critical time to be able to differentiate among overwintering, noneconomic, and problem species. Most grasshopper control decisions take increased numbers of problem species into account.

Intermediate-Hatching Species

This group includes a number of species that hatch over an extended period of time, mainly because of a number of environmental conditions. Most species in this group begin appearing in late May or early June.

Late-Hatching Species

This group includes several late-hatching species and many that could fall into the intermediate-hatching group. Grasshopper species in this group appear slightly later than intermediate-hatching species and reach adulthood late. Both the intermediate- and late-hatching species need further study.

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Selected References

Brooks, A. R. 1958. Acridoidea of southern Alberta, Saskatchewan and Manitoba (Orthoptera). Suppl. 9. Canadian Entomologist 90: 1–92.

Capinera, J. L.; Sechrist, T. S. 1981. Grasshoppers (Acrididae) of Colorado, identification, biology and management. Exp. Stn. Bull. 584s. Fort Collins, CO: Colorado State University.

Handford, R. H. 1946. The identification of nymphs of the genus *Melanoplus* of Manitoba and adjacent areas. Scientific Agriculture 26: 147–178.

Newton, R. C.; Esselbaugh, C. O.; York, G. T.; Prescott, H. W. 1954. Seasonal development of range grasshoppers as related to control. Bull. E-873. Washington, DC: U.S. Department of Agriculture, Agricultural Research Service, Bureau of Entomology and Plant Quarantine.

Onsager, J. A.; Henry, J. E. 1977. A method for estimating the density of rangeland grasshoppers (Orthoptera: Acrididae) in experimental plots. Acrida 6: 231–237.

Unpublished Reference

Mazuranich, Philip. 1987. Seasonal history of common Montana grasshoppers. (Mimeo, 4 p.)

Table 1—Seasonal history of common western North Dakota grasshoppers

Overwintering species	April			May			June			July			August		
	E[1]	M	L	E	M	L	E	M	L	E	M	L	E	M	L
Arphia conspersa[2]			▬	▬	A[3]	▬	▬				[4]1	2	3	4	
Chortophaga viridifasciata		5 ▬	▬	▬	A	▬	▬			1	2	3	4	5	
Eritettix simplex		5 ▬	▬	▬	▬	A	▬	▬	▬		1	2	3	4	
Psoloessa delicatula		5 ▬	▬	▬	A	▬	▬	▬			1	2	2	2 3	—[5]
Pardalophora haldemani		—	—	—	4	5		A	▬						
Xanthippus corallipes		4 5	▬	▬	▬	A	▬	▬	▬		1	2	2 3		
Early-hatching species															
Aeropedellus clavatus				1	2	3	—	4 ▬	▬	▬	A	▬	▬	▬	
Acrolophitus hirtipes					1	2	3	—	4	5 ▬	▬	A	▬	▬	
Ageneotettix deorum					1	—	2	3	4	5 ▬	▬	A	▬	▬	
Amphitornus coloradus[6]						1	2	—	3 4	5	▬	A	▬	▬	
Aulocara elliotti					1	2	3	4	5	▬	▬	A	▬	▬	
Camnula pellucida						1	2	3 4	—	5 ▬	▬	A	▬	▬	
Circotettix carlinianus						1	2	3	4	5 ▬	▬	A	▬	▬	
Chloealtis conspersa					—	—	—	—	5	▬	▬	A	▬	▬	
Melanoplus bivittatus						1	2	3	4	5 ▬	▬	A	▬	▬	
Melanoplus confusus				1	2	3	4	5 ▬	▬	▬	A	▬	▬	▬	
Melanoplus infantilis						1	2 3	4	5 ▬	▬	▬	A	▬	▬	
Melanoplus occidentalis				—	1	2	3	4	5 ▬	▬	▬	A	▬	▬	

Table 1—Seasonal history of common western North Dakota grasshoppers (continued)

Early-hatching species (cont’d.)	April			May			June			July			August		
	E[1]	M	L	E	M	L	E	M	L	E	M	L	E	M	L
Melanoplus packardii						1	2	—	3	4	5 ▬	A	▬	▬	
Melanoplus sanguinipes						1	—	2	3	4	5 ▬	▬	A	▬	
Trachyrhachys kiowa						1	—	—	2	3	4	5	▬	A	
Intermediate-hatching species															
Aeoloplides turnbulli						1	2	3	4	5	▬	A	▬	▬	
Aulocara femoratum								1	2	3	4	5	A	▬	
Boopedon nubilum						1	—	2	3	4	—	5 ▬	A	▬	
Chorthippus curtipennis							1	2	3	4	—	—	A		
Derotmema haydeni								1	2	3	4 ▬	▬	A	▬	
Hesperotettix viridis						1	—	2	3	4	5 ▬	▬	A	▬	
Melanoplus femurrubrum						1	—	1	2	—	3 4		5 ▬	A	
Melanoplus bowditchi							1	2	3	4	5	▬	A	▬	
Metator pardalinus							1	2	3	4	5 ▬	▬	A	▬	
Spharagemon equale						—	2	3	—	4	—	5 ▬	▬	A	
Stenobothrus brunneus					—	1	2	3	4	5	▬	A	▬	▬	
Late-hatching species															
Dissosteira carolina							—	2	3	4	5 ▬	▬	A	▬	
Hadrotettix trifasciatus						—	2	—	—	3	4	5	▬	A	

Late-hatching species (cont'd.)	April			May			June			July			August		
	E[1]	M	L	E	M	L	E	M	L	E	M	L	E	M	L
Hypochlora alba								1	2	—	3	4	5 ■	A	
Melanoplus dawsoni									1	2	3	4	5	A	
Phlibostroma quadrimaculatum									1	2	3	4 5	A	■	
Spharagemon collare								1	2	3	4	5 ■	A	■	
Arphia pseudonietana									1	2 3	4	5 ■	A	■	
Encoptolophus costalis									1	2	3	4	5 ■	A	
Melanoplus keeleri								1	2	—	2	—	3	4 5 A	
Mermiria bivittata							1	—	2	3	—	4	5 ■	A	
Opeia obscura[6]							1	2	3	4	■	5	A	■	
Orphulella speciosa								1	—	2 3	4	5 ■	A	■	
Phoetaliotes nebrascensis									1	2	3 4	5 ■	A	■	
Melanoplus gladstoni								1	2	—	3 4	5 ■	A	■	
Dactylotum pictum									1	2 3	4	■	A	■	
Schistocerca lineata								—	—	—	—		5	A	
Melanoplus angustipennis							1	2	3	4	5 ■	■	A	■	

[1] E = early part of month, M = midmonth, L = latter part of month.
[2] Overwintering immatures of *Arphia conspersa* and *Chortophaga viridifasciata* usually hatch near the second week of July.
[3] A = adult grasshopper.
[4] Numerals 1 through 5 refer to grasshopper instar.
[5] — = little or no data about instar stage.
[6] *Amphitornus coloradus* and *Opeia obscura* exhibit like early instar characteristics and colors, but *Amphitornus coloradus* usually hatches at least 10 days before *Opeia obscura*.

Table 2—Species collected in northwestern North Dakota, 1987–93

Acrolophitus hirtipes (Say)
Aeoloplides turnbulli (Candell)
Aeropedellus clavatus (Thomas)
Ageneotettix deorum (Scudder)
Amphitornus coloradus (Thomas)
Arphia conspersa (Scudder)
Arphia pseudonietana (Thomas)
Aulocara elliotti (Thomas)
Aulocara femoratum (Scudder)
Boopedon nubilum (Say)
Camnula pellucida (Scudder)
Chloealtis conspersa (Harris)
Chorthippus curtipennis (Harris)
Chortophaga viridifasciata (DeGeer)
Circotettix carlinianus (Thomas)
Dactylotum pictum (Thomas)
Derotmema haydeni (Thomas)
Dissosteira carolina (L.)
Encoptolophus costalis (Scudder)
Eritettix simplex (Scudder)
Hadrotettix trifasciatus (Say)
Hesperotettix viridis (Thomas)
Hypochlora alba Dodge
Melanoplus angustipennis (Dodge)
Melanoplus bivittatus (Say)
Melanoplus bowditchi (Scudder)
Melanoplus confusus Scudder
Melanoplus dawsoni (Scudder)
Melanoplus femurrubrum (DeGeer)
Melanoplus foedus Scudder
Melanoplus gladstoni Scudder
Melanoplus infantilis Scudder
Melanoplus keeleri (Thomas)
Melanoplus occidentalis (Thomas)
Melanoplus packardii Scudder
Melanoplus sanguinipes (Fabricius)
Mermiria bivittata (Serville)
Metator pardalinus (Saussure)
Opeia obscura (Thomas)
Orphulella speciosa (Scudder)
Pardalophora haldemani (Scudder)
Phlibostroma quadrimaculatum (Thomas)
Phoetaliotes nebrascensis (Thomas)
Pseudopomala brachyptera (Scudder)
Psoloessa delicatula (Scudder)
Schistocerca lineata Scudder
Spharagemon collare (Serville)
Spharagemon equale (Say)
Stenobothrus brunneus Thomas
Trachyrhachys kiowa (Thomas)
Trimerotropis agrestis McNeill
Trimerotropis campestris McNeill
Trimerotropis gracilis (Thomas)
Trimerotropis latifasciata Scudder
Trimerotropis pallidipennis (Burmeister)
Trimerotropis sparsa (Thomas)
Xanthippus corallipes (Haldeman)

Table 3—The 15 most abundant grasshopper species encountered on rangeland during the study in North Dakota, in alphabetical order

Aeropedellus clavatus (Thomas)	Clubhorned grasshopper
Ageneotettis deorum (Scudder)	Whitewhiskered grasshopper
Amphitornus coloradus (Thomas)	Striped grasshopper
Aulocara elliotti (Thomas)	Bigheaded grasshopper
Camnula pellucida (Scudder)	Clearwinged grasshopper
Melanoplus bivittatus (Say)	Twostriped grasshopper
Melanoplus confusus Scudder	Pasture grasshopper
Melanoplus femurrubrum (De Geer)	Redlegged grasshopper
Melanoplus gladstoni Scudder	Gladston grasshopper
Melanoplus infantilis Scudder	Little spurthroated grasshopper
Melanoplus packardii Scudder	Packard grasshopper
Melanoplus sanguinipes (Fabricius)	Migratory grasshopper
Metator pardalinus (Saussure)	Bluelegged grasshopper
Phlibostroma quadrimaculatum (Thomas)	Fourspotted grasshopper
Trachyrhachys kiowa (Thomas)	Kiowa grasshopper

VI.9 Geographic Information Systems (GIS) and Integrated Pest Management of Insects

W. P. Kemp, D. McNeal, and M. M. Cigliano

Space and Pests

An understanding of the geographic variability in distributions and densities of pests is required for any integrated pest management (IPM) program. Pest densities influence the intensity of sampling required to define the area infested and the timing and economics of various control options. However, until recently there has been a general lack of analytical and data management tools that pest managers and researchers could use in IPM planning and execution.

Among several new technologies evaluated and demonstrated by Grasshopper Integrated Pest Management (GHIPM) Project participants, the geographic information system (GIS) and Global Positioning System (GPS) technologies appear to be sufficiently well developed to be integrated into existing IPM programs for rangeland grasshoppers in the Western United States. Although the primary focus of this chapter is GIS, we have chosen to include additional information on GPS because of the obvious link between the two technologies.

First Consider GPS

GPS refers to an advanced navigational system that was developed primarily for military applications. GPS consists of a number of satellites orbiting the Earth. These satellites have the ability to communicate with any appropriately equipped plane, ship, vehicle, or individual and indicate the geographic position on the face of the Earth and the elevation of the receiver. Position accuracy within feet may be obtained with appropriate equipment.

Because of the obvious improvements in guiding or tracking for commercial uses, some portions of the GPS have been made available to the public. Hand-held GPS receivers (fig. VI.9–1) are finding wide usage throughout

Figure VI.9–1—One of the newest tools to aid pest managers is a hand-held Global Positioning System (GPS) instrument. GPS provides accurate latitude and longitude coordinates, aiding the process of mapping locations of grasshopper populations. (APHIS photo by Mike Sampson.)

the public and private sectors. For the purposes of IPM, the GPS offers several capabilities. The most highly developed aspect of GPS that has been exploited by the participants of the GHIPM Project is aircraft guidance (see II.22). We focus the following discussion of GPS application on field scouting and the obvious link to the GIS.

Those involved with pest management of rangeland grasshoppers have struggled with the problem of locating their position on a map. Agencies often use the U.S. Geological Survey 7.5 Minute Quadrangle Series Maps, frequently referred to as simply "topo maps" or "quad sheets," where 2 inches on the map represents 1 mile on the surface of the Earth. Using 2 inches = 1 mile map scale as an example, consider what a scouting activity frequently involves. Whether sampling for Mediterranean fruit fly in California or for grasshoppers in Montana, the problem is the same—how to mark a place on a map that represents the location of a sample site?

Over the years, most scouts develop experience, which helps them locate their position on a map quickly and accurately. Scouts usually become good "mappers." However, learning to read maps is an acquired skill, and new scouts cannot be expected to be able to locate their position at all times quickly and accurately (accuracy is possible, but most novices cannot work quickly). Furthermore, scouts vary in their ability to read maps. As with any human activity, some scouts are simply better mappers than others.

Currently, a number of GHIPM Project participants use hand-held GPS receivers (some of which are about the size of a large pocket calculator), which can provide positional accuracies of plus or minus 100 feet in normal operational mode or plus or minus a few feet when operating in an optional mode. The positional accuracy possible in point location and block location (for example, the location of an infestation of insect A) via GPS goes a long way toward reducing errors and helps minimize the differences between scouts in mapping activities. Furthermore, many of the currently available GPS receivers can be connected directly to microcomputers or field data recorders. These can manage data in standard GIS formats, so scouting information can be examined very rapidly and thoroughly.

On to GIS

A GIS is a set of computer programs that can store, use, and display information about places of interest. Examples of places of interest to a grasshopper pest manager might be a 20-acre field, a 20,000-acre watershed, or the 2 million square miles of rangeland in a particular State. Examples of information for any place of interest are soil types, rainfall and temperature patterns, land use, ownership patterns, roads, vegetation types, and topography (landform). A GIS stores two types of data that are found on a map, the geographic definitions of Earth's surface features (spatial reference) and the attributes or qualities that those features possess. It is generally agreed that a true GIS is capable of several characteristic activities: (1) the storage and retrieval of information with a spatial reference (point A is located in Section 20 of Township 5, Range 8 and has soil type B), as well as (2) the input, (3) analysis, and (4) reporting of spatially referenced information in digital form.

GIS Storage and Retrieval

A basic feature of any of the hundreds of GIS products available today is the ability to represent map information in a form that a computer can use. In the world of information management, people generally reserve the term "map" for paper, acetate, or Mylar™ maps, whereas the representation of the map in the GIS is called a "coverage" or "map layer." For the sake of simplicity, we will use "coverage" throughout for the GIS representation of a paper map. Of the approaches used by various GIS products, the two most often heard about are "raster" and "vector."

A GIS that uses a raster approach is similar to observing an attribute such as soil type through a grid or to the view that one has of the world through a screen door. With raster-based GIS products, a coverage of the frequency of grasshopper outbreaks in Montana consists of hundreds of tiny cells each with only one value for the number of years when outbreaks were observed (fig. VI.9–2). Raster-based GIS products keep track of the arrangement of each cell. Each cell and its unique outbreak frequency value have one and only one correct location on the coverage, so when pest managers want to view the grasshopper outbreak frequency coverage of Montana, the GIS always displays the same arrangement of the cells.

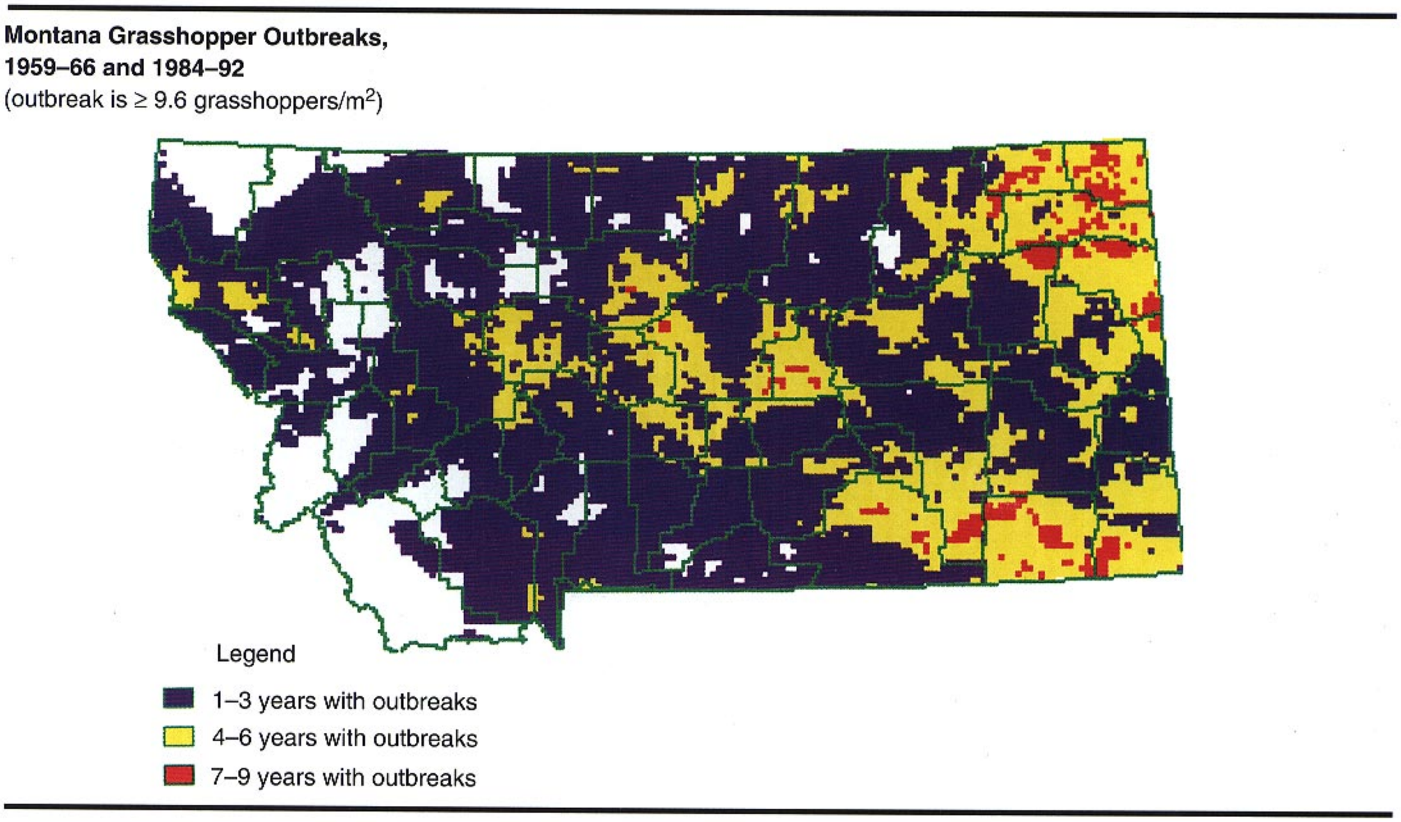


Figure VI.9–2—Rangeland grasshopper outbreak frequency in Montana, an example of a raster-based GIS product.

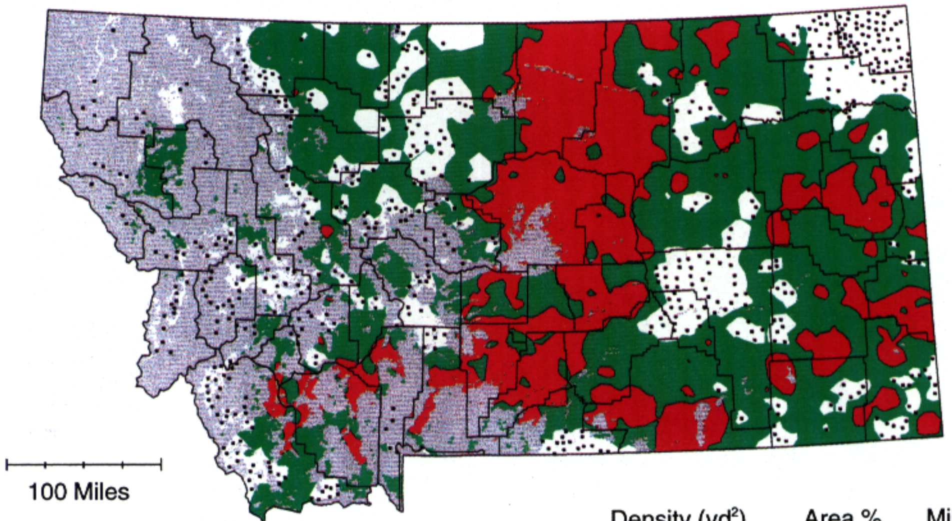
A GIS that uses a vector approach stores information in a somewhat different manner. For example, rather than viewing grasshopper densities as a collection of discrete cells that, when taken together, make up the entire image (the raster-based GIS approach), vector-based GIS products keep track of borders. Vector-based GIS products then associate a particular density to each unique area or polygon area found on the coverage (fig. VI.9–3). With vector representation, the boundaries of the features are defined by a series of points that, when joined with straight lines, form the graphic representation of that feature. The attributes (information) of features are then stored within a standard data-base management software program. The vector-based method is similar to what pest managers do when they draw insect-infested areas on a map in pencil.

Although some applications are more logically approached with either a raster or vector GIS product, in reality it is possible to convert map coverages from raster to vector format and vice versa. If one has purchased a raster-based GIS, he or she is not limited from obtaining a coverage from a vector-based GIS. Whether the basic unit of a coverage is a raster or a polygon, it is not uncommon to have more than one attribute (for example, soil type, vegetation type, or elevation) associated with it. The way that this task is accomplished varies from one GIS product to another.

Data Input and Spatial Analyses

An obvious, yet underappreciated (see more on this below in GIS—The Growth Years), GIS activity is getting the information on the map that you have in front of you into the GIS. In reality, there are a variety of data types that GIS products (paper maps showing point samples or infested areas, digital line graphs, or remotely sensed data) can use. With, for example, a soil type map resting on your desk, you have two logical ways, either "digitizing" or "scanning," of getting the information from that map into the GIS that resides on your desktop microcomputer or workstation.

1994 Montana Rangeland Adult Grasshopper Survey



Data source:
USDA Animal and Plant Health Inspection Service
Plant Protection and Quarantine
Billings, MT 59101

Prepared by:
USDA Agricultural Research Service
Rangeland Insect Laboratory
Bozman, MT 59717-0366

	Density (yd^2)	Area %	Million acres
☐ Uninfested	<3	16.4	15.4
🟩 Threatened	3–8	35.8	33.7
🟥 Infested	>8	18.5	17.5
▨ Forested		29.3	27.6

⊡ Sample locations in uninfested areas

Figure VI.9–3—The 1994 annual adult grasshopper survey for Montana, an example of a vector-based GIS product used in integrated pest management.

A digitizer device connected directly to your GIS by a cable from your computer may be as small as the blotter on your desk or as big as a draftsman's table. The digitizer has a device, called a "puck," that looks like the mouse on your PC (personal computer), but has more buttons as well as a set of cross hairs to allow you to trace the outlines of soil types on your paper map. The tracing process and some additional steps taken with your GIS successfully convert the information from your paper map into an electronic version or coverage, as we explained above. This process should sound a lot like the vector-based GIS approach we discussed above.

A scanner, on the other hand, performs a task much like a facsimile machine in a home or office and may range in size from a small hand-held device to the large-format photocopy machines that you have seen in photocopy shops. A scanner simply performs a raster (grid) scan of the map that you insert and senses and records the light reflectance of each raster cell. This information is stored in a file format the GIS on your computer can read and convert into a desired coverage. As mentioned, although digitizing and scanning are two commonly used methods for getting map data into a GIS, digital line graphs (DLG), published electronically by government agencies such as the U.S. Geological Survey, provide information in GIS-ready formats. Formats include attributes like elevation, political boundaries, highways, soils, land use, and more.

As mentioned, when people discuss GIS applications or the potential of the technology, they frequently gloss over the "minor details" of getting data into a GIS and concentrate on what we call the spatial analysis capabilities of GIS. Perhaps the most important process common to all true GIS products is the "overlay." An overlay is simply a GIS procedure where two or more coverages (perhaps vegetation type, river courses, and primary highways) are combined and the result is a new coverage that represents a combination of the originally separate coverages. In another example, one coverage (environmentally sensitive areas, for example) may be used to mask out portions of a second coverage. Lastly, it is possible to compute the sum of specific attributes from a series of yearly coverages to compute, for example, the number of years each county in Utah has seen problem populations of rangeland grasshoppers or Mormon crickets.

In addition to the overlay, most GIS products offer a variety of spatial measurement techniques or area analyses. Examples include calculating the area of rangeland in a particular county with more than 20 grasshoppers/yd^2, estimating the area of a lake, or computing the proportion of a chemical control block devoted to buffer zones. All true GIS products also offer solutions to people interested in overlaying coverages of different scales (and projections—although we have chosen for the sake of simplicity to discuss only different scales). Consider, for example, a situation where you want to identify those vegetation types in a particular county where grasshopper densities exceeded 20 grasshoppers/yd^2. If scouts collected density data on maps with a scale of 2 inches = 1 mile (a 7.5-minute quad) and vegetation data was mapped at a scale of 1 inch = 1 mile (a 15-minute quad), you can use the capabilities of a GIS to rescale one map or the other. You could produce a correct overlay to depict only those vegetation types with more than 20 grasshoppers/yd^2.

Maps, Graphs, and Tables

GIS products offer a bewildering array of report types. Reports can consist of paper maps, tables, charts, graphs, or computer images. Selecting which report type is the most useful will depend on your particular application (see Cigliano et al. 1995). For viewing an overlay consisting of vegetation type, land use, rivers, and roads, you would likely choose a simple paper map presentation. If you wanted to forecast grasshopper densities throughout a State for next year, you could select options that would produce a contour map (for example, fig. VI.9–3). In short, GIS offers pest managers a great deal of flexibility in the presentation of information.

GIS Applications and IPM of Insects

Liebhold et al. (1993) described GIS's as "enabling technology." As previously stated, a GIS provides pest managers with the capabilities to store, retrieve, process, and display spatially referenced data. It seems only logical that GIS technology will be rapidly embraced because so many questions from insect ecology to pest management have a spatial component. Whether studying the patch dynamics of host and herbivore or predicting multi-State pest hazards, GIS technology provides today's researchers and pest managers with the ability to answer questions that frustrated their predecessors.

Now it is possible to identify two general areas where GIS technology has been used in entomology—applied insect ecology research and insect pest management. Within the general area of applied insect ecology, perhaps the major use of GIS is in the relation of insect outbreaks to environmental features of the landscape. Using grasshoppers as an example, investigators in Canada used GIS products to examine the relationship between historical grasshopper outbreaks and soil characteristics (Johnson 1989a) and between weather and survey counts (Johnson and Worobec 1988). From these geographically referenced data, Johnson (1989a) found that grasshopper abundance in Alberta was related to soil type but not to soil texture. Furthermore, a significant association was found between rainfall levels and grasshopper densities. Populations tended to decline in areas receiving above average rainfall (Johnson and Worobec 1988).

Future efforts to characterize habitat susceptibility probably will use remotely sensed data extensively because of its high spatial resolution and its availability in virtually every portion of the globe (for a complete review of remote sensing in entomology, see Riley 1989). For example, Bryceson (1989) used Landsat satellite data to determine areas in New South Wales, Australia, that were likely to have egg beds of the Australian plague locust. Through the use of an index that indicated the general greenness levels of local vegetation, Bryceson was able to identify resulting nymphal bands geographically through changes in the greenness index that resulted from rains during March (nymphal bands tend to be associated with "green" areas that result from rain).

Similar "greenness mapping" exercises have been conducted in Africa for grasshoppers and locusts (Tappan et al. 1991). In addition to illustrating the apparent ecological association between nymphal bands of grasshoppers or locusts in Australia and Sahelian Africa and changes in greenness indices, studies of Bryceson (1989) and Tappan et al. (1991) have immense practical utility because they produce rapid estimates of the location and extent of potential pest problems. Through such methods, it has been possible to improve sampling efficiency vastly for detection of problems as well as to reduce the guesswork involved with planning and execution of pest management programs.

The second major area where GIS products have been used is for compilation and analysis of insect census data that are collected regularly by U.S. Department of Agriculture, Animal Plant Health Inspection Service (USDA, APHIS). One example of this application for rangeland insects in the United States is the use of a GIS for developing a distribution atlas for grasshoppers and Mormon cricket in Wyoming (Lockwood et al. 1993). Additionally, Kemp et al. (1989) and Kemp (1992 unpubl.) provide methods for the development of rangeland grasshopper GIS coverages and hazard forecasts, using annual survey data collected on adult grasshoppers in Montana. (See Johnson [1989b] for similar studies for grasshoppers in Canada.)

The compilation and interpretation of spatially referenced insect and habitat data is a complex process, if for no other reason than the sheer volume of information. Although GIS software is designed to handle this complexity successfully, these systems often are not easy to use. In order to make a GIS more accessible to applied problems, GIS is increasingly being linked as a part of a larger decision support system (DSS). These systems typically use a GIS to manage habitat, geophysical, political, and census data. The DSS uses these data, along with other data, as input to mathematical models and other modeling methods to produce useful abstractions or recommendations (Power 1988). These outputs might be maps of high damage hazard or even maps of proposed control areas. Hopper, the DSS for rangeland grasshoppers being developed by the GHIPM Project (Berry et al. 1991; see chapter VI.2), currently has the ability to display density coverages. Future plans include a closer link to GIS procedures. Coulson et al. (1991) use the term “intelligent geographical information system” (IGIS) to describe systems that use a GIS and rule-based models to combine landscape data and knowledge from a diversity of scientific disciplines.

GIS—The Growth Years

GIS brings a great deal of analytical horsepower to the complex tasks associated with managing America’s natural resource base. However, expectations frequently associated with bringing GIS activities into the IPM realm frequently result in frustration for both pest managers and GIS professionals. Two major reasons why frustrations develop already have been mentioned: (1) People generally underestimate the resources required to get information into a GIS, and (2) GIS products are, at present, frequently complex enough to require specialized training. Another confounding problem that we should add is communication. Pest managers frequently lack indepth familiarity with computer systems and at times may distrust all the apparent complexity involved with GIS activities. GIS technicians, on the other hand, frequently lack the biological expertise necessary to assist the pest managers with creative solutions to a particular problem. These communication problems can be frustrating to those on both sides of the table and may result in little advancement toward the solution to the current pest management problem.

At this time, to expect pest management professionals, for example APHIS, Plant Protection and Quarantine (PPQ) plant health directors, to be trained as GIS technicians is no more realistic than expecting them to be able service their personal computers. Rather, it indeed is logical to provide plant health directors or similar professionals with general training that highlights GIS capabilities, so they can in turn direct the activities of GIS technicians or cooperators. At present, the revamped APHIS, PPQ Cooperative Agriculture Pest Survey (CAPS) is being used by a number of plant health directors from individual States to contract small GIS projects, frequently involving rangeland grasshoppers. This is a way of exploring the uses of GIS products with minimal investment and an attempt to become more knowledgeable about potential GIS applications in other pest management problems.

In general, GIS–pest management activities coordinated through the CAPS program have received good reviews from the participants largely because of the ability of plant health directors from individual States to specify the types of GIS products best suited to their particular needs. For the future of GIS and rangeland grasshopper IPM, today's interactions among plant health directors, GIS technicians, and researchers will be the basis for tomorrow's creative solutions.

References Cited

Berry, J. S.; Kemp, W. P.; Onsager, J. A. 1991. Integration of simulation models and an expert system for management of rangeland grasshoppers. AI Applied Natural Resource Management 5: 1–14.

Bryceson, K. P. 1989. Use of Landsat MSS data to determine the distribution of locust egg beds in the Riverina region of New South Wales, Australia. International Journal of Remote Sensing 10: 1749–1762.

Cigliano, M. M.; Kemp, W. P.; Kalaris, T. M. 1995. Spatiotemporal characteristics of rangeland grasshopper (Orthoptera: Acrididae) regional outbreaks in Montana. Journal of Orthoptera Research 4: 111–126.

Coulson, R. N.; Lovelady, C. N.; Flamm, R. O.; Spradling, S. L.; Saunders, M. C. 1991. Intelligent geographic information systems for natural resource management. In: Turner, M. G.; Gardner, R. H., eds. Quantitive methods in landscape ecology. New York: Springer Verlag: 153–172.

Johnson, D. L. 1989a. Spatial analysis of the relationship of grasshopper outbreaks to soil type. In: McDonald, L. L., et al., eds. Estimation and analysis of insect populations: proceedings of a conference; 25–29 January 1988, Laramie, WY. New York: Springer Verlag: 347–359.

Johnson, D. L. 1989b. Spatial autocorrelation, spatial modeling, and improvements in grasshopper survey methodology. Canadian Entomologist 121: 579–588.

Johnson, D. L.; Worobec, A. 1988. Spatial and temporal computer analysis of insects and weather: grasshoppers and rainfall in Alberta. Memoirs of the Entomological Society of Canada 146: 33–48.

Kemp, W. P.; Kalaris, T. M.; Quimby, W. F. 1989. Rangeland grasshopper (Orthoptera: Acrididae) spatial variability: macroscale population assessment. Journal of Economic Entomology 82: 1270–1276.

Liebhold, A. M.; Rossi, R. E.; Kemp, W. P. 1993. Geostatistics and geographic information systems in applied insect ecology. Annual Review of Entomology 38: 303–327.

Lockwood, J. A.; McNary, T. J.; Larsen, J. C.; Cole, J. 1993. Distribution atlas for grasshoppers and the Mormon cricket in Wyoming 1988–92. Misc. Rep. B-976. Laramie, WY: University of Wyoming and Wyoming Agricultural Experiment Station. 117 p.

Power, J. M. 1988. Decision support systems for the forest insect and disease survey and for pest management. Forestry Chronicle 64: 132–135.

Riley, J. R. 1989. Remote sensing in entomology. Annual Review of Entomology 34: 247–271.

Tappan, G. G.; Moore, D. G.; Knausenberger, W. I. 1991. Monitoring grasshopper and locust habitats in Sahelian Africa using GIS and remote sensing technology. International Journal of Geographical Information Systems 5: 123–135.

Reference Cited—Unpublished

Kemp, W. P. 1992. Annual report for grasshopper population dynamics. In: Cooperative Grasshopper Integrated Pest Management Project, 1992 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 39–44.

VI.10 Assessing Rangeland Grasshopper Populations

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Introduction

Land managers need accurate and comprehensive methods for assessment of rangeland grasshopper populations to make appropriate management decisions and to support research. Some of the needed information at known locations includes grasshopper density, developmental stage, and species composition.

One option is to count and identify every grasshopper in an area. This procedure is called a census. Obviously, a complete census of grasshoppers in a State, a county or even a small ranch is impossible. Therefore, managers must have methods to sample a limited number of the grasshoppers in order to estimate the status of entire grasshopper populations over large and often remote geographic areas where rangeland grasshoppers occur. The result of sampling large areas to estimate grasshopper populations is called a survey. In this chapter, we will explore techniques and issues related to sampling and surveying rangeland grasshoppers.

Overview of Types and Purposes of Surveys

Nymphal Survey.—This is an early season survey to identify areas with high densities of grasshoppers. The nymphal survey notes grasshopper density, species, and developmental stages at recorded sites on all rangeland areas where grasshoppers may be a problem in a State. Developmental stage data are useful for timing the adult survey later in the year (discussed later in this chapter). In years when resources and time are limited for the nymphal survey, areas associated with a greater risk of grasshopper outbreak (such as a potential treatment block) should receive a greater priority for survey. Priority can be determined using previous year adult survey maps, other historical data, and cooperator reports, including requests from and discussions with local people. Other considerations include current conditions, weather (drought or above normal precipitation), cattle prices, range conditions, economics (benefit–cost), species composition, and politics.

Nonoutbreak Years/Areas.—In general, survey sites should be 5 miles (7.65 km) apart on accessible routes. Another alternative is to use sentinel sites (fixed locations) that have been proven as predictive indicator locations. All areas will have uniform priority.

Outbreak Years/Areas.—Deploy survey sites first to high-priority areas as discussed above. Within a potential treatment block (highest priority), survey sites may be a quarter to a half mile (0.4–0.8 km) apart (an area probably less than the entire infestation). These data can be used to establish density estimates for management decisions for the block, including use in the Hopper Decision Support System (Hopper). Grasshopper populations that lie outside but near the potential treatment block are of secondary priority. These areas may not be sampled, but you can collect data in them later during the adult survey.

Proposed Treatment Areas.—A proposed treatment area is one where grasshopper densities exceed the economic threshold (ET, determined by Hopper) for a given treatment, or where land owners or managers have indicated a desire for their lands to be treated (escrow accounts established, letters of request on file, and cooperative agreements in place). For management purposes, a single average grasshopper density is needed for the proposed treatment block. You can combine estimated grasshopper densities over all sample stops within the proposed treatment block to obtain this single average grasshopper density. This average density is useful for the decision-support process, which may include economic analysis with Hopper.

Delimiting Survey.—The purpose of a delimiting survey is to determine the perimeter of the area infested with economically important densities of grasshoppers. (The economic density can be estimated using Hopper.) Often, delimiting surveys are a continuation of the nymphal survey, and they also may be used in the adult survey to collect additional data for forecasting. These data also should be sufficient to support a single density estimate for a proposed treatment area for use in Hopper (to determine the ET). Surveyors can record key grasshopper species composition and developmental stages during the delimiting survey. Survey sites may be one-quarter to one-half mile apart. Concentrate sampling effort in the transition between high-density areas and lower density areas to delineate the perimeter of a treatment block.

Adult Survey.—This is a midseason forecasting survey timed to evaluate *economic species* (5 to 10 in each State) in prime reproductive stage (fifth instar through early adult stage) to predict hazard for the following season. Record grasshopper density, species composition, and developmental stages at survey sites. Determine priorities for survey areas to sample by using nymphal survey maps and other historical data and cooperator concerns (requests from and discussions with local people). In general, survey sites should be 5 miles apart on accessible routes. Sample areas containing grasshopper densities of the greatest concern should be sampled with more survey sites (delimit high-density areas) to provide more information for hazard prediction.

Common Data Set Survey.—These data are used to provide regional- and national-level hazard maps. A data base can be developed (and saved) for improving existing models for predicting hazard. For example, while trained surveyors frequently refer to differences in vegetation and grasshopper dynamics throughout the 17 Western United States, so far surveyors have collected little data to confirm these impressions. In an effort to describe just how different outbreak dynamics can be throughout the West, it is necessary to collect data on both density and grasshopper species composition. These data will be used to develop a better understanding of grasshopper dynamics in different ecoregions (biologically similar areas) throughout the West and provide a mix of strategic planning maps that will be valuable at regional and national scales.

These data are collected as part of the normal adult survey. In general, sample sites are at least 5 miles apart on accessible routes with uniform priority. For States that survey more than 1,000 sites, 10 percent of the sites are used for the common data set. All other States should provide data for about 100 sites.

General Guidelines for Surveying Large Areas

Each year, the U.S. Department of Agriculture's Animal and Plant Health Inspection Service (USDA, APHIS) conducts the preceding surveys of grasshopper populations throughout the rangelands of the Western United States. The surveys are managed within each State to meet local, State, and Federal needs for the information. Planning begins each fall for the surveys to be conducted the next summer. The survey manager determines the areas that need to be surveyed, when to begin and end each survey, survey site intervals, method of determining population, and logistics of completing the survey.

Area To Be Surveyed.—The criteria for deciding what areas to survey vary from State to State. Historical and recent information on the outbreaks of grasshopper and control activities provide the best guide to the areas that need to be surveyed. Priority is given to areas that have frequent outbreaks that tend to persist over several years. These are the areas where control is most likely to be requested.

Nymphal survey concentrates on areas that had high grasshopper densities the preceding fall and on areas that cooperators indicate may need treatment during the current season. Information from the nymphal survey is useful for making management decisions during the current season. Adult grasshopper surveys cover the general area where grasshoppers occur because information from these surveys is targeted for predicting future trends and recording historical information.

Survey managers consider many other factors when determining what areas within a State to survey. The amount of rangeland versus cropland is important in some States. Likewise, the amount of rangeland versus forested or mountainous areas is important. In recent years, Conservation Reserve Program (CRP) land is included as part of the surveyed area in some States.

The survey in Nevada targets areas where large parcels of the rangeland have burned, removing much of the sagebrush. Much of the rangeland in southwestern Wyoming is not surveyed because historical records show that, even if an outbreak occurs, it is usually short lived and grasshopper populations collapse on their own. Other States may concentrate surveys on rangeland that is sufficiently productive so that the costs of treatment can be recovered and leave out areas of low forage productivity.

Survey Timing.—The objectives of each survey are considered while planning the surveys. Weather strongly influences when each species of grasshopper will hatch. Nymphal surveys are timed to occur after the majority of the potential pest species hatch but must be completed in a timely manner, allowing management decisions to be made for effective management and forage protection. Adult surveys are timed to include the period when most individuals of the potential pest species are nearing reproductive maturity but before the seasonal population decline. This timing gives results that yield the best indication of the reproductive potential of the grasshopper populations.

Survey Site Interval.—The standard interval between survey sites used in APHIS grasshopper surveys is 5 miles, but each State office adjusts this distance to meet its own needs. When habitat or populations are homogeneous (similar) over large expanses the distance between sites can be lengthened beyond 5 miles without detriment to survey quality. If the rangeland is interrupted by crops, forest, river, or other features or the habitat or grasshopper population are localized, then shorter survey site intervals may become necessary. Often the availability of roads dictates the interval between sites.

Method of Estimating Grasshopper Density.—The 18-ft^2 sample method used by many APHIS offices in the Western United States is a simple and quick way of determining the density of grasshoppers on rangelands. (A few States use a less reliable method correlating the number of grasshoppers caught in a sweep net to a population density.) At each survey site, choose a sample area typical of the rangeland to be surveyed. Next, look ahead and determine the approximate route you will walk (fig. VI.10–1). Pick a spot on the ground about 10 paces in front of you. Choose the spot before you determine if any grasshoppers are actually present there.

Visualize a sample area surrounding the spot that is equal to 1 ft^2 on the ground. You can use landmarks such as a stick, pebble, tuft of grass, or flower to help keep your eye focused on the sample area chosen. Once the area is set in your mind, walk slowly toward the area and determine the number of grasshoppers that are in the area by counting the grasshoppers as they flush out of the visualized sample area.

Do not count individuals that hop into the sample area while counting. When you reach the spot, probe the area with the handle of your insect net or other suitable object to make sure all individuals have flushed and been

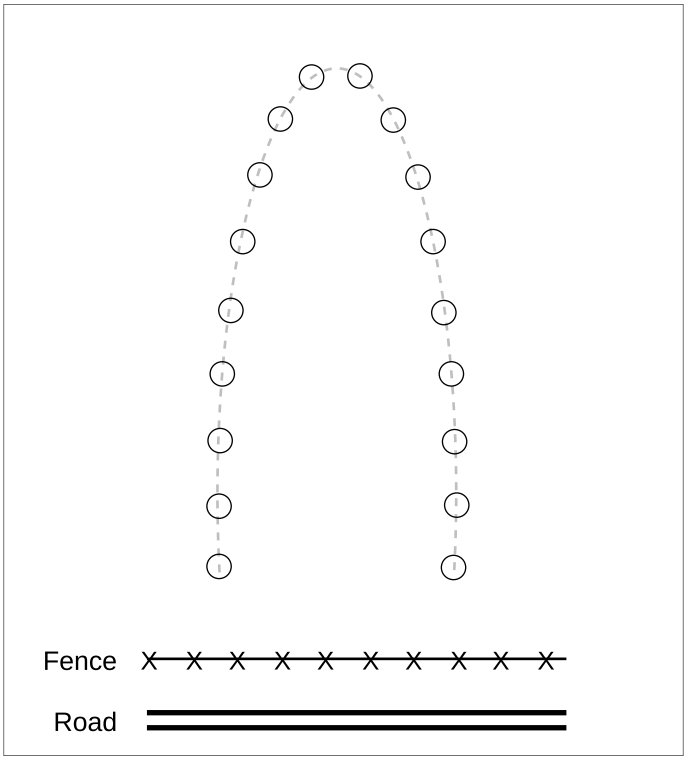

Figure VI.10–1—Configuration of the 18 1-ft^2 sample areas counted during a grasshopper surver on rangeland.

Figure VI.10–2—Using a prod can help flush grasshoppers out of the 0.1-m^2 counting rings. (APHIS photo.)

counted (fig. VI.10–2). Record the number counted and repeat the count at a total of 18 sample areas. The total number of grasshoppers counted in the 18 1-ft^2 sample areas, divided by 2, gives you the number of grasshoppers per square yard.

Logistics of Completing a Survey

After determining the area to be surveyed, survey timing, survey site interval, and the method to determine grasshopper density, you can decide the logistics for completing the survey. A combination of the size of the area to be surveyed and the site interval determines the total number of sites to be visited. For example, if the area to be surveyed is 30 million acres and the site interval is 5 miles, you will need approximately 1,875 survey sites. Plan 10 percent more survey sites for a delimiting survey where needed. For this example, the total number of survey sites is now 2,062.

Next, calculate the time it takes to sample each survey site. Include the time to actually complete the count at a survey site, plus time to record the data, travel between sites, travel to the area, contact cooperators and landowners, time lost to bad weather, and vehicle servicing and repair. This time ranges from 45 minutes to an hour and 15 minutes per site in the States surveyed by APHIS. For example, if you allot 1.1 hours for each site, to complete a survey of 2,062 sites takes 2,268 hours. If the time window to complete the survey is 6 weeks (240 work hours), 10 surveyors are needed to complete the survey. Other examples are outlined in table VI.10–1.

Issues Related to Sampling Error

Sample Accuracy, Precision, and Bias.—There are two broad criteria for evaluating sampling procedures: accuracy and precision. Both are important, and both must be present in some degree of balance.

To illustrate accuracy, imagine a person shooting a rifle at a target. If all hits are in the bull's-eye, these hits are accurate. If, however, the sights are not properly aligned, the hits will be outside of the bull's-eye. In statistical language, these hits are inaccurate, and the degree to which they miss the bull's-eye is called bias. Specifically, bias is the distance from where hits should fall to where they do fall. In terms of grasshopper sampling, accurate counts are those that include all grasshoppers that are within the correctly envisioned area. If the sampler consistently counts fewer or more grasshoppers than what are there, and/or if the sampler is envisioning an area that is smaller or larger than it should be, then the counts will be biased.

Notice that accuracy requires hits to fall in the bull's-eye, but is not concerned with size of the bull's-eye. In order to hit a very small bull's-eye consistently, surveyors need very high precision. In terms of grasshopper sampling, low precision might allow one to accurately estimate an infestation at 10–50 grasshoppers/yd^2, but high precision could accurately fine-tune the estimate to 28–32/yd^2.

Table VI.10–1—Example of logistics for completing a grasshopper survey over a large area

Survey type	Thousand acres surveyed	Stop interval (miles)	Acres represented per stop	+ 10% No. of stops	Hours to for delimiting	Hours for each stop	complete survey	Survey window	Surveyors needed
Adult	30,000	5	16,000	1,875	2,062	1.1	2,268	6 wk	10
Adult	10,000	3	5,760	1,736	1,909	1.0	1,909	5 wk	10
Nymphal	5,000	5	16,000	313	344	1.1	278	3 wk	3
Nymphal	25,000	10	64,000	390	430	1.2	516	2 wk	7
Delimiting	25	0.5	160	156	N/A	0.5	78	3 d	4
Delimiting	100	2	2,560	39	N/A	0.75	30	2 d	2

Land managers realistically can desire both accuracy and a certain minimum level of precision. Accuracy of grasshopper sampling can be affected by a number of factors will be discussed here. As far as we know, however, there is only one way to increase precision (estimate density within a narrower range), and that will be the subject of the next two paragraphs.

Rangeland grasshoppers generally appear to be distributed at random, with predictable probabilities of occurrence within samples taken at reasonably homogeneous sites. In mathematical terms, grasshoppers follow a “Poisson” distribution (a probability function which offers a description of a number of possible outcomes), which is not typical of most insects. Therefore, grasshopper sampling requires some atypical rules.

For all practical purposes, surveyors can increase sampling precision only by accurately counting more grasshoppers. This can be accomplished only by taking more samples in an accurate manner because an individual sample area cannot be increased without an accompanying loss in accuracy. In 1981 Onsager published a simple relationship between the counts and precision. In general, rapid gains in precision are made by continuing to examine samples until at least 40–60 total grasshoppers have been counted. On the other hand, there is little to be gained in precision by sampling after 150–200 grasshoppers have been counted.

Estimated (Visualized) Versus Delineated Samples.— For all but the most experienced persons, samples that are mechanically delineated (by wire frames or hoops) should yield greater accuracy and consistency between different individuals than visualized or estimated samples (fig. VI.10–3). Delineated samples are inconvenient in that templates should be placed about a day before they are examined (necessitating two trips to each survey site) and they require investment in bulky, single-purpose equipment. However, during the training process or when high accuracy is very important, the extra effort associated with delineated samples is worthwhile.

Sample Area Size.—Experiments have shown that examination of sample areas as large as 1.08 ft^2 (0.1 m^2) tends to detect only about 90 percent of the true density estimated by less subjective but more labor-intensive methods of sampling. Successively larger sample areas detect successively lower percentages of the true density, so the 1-ft^2 sample area is about as large as even a well-experienced sampler should attempt to examine. Experiments found that persons with moderate experience were able to count grasshoppers accurately in 0.06-ft^2 (0.05-m^2) rings, even when densities exceeded 125/yd^2. That area is approximately the size of a 9-inch pizza pan (about 1/20 of a square yard) or an 8 1/2- × 8 1/2-inch square (about 1/18 of a square yard).

Figure VI.10–3—One of the most valuable tools in field surveys is the 0.1-m^2 counting ring. Counting the number of grasshoppers in a series of rings provides an accurate count of grasshoppers per square meter or square yard. (USDA photo.)

Bias in Selecting a Site.—Sample sites must be representative of the general area. Atypical vegetation or topography could influence grasshopper density and species composition. For example, surveyors should avoid sites near roads, cattle trails, ditchbanks, fencelines, or any features not representative of the general habitat in the area.

Bias in Selecting a Visualized Sample Area.—Even a slight bias may seriously affect the outcome of the survey. If a sampler counted only 1 more grasshopper per sample than was actually present, the density estimate would be increased by 9 grasshoppers/yd^2 (assuming that

9 samples/yd^2 are taken at each survey site). Subconsciously, a sampler may choose movement by a grasshopper to be the center or edge of the area that will be visually delimited and counted. To demonstrate the potential for bias, one need only consistently use the last grasshopper movement as the edge of the visualized area and not include that grasshopper in the count. Such counts are obviously low estimations of actual densities. To prevent inaccuracy, exercise great care to select a point, patch of vegetation, pebble, or small topographic feature from which to base the boundaries of the visualized sample area. These boundaries must be established before the counting begins.

Sample Area Shape.—Most experienced samplers agree that the best sample area shape is the one they were taught to use. Some prefer squares while others prefer circles, and both can defend their viewpoint. Advantages of squares are that standard areas are easily visualized, and a variety of standard templates are easily found or constructed. For example, the suggested 8 1/2- × 8 1/2-inch square template can be made from a standard sheet of writing paper. However, a visualized square entails keeping mental track of four 90-degree corners that are equidistant from each other and connected by straight lines.

The advantage of circles is that a sampler can concentrate on one central point plus a constant omnidirectional radius without shifting focus. However, a circular standard area is not easy to visualize without studying a standard template, and round templates usually are not available in a variety of convenient dimensions. For example, a 0.5-ft^2 circular template would require a diameter of 9.57 inches.

Effects of Weather.—Variations in daily weather conditions probably contribute more to sampling error than any other single factor like size or shape of typical samples, visualized versus delineated sample areas, or total area sampled. Cool temperatures reduce grasshopper mobility, and lack of mobility can make smaller grasshoppers inconspicuous and larger ones relatively easier to spot before they flush. Cool weather most often occurs during the nymphal stages, when their small size makes grasshoppers most difficult to see. Under such conditions, additional prodding with a stick or pole is required to provoke movement and ensure that all grasshoppers in the sample area are counted. Under extreme conditions, the sampler will have to stoop and brush the ground with a hand to ensure a more accurate count. Warm temperatures are generally the best condition for conducting surveys because of the increased activity of grasshoppers and ease with which they are seen. However, because of this increase in activity, the sampler must begin concentrating on the sample area from a greater distance. Higher temperatures are usually associated with sunny conditions, which can cause the sampler's own shadow to become a factor. The sampler must approach the sample so the shadow will not flush grasshoppers prematurely.

Cloudy conditions reduce general visibility and can make some inconspicuous grasshopper species even more difficult to detect. Rain or mist may reduce the activity of grasshoppers even more than cool temperatures. In addition, rain or mist causes grasshoppers to hide and may prevent movement even when prodded. When counts are conducted in the rain, even with extra care, they are generally lower than the actual density of grasshoppers. Therefore, grasshopper surveys should not be conducted under these conditions.

Wind can be particularly troublesome when it is strong enough to provide a lot of background movement within the plant canopy, to alter the normal trajectory of grasshoppers that hop in the vicinity of the sample, or to whisk away grasshoppers that take flight. Under these conditions, probing with a stick to flush grasshoppers may also dislodge seeds or other dry pieces of vegetation, which blow in the same direction as most disturbed grasshoppers. When this happens, some seeds (those that appear to be grasshoppers) will need to be followed and probed again to determine if they were grasshoppers.

In itself, wind can become a major distraction to the concentration of the sampler. Wind moves clothing, equipment, and other items near the site and/or the sampler. If collections of grasshoppers are required in addition to the count, the consistent operation of a sweep net sometimes may become almost impossible. Wind generally is accompanied by other adverse conditions and tends to further aggravate less-than-ideal conditions already present. Walking at an angle to the wind is helpful, but going slower, concentrating harder, and spending more time at each sample are requirements for achieving accurate counts under windy conditions.

When weather conditions become increasingly unfavorable, it is critical that a sampler apply an increasing level of concentration if survey data are to have meaning. Nevertheless, in spite of the highest degree of concentration, if foul-weather sampling should yield high densities near some pivotal action threshold, it would be wise to verify some of the results later during favorable weather.

Effects of Habitat.—The nature of the vegetative canopy can affect sampling results. A short, sparse, and uniform canopy is easiest to sample accurately. A classic example would be crested wheatgrass that has been mowed or subjected to moderate grazing pressure. As vegetation becomes taller, the vertical dimension increases the volume you must examine simultaneously for grasshoppers. When vegetation becomes more dense, as when the sampler goes from bunchgrass to sod, it becomes easier to overlook smaller nymphs or species.

Where vegetation is strongly clumped, it becomes more difficult to apply representative sampling intensity to occupied and unoccupied portions. Habitats dominated by tall, thick, well-spaced clumps of shrubs are the most difficult to sample. Sample areas with dense vegetation require thorough probing with a stick, even under the best weather conditions.

Other Insects.—You may confuse other insects with grasshoppers as the other insects move from a sample area when the sampler approaches, probes, or brushes the area by hand. Most often, these insects are leafhoppers. During nymphal surveys, leafhoppers can be about the same size as very young grasshoppers. At low densities, you can follow these small insects and flush them again to determine if they are grasshoppers. Grasshoppers and other insects that move ahead of the sampler may land and flush new grasshoppers from a sample area before they can be counted. Be aware of this possibility, especially during the adult survey.

Disturbance of Sample Area.—Sample areas undisturbed for 24 hours before survey can produce accurate counts. Disturbance of sample areas just prior to or during counting can reduce the density estimate significantly. Cattle grazing or moving through the site are the most frequent source of direct disturbance. Vehicles driven by the sampler or others through or near the site also can affect the count. Nearby farming activity, such as harvesting or irrigation, may cause local movement of grasshoppers, and that can affect the counts. If densities at sites near these activities yield results that are of concern, additional counts at a later date may be required.

Dense Grasshopper Populations.—When finding grasshoppers at densities of 1 per square foot or fewer, counting is relatively easy. In denser populations where you flush several grasshoppers from each sample area, take greater care. When this happens, the sampler should take a mental picture of the action in the sample area to estimate the number of grasshoppers.

Concentration of the Sampler.—Concentration plays the central role in dealing with all factors that affect survey and can become critical at the end of a long day for a tired sampler. Many of the factors that complicate surveying are uncontrollable, but you can practice and improve concentration. A sampler may take several actions to maintain good concentration. A sampler continually using visualized sample areas can recalibrate by frequently referring to a physical template the size of the visualized area to be counted.

Removal of as many distractions as possible during the actual counting can help greatly. Wearing a billed hat or cap not only shades the eyes from the sun but can help focus the attention toward the ground and reduce distraction. The use of a long probing stick helps flush grasshoppers from the sample area. By simply slowing down while approaching and counting sample areas, you can reduce or eliminate many problems.

Training New Scouts

In the past, it was common practice for an experienced sampler to line up a class of novices, have everyone count grasshoppers in a certain number of visualized sample areas, compare results, and repeat the process until counts by the novices approximated those by the expert. There are three major disadvantages to this system. First, the expert may have unknown biases that are then passed on to the trainees. Second, a trainee cannot verify or recalibrate density estimates in the absence of an expert. Third, the system cannot be used for self-instruction.

A novice must learn to overcome two major tendencies that contribute to sampling error. The first is a tendency to overestimate size of the sample area. The second is a tendency to count all grasshoppers that are moving in the general vicinity of the sample area, even though there is uncertainty whether the movement originated inside or outside of the sample area. Both of these negative tendencies can be minimized by starting trainees out with delineated samples (all sample areas marked with wire rings or squares). When the trainees show proficiency with that setup, they can advance to using visualized sample areas and then carry one standard template along for periodic confirmation or recalibration of proper sample area size. To obtain accurate counts, sample areas should be small enough to be totally comprehended without shifting the focus of attention (preferably about 0.5 ft^2 each, but not over about 1 ft^2; see Issues Related to Sampling Error, Sample Area Size in this chapter).

The Importance of Species Composition and Developmental Stage

Information on species composition and average stage of development is necessary to take maximum advantage of biological relationships that are considered in Hopper (see VI.2). Useful information may include proportions and developmental stage of grasshopper infestations made up of known pest species, grass feeders, mixed feeders, forb feeders, or bait feeders. Environmental assessments of proposed management activities also may require such documentation.

Determine species composition by collecting with a sweep net (fig. VI.10–4) and identifying at least 50 grasshoppers from what is judged to be representative habitat. Other chapters in section VI of the User Handbook provide help in identifying grasshoppers. Because issues about habitat representation are beyond the scope of this chapter, our concern is largely reduced to the question, “How many grasshoppers do we need to identify?” We can develop some intuitive guidelines through examination of binomial confidence limits (mathematical description of confidence associated with an estimate) if we can agree on some useful examples of proportions that we will regularly encounter.

Figure VI.10–4—Catching grasshoppers in a sweep net is the first step in determining which of many species are active in a given area. (APHIS photo.)

In our experience, three to six pest species usually dominate extensive outbreaks of grasshoppers. As troublesome infestations build up over a time scale of several seasons, sweep-net samples tend to recover an increasing total number of species. Nevertheless, the proportion of individuals in the samples that are known pest species also tends to increase. Let’s consider two normal examples. First, assume that 90 percent of the grasshoppers are pest species. Second, assume that 50 percent of these grasshoppers are bait feeders (bait treatment probably will not be effective under these conditions).

Figure VI.10–5 shows 95 percent confidence limits for composition of 50 percent and 90 percent based on sample sizes ranging from 50 to 800 total grasshoppers. Notice that the highest proportion obviously is the easiest one to estimate precisely. For example, if 90 percent of a sample of 50 grasshoppers (45 of them) from 1 sample site are pest individuals, figure VI.10–5 suggests that the true proportion likely is somewhere between 78 percent and 97 percent, a range of 19 percentage points. If half of them (25) are bait feeders, the figure suggests that the true proportion is somewhere between 36 percent and 64 percent, a range of 28 percentage points.

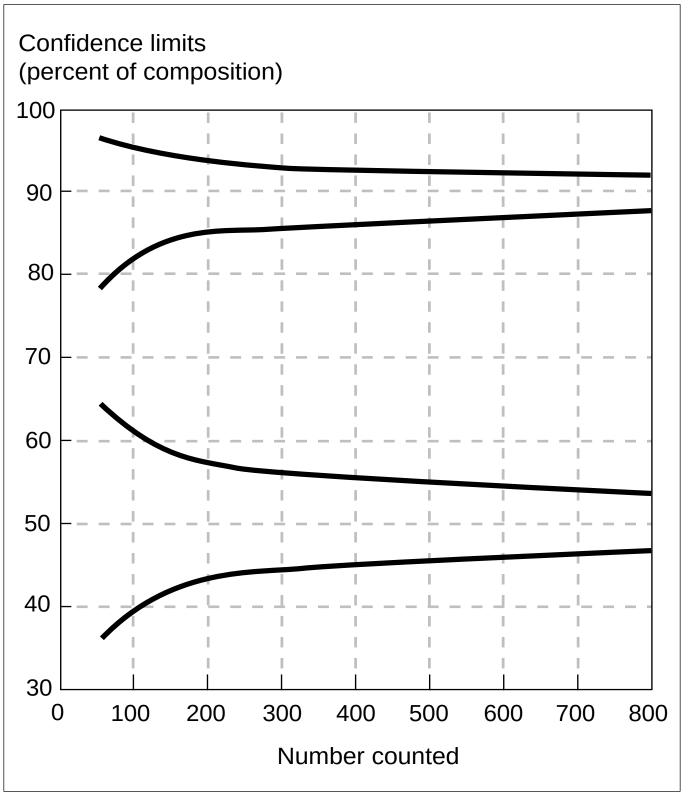

Figure VI.10–5—Confidence limits in relation to numbers of grasshoppers counted.

If those broad ranges do not inspire sufficient confidence to support a management decision, then we need to examine a larger sample or sample more sites. If our estimate of 90 percent pest species was from examination of 50 grasshoppers from each of 16 sites (720 out of 800), then the true composition is likely between 88 percent and 92 percent, a range of only 4 percentage points. Notice in figure VI.10–5 that our confidence intervals improve rapidly as sample size increases to about 200–300 grasshoppers. Notice also that minor improvements require major increases in effort when counts exceed about 400 grasshoppers.

Average stage of development usually is estimated as the summation of each observed instar number (adults are considered sixth instar for this purpose) divided by the number of individuals. Thus, for 20 fifth instars and 30 adults, the average stage is

$$\frac{(20 \times 5) + (30 \times 6)}{(20 + 30)} = \frac{100 + 180}{50} = 5.6.$$

During the nymphal survey, the stage of development is important for at least four major reasons. First, it is an indication of whether egg-hatch is completed. When very early instars predominate, it is possible that continued hatch will cause future increases in density. Second, knowing the stage of development helps to establish viable action windows. For example, if average life stage is 5.0, we know we have about 24 days until egg laying seriously begins to negate the opportunity for reducing next year's population. Third, the developmental stage is used to estimate the amount of forage destruction that can be prevented by a treatment. For any given treatment, application early in the action window should be more economical than late in the action window. Fourth, ascertaining the developmental stage correctly makes it possible to time the adult survey accurately.

In certain cases, it may be advisable to exclude particular species from the calculation of average stage of development. For example, in predicting the expected short-term response to a bait treatment, the developmental stage of grasshopper species that do not eat bait is irrelevant. Similarly, in estimating the economic benefits of a spray treatment, the developmental stage of nontarget species is not a consideration.

Future Considerations: The Potential for Sequential Sampling

Sometimes the number of grasshoppers per square foot is so low or so high that taking the full complement of required samples is a ridiculous waste of time. Under these circumstances, ranchers, university Cooperative Extension personnel, weed and pest district supervisors, and even USDA, APHIS grasshopper scouts could spend more of their sampling time on other tasks. Further, some scouts might intuitively leave a survey site before examining all samples when grasshopper densities are very low or extremely high. This is could be a perfectly valid thing to do for very busy people; in fact, it represents a crude form of something we call sequential sampling.

What is sequential sampling and how can it be used to sample grasshoppers? Well, it is the process of classifying grasshopper infestations into "high," "low," or "too close to call" categories, in sequence, from one sample to the next. Sequential sampling can save a lot of time by allowing you to stop sampling at a site when it has been determined, by a sequential sampling plan, that grasshopper densities are very low or extremely high. The technology for developing and using sequential sampling has been around for a long time, but is just now being proposed for use in grasshopper sampling.

Lower and upper grasshopper densities levels must be specified to use a sequential sampling plan. For example, we could specify grasshopper densities below which infestations are of no economic concern and above which economic concern may be justified. The computer program Hopper will allow you to calculate economic thresholds so that you can generate these upper and lower density levels.

Using sequential sampling, three possibilities exist after each sample: (1) density could be declared less than a lower level, say, 8/yd^2; (2) density could be declared greater than an upper level, say, 16/yd^2; or (3) no such decision may be concluded. When the first or second decision is made, sampling can stop because the infestation has been classified. When the third situation occurs, examination of another sample is mandated.

If a classification is not made within some arbitrary number of samples (say, within 18 samples), then sampling can stop and the grasshopper infestation is declared as being between the two levels. If this third decision occurs at most survey sites, use sequential sampling at a later date to determine whether the population has changed. Note that the total number of sample areas at 1 survey site can range from 1 to 18 in our example.

The advantages sequential sampling are several:

- It will save time when actual densities are either well above or well below the upper and lower levels.
- It reduces the number of samples at most survey sites.
- It allows the sampler to predetermine the proportions of decisions that will be correct. For example, a person could specify that at least 9 of 10 sites be correctly classified.
- It can be used to delimit the borders of grasshopper infested areas.

But sequential sampling also has some disadvantages:

- Density estimates will be less precise if sequential sampling is used and a classification is reached with a low number of samples.
- Some erroneous classifications cannot be avoided.
- A table must be consulted to know when to stop sampling.

How To Conduct a Sequential Sampling Effort.— Sequential sampling can be conducted by either counting all grasshoppers or by simply noting their presence or absence (presence–absence sequential sampling) in successive samples. Here, we offer an example of the presence–absence method.

In Wyoming, there is a need to develop a grasshopper sampling plan for use by ranchers, Cooperative Extension system personnel, and weed and pest district employees. The objective is to help these individuals rapidly decide if grasshopper densities are less than 8/yd^2 (no cause for concern), greater than 16/yd^2 (potential cause for concern), or in between (worth watching). These levels of grasshopper densities may be referred to as the lower and upper thresholds, respectively. Also, we can set these thresholds to any values that are appropriate for a specific situation.

In this example, we will use a visualized sample area defined by folding a sheet of 8 1/2- × 11-inch paper into an 8 1/2- × 8 1/2-inch square (0.5 ft^2). Once you have calibrated your eyes to the 8 1/2- × 8 1/2-inch square, take a copy of table VI.10–2 and examine the first sample at a survey site. If it contains no grasshoppers, write a zero in the “Running total” slot opposite sample number 1 (as shown in table VI.10–3, example A).

If there are no grasshoppers present in the second sample area, then add zero to the previous running total and enter zero in the “Running total” slot for “Sample area” number 2, as shown in table VI.10–3, example A. However, if at least one grasshopper is present in the second sample area, then add 1 to the previous running total and enter 1 in the “Running total” slot for “Sample area” number 2, as shown in table VI.10–3, example B. This new running total is then compared to the lower and upper stop values. Each time a sample area contains at least one grasshopper, add 1 to the running total. A minimum of four

Table VI.10–2—Presence–absence sequential sampling stop values for levels of 8 and 16 grasshoppers/yd^2, assuming samples areas are 0.5 ft^2 each. Note that other sample area sizes cannot be used with this table.

Sample number	Lower stop value	Running total	Upper stop value
1	—	_____	3
2	—	_____	3
3	—	_____	4
4	0	_____	4
5	0	_____	5
6	1	_____	5
7	1	_____	6
8	1	_____	6
9	2	_____	7
10	2	_____	7
11	3	_____	8
12	3	_____	8
13	4	_____	8
14	4	_____	9
15	5	_____	9
16	5	_____	10
17	6	_____	10
18	6	_____	11

samples is needed in this case to yield a running total that is potentially less than or equal to the lower stop value or is greater than or equal to the upper stop value. If either case is true, you can stop sampling and declare the infestation as being 8 or fewer per square yard or 16 or more per square yard, respectively. Thus, the sampling process repeats itself until one of the following occurs:

- The running total is equal to or less than the lower stop value (table VI.10–3, example A),
- The running total is equal to or greater than the upper stop value (table VI.10–3, example B), or
- A density classification has not been made after the 18 samples have been examined (table VI.10–3, example C).

Corresponding decisions about grasshopper infestations for this example may be found at the bottom of table VI.10–3.

As mentioned, you also can do sequential sampling by counting each grasshopper in each sample area. If this is done, the sampler must keep a running total of the number of grasshoppers counted, and the stop values used are different from those shown in table VI.10–2. This kind of sequential sampling would be useful in delimiting surveys where grasshopper density estimates are needed.

If sequential sampling is to be used throughout a State or region, then flexible methods for choosing realistic lower and upper thresholds must be developed.

Future Considerations: Electronics

Electronic mapping, using geographic information systems (GIS) (see VI.9) may be very useful for grasshopper survey. For example, maps produced using GIS are useful for historical perspectives, analyses of ecological correlates (such as topography, vegetation, and soil), planning surveys, and allocating limited resources. GIS also will allow maps to be updated daily during a survey. We can use these maps to focus the survey effort on the most important areas as the season unfolds.

Computer-interpolated maps of grasshopper densities can be combined with land-use maps, ecological buffer zone maps, and land ownership maps to produce final treatment area maps. GIS software also can calculate the size of any defined area on an electronic map. These maps can be printed on paper to be used in the field or for display at meetings.

Economical battery-powered, hand-held computers hold much promise for grasshopper surveys. Scouts recently have used these types of computers in the field to enter and store data. These data can be transmitted through normal telephone lines to a computer centrally located in each State. Sequential sampling protocols, described earlier in this chapter, could be programmed into these computers. The user would simply enter the number of grasshoppers in each sample area, and the computer could store and analyze the data and notify the user when to stop sampling.

Other types of electronic data-collection equipment being used at some sites store environmental data important for

Table VI.10–3—Three examples of using a presence–absence sequential sampling plan

Example A				Example B				Example C			
Sample area	Lower stop value	Running total	Upper stop value	Sample area	Lower stop value	Running total	Upper stop value	Sample area	Lower stop value	Running total	Upper stop value
1	—	0	3	1	—	0	3	1	—	0	3
2	—	0	3	2	—	1	3	2	—	0	3
3	—	0	4	3	—	2	4	3	—	0	4
4	0	0	4	4	0	3	4	4	0	1	4
5	0	[quit]	5	5	0	4	5	5	0	2	5
6	1		5	6	1	5	5	6	1	2	5
7	1		6	7	1	[quit]	6	7	1	2	6
8	1		6	8	1		6	8	1	3	6
9	2		7	9	2		7	9	2	4	7
10	2		7	10	2		7	10	2	4	7
11	3		8	11	3		8	11	3	4	8
12	3		8	12	3		8	12	3	5	8
13	4		8	13	4		8	13	4	5	8
14	4		9	14	4		9	14	4	6	9
15	5		9	15	5		9	15	5	6	9
16	5		10	16	5		10	16	5	7	10
17	6		10	17	6		10	17	6	7	10
18	6		11	18	6		11	18	6	8	11
Decision: Infestation is less than 8 grasshoppers/yd^2.				Decision: Infestation is greater than 16 grasshoppers/yd^2.				Decision: Infestation is between 8 and 16 grasshoppers/yd^2.			

grasshopper research and management. These devices automatically log information, such as temperature and precipitation, for weeks at a time without human intervention. Technology that allows a computer to read hand-written data directly from data sheets is also becoming available. A scout could use a standard pen and clipboard to record the data on a printed data sheet in the field. The data sheet could then be faxed directly to a waiting computer or delivered to a site with a page scanner and scanned into a computer. In both cases, software could read the image made from the data sheet, interpret the information, and automatically store it in a data base that corresponds to the specific data sheet. Paper data sheets would be inexpensive, familiar, and highly reliable for field data entry. Data still could be rapidly acquired and distributed for use in management decisions.

Another technology that is already showing usefulness for rangeland grasshopper management is Global Positioning System (GPS). With GPS, hand-held units receive information from navigational satellites and calculate the location coordinates of the unit. Surveyors can obtain latitude and longitude coordinates even for the most remote sites where there are no distinguishing landmarks. A computer can use these coordinates to map any data collected at the site. Also, the hand-held units help a person navigate back to a site.

High-quality survey data always will be the basis for sound management decisions. Most of these data will be collected by humans working under various conditions in the field. This chapter provides reference for current survey activities and a starting place for future innovations in survey technology.

VI.11 Major Grasshopper Species of the Western Rangeland States and Alaska

R. Nelson Foster

On rangeland, the number of grasshopper species that occur across an area of several thousand acres typically ranges from about 15 to 40. Assemblages of grasshopper species in each of the western rangeland States can differ considerably. The makeup of these assemblages also can vary between locations within a State and from year to year at the same location.

To make wise management and treatment decisions requires a knowledge of the species that make up the populations of concern. To aid land managers and pest managers in making their decisions, the Animal and Plant Health Inspection Service plant health directors in the rangeland States recently provided a listing of major grasshopper species in the States.

The listing is a combination of responses to two questions asked of each plant health director on separate occasions: (1) What are the 10 most important grasshopper species in your State? and (2) what are the top 10 pest species of grasshoppers in your State? Species are listed alphabetically in table VI.11–1 with full names, and listed by occurrence in States in table VI.11–2. The listings will be especially useful in combination with Pfadt's "Field Guide to Common Western Grasshoppers" (described in VI.5) and Hopper Helper (VI.7).

The two species that occurred most frequently (16 out of 18 States) in responses are *Ageneotettix deorum* and *Melanoplus sanguinipes.* Next in terms of frequency are *Aulocara elliotti* (in 14 out of 18 States) and *Camnula pellucida* (13 out of 18 States). Four other species—*Melanoplus bivittatus* and *Melanoplus femurrubrum* (both 11 out of 18 States) and *Amphitornus coloradus* and *Phlibostroma quadrimaculatum* (both 10 out of 18 States)—are of concern in a majority of rangeland States. All other species in these surveys were of concern in fewer than 10 States.

The lists are not limited to species that cause economically unacceptable levels of damage. Grasshoppers noted on the lists include the most commonly encountered species in each State, some of which may not be considered economically damaging to rangelands.

Some species usually considered nonpests are included because they may occur in significant numbers at some

Table V.11–1—Grasshopper species most frequently encountered and pest species (with full names), listed alphabetically

Aeropedellus clavatus (Thomas)
Ageneotettix deorum (Scudder)
Amphitornus coloradus (Thomas)
Arphia conspersa Scudder
Aulocara elliotti Thomas
Aulocara femoratum (Scudder)
Camnula pellucida Scudder
Campylacantha olivacea (Scudder)
Conozoa sulcifrons Scudder
Cordillacris crenulata (Bruner)
Cordillacris occipitalis (Thomas)
Eritettix simplex (Scudder)
Hesperotettix viridis (Scudder)
Melanoplus angustipennis (Dodge)
Melanoplus bivittatus (Say)
Melanoplus borealis (Fieber)
Melanoplus confusus Scudder
Melanoplus cuneatus Scudder
Melanoplus devastator Scudder
Melanoplus differentialis (Thomas)
Melanoplus femurrubrum (DeGeer)
Melanoplus foedus Scudder
Melanoplus gladstoni Scudder
Melanoplus infantilis Scudder
Melanoplus marginatus (Scudder)
Melanoplus occidentalis (Thomas)
Melanoplus packardii Scudder
Melanoplus rugglesi Gurney
Melanoplus sanguinipes (Fabricius)
Mermiria bivittata (Serville)
Metator pardalinus (Saussure)
Oedaleonotus enigma (Scudder)
Oedaleonotus pacificus (Scudder)
Opeia obscura (Thomas)
Orphulella speciosa (Scudder)
Phlibostroma quadrimaculatum (Thomas)
Phoetaliotes nebrascensis (Thomas)
Psoloessa delicatula Scudder
Schistocerca emarginata (Scudder)
Syrbula admirabilis Uhler
Trachyrhachys kiowa Thomas
Xanthippus corallipes Haldeman

Table VI.11–2—Major grasshopper species of the western rangeland States and Alaska

Species	AK	AZ	CA	CO	ID	KS	MT	NB	NV	NM	ND	OK	OR	SD	TX	UT	WA	WY
Gomphocerinae																		
Aeropedellus clavatus											x							
Ageneotettix deorum		x		x	x	x	x	x	x	x	x	x	x	x	x	x	x	x
Amphitornus coloradus		x				x	x			x	x	x		x	x		x	x
Aulocara elliotti		x			x	x	x		x	x	x	x	x	x	x	x	x	x
Aulocara femoratum			x	x		x				x		x			x			
Cordillacris crenulata				x						x								
Cordillacris occipitalis				x						x								x
Eritettix simplex		x						x			x							
Mermiria bivittata		x																
Opeia obscura				x		x		x			x			x				
Orphulella speciosa						x												
Phlibostroma quadrimaculatum				x	x	x	x	x		x	x			x	x			x
Psoloessa delicatula		x						x										
Syrbula admirabilis						x												
Oedipodinae																		
Arphia conspersa			x															
Camnula pellucida	x	x	x	x	x		x		x	x	x		x			x	x	x
Conozoa sulcifrons																	x	
Metator pardalinus		x																
Trachyrhachys kiowa				x			x				x	x		x				x
Xanthippus corallipes			x													x		
Melanoplinae																		
Campylacantha olivacea								x										
Hesperotettix viridis										x			x					
Melanoplus angustipennis								x										
Melanoplus bivittatus			x	x	x		x		x	x		x	x		x	x	x	
Melanoplus borealis	x																	
Melanoplus confusus											x							
Melanoplus cuneatus		x								x								
Melanoplus devastator			x															
Melanoplus differentialis			x		x							x			x	x		
Melanoplus femurrubrum	x		x		x			x	x			x	x	x	x	x	x	
Melanoplus foedus					x							x	x		x		x	
Melanoplus gladstoni								x										
Melanoplus infantilis					x		x				x			x				x
Melanoplus marginatus			x															
Melanoplus occidentalis																		x
Melanoplus packardii			x		x	x	x		x	x			x			x	x	
Melanoplus rugglesi									x									
Melanoplus sanguinipes	x	x	x	x	x		x		x	x	x	x	x	x	x	x	x	x

Table VI.11–2—Major grasshopper species of the western rangeland States and Alaska (Continued)

Species	AK	AZ	CA	CO	ID	KS	MT	NB	NV	NM	ND	OK	OR	SD	TX	UT	WA	WY
Oedaleonotus enigma			x		x				x				x			x	x	
Oedaleonotus pacificus			x															
Phoetaliotes nebrascensis						x		x						x				
Cyrtacanthacridinae																		
Schistocerca emarginata			x															

Note: The importance of some species in some States has changed over the years. For a comparison with a 1969 listing of species and their potential for damage by State, see: Grasshopper Survey: A Species Field Guide, published in 1969 by the U.S. Department of Agriculture, Animal and Plant Health Inspection Service, Plant Protection and Quarantine unit. Copies of the 1969 publication are available from the National Technical Information Service, U.S. Department of Commerce, P.O. Box 1425, Springfield, VA 22151. The publication, number P95241436, is available in print for $19.50 and on microfiche for $9.00.

sites during survey. For example, overwintering species such as *Psoloessa delicatula, Eritettix simplex, Xanthippus corallipes,* and *Arphia conspersa*—which rarely if ever cause concern—may occur in significant numbers late in the summer. The early hatching species, *Aeropedellus clavatus* and *Melanoplus confusus,* feed at a time of year when forage removal is generally irrelevant. These two species are included because they are common in some areas and signal the awakening of the grasshopper season.

Hesperotettix viridis, which feeds on broom snakeweed and burrowweed, is considered a beneficial species but is included because it can occur in high numbers at some locations. In addition, some species usually considered to be cropland species—such as *Melanoplus bivittatus, M. differentialis*, *M. femurrubrum,* and *M. packardii*—are frequently found on rangeland and hence are included.

The circumstances under which a species or a combination of species occurs is what determines the economic importance of a particular species at a particular time. By themselves, many of the species listed here would not be economic pests, but together with other species, the population may cause damage.

A knowledge of the most commonly encountered species in each State will promote a better understanding of the grasshopper populations and will provide the foundation for making good management and pest treatment decisions involving rangeland grasshoppers.

VII. Future Directions

Grasshoppers that die from exposure to fungi in the *Entomophaga* genus characteristically do so in a heads-up position. The GHIPM Project studied the use of *Entomophaga grylli* in the hope of lessening overall dependence on chemical control to manage rangeland grasshopper populations. (APHIS file photo.)

VII.1 Future Directions in Grasshopper Management—An Introduction

Jerome A. Onsager

The following chapters about future directions in grasshopper management fall into three general categories. These categories can be described as potential new agents for grasshopper suppression, emerging new ecological information that could be integrated into grasshopper management systems, and issues that could affect grasshopper management priorities, especially on public lands.

Chapters VII.2, 3, 5, 6, 7, and 8 discuss a number of promising new candidate agents for conventional short-term control of economic infestations of grasshoppers. The agents' eventual viability will be dictated primarily by the same practical elements that affect current control tactics. These elements include

- approval by regulatory agencies,
- reasonable production costs plus economical volume for the producer,
- reasonable shelf life and consistency of demand for the distributor, and
- satisfaction plus reasonable profit for the consumer.

A candidate agent that is deficient in any critical element will not compete strongly with current technology until the deficiency is corrected.

Chapters in this section also discuss two exotic biological control agents that were considered by the Federal Government for nonconventional long-term suppression of grasshopper populations. Grasshopper Integrated Pest Management (GHIPM) Project scientists evaluated a fungal pathogen (chapter VII.4) and an egg parasite (chapter VII.9) from Australia as candidates for release in the United States to build a reservoir of biological control. Such a strategy, called inoculative release, appeals to some pest managers because the organisms could become self-perpetuating and therefore permanent deterrents to grasshopper populations. Conversely, inoculative release is worrisome to others because it could produce undesirable side effects that also could become permanent. At this time, it appears unlikely that current regulatory guidelines will allow the release of the two exotic agents.

Chapters VII.11, 13, 14, and 15 discuss areas of unfinished long-term research on grasshopper ecology. Hopefully, the finished products someday will be incorporated into improved land-management systems. An understanding of how grasshoppers respond to controllable attributes of habitat can be exploited in management systems that reduce the frequency and intensity of grasshopper depredation.

Finally, this handbook would be incomplete without some direct input into the complex and competing social, political, and environmental issues that affect grasshopper management on public lands. Chapters VII.10, 12, and 16 are contributions that obviously are within the competence and responsibility of GHIPM and are of interest to the Project. The information is intended not to provide definitive solutions to problems but rather to be available when conflicts of interest must be resolved.

VII.2 Dimilin® Spray for Reducing Rangeland Grasshopper Populations

R. N. Foster and K. Christian Reuter

Introduction

The insecticides used to control outbreaks of grasshoppers on rangeland are active against a broad spectrum of insects, in both adult and immature stages. For rangeland use in Animal and Plant Health Inspection Service (APHIS) cooperative programs, pest managers apply insecticides at doses and in formulations that have a minimal but acceptable impact on nontarget insects while substantially reducing grasshoppers. Because their activity is broad, these insecticidal sprays sometimes reduce some nonpest insect species in the target areas. However, populations of nontargets have been seen to rebound relatively quickly following treatments on rangeland, even over large areas (see chapter III.3, "Impact of Control Programs on Nontarget Arthropods"). While undesirable, the effects of these sprays on nontarget insects are acceptable. Short-term reductions in nontargets are part of the price pest managers currently pay for artificially bringing an outbreak of grasshoppers back to a normal level.

The goals of insect control today are rapidly expanding. It is environmentally advantageous to reduce the minimal effects of sprays on nontargets even further. Increasing protection to nontargets, particularly those that naturally work to keep grasshopper populations in balance, supports basic integrated pest management (IPM) objectives that encourage and emphasize the use of naturally occurring organisms.

Some insecticides, called insect growth regulators, have a narrower spectrum of activity and cause death in a manner different from most broad-spectrum insecticides. The Dimilin® brand of diflubenzuron, (1-(4-clorophenyl)-3-(2,6 diflourobenzoyl)-urea, is one of these growth regulators. It inhibits chitin biosyntheses and thereby interferes with the formation and deposition of the chitin in the cuticle in an insect exoskeleton. This disruption of normal development may result in death to the insect when molting is attempted.

Diflubenzuron has been shown to be effective against immature stages of several insect pests and is registered in the United States for control of beet armyworm, fall armyworm, and boll weevil on cotton, several insects on soybean, several forest pest insects, and in California on mosquito larvae. Because of its mode of action, nonchitin-forming animals and adult insects and spiders enjoy a reduced risk compared to that of conventional insecticides.

Several studies have been conducted with Dimilin formulated into a bran-based bait for grasshoppers. Wang and Fuller (1991) demonstrated the effectiveness of 1 and 2 lb of 1 percent diflubenzuron bran bait per acre against rangeland grasshoppers on 12-acre plots in southwestern South Dakota. Bomar and Lockwood (1991) demonstrated the effectiveness of the same formula and rate against rangeland grasshoppers on 10-acre plots in eastern Wyoming. Both of these studies utilized ground equipment for application. In two 2-year studies where bait was aerially applied to replicated 40-acre plots, Jech et al. (1993) showed diflubenzuron and carbaryl bran bait treatments to be equally effective on mixed populations of grasshoppers. (Figures VII.2–1 and –2 illustrate technical challenges in using bran materials in aerial spray programs.) However, the study indicated that the species *Phlibostroma quadrimaculatum* (Thomas) could be controlled with diflubenzuron when not controlled with carbaryl bait.

Results of these studies are very promising. However, some damaging species of grasshopper do not readily accept the bran baits and may remain at undesirable levels (Jech et al. 1989 unpubl., 1992 unpubl., and 1993; Onsager et al. 1990; Quinn et al. 1989). Additionally, levels of reduction with all bran-based baits on susceptible species tend to be lower when compared to spray treatments that are deposited directly on both the pest and the preferred food of the pest.

In an effort to take advantage of the desirable qualities of Dimilin while avoiding the general limitations of bran baits, APHIS scientists at the Phoenix Methods Development Center studied spray formulations. Compared to currently used broad-spectrum insecticides, Dimilin should lessen the impact on those nontarget insects and arachnids that are in an adult stage at the time the grasshoppers are treated.

Figure VII.2–1—A load of bran is delivered for onsite mixing with chemicals or insect growth regulators at an airstrip in the Dakotas. (Agricultural Research Service photo by John Kucharsky.)

Evaluating Potential Treatment Rates—A Field Study

In 1991 we conducted a detailed study to (1) generally evaluate an aerially sprayed formulation for control of grasshoppers on rangeland, (2) determine the most effective dose of three candidate doses for achieving immediate and seasonlong effectiveness on both the total grasshopper population and the individual component species of the population, and (3) determine the usefulness of the treatments for suppression or controlling migration into the treated area during the season of treatment.

In this study, we applied three doses of Dimilin 25W spray in volumes of 32 oz/acre to 40-acre mixed-grass rangeland plots in western South Dakota. Three sets of plots were treated with Dimilin spray at 0.015, 0.030, and 0.045 lb active ingredient (AI) per acre. An additional set of plots was sprayed with the standard carbaryl rangeland treatment (Sevin®-4 Oil ULV at 0.5 lb AI/acre) for comparison. A fifth set of plots was left untreated. When applications were made, most grasshoppers were in the second or third instar.

We found that all three dosages of Dimilin caused reduction as great as the standard carbaryl treatment after 1 week. After 2 weeks, all treatments showed reduction in the range of 94 to 96 percent. Reductions continued to increase to the end of the study and 9 weeks after treatment ranged from 96 to 98 percent.

Overall, we found no differences in the effects of Dimilin and carbaryl. Dimilin showed almost immediate acceptable reduction of grasshoppers within 7 days and continued to be effective throughout the season of treatment. Measurable migration into the Dimilin-treated plots was undetectable. Surviving hatch that might have occurred was also undetectable. In this study, in terms of providing acceptable control, Dimilin proved to be an excellent alternative for consideration when treating grasshoppers on rangeland.

Figure VII.2–2—The treated bran bait is sacked and then dumped into a chamber in the fuselage of the spray plane. Inside that chamber, APHIS-developed aerating equipment keeps the bran bait from clumping, which would cause uneven applications of product. (Agricultural Research Service photo by John Kucharsky.)

Use of Dimilin Spray Under Operational Conditions

In 1993, we conducted a study to evaluate the usefulness of two formulations of Dimilin for control of grasshoppers on rangeland under operational conditions that could be experienced during a large-scale grasshopper control program. In this study, we aerially applied Dimilin 25W, Dimilin 2F, and carbaryl (Sevin-4 Oil ULV) to mixed-grass rangeland plots in western North Dakota. All three formulations were sprayed in a diesel carrier. We applied each treatment to a square 640-acre block. Both Dimilin treatments were applied at the dose of 0.0156 lb AI/acre in 32 fluid oz of mix. The carbaryl treatment was applied in 20 fluid oz of mix per acre at the dose of 0.5 lb AI and was used as a standard rangeland treatment for comparison. We compared reduction in grasshopper populations within the operational plots to populations of untreated grasshoppers in adjacent areas surrounding the treated plots. Most grasshoppers treated were in the second or third instar.

We found that the standard (Sevin-4 Oil ULV) treatment caused greater reductions in grasshoppers after 1 week than the Dimilin treatments. After 2 weeks, all three treatments caused reductions in grasshoppers that would be acceptable in large-scale program efforts. However, the Dimilin 2F and carbaryl treatments were causing greater reductions than the Dimilin 25W. Mortality at 3 weeks after application showed that all three treatments were performing equally well. After 4 weeks, we found that the Dimilin 2F formulation caused greater reductions in grasshoppers compared to the other treatments. Trends in our study showed that mortality increased over the 4 weeks after treatment with Dimilin 2F and started to decline with Dimilin 25W and Sevin-4 Oil ULV between the second and third week after treatment.

From a cursory examination of the study area 16 weeks after treatment, we found that no obvious additional hatch had survived, nor had any migration into the treated area occurred. Densities of grasshoppers were no greater than at 4 weeks after treatment.

In our operational study, the 2F formulation proved to be more compatible with the spraying system. The 25W formulation mixed with diesel resulted in a precipitant that could potentially cause a clogging problem with the spraying system and made cleanup significantly more difficult.

Results from our study demonstrated that a low amount of Dimilin active ingredient per acre with the 2F formulation can be used in a large-scale control program manner for control of grasshoppers on rangeland. Upon final Environmental Protection Agency registration, Dimilin—because of its mode of action and its reduced spectrum of activity—could be an attractive option to be considered for controlling grasshoppers on rangeland.

References Cited

Bomar, R.; Lockwood, J. A. 1991. Evaluation of Dimilin bran-bait for rangeland grasshopper control, 1990. Insecticide and Acaricide Tests 16: 212–213.

Jech, L. E.; Foster, R. N.; Colletto, D.; Walgenbach, D. D.; Roland, T. J.; Rodriguez, G. D.; Bohls, R.; Houston, R. D.; Meeks, W. K.; Queener, R. L.; Jackson, C. L.; Dines, J. L.; Puclik, M. J.; Scott, A. K. 1993. Field evaluation of diflubenzuron and carbaryl bran baits against grasshopper (Orthoptera: Acrididae) populations in South Dakota. Journal of Economic Entomology 86: 557–565.

Onsager, J. A.; Henry, J. E.; Foster, R. N.; Staten, R. T. 1980. Acceptance of wheat bran bait by species of rangeland grasshoppers. Journal of Economic Entomology 73: 548–551.

Quinn, M. A.; Kepner, R. L.; Walgenbach, D. D.; Bohls, R. A.; Pooler, P. D.; Foster, R. N.; Reuter, K. C.; Swain, J. L. 1989. Immediate and 2nd-year effects of insecticide spray and bait treatments on populations of rangeland grasshoppers. Canadian Entomologist 121: 589–602.

Wang, T.; Fuller, B. W. 1991. Evaluation of Dimilin 1% bran bait for grasshopper control on rangeland, 1990. Insecticide and Acaricide Tests 16: 213.

References Cited—Unpublished

Jech, L. E.; Foster, R. N.; Colletto, D.; Walgenbach, D. D.; Bohls, R.; Rodriguez, G.; Burgess, J.; Meeks, W. K.; Queener, R. 1989. Field trials of chlorpyrifos and carbaryl bran baits for control of grasshoppers on rangeland near Buffalo, South Dakota, 1988. In: Cooperative Grasshopper Integrated Pest Management Project, 1989 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 194–206.

Jech, L. E.; Foster, R. N.; Reuter, K. C.; Hirsch, D.; Cushing, W.; Walgenbach, D. D.; Bohls, R.; Herbaugh, L. L. 1992. Response of grasshopper species to carbaryl bran bait treatments. In: Cooperative Grasshopper Integrated Pest Management Project, 1992 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 83–89.

VII.3 Impact of Dimilin® on Nontarget Arthropods and Its Efficacy Against Rangeland Grasshoppers

Michael A. Catangui, Billy W. Fuller, and Arnie W. Walz

Introduction

Dimilin® (diflubenzuron) is a chitin-synthesis inhibitor and causes death in insects during the molting process (van Daalen et al. 1972, Post and Vincent 1973). Chitin, a nitrogenous polysaccharide, is the organic foundation of the exoskeleton of all insects and the entire phylum Arthropoda (Snodgrass 1935). Therefore, some concern exists that widespread use of diflubenzuron may affect not only the target insect pest but also nontarget arthropods that are essential for the diversity and stability of rangeland ecosystems. Other studies have shown the potential of diflubenzuron against rangeland grasshoppers (Foster et al. 1991 unpubl. and 1993 unpubl.).

Our key research objective was to determine whether diflubenzuron negatively affected the abundance and diversity of nontarget arthropods (including ants, spiders, predatory beetles, and pollinator bees) in rangelands, and if so to determine if the effect was greater than the effect of one of the current standard treatments. Another research objective was to develop additional data on the potential of diflubenzuron as an alternative insecticide against rangeland grasshoppers.

Previous studies indicate that diflubenzuron spares most nontarget arthropods. Ables et al. (1975) reported diflubenzuron to be harmless to a pupal parasitoid of the house fly. Compared to dimethoate-treated poultry farms in North Carolina, diflubenzuron-treated farms had greater parasitoid abundance and species diversity. In cotton fields, Keever et al. (1977) observed that arthropod predators belonging to orders Hemiptera, Coleoptera, and Neuroptera were not affected by diflubenzuron when it was sprayed aerially at 0.12 lb active ingredient (AI) per acre (0.14 kg AI per hectare). Wilkinson et al. (1978) evaluated various rates and formulations of diflubenzuron on adult and immature stages of selected parasitoids and predators found in cotton fields. The authors found test insects to be unaffected by diflubenzuron even at high concentrations except for immatures of a lacewing species.

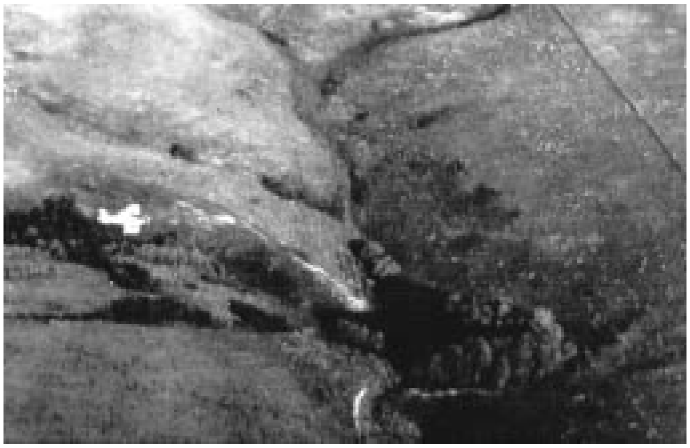

Figure VII.3–1—To minimize insecticide drift, spray booms are turned off well before this plane flies over a pond in the Great Plains. (Agricultural Research Service photo by John Kucharsky.)

In contrast, diflubenzuron may be detrimental to some freshwater crustaceans and immature aquatic insects (fig. VII.3–1). Miura and Takahashi (1974, 1975) observed temporary population reductions in tadpole shrimp, clam shrimp, water fleas, copepods, cladocerans, mayfly naiads, and midge larvae in treated (0.1 lb AI per acre) mosquito breeding areas in California. Adult aquatic beetles, spiders, and mosquito fish were not affected by diflubenzuron even at the highest rates tested. Farlow et al. (1978) studied the impact of diflubenzuron on nontarget organisms of a Louisiana coastal marsh. Those authors reported significant reductions in amphipods, dragonfly naiads, nymphs of corixid and notonectid bugs, as well as adult hydrophilid beetles in marshlands treated six times with 0.025 lb AI per acre (28 g AI per ha) over an 18-month period. On the other hand, significant increases were observed among mayfly naiads, larvae of noterid and dytiscid beetles, adult corixid bugs, and mosquito fish. Numerous immature and adult insects were listed as unaffected by the diflubenzuron treatments.

The environmental fate and degradation of diflubenzuron in a laboratory model ecosystem, a soil bacterium, sheep liver microsomes, and ultraviolet light were investigated by Metcalf et al. (1975). They found diflubenzuron to be moderately persistent in organisms such as algae, snails, caterpillars, and mosquito larvae but efficiently degraded by mosquito fish, however. Ecological magnification may not be a problem: the lowest concentration of diflubenzuron was found in the mosquito fish, at the top of the model food chain. Sheep liver microsomes and the soil bacterium were not able to degrade diflubenzuron under the experimental conditions imposed.

Other studies have shown the potential of diflubenzuron against rangeland grasshoppers. Foster et al. (1991 unpubl.) reported aerial treatments of diflubenzuron spray at 0.015, 0.030, and 0.045 lb AI per acre to reduce second- and third-instar grasshoppers as well as a standard treatment of carbaryl (0.5 lb AI per acre) after 1 week. Foster's team showed reductions for all treatments in the range of 94 to 96 percent after 2 weeks. Under simulated control program operational conditions, Foster et al. (1993 unpubl.) reported two formulations of diflubenzuron at 0.0156 AI per acre and a carbaryl standard performed equally well (control ranged from 87 to 91 percent).

Our Study in South Dakota

Different rates and formulations of were tested in an open rangeland near Ludlow (Harding County), SD, during the 1993 season. Dimilin 2F (0.0075 and 0.015 lb AI/acre) and Dimilin 25W (0.015 lb AI/acre) were compared with Sevin® 4-Oil (0.5 lb AI/acre) and untreated plots. The lower rate of Dimilin 2F was evaluated only for efficacy against grasshoppers. The remaining treatments were evaluated for impact on nontarget arthropods *and* efficacy against grasshoppers. We used a completely randomized design with each treatment replicated four times. A fixed-wing airplane applied chemical treatments over 40-acre plots from July 2 to July 7, 1993.

Our study used pitfall traps to sample soil surface-associated nontarget arthropods (ants, spiders, predatory beetles, and scavenger beetles). A pitfall trap consisted of a wide-mouth 1- qt canning jar filled with approximately 4 inches of mineral oil. Each pitfall trap was buried so that the opening was flush with the soil surface. The oil killed and temporarily preserved crawling insects that fell into the traps. Six pitfall traps spaced 15 ft apart and arranged in hexagonal pattern were installed near the center of each 40-acre plot.

Malaise traps were used to sample flying nontarget arthropods such as parasitic and predatory wasps, lacewings, flies, and pollinator bees. Each malaise trap was a 12- by 4- by 6-ft rectangular tent made of nylon screen that intercepted and directed flying insects to killing jars. Two malaise traps were placed near the center of each 40-acre plot.

We used rings to count live grasshoppers (fig. VII.3–2). Forty aluminum rings, each 0.1 m^2, were arranged in grids near the center of each plot. We counted grasshoppers within each ring using a tally counter. Sweep-net samples determined grasshopper species and their age composition.

Figure VII.3–2—A grasshopper's eye view of the kind of ring field crews use to delimit a sampling spot before counting resident 'hoppers. (Agricultural Research Service photo by John Kucharsky.)

Sampling for nontarget arthropods was carried out before and after treatment application. The malaise and pitfall traps were run a week before treatment, then resumed 1 week after the last chemical treatment application. Traps were maintained continuously thereafter, and catches were collected at weekly intervals for 10 weeks from July to September. Plot and trap location markers remained onsite over the winter months, and an additional sample was collected about 1 year after treatment. We took grasshopper counts from rings and sweep-net samples (fig. VII.3–3) once before chemical treatment and at weekly intervals for 7 weeks after treatment. Additional grasshopper counts and samples were taken the end of season (11 weeks after treatment).

We sorted nontarget arthropod samples and counted them in the laboratory. Arthropods were identified to family level then grouped according to their biological function (such as predator, parasite, scavenger, or pollinator). Identification of ants to the species level (Wheeler and Wheeler 1963) was used to calculate a measure of species diversity referred to as the probability of interspecific encounter (PIE) (Hurlbert 1971, Washington 1984).

Figure VII.3–3—Sweep-netting grasshoppers is a labor-intensive but time-tested method for sampling insect populations. (Agricultural Research Service photo by John Kucharsky.)

Hurlbert defined PIE as the probability that two individuals encountered at random in a community will belong to different species. In our present paper, PIE may be interpreted as the probability that two individual ants randomly encountered in rangeland will be of different species. The higher the probability, the more diverse, and presumably more stable, is the ant community.

Findings and Discussion

Arthropods collected from the experimental site were grouped arbitrarily as follows: (1) soil surface-associated nontarget arthropods, (2) flying nontarget arthropods, and (3) grasshoppers. Each group was sampled using techniques appropriate for their mobility and biological characteristics.

Impact of Dimilin on Soil Surface-Associated Nontarget Arthropods.—There were four major groups of soil surface-associated arthropods: (1) ants (order Hymenoptera: family Formicidae), (2) spiders (order Araneae: families Agelenidae, Amaurobiidae, Clubionidae, Dictynidae, Gnaphosidae, Hahniidae, Lycosidae, Mimetidae, Philodromidae, Salticidae, Tetragnathidae, Theridiidae, and Thomisidae), (3) predatory beetles (order Coleoptera: families Carabidae, Cicindelidae, Histeridae, Meloidae, and Staphylinidae), and (4) scavenger beetles (order Coleoptera: families Scarabaeidae, Silphidae, and Tenebrionidae).

In terms of biological function on the rangeland ecosystem, ants may be regarded as both general predators and scavengers (Wheeler and Wheeler 1963). All spiders are predators (Kaston 1972). Beetles belonging to families Carabidae (ground beetles), Cicindelidae (tiger beetles), Staphylinidae (rove beetles), and Histeridae (hister beetles) are also general predators (Borror and DeLong 1964). Blister beetle (Meloidae) larvae feed on grasshopper eggs, but adults are considered pests of certain crops. Scavengers were composed of families Scarabaeidae (scarab beetles), Silphidae (carrion beetles), and Tenebrionidae (darkling beetles). Certain scarabs like the dung beetle feed on cattle manure; carrion beetles feed on dead animal carcasses. Darkling beetles feed on decaying plant materials but some, like the false wireworms, feed on the roots of wheat and are considered pests. All arthropods mentioned above are important components of the rangeland food chain because they are potential food for vertebrate animals like birds, frogs, mice, moles, and shrews.

In general, Dimilin 2F (0.015 lb AI/acre), Dimilin 25W (0.015 lb AI/acre), and Sevin 4-Oil (0.5 lb AI/acre) did not significantly reduce the number of ants, spiders, predatory beetles, or scavenger beetles from 7 to 76 days after treatment (DAT). Even at 1 year after treatment (350 to 357 DAT), no significant reductions in any of the soil surface-associated arthropods were detected. Ant numbers temporarily (49 to 55 DAT) declined after Dimilin 2F and Sevin 4-Oil treatments by 43 and 56 percent, respectively. The temporary decline in ant numbers may or may not be due to chance alone. What is important is that ant numbers rebounded immediately and that in most of the sampling periods, the Dimilin and Sevin treatments were consistently shown to have no detrimental effects on ant numbers. Additionally, ant diversity (based on PIE calculations) was not significantly affected by the Dimilin or Sevin treatments from 7 to 357 DAT. This result may indicate that no ant species was particularly susceptible to the Dimilin and Sevin treatments at the dosages studied.

Impact of Dimilin on Flying Nontarget Arthropods.— The arthropods collected in malaise traps were subdivided into the following 3 groups: (1) pollinator bees (order Hymenoptera: families Apidae, Halictidae, Colletidae, Andrenidae, and Megachilidae), (2) predators (order Hymenoptera: families Sphecidae, Pompilidae, and Vespidae; order Diptera: families Asilidae and Therevidae; order Coleoptera: family Coccinelidae; order Neuroptera: families Chrysopidae, Hemerobiidae, and Myrmeleontidae), and (3) parasites (order Hymenoptera: families Ichneumonidae, Braconidae, Tiphiidae, Chalcididae, Chrysididae, Mutillidae, Proctotrupidae, and Pteromalidae; order Diptera: families Bombyliidae and Nemestrinidae).

In general, no significant reductions in flying nontarget arthropods were observed in the Dimilin 2F, Dimilin 25W and Sevin 4-Oil treatments. Dimilin 25W reduced predator numbers during the 15- to 20-DAT period by 59 percent. Predator numbers subsequently recovered, and in most of the sampling periods, no significant reductions in predator numbers were observed. A temporary decline of 18 percent in parasite numbers was recorded in the Dimilin 2F treatment at 35 to 41 DAT. No significant reductions were observed in the number of pollinator bees. About 1 year after treatment (350 to 357 DAT), no significant reductions in numbers of predators, parasites or pollinators were observed for any treatment.

Efficacy of Dimilin Against Rangeland Grasshoppers.—Nineteen grasshopper species were present on the 800-acre experimental area immediately before spraying (0 DAT). *Melanoplus sanguinipes* F., *M. infantilis* Scudder, and *Trachyrhachys kiowa* Thomas were the dominant grasshopper species. Grasshopper age structure was 46.8, 24.6, 23.5, 3.7, 0.2, and 0.1 percent for 1st, 2d, 3d, 4th, and 5th instars and adults, respectively, at 0 DAT. This age composition was ideal for a chitin-synthesis inhibitor like Dimilin because the majority of grasshoppers had several molts remaining in their life cycle.

All Dimilin treatments were comparable to Sevin 4-Oil starting at 14 DAT. From 14 DAT to 49 DAT, grasshopper numbers in the Dimilin- and Sevin-treated plots were significantly lower than those of the untreated plots. Dimilin provided consistent grasshopper control from 14 DAT to 49 DAT; Sevin-treated plots revealed temporarily elevated grasshopper numbers at 35 DAT and 42 DAT. No differences between plots treated with Dimilin at different rates or formulations were detected after 14 DAT.

Dimilin was not as effective as Sevin at 7 DAT. This delayed response is most likely due to its mode of action. Dimilin exerts its effect at molting while Sevin (a cholinesterase inhibitor) acts at any time of development. Grasshopper population reductions (adjusted for natural population changes) in Dimilin-treated plots ranged from 65 percent to 90 percent from 14 DAT to 49 DAT. In this study, all treatments lost effectiveness against grasshoppers by 76 DAT. For more information about diflubenzuron efficacy on rangeland grasshoppers, see chapter VII.2, “Dimilin Spray for Reducing Rangeland Grasshopper Populations.”

In summary, our study showed that Dimilin and Sevin sprays did not appear to significantly reduce the abundance of soil-surface-associated or flying nontarget arthropods while providing good grasshopper control in rangeland. Our observations extended only through about 1 year after treatment. Interpretation of our results is limited to this period.

References Cited

Ables, J. R.; West, R. P.; Shepard, M. 1975. Response of the house fly and its parasitoids to Dimilin (TH-6040). Journal of Economic Entomology 68: 622–624.

Borror, D. J.; DeLong, D. M. 1964. Introduction to the study of insects. New York: Holt, Rinehart and Winston. 819 p.

Farlow, J. E.; Breaud, T. P.; Steelman, C. D.; Schilling, P. E. 1978. Effects of the insect growth regulator diflubenzuron on non-target aquatic populations in a Louisiana intermediate marsh. Environmental Entomology 7: 199–204.

Hurlbert, S. H. 1971. The nonconcept of species diversity: a critique and alternative parameters. Ecology 52: 577–586.

Kaston, B. J. 1972. How to know the spiders, 3d ed. Dubuque, IA: Wm. C. Brown Co. 272 p.

Keever, D. W.; Bradley, J. R.; Ganyard, M. C. 1977. Effects of diflubenzuron (Dimilin) on selected beneficial arthropods in cotton fields. Environmental Entomology 6: 732–736.

Metcalf, R. L.; Lu. P.; Bowlus, S. 1975. Degradation and environmental fate of 1-(2,6-diflurobenzoyl)-3-(4-chlorophenyl) urea. Journal of Agricultural Food Chemistry 23: 359–364.

Miura, T.; Takahashi, R. M. 1974. Insect developmental inhibitors. Effects of candidate mosquito control agents on nontarget aquatic organisms. Environmental Entomology 3: 631–636.

Miura, T.; Takahashi, R. M. 1975. Effects of the IGR, TH6040, on nontarget organisms when utilized as a mosquito control agent. Mosquito News 35: 154–159.

Post, L. C.; Vincent, W. R. 1973. New insecticide inhibits chitin synthesis. Naturwissenschaften 60: 431–432.

Snodgrass, R. E. 1935. Principles of insect morphology. New York: McGraw-Hill. 667 p.

van Daalen, J. J.; Meltzer, J.; Mulder, R.; Wellinga, K. 1972. Selective insecticide with a novel mode of action. Naturwissenschaften 59: 312–236.

Washington, H. G. 1984. Diversity, biotic and similarity indices: a review with special relevance to aquatic ecosystems. Water Research 18: 653–694.

Wheeler, W. P.; Wheeler, J. 1963. The ants of North Dakota. Grand Forks, ND: University of North Dakota Press. 326 p.

Wilkinson, J. D.; Biever, K. D.; Ignoffo, C. M.; Pons, W. J.; Morrison, R. K.; Seay, R. S. 1978. Evaluation of diflubenzuron formulations on selected insect parasitoids and predators. Journal of the Georgia Entomological Society 13: 226–236.

References Cited–Unpublished

Foster, R. N.; Reuter, K. C.; Wang, T.; Fuller, B. W. 1991. Field evaluations of diflubenzuron (Dimilin) spray for control of grasshoppers on rangeland. In: Cooperative Grasshopper Integrated Pest Management Project, 1991 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 79–86.

Foster, R. N.; Reuter, K. C.; Black, L.; Daye, G.; Vigil, E. L.; Walz, A.; Radsick, B. 1993. Field evaluation of aerial application of two formulations of diflubenzuron for rangeland grasshopper control under simulated control program operational conditions. In: Cooperative Grasshopper Integrated Pest Management Project, 1993 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 93–100.

VII.4 An Attempt at Classical Biological Control of Rangeland Grasshoppers With *Entomophaga grylli,* Pathotype 3

Donald L. Hostetter

The primary objective of this project, conceived and developed by R. I. Carruthers, was to develop and implement a classical biological control program against rangeland grasshoppers using an Australian isolate (pathotype 3) of the grasshopper obligate *Entomophaga grylli* (Zygomycetes: Entomophthorales) species complex (Ramoska et al. 1988). Pathotype 3 was isolated from *Praxibulus* sp. grasshoppers in Australia in 1985 by R. S. Soper and R. J. Milner during an epizootic (grasshopper outbreak) (Milner 1985).

The project was based on the collaborative findings of Soper and Milner and a 5-year study of the two native species designated pathotype 1 and 2 in Arizona and New Mexico (Carruthers and Humber 1988 unpubl.).

Implementation of the attempt (Carruthers and Humber 1988 unpubl.) was through the U.S. Department of Agriculture, Animal and Plant Health Inspection Service, Plant Protection and Quarantine (USDA, APHIS, PPQ), Grasshopper Integrated Pest Management (GHIPM) Project initiated in 1987.

The proposed and pursued approach was the introduction, establishment, evaluation, and dispersal of an Australian isolate, pathotype 3, to augment the two native pathotypes (1 and 2). Previous laboratory studies indicated that pathotype 3 had a wider host range than either of the native species plus other attributes that led to its selection (Ramoska et al. 1988).

These fungi, along with other biotic agents could theoretically provide long-term, nonchemical suppression of grasshoppers between outbreaks.

An ecological survey of sites with histories of grasshopper populations and densities suitable for introduction was made within the 17 Western States. The Little Missouri National Grasslands in McKenzie County, ND, was selected as the initial study area for field evaluation of pathotype 3 (Carruthers et al. 1989 unpubl.).

The use of biological control methods for grasshopper management, and specifically the introduction of the Australian fungus, was supported by the membership of the McKenzie County Grazing Association, Watford City, ND.

Administrative policies and technical procedures within Federal agencies and the State of North Dakota in effect at the time were observed and provided guidance for introduction. Permission was granted for field studies in North Dakota (Carruthers et al. 1989 unpubl., and in press).

The goals of proposed releases were to reduce populations of economically important grasshoppers on western rangeland to, or below, threshold densities; to establish pathotype 3 as a biorational agent that would augment native fungi; and to determine the plausibility of future large scale releases throughout the Western United States by PPQ's Plant Protection Laboratories.

Pathotype 3 was introduced into susceptible grasshopper populations at several sites in McKenzie County in 1989, 1990, and 1991 and at two sites near Delta Junction, AK, in 1990 (Carruthers et al. 1989 unpubl., 1990 unpubl., 1991 unpubl.).

Introduction was by randomly releasing laboratory-infected fifth-instar and adult *M. differentialis* (Thomas), each injected with 10 μl of 10^4 pathotype 3 protoplasts, into grasshopper populations in alfalfa/mixed grass or crested wheatgrass fields with no history of pathotype 1 or 2 fungus infection. Each field was about 44.5 acres (18 ha) in size. Releases were made at 2- to 3-day intervals 3 days postinjection (just prior to death of the grasshopper). Weekly releases in lots of 500 infected grasshoppers totalled from 500 to 3,500 at each site.

The initial release of pathotype 3 was made July 24, 1989. Five hundred (500) infected grasshoppers were released in an alfalfa/mixed-grass hayfield at Wold's ranch (T153N, R97W, Sec. 33), 25 miles north of Watford City, ND. Incidence of fungus infection among grasshoppers within this release site was 13 percent 2 weeks after the release (Carruthers et al. 1989 unpubl.).

Additional releases of ca. 500 per day were made at Wold's on July 8, 11, 15, 19, 25, and 30, 1990. A 20-percent incidence of infection was observed at this site within 2 weeks of the 1990 releases. No additional releases were made at this site after 1990.

Similar releases were made in crested wheatgrass fields at three other sites in McKenzie County, ND, during 1990. Incidence of fungus infection among grasshoppers at these locations was less than 3 percent. Low incidence of infection in these fields was attributed to the open canopy of the crested wheatgrass, which likely resulted in a less favorable habitat for the fungus (Carruthers et al. 1990 unpubl.). Seasonal monitoring of grasshopper populations at these sites (1991–94) has failed to detect fungus-infected individuals.

The releases of pathotype 3 into *M. sanguinipes* populations at two sites in Alaska were considered unsuccessful in that only a single sporulating cadaver was recovered 2 weeks after release. Grasshopper populations at these release sites have been monitored annually for incidence of fungal infection.

Overwintering of pathotype 3 was thought to occur in Wold's field based on recovery of sporulating *M. bivittatus* (Say) cadavers in June, 1991. Fungal mortality among grasshoppers at this site reached 26 percent in 1991 even though no additional introductions were made (Carruthers et al. 1991 unpubl.).

Releases of infected grasshoppers (500–1,000 each) were made on land managed by the U.S. Army Corps of Engineers near Lake Sakakawea (T154N, R95W, Sec. 32) on June 6, 8, 11, and 13, 1991. The incidence of fungus infection at this location reached 25 percent 2 weeks after the last release. No additional releases were made after June 13, 1991. Grasshopper populations at this site continued to be monitored for incidence of fungal disease through 1994. Populations and incidence of fungal infection have been diminishing since 1991.

The initial success in North Dakota was encouraging, and a plan for additional releases of 150,000 infected *M. differentialis* (10,000 per week per location for 5 consecutive weeks) at 3 other locations was drafted. Additional releases were contingent upon production and supply of suitable hosts by a commercial insectary in Colorado. The number of sites and infected grasshoppers to be released was based on available human and fiscal resources as well as host population densities.

This project and plans for future releases of nonnative pathogens and parasites within the GHIPM Project caused intense debate among certain researchers and between agency administrators in 1991 (Bomar and Lockwood 1991, Lockwood 1993a and b, Carruthers and Onsager 1993). In August 1991, amid the beginning controversy of the legality and wisdom of this approach, the principal investigator (R. I. Carruthers) was reassigned, and the project was transferred from Ithaca, NY, to me at Kimberly, ID.

Additional documentation was drafted and submitted (April 1992) and revised and resubmitted (October 1992) seeking a policy decision on the need for an environmental assessment (EA) before proceeding with additional releases of pathotype 3.

Additional releases of pathotype 3 are stalemated. No releases of pathotype 3 have been made since June 1991. Efforts since that time have been relegated to monitoring (population densities, composition, species fluctuations, incidence of mortality due to fungus infection, dispersal studies) in the release field and surrounding areas.

Laboratory studies were conducted to establish basic parameters of conidia production, germination and viability, and dose/mortality curves, as well as mass inoculation methods that would be required if the project was to be assumed and enlarged by PPQ's Plant Protection Laboratories.

The development of DNA probe technology for separation and identification of three *Entomophaga* spp. of the *E. grylli* complex has also been successful. Cooperation between USDA's Agricultural Research Service staff scientists at Ithaca, NY, and Kimberly, ID; the Boyce Thompson Institute for Plant Science; and the University of Toronto, Scarborough campus led to the development of a positive DNA identification probe whereby pathotypes 1, 2, and 3 can be separated and positively identified (Bidochka et al. 1995). This is a critical accomplishment and provides a tool necessary to delineate dispersal and distribution of pathotype 3 in the field.

References Cited

Bidochka, M. J.; Walsh, S.R.A.; Ramos, M. E.; St. Leger, R. J.; Silver, J. C.; Roberts, D. W. 1995. Pathotypes in the *Entomophaga grylli* species complex of grasshopper pathogens differentiated by molecular methods. Journal of Applied and Environmental Microbiology 61: 556–560.

Bomar, C. R.; Lockwood, J. A., eds. 1991. Biological control of western rangeland grasshoppers with exotic predators and parasitoids: potential benefits, costs, and alternatives. Bull. B-958. Laramie, WY: Wyoming Agricultural Experiment Station.

Carruthers, R. I.; Onsager, J. A. 1993. Perspective on the use of exotic natural enemies for biological control of pest grasshoppers (Orthoptera: Acrididae). Environmental Entomology 22: 885–904.

Carruthers, R. I.; Ramos, M. E.; Larkin, T. S.; Hostetter, D. L.; Soper, R. S. [In press.] The *Entomophaga grylli* (Fresenius) Batko species complex: its biology, ecology and use for biological control of pest grasshoppers. Memoirs of the Entomological Society of Canada.

Lockwood, J. A. 1993a. Environmental issues involved in biological control of rangeland grasshoppers (Orthoptera: Acrididae) with exotic agents. Environmental Entomology 22: 503–518.

Lockwood, J. A. 1993b. Benefits and costs of controlling rangeland grasshoppers (Orthoptera: Acrididae) with exotic organisms: search for a null hypothesis and regulatory compromise. Environmental Entomology 22: 904–914.

Milner, R. J. 1985. Field tests of a strain of *Entomophaga grylli* from the USA for biocontrol of the Australian wingless grasshopper, *Phaulacriduim vittatum*. In: Chapman, R. B., ed. Proceedings of the 4th Australian conference on grassland invertebrate ecology; 13–17 May 1985; Canterbury, UK. Christchurch, NZ: Caxton Press: [inclusive pagination unknown].

Ramoska, W. A.; Hajek, A. E.; Ramos, M. E.; Soper, R. S. 1988. Infection of grasshoppers (Orthoptera: Acrididae) by members of the *Entomophaga grylli* species complex (Zygomycetes: Entomophthorales). Journal of Invertebrate Pathology 52: 309–313.

References Cited—Unpublished

Carruthers, R. I.; Humber, R. A. 1988. Development of *Entomophaga grylli* as a biological control agent of grasshoppers. In: Cooperative Grasshopper Integrated Pest Management Project, 1988 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 330–335.

Carruthers, R. I.; Humber, R. A.; Ramos, M. E. 1989. Development of *Entomophaga grylli* as a biological control agent of grasshoppers. In: Cooperative Grasshopper Integrated Pest Management Project, 1989 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 229–234.

Carruthers, R. I.; Humber, R. A.; Ramos, M. E. 1990. Development of *Entomophaga grylli* as a biological control agent of grasshoppers. In: Cooperative Grasshopper Integrated Pest Management Project, 1990 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 220–231.

Carruthers, R. I.; Humber, R. A.; Ramos, M. E. 1991. Development of *Entomophaga grylli* as a biological control agent of grasshoppers. In: Cooperative Grasshopper Integrated Pest Management Project, 1991 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 185–189.

VII.5 Lab Studies and Field Trials With the Fungus *Beauveria bassiana* Against Grasshoppers

R. Nelson Foster, K. Christian Reuter, Jim Britton, and Cliff Bradley

More than 150 years ago, the Hyphomycete fungus *Beauveria bassiana* was recognized as the cause of a disease fatal to insects (Steinhaus 1967). *B. bassiana* is a common insect pathogen (an agent that causes disease) found on all continents except Antarctica (Humber 1992). Hundreds of isolates of the fungus, including five from grasshoppers, are listed in the U.S. Department of Agriculture (USDA) collection of Entomopathogenic Fungal Cultures (Humber 1992).

In the fungus' life cycle, conidia (spores) adhere to the grasshopper cuticle (part of the exoskeleton). The conidia germinate, and the germ tube penetrates the cuticle. The fungi replicate inside the insect haemocoel (body cavity) in the form of blastospores (spores produced by a budding process). Degradative enzymes destroy the internal structures of the grasshopper.

When in sufficient quantity, the fungus causes sickness within 3 days. The grasshopper reduces its feeding and becomes immobile. Typically, infected grasshoppers die between 4 and 10 days after infection depending on their species, age, and size, and the dose of conidia. After death, under conditions of high humidity, blastospores form hyphae (filaments of the vegetative structure of the fungus) that emerge through the insect's cuticle, sporulate (produce spores), and cover the insect in a characteristic white growth (fig. VII.5–1).

Figure VII.5–1—An immature rangeland grasshopper, *Melanoplus sanguinipes,* exhibits the fungus *Beauveria bassiana,* which caused its death. (Photo by K. Christian Reuter.)

In 1987, Mycotech Corporation in Butte, MT, isolated, from an infected grasshopper found in Montana, a strain of *Beauveria bassiana* that is virulent (disease-causing) to several grasshopper species in laboratory bioassays. Since that time, Mycotech has developed and refined production capabilities to the point that large-scale commercialization is planned upon the final development of an acceptable formulation for delivering the pathogen to grasshoppers in the field. The following summarizes some of the research conducted since early 1991 in the development of formulations of *Beauveria bassiana* usable against grasshoppers on rangeland.

Laboratory Studies, 1991–93

During this period, we conducted more than 20 different replicated studies. The objectives provided for (1) developing equipment and procedures for our laboratory studies, (2) studying the effect of *Beauveria bassiana* on different age groups of grasshoppers, (3) comparing of formulations, and (4) comparing the virulence of different batches of commercially produced *B. bassiana*.

Test formulations were sprayed from a tower apparatus in the lab to simulate aerially applied sprays (fig. VII.5–2). Applications were conducted according to a detailed standard operating procedure (Foster and Reuter 1991 unpubl.). Laboratory-reared *Melanoplus sanguinipes* grasshoppers supplied by South Dakota State University were used for all studies. All tests focus on a dose of 1×10^{13} (1 trillion) spores/acre as a standard. Depending on the specific test protocol, we sprayed grasshoppers and/or live vegetation upon which the grasshoppers were to be confined.

When grasshoppers were sprayed, third instars through adult stages were sprayed singly or in groups consisting of from 5 to 20 grasshoppers per group. After spraying, the grasshoppers were monitored daily for death, usually for 2 weeks. In tests where grasshoppers were sprayed, fresh food was provided to surviving grasshoppers daily, and dead grasshoppers were held singly under high humidity conditions for observance of sporulation.

Figure VII.5–2—Spray tower used to simulate aerially applied sprays for bioassaying grasshoppers in the laboratory. (APHIS photo by Lonnie Black.)

Initial studies demonstrated the superiority of an oil formulation over a water formulation. A typical example of results from one of these tests is shown in figure VII.5–3. In later studies where candidate field formulations were compared, we focused primarily on different oil types with various additives selected for ultraviolet light protection and emulsion stabilization (formulation stability). Two petroleum oils performed equally well as base carriers; however, one is significantly less expensive. We found that formulations involving emulsifiable concentrates tend to be more difficult to spray consistently in the laboratory. However, our results indicate that such compounds may provide higher mortality in field application.

In studies where untreated grasshoppers were confined on sprayed vegetation, we showed a significant decrease in mortality on vegetation that had been exposed to sunlight for longer than 24 hours (fig. VII.5–4). However, two formulations currently under development show promise for extending protection beyond 24 hours.

Third-, fourth-, and fifth-instar grasshoppers were easily infected and very susceptible to sprays equivalent to 1×10^{13} spores/gal/acre. However, compared to these results, two separate studies with adult grasshoppers showed a greatly reduced level of mortality at the same dose. Subsequent studies in which adults with amputated wings were sprayed showed that reduced mortality in adults cannot be attributed to physical protection provided by wings, which shield a major portion of the abdomen from the spray.

We conducted several studies to compare spores from different productions and to evaluate shelf life. Spores stored in oil for up to 1 year performed as well as dry conidia powder stored for an equal period. A 1992 spring production as well as a new isolate both performed similarly to spores produced in 1991. However, a 1992 fall production sampled resulted in some inconsistencies during the physical spraying. Slightly cooler temperatures during the spray operation may have affected the sprayability of the formulation. Also, a new harvesting method at the production facility resulted in some larger particles of spore powder, increasing spray problems.

Field Studies—1991

A 9-acre rangeland plot near Edgemont, SD, infested with predominantly second- and third-instar grasshoppers of mixed species, was aerially sprayed with an oil formulation containing 8×10^{12} spores/gal/acre (fig. VII.5–5). Grasshopper moralities measured in this plot were compared to a similar untreated adjacent plot (Foster et al. 1991 unpubl.).

We evaluated mortality on six grasshopper species by collecting grasshoppers from both plots after application and confining them in (1) small rearing cups (fig. VII.5–6), which we moved to the laboratory for daily monitoring, and (2) bottomless field cages (fig. VII.5–7) established after treatment in both plots. Additionally, 0.1-m^2 rings (Onsager and Henry 1977) were used to delimit counting areas for estimating total field populations of grasshoppers.

Beauveria bassiana caused mortality in all six species of the grasshoppers tested. Both grasshoppers held in rearing cups in the laboratory and those caged on native vegetation in the field demonstrated significant mortality in treated populations compared to untreated populations. Some species were killed faster than others, but we do not know if this is due to inherent susceptibility or behavioral differences between the species.

In rearing cups, the average reduction of all species combined in treated populations was about 96 percent at 8 days after treatment. Mortality in the controls during the same period was about 34 percent. In field cages, the mean reduction of all species combined was 79 percent and 11 percent for treated and untreated populations, respectively, at 9 or 10 days after treatment.

In field plots, counts of unconfined populations in treated and untreated plots showed average differences in mortality that ranged from about 39 percent to 63 percent at 3 to 15 days after treatment (fig. VII.5–8).

We also used field cages to determine the general manner in which grasshoppers pick up the spores. Immediately after application, grasshoppers from the untreated plots were collected and caged in the treated area to determine pickup through feeding activity. Treated grasshoppers were caged in the untreated plot to determine the mortality associated with direct contact. Treated grasshoppers were caged in the treated plot to determine the total mortality, and untreated grasshoppers were caged in the untreated plot as a control.

At 11 days after treatment, there were no significant differences in grasshopper mortality between the direct deposition, feeding activity, or combined direct deposition/feeding activity treatments. All three treatments showed significantly greater mortality than the untreated

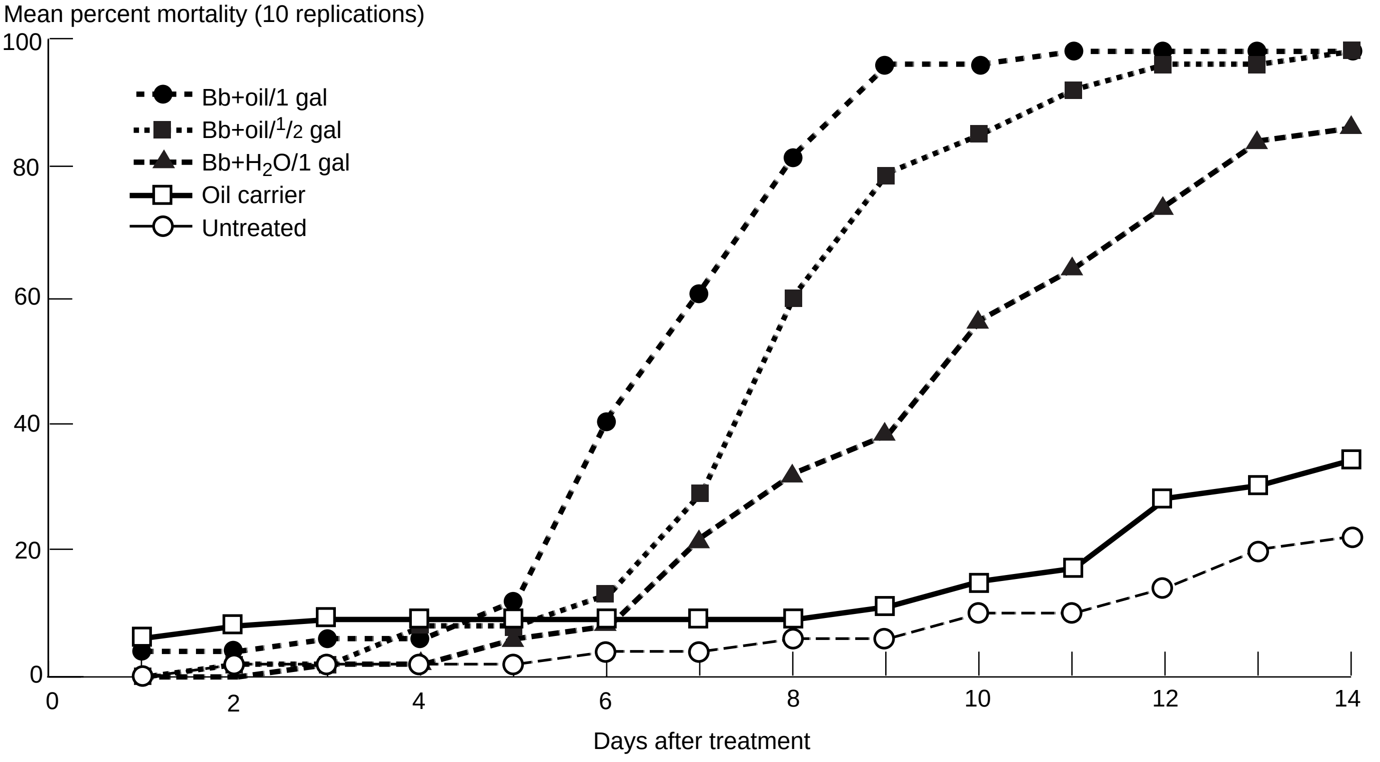

Figure VII.5–3—Mortality of caged grasshoppers treated with experimental formulations of *Beauveria bassiana* at 1 × 10^{13} conidia per acre.

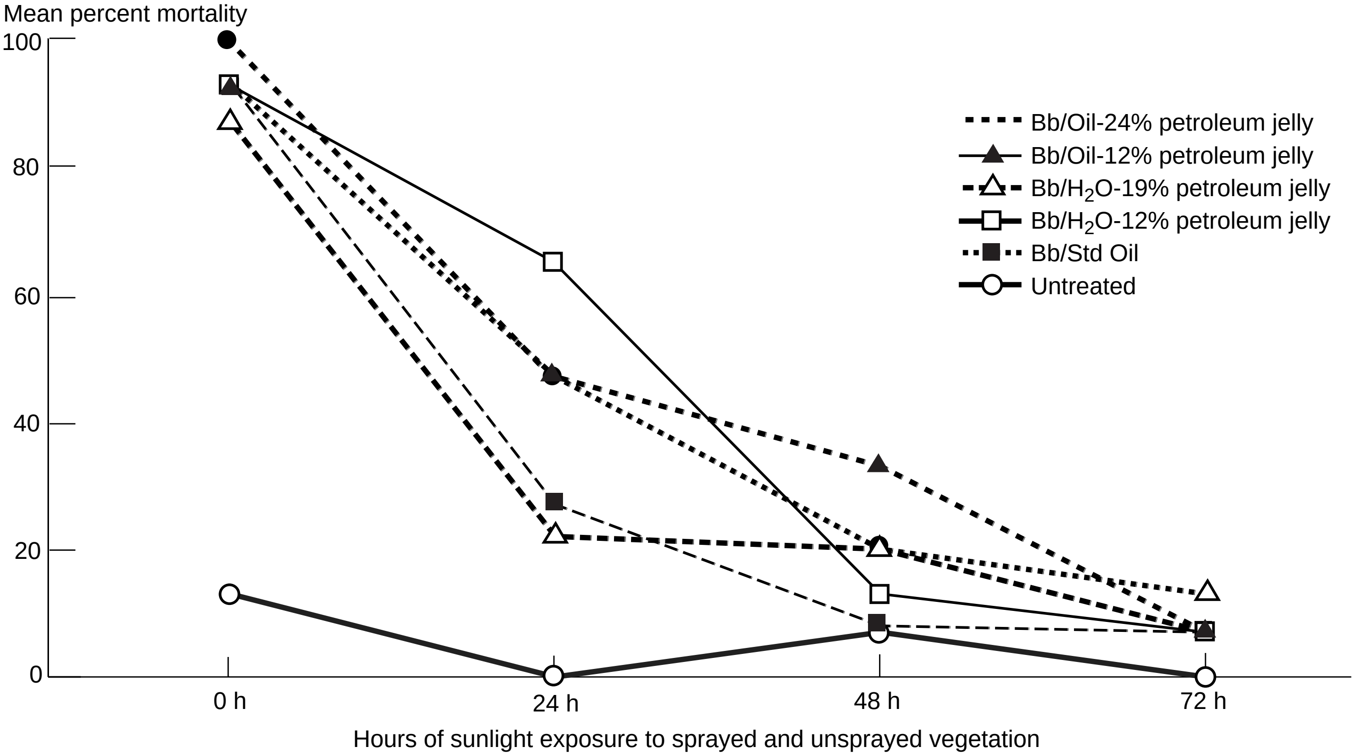

Figure VII.5–4—Effect of grass treated with selected formulations of *Beauveria bassiana* and exposed to several periods of sunlight on grasshopper survival after 9 days. All treatments were applied at a volume of 1 gal/acre containing 1×10^{13} spores.

Figure VII.5–5—The first aerial application of the fungus *Beauveria bassiana* was applied at 1 gal/acre to a rangeland plot near Edgemont, SD in 1991. (Photo by Cliff Bradley.)

Figure VII.5–6—Four-ounce rearing cups used to confine test grasshoppers after they have been treated. (APHIS photo by R. Nelson Foster.)

check. Our data indicate that pickup may occur through either direct impingement (direct striking by spray droplet) or feeding activity. We do not know if the feeding activity component is simply due to contact with the mouthparts of the grasshopper during feeding or actual ingestion of spores.

We evaluated the short-term residual activity of the spores by caging untreated grasshoppers approximately 10 hours after treatment in the treated plot. Survival of the conidia on vegetation was evaluated in the sprayed plot by taking vegetation samples at three posttreatment intervals. These samples were washed, diluted, and placed on selective agar plates, where fungus colonies developed from each colony-forming unit. The colonies then were counted to estimate the number of viable (living) conidia.

Figure VII.5–7—Bottomless field cages used to confine test grasshoppers in the field are inspected carefully to determine the daily insect mortality. (APHIS photo by R. Nelson Foster.)

Figure VII.5–8—Mortality of unconfined field populations of grasshoppers is estimated by counting grasshoppers in metal rings. (APHIS photo by R. Nelson Foster.)

Untreated grasshoppers exposed to the treated vegetation in the field approximately 10 hours after application died at about 3.3 times the mortality rate of untreated grasshoppers over the same period of time, 11 days. The delayed exposure demonstrates the infectivity of spores at least 10 hours after field application and indicates that, in field situations, at least several hours are available for a grasshopper to become infected with the fungus. Results of the study to determine survival of conidia on vegetation in the field showed relatively uniform coverage in the plot and indicated no loss of activity over at least the first 10 hours after application.

Field Studies—1992

Three adjoining 9-acre rangeland plots near Amidon, ND, infested with predominately fourth- and fifth-instar grasshoppers of mixed species were the basis for studies in 1992. One plot was aerially sprayed with 9.5×10^{12} spores/64 oz/acre. One plot was sprayed with 64 oz/acre of the oil carrier (without spores), and the other plot was left untreated for comparison (Foster et al. 1992 unpubl.).

Mortality evaluations were conducted as in 1991, by confining, after treatment, the six predominant grasshopper species in cages held in the laboratory or in the field. The methods used for maintaining the cages and confirming fungus-induced death by sporulation were similar to those employed in 1991. Reduction in the total field population was again estimated by using 0.1-m^2 rings to delimit counting areas.

In this study, the aerial application of *B. bassiana* resulted in substantial mortality of all six species of grasshoppers evaluated. Both grasshoppers held in rearing cups in the laboratory and those caged on native vegetation in the field demonstrated significant mortality in fungus-treated populations compared to untreated populations and populations treated with oil only. These results were generally similar to those obtained in 1991, and again time to mortality varied among species, beginning in as little as 3 days for some species and as much as 4 to 6 days for other species. These differences may be attributed to individual species susceptibility or a result of behavioral avoidance, which limits physical exposure of individual species to direct impingement of the spray droplet.

In rearing cages, the mean reduction of all species combined in treated populations was 95 percent at 8 days after treatment. During the same time period, mortality in the untreated population and the population treated only with oil was 10 percent and 4 percent, respectively. Three species common to both the 1991 and 1992 studies demonstrated very similar responses to the aerially applied *B. bassiana* treatment.

In field cages, the mean reduction for 5 of the 6 species confined in treated populations was 91 percent at 15 to 17 days following treatment. This reduction compared to mortality during the same period in the untreated population and the population treated only with oil of 23 percent and 11 percent, respectively. The sixth species in the study was reduced much quicker: 100-percent mortality occurred by the eleventh day. Its counterparts in the untreated plots and the plots treated with oil showed 26 percent and 16 percent reduction during the same period.

Comparisons of the in-field posttreatment population estimates in single, small plots are difficult to interpret. High densities of grasshoppers, sparse vegetation, small plot size, and local movement all contribute to confounding estimates of nonrestricted in-field populations. Compared to 1991, in-field mortality was lower in this study. In 1992, apparent mortality at 9 days after treatment was only about 20 percent. We did note that vegetation in the 1992 study was much sparser than in the 1991 study and may have offered the spores less protection from sunlight. Using large field plots in future studies should reduce many of the difficulties commonly encountered when comparisons of in-field grasshopper populations on rangeland are attempted.

Field Studies—1993

We focused studies for the first time in 1993 on larger plots than previously used (Foster et al. 1993 unpubl.). That year, we aerially sprayed 24 adjoining 40-acre rangeland plots located near Amidon, ND, infested with predominantly second-, third-, and fourth-instar stages of grasshoppers of mixed species. Two formulations of *Beauveria bassiana* spores were each applied to eight plots. One treatment consisted of 9.9×10^{12} spores/64 oz/acre in an oil formulation, and the other treatment consisted of 9.4×10^{12} spores/64 oz/acre in an oil plus additive (adjuvant) formulation. An oil-only treatment was applied at 64 oz/acre to four plots. Carbaryl was sprayed at 20 oz/acre (0.5 lb/active ingredient [AI] per acre) to four plots as a standard treatment for comparison. Four plots were left untreated to determine the natural changes in the grasshopper population and for comparison with all applied treatments.

In field populations, estimates were again made using 0.1-m^2 rings. A monitoring site located near the center of each 40-acre plot consisted of 40 rings arranged in a circle with rings separated by 5 paces. Field cages were placed adjacent to the ring site in each plot after the treatment was sprayed. Sprayed grasshoppers of two of the dominant species were confined in these cages in a manner similar to that employed in 1991 and 1992 field studies.

Additional field cages were set up in each fungus- and oil-only treated plot and in the untreated plots. These cages were used to study the residual activity of *Beauveria bassiana* over a 5-day period after treatment. Untreated grasshoppers were confined in some cages on the day of treatment and on each of the 5 days following treatment.

Unfortunately, the study's value was lessened by measurable rain (heavy at times) that occurred on 9 of the 13 days that population estimates were made. During the entire study, measurable rain was recorded on 15 of 21 days.

Although incomplete, analysis of counts from rings to date shows that the carbaryl standard was statistically superior to all other treatments at each of the posttreatment interval readings. Good performance of carbaryl under these conditions was expected and is consistent with two of our previous studies where carbaryl was used (Foster et al. 1991 unpubl. and Foster et al. 1993 unpubl.). All other experimental treatments (including the untreated checks) showed erratic results, undoubtedly confounded by the weather conditions experienced during the study, and were statistically inseparable.

Results from the field cages for the two species studied at 15 days after treatment indicated that both fungus treatments and the carbaryl treatment produced mortality significantly greater than what occurred in the untreated populations. However, mortality in the field cages was somewhat lower than in 1991 and 1992 for the one species that was common to studies in all 3 years.

Residual activity was evident only during the day of treatment. Beyond 1 day, no significant differences in mortality were detected between fungus-treated or untreated grasshoppers.

Under the conditions of this study, evaluations of unproven formulations are confounding and inconclusive at best. However, there is no doubt that carbaryl performed well under these conditions and that the current formulation of *Beauveria bassiana* will need to be improved if it is to be employed under these conditions, or excluded from use under such conditions. Additional replicated studies to obtain information on the original objectives of the 1993 field study and new formulation evaluations are planned for the future.

Summary of Additional Foreign Studies

During the past 5 years, Mycotech has been working to develop fungal pathogens of locusts and grasshoppers for use in integrated pest management (IPM) programs in Africa. This work is in collaboration with Montana State University, the U.S. Agency for International Development, and several African government agencies. These efforts were undertaken to devise alternatives to chemical grasshopper/locust control measures commonly used in Africa. Fungi can fit well into an IPM scheme because they provide control alternatives where chemical insecticides are inappropriate. In fact, because of their relatively slow action, fungi will work best as part of a continuous pest-control strategy, where they can be applied before populations are able to reach damaging levels.

A Mycotech strain of the fungus *Beauveria bassiana* has been tested against grasshoppers and locusts in several small-plot field trials in the west African countries of Cape Verde and Mali. Fungal spores were applied at a rate of 1×10^{13} per acre. Low-volume application of an oil-based formulation (27 ounces to 2 quarts per acre) was made with hand-held spinning disc sprayers. High-volume application of an emulsifiable formulation (2–10 gal/acre) was made with motorized or hand-pumped backpack sprayers. Spores were also formulated on wheat bran bait with a molasses sticker.

In all trials, 80 to 100 percent of treated, caged insects died from *Beauveria bassiana* infection after 7 days. More significantly, replicated 5-acre blocks in Cape Verde, treated with either oil-formulated or emulsion-formulated fungus, showed approximately 50 percent population density reductions measured in the field after 7 days. It is quite encouraging that the insect population in these tests consisted primarily of older nymphs and adults, which have demonstrated more resistance to the fungus in laboratory bioassays.

Mycotech and Montana State University have taken part in an expedition to Madagascar to collect new fungal pathogens of locusts and grasshoppers. The fungi isolated from infected insects are presently being examined for virulence, target specificity, production characteristics, and impact on mammals. The government of Madagascar is particularly interested in using fungi to treat locust populations before the insects expand out of their recessionary (nonoutbreak) areas. When a suitable fungus is identified, field trials will begin.

These promising results indicate that fungal insecticides may be able to play an important role in grasshopper/locust control. This field experience in the harsh African conditions will continue to yield information valuable to the development of fungal insecticides for North America.

Summary and Conclusion

A strain of the entomopathogenic fungus *Beauveria bassiana* has been isolated from U.S. grasshoppers by Mycotech Corporation. Development of mass production capabilities with a potential for large-scale commercialization has resulted in extensive testing of the commercially produced fungus for use against grasshoppers and locusts. Laboratory studies have demonstrated the insecticidal value of the fungus against several species of grasshoppers and locusts. In 1991, 1992, and 1993, we conducted field studies using cages to demonstrate successful control of several species of confined grasshoppers in the United States when liquid formulations of *Beauveria bassiana* were aerially applied with conventional commercial application equipment. Results of field studies with unconfined grasshoppers in this country are inconclusive to date. Foreign field studies on unconfined populations showed good potential for providing control. Results from the last 3 years suggest the potential for controlling several species of grasshoppers and locusts using a liquid formulation of *B. bassiana,* as a bioinsecticide, and applied with conventional aerial application equipment.

References Cited

Humber, Richard A. 1992. Collection of entomopathogenic fungal cultures. ARS-110. Washington, DC: U.S. Department of Agriculture, Agricultural Research Service. 177 p.

Onsager, J. A.; Henry, J. H. 1977. A method for estimating the density of rangeland grasshoppers (Orthoptera: Acrididae) in experimental plots. Acrida 6: 231–237.

Steinhaus, E. A. 1967. Insect microbiology. New York and London: Hafner Publishing Co. 763 p.

References Cited—Unpublished

Foster, R. N.; Reuter, K. C. 1991. Using a spray tower to simulate aerially applied liquid sprays for bioassaying insects in the laboratory: standard operating procedure. Phoenix, AZ: U.S. Department of Agriculture, Animal and Plant Health Inspection Service, Methods Development, Rangeland Section. 5 p.

Foster, R. N.; Reuter, K. C.; Black, L. R.; Vigil, E.; Daye, G.; Walz, A.; Radsick, B. 1993. Field evaluation of aerial applications of two formulations of diflubenzuron for rangeland grasshopper control under simulated control program operational conditions. In: Cooperative Grasshopper Integrated Pest Management Project, 1993 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 93–100.

Foster, R. N., Reuter, K. C.; Bradley, C.; Britton, J.; Black, L.; Drake, S.; Leisner, L.; Tompkins, M.; Fuller, B.; Hildreth, M.; Kahler, E.; Radsick, B. 1992. Development of the fungus *Beauveria bassiana* as a bio-insecticide for grasshoppers on rangeland. In: Cooperative Grasshopper Integrated Pest Management Project, 1992 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 207–215.

Foster, R. N.; Reuter, K. C.; Bradley, C. A.; Wood, P. P. 1991. Preliminary investigations on the effect of *Beauveria bassiana* on several species of rangeland grasshoppers. In: Cooperative Grasshopper Integrated Pest Management Project, 1991 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 203–210.

Foster, R. N.; Reuter, K. C.; Tompkins, M. T.; Bradley, C.; Britton, J.; Underwood, N.; Black, L. R.; Vigil, E.; Daye, G.; Radsick, B.; Fuller, B.; Brinkman, M.; Kahler, E. 1992. Development of the fungus *Beauveria bassiana* as a bio-insecticide for grasshoppers on rangeland. In: Cooperative Grasshopper Integrated Pest Management Project, 1993 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 233–238.

Foster, R. N.; Reuter, K. C.; Wang, T.; Fuller, B. W. 1991. Field evaluation of diflubenzuron (Dimilin) spray for control of grasshoppers on rangeland. In: Cooperative Grasshopper Integrated Pest Management Project, 1991 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 79–86.

VII.6 *Beauveria bassiana* for Mormon Crickets

D. A. Streett and S. A. Woods

Introduction

The first crops planted by the Mormon settlers in Utah were damaged by the insect now referred to by the common name "Mormon cricket" (Cowan 1990). The Mormon cricket, *Anabrus simplex* Haldeman, is not a cricket at all but a longhorned grasshopper from the family Tettigoniidae (fig. VII.6–1). This pest can reach outbreak levels before Mormon crickets begin migrating into range and cropland. Mormon crickets can cause significant damage when bands of huge numbers of insects move onto cropland in the Western United States (Pfadt 1991, MacVean 1990, Swain 1944). Our studies evaluated the effectiveness of a fungal pathogen, *Beauveria bassiana*, to suppress Mormon cricket populations.

Figure VII.6–1— The Mormon cricket is mainly a pest on rangelands but sometimes moves into planted crops and causes economic damage. (Agricultural Research Service file photo K4797–1.)

How *Beauveria bassiana* Works

Interest in insect–fungi interactions has centered, for the most part, on the pathogenic (disease-causing) nature of fungi and their use as microbial control agents. Unlike other insect pathogens that must be eaten to infect insects, fungi can infect an insect through its cuticle (outer skin). The development of fungi pathogenic to insects typically follows this pattern:

(1) Attachment of an infectious stage (called a conidium or spore) to the insect cuticle,

(2) Germination of the conidium and penetration of the insect cuticle by a germ tube from the conidium,

(3) Growth of the fungus inside the insect body (hemocoel) and eventual death of the insect,

(4) Penetration of the fungus to the surface of the dead insect and formation of conidia (plural of conidium) under conditions of high relative humidity, and

(5) Dispersal of the conidia to locations where they may encounter susceptible insects and start the process again.

Among the insect-pathogenic fungi that follow this pattern of development is *Beauveria bassiana*. It is commonly known as the white-muscardine fungus because of the characteristic white covering of conidia (spores) found on the surface of dead insects. Insect cadavers infected with the fungus are transformed into white, mummified bodies resembling in appearance a bonbon candy ("muscardin" means "bonbon" in French [Steinhaus 1949]).

Isolate of *B. bassiana* for Mormon Cricket

The *B. bassiana* strain used in these studies was originally obtained from Mycotech Corporation in Butte, MT. Mycotech has obtained Environmental Protection Agency registration of this *Beauveria* strain for the suppression of several insect pests, including grasshoppers and Mormon crickets. Mycotech recently developed a solid culture system for the production of *B. bassiana* conidia (Goettel and Roberts 1992). Mycotech prepared and supplied a *B. bassiana* dry conidia powder for the laboratory studies and *B. bassiana* formulated in oil (OF) and in an emulsible suspension (ES) for the 1992 and 1993 Idaho field trials (Onsager et al. 1992, Kemp and Streett 1993).

Laboratory Studies

Conidia were suspended in ES1 and ES2 oil and applied to Mormon crickets as 0.08 µL (microliter) droplets beneath the pronotum (on the thorax) at dosages ranging from 0 to 10^6 spores per Mormon cricket. Mormon crickets were reared individually in plastic cups and maintained in an incubator at 77 °F (25 °C). Mormon crickets were fed every 2 days with romaine lettuce, kale, and wheat bran. Mortality was recorded during feeding, and a damp cotton ball was added to cups containing cadavers. The cadavers were then stored at room temperature for 4–6 days to diagnose *Beauveria* infection by observing the characteristic white muscardine appearance on the insect surface.

The median lethal dose (LD_{50}) is commonly used to assess the infectivity of a pathogen. The LD_{50} for the *B. bassiana* isolate against fifth-instar Mormon crickets at 12 days was 1,000 conidia (fig. VII.6–2). The two oil formulations that were compared in laboratory assays showed no consistent differences in overall mortality or percentage of Mormon crickets with confirmed infections (table VII.6–1).

Four replicates of 200 adult Mormon crickets each were treated with 5×10^5 or 5×10^6 conidia in oil according to the procedures described by Kemp and Streett, 1993. A check preparation consisting of oil without conidia and an untreated control were included for each replicate. Each treatment within a replicate was separated into two groups and reared either individually in an incubator at 77 °F or transferred to field enclosures. Four field enclosures 16 ft^2 (1.5 m^2) for each treatment were stocked with 25 Mormon crickets. Mormon crickets were fed lettuce daily. Counts of Mormon crickets were made for each cage, and cadavers were collected for incubation in cups with a moistened cotton ball to diagnose *Beauveria* infection (Kemp and Streett 1993).

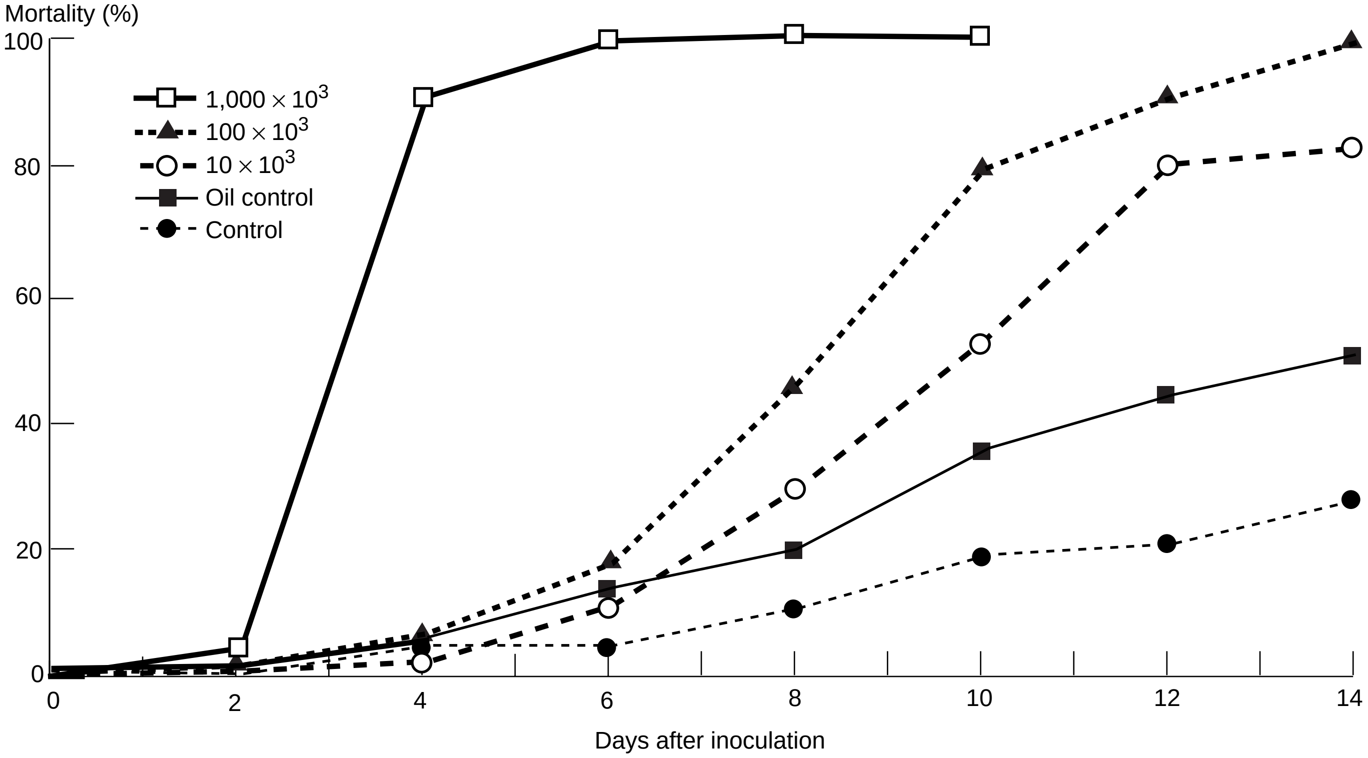

Figure VII.6–2—Cumulative mortality among fifth-instar Mormon crickets in a bioassay of *Beauveria bassiana.*

Table VII.6–1—Laboratory comparison of ES1 versus ES2 oil as a carrier for *Beauveria bassiana.* Cumulative mortality and incidence of infection for Mormon crickets.

	Mortality		Infection	
Dose	[1]ES1	ES2	ES1	ES2
Conidia/ *grasshopper*	— — — — — — *Percent*	— — — — — — —		
0	34	46	8	12
10^2	50	38	20	18
10^3	71	87	42	44
10^4	90	98	65	62

[1] ES = emulsifiable suspension.

Adult Mormon crickets that were inoculated with 5×10^6 conidia per Mormon cricket showed a significant difference in mortality in laboratory versus field cages (fig. VII.6–3). Adult Mormon crickets reared in the field enclosures survived more than 3 weeks longer than Mormon crickets reared in the laboratory. One possible explanation for these results is that Mormon crickets in the field use a behavioral thermoregulation to increase body temperature to a point that restricts fungal development and allows the insect to survive.

Field Studies

Field trials against Mormon crickets were conducted near St. Anthony, ID. Oil (ES1 oil) and clay–oil–water (COW)—100 g clay: 1 liter (L) oil: 2 L water)—formulations were applied at rates of 4.9 (10^{11} and 4.9×10^{12} conidia/acre (1.2×10^{12} and 1.2×10^{13} conidia per ha) and application volumes of 0.9 and 2.7 qt/acre (2.5 and 7.5 L/ha). Each replicate consisted of 10 arenas of 14.4 yd^2 (12 m^2) constructed of aluminum flashing approximately 10–18 inches (25–45 cm) in height. Each arena was stocked with more than 250 Mormon crickets prior to application. Treatments were replicated four times, and treatments within each replicate were applied on the same day (weather permitting) in the sequence outlined by Onsager et al. (1992). An ultralow-volume sprayer (North American Micron) was used for the applications. After application, Mormon crickets were collected from each arena for rearing. Approximately 30–50 Mormon crickets per arena were reared individually in the laboratory; mortality and infection data were recorded as described earlier. Three field cages (16 ft^2/cage) were each stocked with 30–50 Mormon crickets from each arena and covered with chicken wire to keep out birds. Mormon crickets were fed lettuce and sagebrush daily. Mormon crickets were counted daily, and cadavers were collected and incubated in cups with a moistened cotton ball to diagnose *Beauveria* infection.

Results differed somewhat between the formulations that were used in the field. The statistical results suggested that the ES1 formulation produced less mortality but similar rates of infection than the OF formulations at the 2.7 qt/acre application volume. There were no differences in overall mortality or infection rates between the

0.9 qt/acre and 2.7 qt/acre application volumes of oil alone formulations. It should be noted that while the differences in mortality between formulations at the 2.7 qt/acre application volume may have been statistically significant, they were not substantial (80 *v.* 74 percent at the low conidia concentration).

The application rate of conidia had a more substantial impact on both the overall mortality and percentage of confirmed infections. Adjusted for controls, overall mortality averaged 55 percent and 89 percent for the low and high conidia concentrations, respectively. All comparisons between conidia concentrations were statistically significant.

Conclusions

A detailed understanding of the disease dynamics of the *B. bassiana* isolate will be necessary before this product can be considered for use in an integrated pest management program. Gaining this understanding will entail both laboratory and field studies to evaluate short-term and longrange impacts of *Beauveria* on Mormon crickets. The effects of cannibalism, behavioral fever, and host behavior will need further evaluation before the potential of *B. bassiana* as a microbial control agent against Mormon crickets can be determined. Formulation of *B. bassiana* for Mormon cricket control will also require additional research.

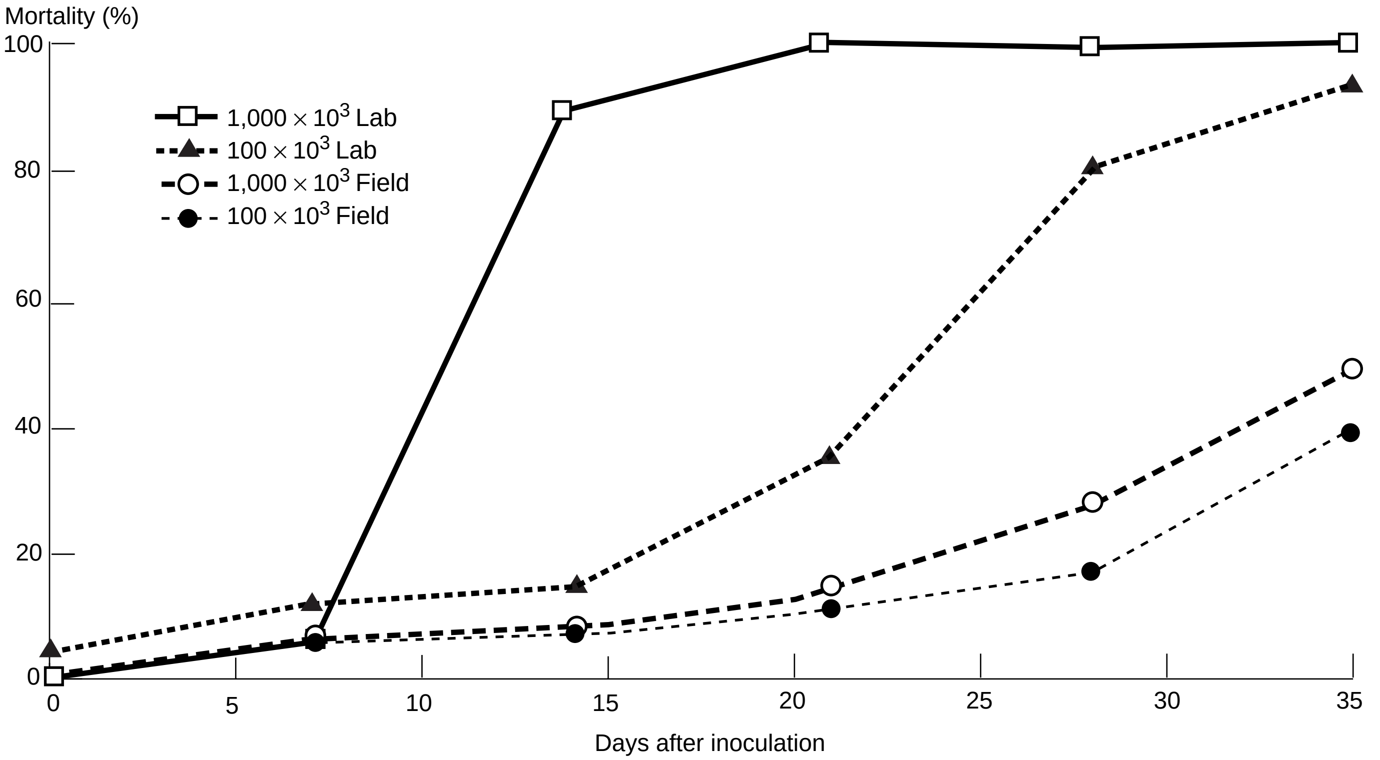

Figure VII.6–3—Cumulative mortality among adult Mormon crickets treated with *Beauveria bassiana* in the lab and reared in the lab or in field cages.

References Cited

Cowan, F. T. 1990. The Mormon cricket story. Spec. Rep. #31. Bozeman, MT: Montana State University and Montana Agricultural Experiment Station. 42 p.

Goettel, M. S.; Roberts, D. W. 1992. Mass production, formulation and field application of entomopathogenic fungi. In: Lomer, C. J.; Prior, C., eds. Biological control of locusts and grasshoppers. Wallingford, UK: CAB International: 230–238.

MacVean, C. 1990. Mormon crickets: a brighter side. Rangelands 12: 234–235.

Pfadt, R. E. 1991. Mormon cricket. In: Field guide to common western grasshoppers. Sta. Bull. 912. Laramie, WY: U.S. Department of Agriculture, Animal and Plant Health Inspection Service and Wyoming Agricultural Experiment Station. 4 p.

Steinhaus, E. A. 1949. Principles of insect pathology. New York: McGraw Hill. 757 p.

Swain, R. B. 1944. Nature and extent of Mormon cricket damage to crop and range plants. Tech. Bull. 866. Washington, DC. U.S. Department of Agriculture. 44 p.

References Cited—Unpublished

Onsager, J. A.; Streett, D. A.; Woods, S. A. 1992. Grasshopper pathogen evaluation. Cooperative Grasshopper Integrated Pest Management Project, 1992 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 179–186.

Kemp, W. P.; Streett, D. A. 1993. Grasshopper pathogen evaluation. Cooperative Grasshopper Integrated Pest Management Project. 1993 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 217–224.

VII.7 Effects of the Fungus *Beauveria bassiana* on Nontarget Arthropods

Mark A. Brinkman, Billy W. Fuller, and Michael B. Hildreth

Introduction

Beauveria bassiana is currently being developed as a potential bioinsecticide alternative to traditional chemical pesticides for controlling grasshopper populations. Currently, *Nosema locustae* is the only other nonchemical treatment registered for control of grasshoppers on rangeland. *B. bassiana* offers at least two major advantages over *N. locustae*: (1) *B. bassiana* appears to kill grasshoppers more rapidly than does *N. locustae* (see VII.5 and I.3), and (2) *Beauveria* does not rely on the ingestion of its spores in a bait formulation by grasshoppers but is capable of directly penetrating through their exoskeleton (Goettel 1992).

Unfortunately, *B. bassiana* may possess at least one potential disadvantage. Unlike the narrow specificity of *N. locustae* for orthopterans (i.e., grasshoppers, locusts and crickets), *B. bassiana* is known to infect a wide variety of insects (Goettel 1992). The wide specificity of *Beauveria* is of concern because distribution of its conidia into the environment also might diminish beneficial insect populations. Attempts have been made to select strains of *B. bassiana* with increased specificity for grasshoppers by selecting stains isolated from grasshoppers (Prior 1992).

Mycotech Corporation (Butte, MT) has mass-produced a strain of *B. bassiana* isolated from an infected grasshopper found in Montana. Laboratory and field studies have indicated that this strain is infectious and lethal in confined populations of several species of grasshoppers (see VII.5). However, no information existed on its virulence in nontarget insects.

In 1993, South Dakota State University (SDSU) assisted the Animal and Plant Health Inspection Service (APHIS) by monitoring the population levels of nontarget arthropods in a *B. bassiana* field study located near Amidon, ND (Brinkman 1995). The grasshopper control data for this study are described in chapter VII.5. Important nontarget arthropods on rangeland include beneficial pollinators (flies and bees), predators (spiders, ants, ground beetles, robber flies, green lacewings, brown lacewings, antlions, ladybird beetles, blister beetles, and wasps), parasites or parasitoids (flies and several hymenopterans) and general scavengers (ants and darkling beetles).

Spray-tower laboratory bioassays as developed by Foster and Reuter (1991) also were used at SDSU to determine the effects of *B. bassiana* on nontarget insects. A spray tower consist of a small airbrush, such as artists use, mounted on a stand and connected to an air pump. A solution of fungal conidia (sporelike stage) can then be injected into the airstream and sprayed onto the insects. This method of conidia application should more closely simulate the field aerial application of conidia than would applying the conidia in a large single drop or by submerging the insects in a solution of conidia (Foster and Reuter 1991).

Adult yellow mealworm beetles *(Tenebrio molitor)* were evaluated with the bioassay because they are easily acquired commercially and have therefore served as research models in many laboratory studies. The species *T. molitor* belongs to the family Tenebrionidae, which is an important group of beetles on western rangeland. This beetle was selected also to represent the many species of beetles evaluated in the field study whose population levels appeared unaffected by the release of *B. bassiana* conidia into their locality.

According to Goerzen et al. (1990), alfalfa leafcutting bees *(Megachile rotundata)* should be considered in evaluations of potential microbial agents. Unfortunately, the low numbers of alfalfa leafcutting bees recovered in field plots prior to the North Dakota study made it impossible to evaluate the effects of *B. bassiana* on this species. Therefore, *M. rotundata* was evaluated in the laboratory bioassay. Spray tower bioassays were first conducted with fourth-instar *Melanoplus sanguinipes* grasshoppers in order to standardize our results with those reported in VII.5.

Field Studies

Methods.—Thirteen days prior to aerial treatments, sampling traps were placed in 4 control plots, 4 carbaryl plots, and 4 plots that were to receive *B. bassiana* at the rate of 9.9 trillion spores/64 oz/acre in oil formulation. Ground-dwelling arthropods were sampled with the use of pitfall traps. Pitfall traps are widemouth quart canning jars placed in the ground with the opening level with the soil surface. Ground-dwelling arthropods were captured, killed, and preserved as they fell into the jars, which contained 70 percent alcohol.

Aerial insects were sampled using malaise traps. Insects were captured by malaise traps as they flew into the netting, and instinctively crawled or flew up into jars at the top. Sampling traps were left in plots for 5 days, and then jars and samples were retrieved. Immediately after treatments, jars were replaced in plots and retrieved every 6 days for the duration of the summer season. Arthropod samples were taken to SDSU to be sorted and identified.

Results.—During the study period, an abnormally high level of precipitation fell on the study plots. The resulting high moisture level was favorable for the natural outbreak of *Beauveria* infections identified in the control grasshoppers from the untreated plots. This natural *Beauveria* outbreak may then have been at least partially responsible for the unexpected erratic results seen in this study in both the treated and untreated plots.

Ant and spider abundance declined in all plots following treatment but rebounded the next week. The sporadic heavy precipitation that occurred following treatment may have resulted in decreased activity of those ground-dwelling arthropods, and thus diminished their chances of falling in the pitfall traps. Therefore, the temporary decrease in ant and spider abundance did not appear to be due to *B. bassiana* or carbaryl treatments. Ground beetle (Carabidae) densities remained stable throughout the summer season.

Flies (Diptera) were the most prevalent aerial insects captured in malaise traps. Abundance of flying Diptera, Hymenoptera, Lepidoptera, Neuroptera, and Coleoptera increased in all plots following treatments. *B. bassiana* and carbaryl applications did not result in any noticeable declines in aerial insect abundance.

Alfalfa leafcutting bees were very rare at the study site. Only three individual Megachilidae were collected in malaise traps during the sampling season. The study site was dominated by mixed grasses, so there was little attraction for pollinating bees. Consequently, we were not able to determine if field applications of *B. bassiana* affected alfalfa leafcutting bees.

Laboratory Studies

Methods.—Fungal conidia (spores) and an oil carrier solution were supplied by Mycotech Corp. Aerial application of *B. bassiana* was simulated in the laboratory with the use of a spray tower. A favorable spray pattern was established in practice tests with the oil solution and the aid of oil-sensitive paper. Procedures, equipment and *B. bassiana* dosages were similar to those described in VII.5 and were selected based on recommendations by Foster and Reuter (1991).

A total of 360 individuals of each species were tested in the laboratory experiments. Prior to each spray event, clean newsprint was placed on the floor of the spray room. In addition, test insects (in groups of 10) were slowed by cooling to 35 °F (1.7 °C). Thirty individuals were sprayed with air for approximately 15 seconds first and were kept as controls. Thirty insects were sprayed with 0.09 mL of the oil carrier. Thirty insects were sprayed with 0.09 mL of oil containing 2.64 billion conidia/mL. Treatments were replicated four times. Insects were then observed for 10 days after treatment.

Results.—Grasshoppers treated with *B. bassiana* began expiring on day 5. After 10 days, more than 73 percent of treated grasshoppers had died. Mortality of beetles treated with *B. bassiana* was extremely low, and beetles did not appear to be susceptible to infection.

B. bassiana was extremely virulent to alfalfa leafcutting bees. Alfalfa leafcutting bees sprayed with *B. bassiana* began expiring on day 4. After 10 days, more than 87 percent of alfalfa leafcutting bees had died. However, mortality of alfalfa leafcutting bees sprayed with oil and air (control) was low. Dead alfalfa leafcutting bees were individually placed in glass vials with a moist cotton ball and were observed for evidence of infection. After approximately 7 days, external sporulation of hyphae (filaments of the vegetative structure of the fungus) was observed on 99 percent of alfalfa leafcutting bees treated with *B. bassiana*.

Conclusions

Treatment of the study sites with *B. bassiana* caused no measurable permanent decrease in populations of any of the monitored beneficial insects. This lack of effect occurred during a time period when moisture levels in the fields were abnormally high, and thus, environmental conditions should have been very good for the spread of the infection into beneficial insects. In fact, even some of the grasshoppers recovered from the control sites also were infected with *Beauveria*, but at low levels and most likely from a natural outbreak.

Spray-tower results on lab-reared grasshoppers were similar to those described in VII.5. The nonsusceptibility of the *Tenebrio molitor* to *B. bassiana* in the spray-tower bioassay was consistent with *Beauveria*'s apparent lack of effect on beetles in the field study. The effects of *B. bassiana* on alfalfa leafcutting bees were evaluated only with the spray-tower bioassay because few bees were recovered in the field. Existing bioassay data indicate that these insects are very susceptible to this strain of *B. bassiana*. Injury to the entire population of alfalfa leafcutting bees might be reduced through management.

B. bassiana conidia can persist if protected from environmental extremes (soil is the natural reservoir for conidia), but become nonviable after only a few hours of exposure to sunlight (Gaugler et al. 1989, see VII.5). Alfalfa leafcutting bees readily accept artificial nesting structures, which could be moved during spray operations and returned later.

References Cited

Brinkman, M. A. 1995. Non-target arthropod impacts and rangeland grasshopper management using *Beauveria bassiana* (Bals.) Vuillemin. Doctoral dissertation. Brookings, SD: South Dakota State University. 62 p.

Foster, R. N.; Reuter, K. C. 1991. Using a spraytower to simulate aerially applied liquid sprays for bioassaying insects in the laboratory: standard operating procedure. Phoenix, AZ: U.S. Department of Agriculture, Animal and Plant Health Inspection Service, Methods Development, Rangeland Section. 5 p.

Gaugler, R.; Costa, S. D.; Lashomb, J. 1989. Stability and efficacy of *Beauveria bassiana* soil inoculations. Environmental Entomology 21(6): 1239–1247.

Goerzen, D. W.; Erlandson, M. A.; Moore, K. C. 1990. Effect of two insect viruses and two entomopathogenic fungi on larval and pupal development in the alfalfa leafcutting bee, *Megachile rotundata* (Fab.) (Hymenoptera: Megachilidae). The Canadian Entomologist 122: 1039–1040.

Goettel, M. S. 1992. Fungal agents for biocontrol. In: Lomer, C. J.; Prior, C., eds. Biological control of locusts and grasshoppers. Wallingford, UK: C.A.B. International: 122–130.

Prior, C. 1992. Discovery and characterization of fungal pathogens for locust and grasshopper control. In: Lomer, C. J.; Prior, C., eds. Biological control of locusts and grasshoppers. Wallingford, UK: C.A.B. International: 159–180.

VII.8 Grasshopper Viruses

D. A. Streett and S. A. Woods

Introduction

Insect poxviruses or “entomopoxviruses” (EPV’s) infect insects from the following five insect orders: Coleoptera (beetles), Lepidoptera (moths and butterflies), Orthoptera (grasshoppers and crickets), Diptera (flies), and Hymenoptera (bees and wasps). The grasshopper EPV’s are found in the genus Entomopoxvirus B, which also includes viruses from Lepidoptera and Orthoptera (Esposito 1991). All grasshopper viruses are physically similar and have roughly the same deoxyribonucleic acid (DNA) size. They differ from EPV’s in other insect orders and other animal poxviruses. Indeed, there is no evidence to suggest any close relationship or similarity between grasshopper entomopoxviruses and other viruses of vertebrate or invertebrates (Langridge 1984).

Virus particles are embedded in a crystalline proteinaceous matrix referred to as an occlusion body (OB). OB’s vary in size from 3 to 12 microns (µm) in diameter and may each contain up to several hundred virus particles. Twelve µm equal about 1/20,000th of an inch. OB’s offer the virus particles some protection from environmental conditions and are thought to be responsible for transmission of a virus from one grasshopper to another. When OB’s are ingested by a grasshopper, the virus particles are released and penetrate through the digestive tract into the body of the grasshopper. Infection by grasshopper EPV’s appears to be restricted to the fat body, a tissue which is used to store food reserves and metabolize food. After the virus particles enter a fat body cell, they replicate and pack the cytoplasm with new OB’s that contain virus particles. Virus particles will also spread to other fat body cells until nearly all the cells in the fat body are infected with virus (Henry et al. 1969, Granados 1981).

EPV’s are the only viruses containing DNA that have been found in field grasshoppers. Typically, an EPV will be named after the host species of the original isolation. Following this convention, there are at least 15 grasshopper EPV’s reported in the literature (Henry and Jutila 1966, Langridge et al. 1983, Oma and Henry 1986, Henry et al. 1985, Wang 1994).

EPV Laboratory Studies

Cross-infection studies have been reported for only seven grasshopper and locust EPV’s (Henry et al. 1985, Oma and Henry 1986, Streett et al. 1990, Lange and Streett 1993). Relative susceptibility of grasshoppers to a given EPV is usually limited to grasshoppers within the same subfamily (Lange and Streett 1993). However, it is interesting to note that some grasshopper EPV’s have been found to infect grasshoppers from several different subfamilies.

Henry and Jutila (1966) isolated the first grasshopper EPV from the lesser migratory grasshopper, *Melanoplus sanguinipes*, a frequent pest on crops and rangeland. The virus, referred to as the *Melanoplus sanguinipes* entomopoxvirus (MsEPV), infects mostly species in the genus *Melanoplus* (Oma and Henry 1986). Grasshoppers infected with a sufficient amount of the virus develop slowly, are sluggish, and die from the effects of the virus (Henry and Jutila 1966).

MsEPV is the only grasshopper EPV that has been grown in vitro (outside the body) (Kurtti et al. 1990 unpubl). The *M. sanguinipes* cell culture lines designated UMMSE–1A, UMMSE–4, and UMMSE–8 have proven susceptible to infection by MsEPV. The UMMSE–4 cell cultures show cytopathic effects (undergo cell changes) when inoculated with MsEPV. The virus produced in vitro is both infectious and virulent (poisonous) against *M. sanguinipes*. Occlusion bodies produced in vitro, though, were somewhat smaller—each about 6 µm in diameter (1/40,000 of an inch)—than occlusion bodies produced in vivo (inside the body). The latter were each about 12 µm in diameter.

In the laboratory, mortality from MsEPV occurs in two distinct timeframes over 5 or more weeks. Infectious OB’s are not present in grasshoppers that die during the first interval of mortality, so these cadavers are of little importance for pathogen transmission. As dosage increases, the proportion of inoculated grasshoppers that die prior to OB formation increases dramatically. Consequently, the proportion of infected grasshoppers that survive long enough to produce OB’s actually decreases with dosage (Woods et al. 1992). These observations suggest that the strategy for using this virus in an integrated pest management program may well depend on the specific objectives at the time of application. Maximum transmission rates are likely to be attained by applying the virus at low rates, and so an EPV treatment may be an appropriate strategy for grasshopper populations that are increasing in density. A high-density population that is already causing significant damage should be treated with high rates to cause substantial early mortality.

Sublethal effects that have been observed for virus-infected grasshoppers include a delay in development, reduction in food consumption, and potential reduction in egg production by the female. All of these sublethal factors can have a profound effect on grasshopper populations.

The delay in development was reported first by Henry et al. (1969) and later by Olfert and Erlandson (1991). In some cases, grasshopper nymphs infected with MsEPV will remain 9 to 18 days longer in an instar. Total food consumption by grasshoppers infected with MsEPV was reduced by 25 percent at 5 days after infection and up to 50 percent at 25 days after infection. This reduction in food consumption in MsEPV-infected nymphs was directly related to dose.

The effects of MsEPV infection on *M. sanguinipes* egg production are unclear. While it has been difficult to thoroughly describe the effects of MsEPV on *M. sanguinipes* egg production, we have observed that development to the adult stage is delayed by infection, and none of the infected adults in our laboratory studies have produced any eggs.

Routes of Transmission

One of the more likely routes of EPV transmission is through the consumption of infected cadavers. Grasshoppers will commonly consume other grasshoppers that are sick or dying. When grasshopper cadavers were placed in the field, nearly 92 percent of the cadavers were almost entirely consumed after 30 minutes (O’Neill et al. 1994). Under high density conditions, there may be considerable competition for these cadavers with the larger individuals successfully defending the resource against smaller intruding grasshoppers (O’Neill et al. 1993). When both infected and uninfected cadavers were placed in the field, there were no significant differences in the number of cadavers that were partially consumed (K. M. O’Neill, unpublished data).

EPV Field Studies

The Environmental Protection Agency granted an Experimental Use Permit (EUP) for field evaluations of MsEPV in 1988. Field evaluations were conducted from 1988 to 1990. Human and domestic-animal safety studies were completed, and no evidence of infectivity was detected in any of the studies. Toxicology data to identify hazards that MsEPV might present to nontarget organisms were also conducted with no evidence of toxicity or pathogenicity (poisonous or disease-related effects) observed in any of the animals examined in these studies. In addition, Vandenberg et al. (1990) did not observe reductions in longevity or pathological effects when MsEPV was tested against newly emerged adult workers of the honeybee, *Apis mellifera.*

Field evaluations of the potential for using MsEPV for grasshopper control were conducted during 1989. Plots were treated with virus that was formulated in starch granules (McGuire et al. 1991). At 13 days after application, prevalence (the number of diseased insects at any given time) was estimated at 14 percent and 23 percent in the plots receiving the low or high application rates, respectively. Prevalence was estimated at 9.2 percent in the control plots at 13 days after application, indicating that considerable dispersal between plots had already occurred (Streett and Woods 1990 unpubl.). Our field studies from 1989 emphasize the problems associated with evaluation of microbial insecticides against insects with considerable dispersal capabilities. That we can infect at least 23 percent of the population with a rate of 10 billion OB’s/acre (24.7 billion OB’s/ha) is clear. The actual infection levels, in view of the dispersal problem and early mortality from the pathogen, are probably much higher.

References Cited

Esposito, J. J. 1991. Poxviridae. In: Francki, R.I.B.; Fraquet, C. M.; Knudson, D. L.; Brown, F., eds. Classification and nomenclature of viruses. Archives of Virology, Supplementum 2: 91–102.

Granados, R. R. 1981. Entomopoxvirus infections in insects. In: Davidson, E. W., ed. Pathogenesis of invertebrate microbial diseases. [Place of publication unknown]: Allanheld, Osman: 101–126.

Henry, J. E.; Jutila, J. W. 1966. The isolation of a polyhedrosis virus from a grasshopper. Journal of Invertebrate Pathology 8: 417–418.

Henry, J. E.; Nelson, B. P.; Jutila, J. W. 1969. Pathology and development of the grasshopper inclusion body virus in *Melanoplus sanguinipes*. Journal of Virology 3: 605–610.

Henry, J. E.; Wilson, M. C.; Oma, E. A.; Fowler, J. L. 1985. Pathogenic micro-organisms isolated from West African grasshoppers (Orthoptera: Acrididae). Tropical Pest Management 31: 192–195.

Lange, C. E.; Streett, D. A. 1993. Susceptibility of Argentine melanoplines (Orthoptera: Acrididae) to entomopoxviruses (Entomopoxvirinae) from North American and African grasshoppers. Canadian Entomologist 125: 1127–1129.

Langridge, W.H.R. 1984. Detection of DNA base sequence homology between entomopoxviruses isolated from Lepidoptera and Orthoptera. Journal of Invertebrate Pathology 43: 41–46.

Langridge, W.H.R.; Oma, E. A.; Henry, J. E. 1983. Characterization of the DNA and structural proteins of entomopoxviruses from *Melanoplus sanguinipes*, *Arphia conspirsa*, and *Phoetaliotes nebrascensis* (Orthoptera). Journal of Invertebrate Pathology 42: 327–333.

McGuire, M. R.; Streett, D. A.; Shasha, B. S. 1991. Evaluation of starch-encapsulation for formulation of grasshopper (Orthoptera: Acrididae) entomopoxviruses. Journal of Economic Entomology 84: 1652–1656.

Olfert, O. O.; Erlandson, M. A. 1991. Wheat foliage consumption by grasshoppers (Orthoptera: Acrididae) infected with *Melanoplus sanguinipes* entomopoxvirus. Environmental Entomology 20: 1720–1724.

Oma, E. A.; Henry, J. E. 1986. Host relationships of entomopoxviruses isolated from grasshoppers. In: Grasshopper symposium proceedings; March 1986; Bismarck, ND. Fargo, ND: North Dakota Extension Service, North Dakota State University: 48–49.

O'Neill, K. M.; Streett, D.; O'Neill, R. P. 1994. Scavenging behavior of grasshoppers (Orthoptera: Acrididae): feeding and thermal responses to newly available resources. Environmental Entomology 23: 1260–1268.

O'Neill, K. M.; Woods, S. A.; Streett, D. A.; O'Neill, R. P. 1993. Aggressive interactions and feeding success of scavenging rangeland grasshoppers (Orthoptera: Acrididae). Environmental Entomology 22: 751–758.

Streett, D. A.; Oma, E. A.; Henry, J. E. 1990. Cross infection of three grasshopper species with the *Melanoplus sanguinipes* entomopoxvirus. Journal of Invertebrate Pathology 56: 419–421.

Vandenberg, J. D.; Streett, D. A.; Herbert Jr., E. W. 1990. Safety of grasshopper entomopoxviruses for caged adult honey bees (Hymenoptera: Apidae). Journal of Economic Entomology 83: 755–759.

Wang, L. Y. 1994. Surveys of entomopoxviruses of rangeland grasshoppers in China. Scientia Agricultura Sinica 27: 60–63.

Woods, S. A.; Streett, D. A.; Henry, J. E. 1992. Temporal patterns of mortality from an entomopoxvirus and strategies for control of the migratory grasshopper *(Melanoplus sanguinipes* (F.). Journal of Invertebrate Pathology 60: 33–39.

References Cited—Unpublished

Kurtti, T. J.; Munderloh, U. G.; Ross, S. E.; Ahlstrand, G. G.; Streett, D. A. 1990. Cell culture systems for production of host dependent grasshopper pathogens. In: Cooperative Grasshopper Integrated Pest Management Project, 1990 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 246–251.

Streett, D. A.; Woods, S.A. 1990. Grasshopper pathogen field evaluation: virus. In: Cooperative Grasshopper Integrated Pest Management Project, 1990 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 210–217.

VII.9 Use of an Australian Parasite of Grasshopper Eggs as a Biological Control Agent

Richard J. Dysart

Introduction

In order to increase the existing mortality level of any pest grasshopper, entomologists are generally limited to two biological control approaches: augmentation or introduction. In the former, some parasite or predator species must be reared in great numbers and distributed evenly over the crop or rangeland to be protected. The augmentation process must be repeated year after year as needed. In the introduction approach, a parasite or predator species, from outside of the system, is imported and colonized, with the intention of obtaining permanent establishment of the natural enemy. Ideally, the natural enemy species would be colonized only once and would spread and distribute itself once established.

Augmentative Approach

In my opinion, using insect parasites or predators augmentatively, as substitutes for chemical insecticides, is not feasible for the control of grasshoppers. The chief obstacle to this approach is the cost. Although certain *Scelio* egg parasites can be reared easily in the laboratory, the rearing process is dependent upon a constant supply of live grasshopper eggs of a certain age. Considering the immense areas that would require treatment with parasites, plus the logistics of rearing and delivery, it is certain that the costs of using *Scelio* wasps augmentatively would be unacceptable.

Classical Introduction Approach

Historical.—According to a worldwide review article by Prior and Greathead (1989), classical biological control of a grasshopper with scelionid wasps has been attempted on only one occasion. The attempt was made in Hawaii, during 1930 and 1931, against the Chinese grasshopper, *Oxya chinensis* (Thunberg), using two parasite species from Malaysia, *Scelio serdangensis* Timberlake and *S. pembertoni* Timberlake (Pemberton 1933, Clausen 1978). *Scelio serdangensis* failed to establish, but *S. pembertoni* became established and is reported to have successfully controlled the pest (Pemberton 1948, Clausen 1978). As pointed out by various authors (Commonwealth Institute of Biological Control 1981, Siddiqui et al. 1986, Greathead 1992), the possibilities for classical introductions against grasshoppers certainly have not been exhausted, particularly with scelionid egg parasites. Worldwide in distribution, the species of the genus *Scelio* are all egg parasites of acridid grasshoppers and there are no host records from any other group of insects (Greathead 1963, Muesebeck 1972, Galloway and Austin 1984).

Rationale for Classical Introduction.—Although there are several native *Scelio* spp. present in western North America, they cause only minor levels of egg mortality. The most abundant and most widespread of our native egg parasites is *Scelio opacus* (Provancher). During an 8-year study in Wyoming, Lavigne and Pfadt (1966) found only trace numbers of *Scelio* parasites in rangeland grasshopper eggs. Results of a long-term study in Saskatchewan (Mukerji 1987) showed that egg parasitism by *Scelio* averaged about 5 percent and had no detectable impact on field populations. In my own field studies in northeastern Montana and northwestern North Dakota from 1988 to 1994, egg-pod parasitism by native *Scelio* spp. averaged 10.7 percent (Dysart 1995), but parasitism of individual eggs was only 4.1 percent (Dysart 1994 unpubl.).

Although the ecological niche is occupied by several native parasites, their total impact on the eggs of pest grasshoppers probably does not affect infestations. Therefore, in 1989, I proposed to the Animal and Plant Health Inspection Service (APHIS) that I try to import and establish an additional species of *Scelio.* If this new parasite became established on one or more of the destructive grasshoppers in the West, it could increase egg mortality and thereby reduce initial densities of nymphs. That scenario could greatly enhance the probability of other indigenous (native) natural enemies maintaining suppression of pest grasshopper densities at or below economic thresholds for greater time intervals.

Periodic outbreaks probably would not be eliminated, but the interval between them might be lengthened or the duration of outbreaks might be shortened. Introduction of exotic parasites to help control indigenous pests is controversial, but as pointed out by Huffaker et al. (1971), there is no pest that should be judged in advance as not amenable to biological control. A good review article on this subject is presented by Carl (1982).

Search for a Candidate *Scelio* in Australia.—In September 1990 and again in 1992, my Australian colleagues and I collected egg-pods of several different grasshoppers and locusts at 10 localities in the States of New South Wales, South Australia, and Western Australia. In September 1992, we made collections in 11 different localities in the same states. A summary of these collections is found in Dysart (1993 unpubl.) and in Baker et al. (in press). In 1990, overall parasitism of egg-pods by *Scelio* spp. was 28 percent (128 of 460 egg-pods), but was highest (36 percent) in Western Australia (66 of 181 egg-pods). During 1990, *Scelio parvicornis* Dodd was the most abundant parasite of the five species reared, and at one locality, Nungarin (Kittyea ranch), in Western Australia, it parasitized about 25 percent of the host egg-pods (Australian plague locust, *Chortoicetes terminifera* [Walker]). Two articles, Baker and Pigott (1993) and Baker et al. (in press), provide additional parasitism and host-range information on *S. parvicornis.* The egg-pod parasitism figures from Australia are considerably higher than those reported above for western North America.

Quarantine Screening in the United States.—Grasshopper egg-pods collected in Australia were kept chilled and were hand-carried to the Montana State University quarantine facility in Bozeman. There the eggs were allowed to hatch, and all Australian grasshopper nymphs were identified and then destroyed. Of the five species of *Scelio* that emerged from the 1990 collections, we investigators selected *Scelio parvicornis* (Nungarin strain) as our primary candidate, based on its dominant position in the Australian collections and its ease of rearing in the quarantine laboratory.

Rearing and Host-Range Tests.—Using nondiapausing eggs of a native pest grasshopper, *Melanoplus sanguinipes* (Fabricius), as hosts, my research team was able to propagate a nondiapausing culture of *S. parvicornis* in the laboratory. Under our lab conditions, we produced a new generation of parasites about every 32 days. In laboratory comparison tests with the native *S. opacus,* females of the Australian *S. parvicornis* were clearly superior: they parasitized more egg-pods and killed more eggs during their respective lifetimes (Dysart 1991 unpubl.). In laboratory host-range tests, we exposed the Australian parasite to about 1,808 egg-pods of 49 species of North American grasshoppers. We obtained emergence of adults of *S. parvicornis* from 33 species, and it failed to emerge from egg-pods of 16 grasshopper species (Dysart 1993 unpubl.). About half of the 33 successful lab hosts of *S. parvicornis* are considered to be our most serious rangeland pests (Hewitt 1977) (see also chapter VI.6).

Plans for Field Releases and Recovery Attempts.— Assuming that permission to release parasites was granted by the Federal and State authorities, I had planned to proceed as follows: colonies of several thousand adult parasites would be released over a period of several weeks at one or more sites in Arizona, Montana, and North Dakota. Prior to releases at proposed sites, screened cages would be erected on sandy soil and furnished with wild female grasshoppers *(M. sanguinipes).* After egg-laying was well under way, adult parasites would be introduced into the cages. The cages would be removed the following spring, and during the next two seasons, egg-pods would be excavated at the site and held for emergence in the laboratory to determine if the Australian parasite had successfully overwintered. If *Scelio parvicornis* is released and becomes established, it will be necessary to conduct additional field studies to assess its impact on pest grasshopper populations.

Addendum.—I made my initial request to U.S. Department of Agriculture, APHIS, Plant Protection and Quarantine, Biological and Taxonomic Support (USDA, APHIS, PPQ, BATS) for permission to release *Scelio parvicornis* in the summer of 1991. Periodically during 1992 and 1993, I provided BATS with revisions and support documents as they continued to prepare their risk assessment (Lakin 1994 unpubl.). The question of whether or not the Australian parasite should be released in North America has been the subject of active debate in the literature, between Lockwood (1993a and b) and Carruthers and Onsager (1993). Lockwood is opposed to the field release of the parasite because he feels that its potential host range is too broad, and he speculates that it might have a detrimental effect on benign, nonpest grasshoppers as well as a few grasshoppers thought to be beneficial because they feed on rangeland weeds. Carruthers and Onsager believe that the release of the Australian egg parasite is warranted and that the risk of harm to nontarget species is negligible at best.

On April 6, 1994, I received word from the permitting agency, USDA, APHIS, PPQ, BATS, that my application for the release of *Scelio parvicornis* had been denied. As a result, I have destroyed the laboratory colony and have abandoned my plans for field releases of the parasite. I still believe that the overall benefits of the proposed biological control introduction would outweigh any potential risks, but for the time being, the outcome will remain a matter of conjecture.

References Cited

Baker, G. L.; Pigott, R. G. 1993. Parasitism of *Chortoicetes terminifera* (Walker) (Orthoptera: Acrididae) eggs by *Scelio parvicornis* Dodd (Hymenoptera: Scelionidae). Journal of the Australian Entomological Society 32: 121–126.

Baker, G. L.; Dysart, R. J.; Pigott, R. G. In press. Parasitism of grasshopper and locust eggs (Orthoptera: Acrididae) by *Scelio* spp. (Hymenoptera: Scelionidae) in southern Australia. Australian Journal of Zoology.

Carl, K. P. 1982. Biological control of native pests by introduced natural enemies. Biocontrol News and Information 3(3): 191–200.

Carruthers, R. I.; Onsager, J. A. 1993. Perspective on the use of exotic natural enemies for biological control of pest grasshoppers (Orthoptera: Acrididae). Environmental Entomology 22: 885–903.

Clausen, C. P. 1978. Orthoptera, Acrididae. In: Clausen, C. P., ed. Introduced parasites and predators of arthropod pests and weeds: a world review. Agric. Handb. 480. Washington, DC: U.S. Department of Agriculture, Agricultural Research Service: 9–10.

Commonwealth Institute of Biological Control. 1981. Possibilities for biological control of grasshoppers (Acridoidea) status paper. Plant Protection Bulletin 33(3&4): 157–160.

Dysart, R. J. 1995. New host records for North American *Scelio* (Hymenoptera: Scelionidae), parasitic on grasshopper eggs (Orthoptera: Acrididae). Journal of the Kansas Entomological Society 68(1): 74–79.

Galloway, I. D.; Austin, A. D. 1984. Revision of the Scelioninae (Hymenoptera: Scelionidae) in Australia. Australian Journal of Zoology. Supplementary Series 99: 1–138.

Greathead, D. J. 1963. A review of the insect enemies of the Acridoidea (Orthoptera). Transactions of the Royal Entomological Society of London 114: 437–517.

Greathead, D. J. 1992. Natural enemies of tropical locusts and grasshoppers: their impact and potential as biological control agents. In: Lomer, C. J.; Prior, C., eds. Biological control of locusts and grasshoppers. Wallingford, UK: C.A.B. International: 105–121.

Hewitt, G. B. 1977. Review of forage losses caused by rangeland grasshoppers. Misc. Publ. 1348. Washington, DC: U.S. Department of Agriculture, Agricultural Research Service: 1–24.

Huffaker, C. B.; Messenger, P. S.; DeBach, P. 1971. The natural enemy component in natural control and the theory of biological control. In: Huffaker, C. B., ed. Biological control. New York: Plenum Press: 16–67.

Lavigne, R. J.; Pfadt, R. E. 1966. Parasites and predators of Wyoming rangeland grasshoppers. Sci. Monogr. 3. Laramie, WY: Wyoming Agricultural Experiment Station: 1–31.

Lockwood, J. A. 1993a. Environmental issues involved in the biological control of rangeland grasshoppers with exotic agents. Environmental Entomology 22: 503–518.

Lockwood, J. A. 1993b. Benefits and costs of controlling rangeland grasshoppers (Orthoptera: Acrididae) with exotic organisms: search for a null hypothesis and regulatory compromise. Environmental Entomology 22: 904–914.

Muesebeck, C.F.W. 1972. Nearctic species of Scelionidae (Hymenoptera: Proctotrupoidea) that parasitize the eggs of grasshoppers. Smithsonian Contribution to Zoology 122: 1–33.

Mukerji, M. K. 1987. Parasitism by *Scelio calopteni* Riley (Hymenoptera: Scelionidae) in eggs of the two dominant melanopline species (Orthoptera: Acrididae) in Saskatchewan. Canadian Entomologist 119: 147–151.

Pemberton, C. E. 1933. Introduction to Hawaii of Malayan parasites (Scelionidae) of the Chinese grasshopper *Oxya chinensis* (Thun.) with life history notes. Proceedings of the Hawaiian Entomological Society 8(2): 253–264.

Pemberton, C. E. 1948. History of the entomology department experiment station, H.S.P.A. 1904–1945. Hawaiian Planters' Record 52: 53–90.

Prior, C.; Greathead, D. J. 1989. Biological control of locusts: the potential for the exploitation of pathogens. Food and Agriculture Plant Protection Bulletin 37: 37–48.

Siddiqui, R. K.; Irshad, M.; Mohyuddin, A. I. 1986. Digest: *Scelio* spp. as biocontrol agents of acridids. Biocontrol News and Information 7(2): 69–76.

References Cited–Unpublished

Dysart, R. J. 1991. Impact of insect parasites and predators on grasshopper populations II. In: Cooperative Grasshopper Integrated Pest Management Project, 1991 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 237–242.

Dysart, R. J. 1993. Impact of insect parasites and predators on grasshopper populations II. In: Cooperative Grasshopper Integrated Pest Management Project, 1993 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 245–254.

Dysart, R. J. 1994. Impact of insect parasites and predators on grasshopper populations II. In: Cooperative Grasshopper Integrated Pest Management Project, 1994 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 223–226.

Lakin, K. R. 1994. Assessment of the proposed release of *Scelio parvicornis* in the United States. Hyattsville, MD: U.S. Department of Agriculture, Animal and Plant Health Inspection Service, Plant Protection and Quarantine, Biological Assessment and Taxonomic Support, April 5, 1994. 34 p.

VII.10 Ongoing Environmental Concerns

L. C. McEwen

NOTE: Acephate is no longer approved by EPA for rangeland grasshopper control.

Perhaps the greatest continuing environmental concern in a Grasshopper Integrated Pest Management (GHIPM) program is providing safeguards and protection for threatened and endangered (T and E) plant and animal species. These problems complicate grasshopper control programs and make them more costly but must be dealt with in a straightforward manner. Plenty of lead time should be allowed to identify species and habitats and to work out solutions with agencies responsible for T and E species' protection and management.

Recognition of the fact that individual vertebrate animals can vary greatly in their sensitivity to a given toxic chemical should help all workers understand that toxic exposure of the T and E species must be kept to a minimum. Toxic hazard is minor for mature animals lightly exposed to the current GHIPM pesticides—carbaryl, malathion, and acephate—but is probably more of a factor for young animals (chicks, nestlings, amphibians, and larval fish). Any toxic mortality would be of concern because species differ in their lower threshold of numbers of animals necessary for maintaining a viable population. Those limits are not known precisely for each species, but land managers should try hard not to cause unnecessary losses with toxic chemicals.

In the larger picture, it would seem that concern for geographic variants that have been given T and E status should not be on the same level as for T and E species that are the sole remaining population or individuals. Technically and legally, however, there is no distinction at this time.

T and E species can be protected in several ways in a rangeland grasshopper cooperative control program. Nonspray buffer zones are one of the main tools (see chapter III.8). Width and size of buffer zones will vary with the T and E species and on the outcome of consultation with managing agencies. Carbaryl bait treatments or other dry baits, including biological control agents such as *Nosema locustae* and *Beauveria bassiana*, can be used safely much closer to the T and E species habitat or even with no buffer zone in some cases.

Baits and biologicals add expense and sometimes cause equipment problems when used but should be recognized and accepted as important and necessary components of many successful programs. The degree of grasshopper reduction will probably be less than where liquid insecticide spray is applied, but the higher densities of grasshoppers remaining after the treatment often will be beneficial to the T and E species.

Another possible option for protecting T and E species is the timing of the grasshopper control program. This aspect can be explored for T and E insects and pollinators of T and E plants (also see chapter III.5). If the T and E insects are in the adult stage for a relatively brief period, then pest managers may conduct treatments safely before or after the adult stage.

For aquatic species, there are significant differences in toxicity among the three chemicals. Acephate is much less toxic to fish than carbaryl or malathion (Johnson and Finley 1980) and is referred to in other publications as practically nontoxic to fish. Acephate is highly effective against grasshoppers at the low application rate of 1.5 oz/acre (0.105 kg/ha) (U.S. Department of Agriculture 1987). Although acephate has been little used in cooperative control programs, it could be an excellent alternative to other pesticides where T and E fish are of concern. Another safety factor for fish would be to use dry bait treatments because less chemical is used per unit area and there is much less potential for drift into aquatic habitat. The entire problem of T and E species protection in GHIPM programs could benefit from further research.

Indirect Effects on T and E Species

The question of indirect effects of grasshopper control programs, primarily reduction or loss of the food base for birds, now comes up more frequently than potential toxic effects. Colorado State University (CSU)-led studies have shown that when grasshopper availability is reduced, birds generally switch to other insects or invertebrates for food and maintain their nesting success and populations (Miller 1993, Miller and McEwen 1995, Miller et al. 1994, George et al. 1995, Fair et al. 1995). Regarding the concern for peregrine prey effects, CSU investigators have shown that total bird population numbers do not decline following a grasshopper control program, even though some individual species might decrease (George et al. 1995). Since peregrines prey on such a wide variety of avian species (DeWeese et al.

1986, Hunter et al. 1988), the decline of one or two species should have no significant effect on their prey base. Use of dry baits, such as carbaryl bait, also could be a safeguard since the baits are selective formulations and consequently leave many unaffected insects for avian food (Adams et al. 1994).

Nevertheless, each T and E species must be examined individually for potential response to GHIPM treatments. The situation is such that T and E species and their habitats cannot be dealt with routinely by generalized procedures. Each T and E situation must be treated as a unique "case history," although as knowledge is acquired, some will be more standardized than others.

New Chemicals and Biologicals

New materials for range grasshopper control, such as Dimilin® (diflubenzuron) and *Beauveria bassiana,* will require close monitoring until their environmental safety is determined. The two materials appear quite safe for terrestrial vertebrates, but final determinations cannot be made until the materials are applied in large-scale operational control programs. Aquatic effects are especially of concern as well as Acridid (grasshopper) specificity and effects on nontarget invertebrates. Any other candidate chemicals and biologicals that are considered for GHIPM must also be closely examined for environmental effects before being approved for large-scale use.

Species of Concern

State and Federal wildlife agencies in recent years have endorsed a philosophy of giving attention to declining species **before** they reach T and E status. If a declining species can be managed for recovery before listing, management efforts are simplified. Declining species may be designated as "species of concern." Some examples are the long-billed curlew *(Numenius americanus),* the western burrowing owl *(Athene cunicularia),* and the ferruginous hawk *(Buteo regalis).* The curlews and burrowing owls use grasshoppers heavily, especially as a source of protein and nutrients important for breeding and for feeding their young. The golden eagle *(Aquila chrysaetos)* is another species of concern in some areas of the West and is a protected species. There is a need to conduct a study of the response of nesting golden eagles to malathion spray as was done with Sevin® 4-Oil. One or more of several species of concern are apt to be present in GHIPM treatment areas and should be treated as T and E species if necessary in the opinion of the biologists and land managers involved.

Gallinaceous birds, such as prairie chickens and sharptailed grouse *(Tympanuchus* spp.), sage grouse *(Centrocercus urophasianus),* chukars *(Alectoris chukar),* and wild turkeys *(Meleagris gallopavo),* also often are considered species of concern. The effects of grasshopper control on the growth and survival of the young chicks and poults is the primary question. More study is needed on the effects of GHIPM programs on species of concern.

Function of Wildlife in a GHIPM System

Scientists and land managers have made a lot of progress in showing the role and benefits of wildlife, especially birds, as important contributors to regulation of grasshopper densities (Joern 1986, Fowler et al. 1991, Bock et al. 1992). However, the overall ecology of native wild vertebrates in preventing insect pest outbreaks is virtually unexplored. The interrelationships of range condition, vegetative cover types, native plants *vs.* introduced species for reseeding (such as crested wheatgrass, *Agropyron cristatum),* and associated wildlife populations need much more investigation. Large expanses of crested wheatgrass become devoid of almost all the breeding avian species (Reynolds and Trost 1980). In the northern Great Plains, grasshopper outbreaks frequently originate in crested wheatgrass, where grasshopper densities are usually higher than on native grass range (Hirsch et al. 1988 unpubl., Kemp and Onsager 1994 unpubl.). This fact should not be surprising because the lack of birds as grasshopper predators is coupled with >40 percent bare ground (compared to <5 percent in native grassland (Dormaar et al. 1995), which is favored by many grasshoppers for egg-laying.

Range condition criteria are currently undergoing review and revision (Task Group on Unity in Concepts and Terminology 1995). Land managers need to relate range wildlife habitat use and populations to condition classes and to grasshopper population fluctuations. Improving range condition is a long, slow process, but range in good condition with a full complement of native wildlife can reduce grasshopper population fluctuations in the central and northern Great Plains (McEwen 1987). Improving the condition of degenerated sagebrush *(Artemisia* spp.) range found farther west is more difficult than improving other range types, but it should be a long-term goal (McEwen and DeWeese 1987). New range management practices (Biondini and Manske 1996; Onsager, in press) should be examined for wildlife responses.

The status and function of wild vertebrates in relation to range condition also need more investigation. Basic knowledge of range wildlife ecology connects with the efforts to improve the vegetative cover on western rangelands. Preventing the extinction of animal and plant species is the goal of conservation biology and will be a benefit of better range condition. This will also be an important factor contributing to grasshopper management in an IPM system.

References Cited

Adams, J. S.; Knight, R. L.; McEwen, L. C.; George, T. L. 1994. Survival and growth of nestling vesper sparrows exposed to experimental food reductions. The Condor 96: 739–748.

Biondini, M. E.; Manske, L. L. 1996. Grazing frequency and ecosystem processes in a northern mixed prairie. Ecological Applications 6: 239–256.

Bock, C. E.; Bock, J. H.; Grant, M. C. 1992. Effects of bird predation on grasshopper densities in an Arizona grassland. Ecology 73: 1706–1717.

DeWeese, L. R.; McEwen, L. C.; Hensler, G. L.; Petersen, B. E. 1986. Organochlorine contaminants in Passeriformes and other avian prey of the peregrine falcon in the Western United States. Environmental Toxicology and Chemistry 5: 675–693.

Dormaar, J. H.; Naeth, M. A.; Williams, W. D.; Chanasyk, D. S. 1995. Effect of native prairie, crested wheatgrass *(Agropyron cristatum)* (L.)(Gaertn.) and Russian wildrye *(Elymus junceus* Fisch.) on soil chemical properties. Journal of Range Management 48: 258–263.

Fair, J. M.; Kennedy, P. L.; McEwen, L. C. 1995. Effects of carbaryl grasshopper control on nesting killdeer in North Dakota. Environmental Toxicology and Chemistry 14: 881–890.

Fowler, A. C.; Knight, R. L.; George, T. L.; McEwen, L. C. 1991. Effects of avian predation on grasshopper populations in North Dakota grasslands. Ecology 72(5): 1775–1781.

George, T. L.; McEwen, L. C.; Petersen, B. E. 1995. Effects of grasshopper control programs on bird populations in western rangelands. Journal of Range Management 48: 336–342.

Hunter, R. E.; Crawford, J. A.; Ambrose, R. E. 1988. Prey selection by peregrine falcons during the nestling stage. Journal of Wildlife Management 52: 730–736.

Joern, A. 1986. Experimental study of avian predation on coexisting grasshopper populations (Orthoptera: acrididae) in a sandhills grassland. Oikos 46: 243–249.

Johnson, W. W.; Finley, M. T. 1980. Handbook of acute toxicity of chemicals to fish and aquatic invertebrates. Publ. 137. Washington, DC: U.S. Department of the Interior, U.S. Fish and Wildlife Service. 98 p.

McEwen, L. C. 1987. Function of insectivorous birds in a shortgrass IPM system. In: Capinera, J. L., ed. Integrated pest management on rangeland: a shortgrass prairie perspective. Boulder, CO: Westview Press: 324–333.

McEwen, L. C.; DeWeese, L. R. 1987. Wildlife and pest control in the sagebrush ecosystem: ecology and management considerations. In: Onsager, J. A., ed. Integrated pest management on rangeland: State of the art in the sagebrush ecosystem. Bull. ARS–50. Washington, DC: U.S. Department of Agriculture, Agricultural Research Service: 76–85.

Miller, C. K. 1993. Responses of nesting savannah sparrows to fluctuations in grasshopper densities in interior Alaska. M.S. thesis. Ft. Collins, CO: Colorado State University. 24 p.

Miller, C. K; Knight, R. L.; McEwen, L. C. 1994. Responses of nesting savannah sparrows to fluctuations in grasshopper densities in interior Alaska. Auk 111: 960–967.

Miller, C. K.; McEwen, L. C. 1995. Diet of nesting savannah sparrows in interior Alaska. Journal of Field Ornithology 66: 152–158.

Onsager, J. A. [In press.] Suppression of grasshoppers in the Great Plains through grazing management. Journal of Range Management. [Accepted for publication December 1, 1999.]

Reynolds, T. D.; Trost, C. H. 1980. The response of native vertebrate populations to crested wheatgrass planting and grazing by sheep. Journal of Range Management 31: 122–125.

Task Group on Unity in Concepts and Terminology. 1995. New concepts for assessment of rangeland condition. Journal of Range Management 48: 271–282.

U.S. Department of Agriculture, Animal and Plant Health Inspection Service. 1987. Rangeland Grasshopper Cooperative Management Program: final environmental impact statement. Washington, DC: U.S. Department of Agriculture, Animal and Plant Health Inspection Service. 221 p.

References Cited–Unpublished

Hirsch, D. C.; Reuter, K. C.; Foster, R. N. 1988. Comparison of grasshopper population characteristics in relation to grassland types in western North Dakota in 1987. In: Cooperative Grasshopper Integrated Pest Management Project, 1988 annual report. Boise ID, U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 244–252.

Kemp, W. P.; Onsager, J. A. 1994. Grasshopper population response to modification of vegetation by grazing. In: Cooperative Grasshopper Integrated Pest Management Project, 1994 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 93–98.

VII.11 Implications of Ecosystem Management and Information-Processing Technologies

W. P. Kemp, D. McNeal, and M. M. Cigliano

Ecosystem Management and Public Lands

A very large portion of the millions of rangeland acres in the 17 Western United States resides within the boundaries of what many refer to as the public land trust, or federally managed lands. Voters have demanded that the public servants who manage these lands employ "ecosystem management" to provide, among other things, a safe food supply while not compromising natural resources like clean air, clean water, productive soils, and biodiversity. Private interests who lease grazing rights from the various public agencies charged with managing our national land treasure must comply with the public's wishes regarding resource management issues or risk losing the opportunity of using those public lands.

At present, agencies involved in managing the natural resources on public lands are struggling to define just what constitutes ecosystem management, how to manage ecosystems whose limits do not agree with political or ownership boundaries, and how to conduct such management with dwindling agency resources. For example, there is general agreement throughout public land-management agencies that an ecosystem focus is desirable in managing the natural resources of public lands. There also is a nagging concern that agencies don't have a very clear vision of just how much information is necessary to meet national objectives. Furthermore, it is obvious that agencies will have to make natural resource management decisions without complete information. Unfortunately, just what constitutes "enough" or "sufficient" ecosystem management will likely emerge only after and as a direct result of a series of court decisions.

Agencies cannot predict with absolute certainty what the result of the ecosystem management consensus-building process will be, nor can they forecast the specific impacts ecosystem management will have on integrated pest management (IPM) of public lands. The executive branch of the Federal Government has provided some expected outcomes, at least in general terms (Gore 1993, National Research Council 1993).

In the case of rangeland grasshopper integrated pest management (IPM), many believe that the amount of information needed to conduct management action (for example, chemical, biological, or cultural control) will be staggering in an effort to satisfy the need of policymakers to feel confident in presenting results for public viewing. Add to this the challenge of a short interval between problem identification and the time when action must be taken if it is to be effective for rangeland grasshopper IPM on public lands. It is clear that scientists and land managers face an information-gathering and -processing crisis. The remainder of this chapter will focus on ways that agencies can address this crisis that is already upon the country.

Present and Future IPM Technologies

In spite of the information crisis faced with IPM on public lands, there are technologies available that agencies managing public lands can use in an attempt to comply with societal mandates. Other chapters in this Handbook discuss global positioning system (GPS) and geographic information systems (GIS) for aircraft guidance (see section II) as well as for IPM in general (see chapter VI.9). The current role of modeling and decision support systems (DSS) also is discussed in the Decision Support Tools section. This chapter will focus on information processing technologies and a new paradigm (example or model) in the context of IPM systems to be developed for rangeland grasshoppers on public lands.

There are at least five areas of information-processing technology that deserve additional attention in the development of IPM systems for rangeland grasshoppers on public lands, under the umbrella of ecosystem management. These are GPS, GIS, remotely sensed information, DSS, and networks. Three of the five areas—GPS, GIS, and remotely sensed information (see details in chapter VI.9) can be classified as technologies that assist land managers in collecting and storing information about the ecosystems that they are responsible for managing. On the other hand, DSS and networks will be central to actually processing the mountains of available information and developing the most appropriate management of a rangeland grasshopper problem on a particular piece of public rangeland.

Fortunately for public land-management agencies, there is a very competitive software and hardware market associated with GPS, GIS, and remote sensing technologies at present. This competition is likely to continue well into the future. Such competition in the private sector of the U.S. economy will result in a steady and timely stream of products for use in collecting and storing information about the ecosystems that must be managed. Similar statements can also be made for the networking industry as everyone anticipates "information highways" of the future.

Perhaps the most serious challenge that agencies face in attempting to implement ecosystem management in general, and rangeland grasshopper IPM in particular, is the development and maintenance of DSS. DSS such as Hopper, developed from funding provided by the Grasshopper Integrated Pest Management (GHIPM) Project, must continually be updated and expanded to have any hope of processing the ecosystem information that is accumulating. In addition to defining who will be responsible for the continued development of DSS, agencies need coordinated planning to ensure that research emerging from Federal, private, and State laboratories will continue to support DSS improvements.

We must note that, although technologies may be sufficiently well developed for implementation and public land-management agencies may be interested in adopting such technologies, costs will increase. This is true because of the significant increase in the information-processing tasks presented by the implementation of ecosystem management on public lands. The efficiencies of operation with the equipment that is available today exceed even wild dreams of 10 years ago. Public land-management agencies are working feverishly to embrace new technologies. There now is uncertainty whether the resources will be forthcoming to do the job right.

Getting Organized

In this section, we offer some specific suggestions on how to coordinate future rangeland grasshopper IPM with Federal land-management agencies. First, the concept of ecoregion—regional areas (fig. VII.11–1) with similar environmental resources, ecosystems, and sensitivities to human impacts (Bailey 1980, Omernik 1987 and 1995) is useful for organizing information concerning all aspects of grasshopper management. This is a somewhat different use of the concept than was discussed in the environmental impact statement that governed the GHIPM Project (U.S. Department of Agriculture, Animal and Plant Health Inspection Service [APHIS] 1987).

Instead of simply acknowledging that there are broad ecological differences in the Western United States, agencies should use the concept of the ecoregion as a fundamental organizational paradigm. Bailey (1980) suggested that the regionalization (for example, fig. VII.11–1) that results from accepting this paradigm helps "(1) planning at the national level, where it is necessary to study management problems and potential solutions on a regional basis; (2) organization and retrieval of data gathered in a resource inventory; and (3) interpretation of inventory data, including differences in indicator plants and animals among regions." In our opinion, the capabilities that agencies have with GIS presently permit them to apply the ecoregion concept in ways that have until now escaped scientists and land managers.

"Ecoregion" relates to the ability of the land to produce goods and services that humans can use. Furthermore, historically sustainable activities related to grasslands have to a large extent been molded by the prevailing conditions—expressed by ecoregion. For example, the differences in ranching styles and associated economics across the Western United States that economists have been talking about are no doubt related to the fact that ranching has evolved in each region in response to the environmental limitations (again, expressed as ecoregion).

Currently, Hopper (see VI.2) has been developed for only a part of the total area over which there is the opportunity to use it. Furthermore, when land managers look at rangeland grasshopper economic injury levels (EIL) for widely separated areas, such as Wyoming and New Mexico, it is becoming more and more clear how important the regional perspective can be. For example, recent results suggest that it may take three to four times as many grasshoppers in New Mexico versus Wyoming before management treatments would be justified economically. In any case, whether agencies call them ecoregions or rename them as management regions for the needs of APHIS, Plant Protection and Quarantine (PPQ) activities, figure VII.11–1 represents a scale that is a good first attempt to capture the variability across the grasslands of the United States without overburdening people with too much detail.

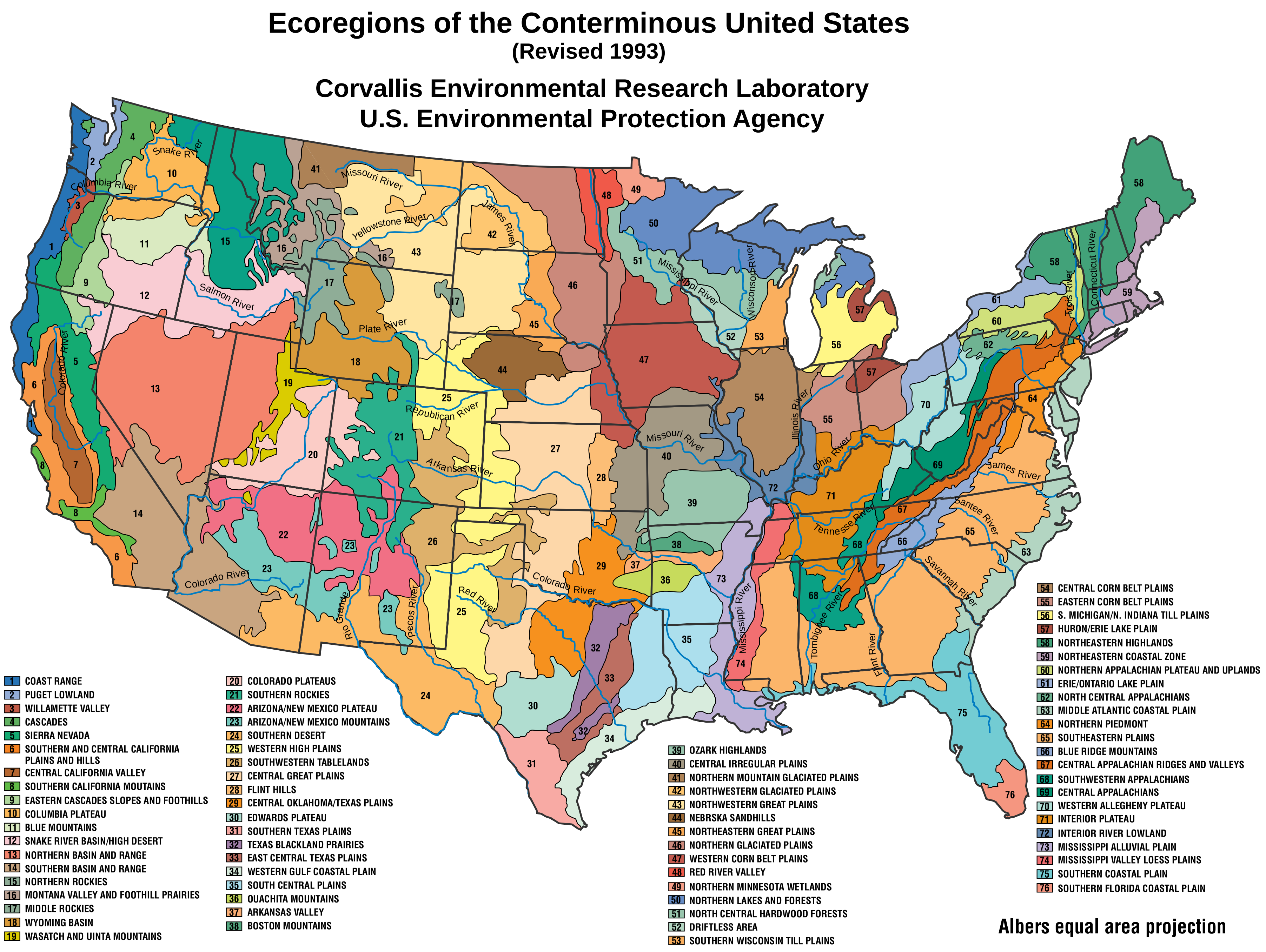

Figure VII.11–1—Ecoregions of the conterminous United States (adapted from a map in Omernik 1987 and 1995.)

The ecoregion concept is useful for exchanging information about environmental conditions, plant production, ranching, and grasshopper ecology and management (from hatching to outbreak frequency and probability and more). There is a credible argument for the use of the concept of ecoregion as a framework for the development of future rangeland grasshopper cooperative management program final environmental impact statements (FEIS's). The ecoregion concept also has potential application for other pest-related issues (for example, noxious weeds) with which APHIS, PPQ and Federal land-management agencies must deal.

In the development of any future FEIS activities, pest managers first should organize rangeland grasshopper IPM activities to be responsive to the situations recognized within each ecoregion. Next, agencies should acknowledge that IPM is the collection of options (including no action) and philosophies most appropriate for addressing grasshopper management. Considering the variation in grassland vegetation and climate depicted in figure VII.11–1 and associated variations in grasshopper populations (for example, Kemp et al. 1990), it is very unlikely that all management options will be equally viable (as viewed by environmentalists, economists, and the public) or of constant efficacy across the rangelands of the 17 Western United States. If this approach to management is acceptable, then there is a logical manner for studying and determining what to emphasize in terms of IPM components at the ecoregion level.

Using this approach as an example, the tabulation in the right column illustrates one way to organize an FEIS.

Organization scheme for a Final Environmental Impact Statement for a Rangeland Grasshopper Cooperative Management Program

Level 1: Ecoregions—regional variations in climate, vegetation, and landform. This is the basis for organizing what agencies know as well as what and how agencies will manage.

Level 2: Things that are likely to be different by ecoregion and that should be considered in any future activities related to the Rangeland Grasshopper Cooperative Management Program FEIS (this list is not meant to be all-inclusive):

- Grasshopper community species composition,
- Likelihood of grasshopper outbreaks,
- Spatial extent of grasshopper outbreaks,
- General insect–animal community composition,
- Grassland plant community composition,
- Forage production on grasslands,
- Economics of ranching and farming (and thus land use and human population density),
- Economics of grasshopper control and EIL,
- Endangered species,
- Soils (and thus water and pesticide movement), and
- Water resources.

1993 Grasshopper Survey

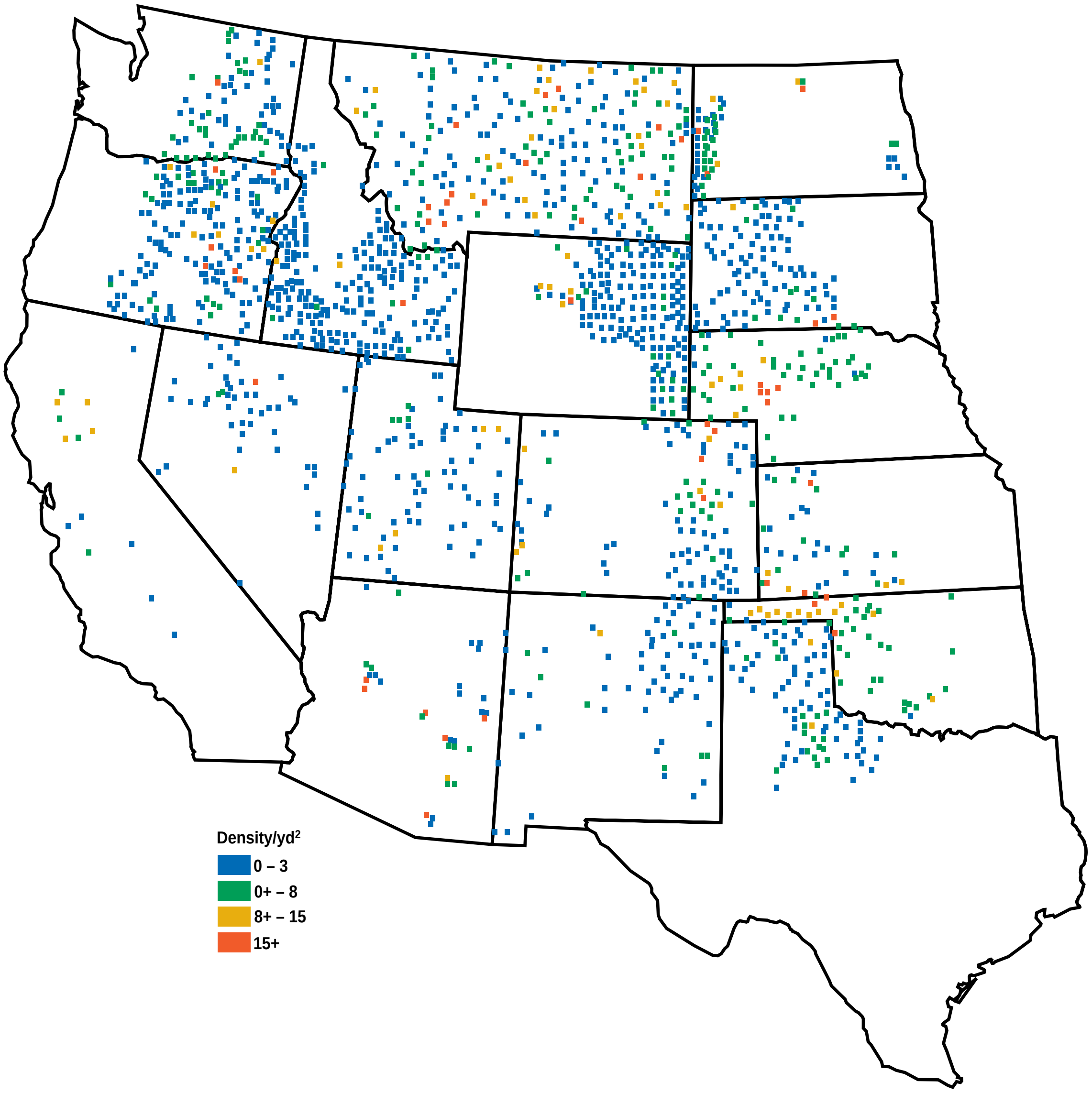

Figure VII.11–2—Locations in the 17 Western United States where (starting in 1993) rangeland grasshoppers were sampled annually for density and species composition by USDA, APHIS, PPQ and cooperators for the Grasshopper Common Dataset Project. Colors indicate grasshopper density at each location in 1993.

The ecoregion paradigm, in addition to being politically and environmentally acceptable (see Gore 1993, National Research Council 1993), can provide Federal land management agencies and APHIS, PPQ with a powerful tool for organizing and interpreting research results relative to rangeland grasshoppers. For example, discussions among a number of GHIPM Project participants and APHIS, PPQ staff eventually resulted in the initiation of the Grasshopper Common Dataset (GCD) during 1993. Scientists now are monitoring rangeland grasshopper communities annually at more than 1,500 locations throughout the 17 Western United States (fig. VII.11–2). Results from ongoing research by GHIPM Project cooperators, with data from the GCD, will tell to what extent grasshopper communities are sensitive to the ecoregion boundaries shown in figure VII.11–1. Given that scientists are able to identify ecological boundaries that are in some way meaningful to the insects, scientists and land managers should apply this concept to assist them in organizing the way that they think about things like rangeland grasshopper management on grasslands west of the 100th meridian of the United States.

In summary, the four main points that we wish to emphasize are

1. GPS, GIS, remote sensing, networking, and DSS will be necessary for ecosystem management of public rangelands.

2. The ecoregion concept is useful, deserves additional consideration by Federal land-management agencies and APHIS, PPQ, and could serve as a useful paradigm for organizing future environmental impact statements related to rangeland grasshoppers (and possibly other insects).

3. By accepting the ecoregion concept, agreeing that IPM is the basis for all grasshopper management, and accepting that IPM consists of all possible alternatives and philosophies as above, agencies eventually could develop ecoregion-specific IPM prescriptions for rangeland grasshopper management.

4. Given 1–3, the regionality provided by the ecoregion concept has great potential for clarifying the goals and objectives of research that Federal land-management agencies and APHIS, PPQ should obtain through contracts and cooperative ventures.

References Cited

Bailey, R. G. 1980. Description of the ecoregions of the United States. Misc. Publ. 1391. Washington, DC: U.S. Department of Agriculture, Forest Service. 77 p.

Gore, A. 1993. Earth in the balance: ecology and the human spirit. New York: Plume. 408 p.

Kemp, W. P.; Harvey, S. J.; O'Neill, K. M. 1990. Habitat and insect biology revisited: the search for patterns. American Entomologist 36: 44-48.

National Research Council. 1993. A biological survey for the Nation. Washington, DC: National Research Council, National Academy Press. 205 p.

Omernik, J. M. 1987. Ecoregions of the coterminous United States. Annual of the Association of American Geographers 77: 118–125.

Omernik, J. M. 1995. Ecoregions: a spatial framework for environmental management. In: Simon, T.; Davis, W., eds. Biological assessment and criteria: tools for water resource planning and decision making. Chelsea, MI: Lewis Publishers: 49–62.

U.S. Department of Agriculture, Animal and Plant Health Inspection Service. 1987. Rangeland Grasshopper Cooperative Management Program: final environmental impact statement. Washington, DC: U.S. Department of Agriculture, Animal and Plant Health Inspection Service. 221 p.

VII.12 Rangeland Environmental Amenities and Grasshopper Management Programs

Melvin D. Skold and Andrew W. Kitts

Range Ecosystems

Rangelands are increasingly recognized as important for their environmental and recreational amenities. Because they are managed much less intensively than many other types of agricultural lands, rangelands are seen to represent closer approximations to natural ecosystems. Rangelands are managed for a variety of outputs; in recent years, the contribution of natural rangeland systems to biological diversity has become increasingly recognized.

Rangelands provide two major values, those associated with use (use values) and those realized in the absence of direct use (existence and option or nonuse values). The major commercial use (use values) of rangelands is livestock grazing to produce food, fiber, and draft animals. Other, less significant, commercial uses such as wild game and bird hunting also are associated with rangeland habitats. In addition, rangelands are viewed as important contributors to watersheds: because rangelands usually have lower rates of soil erosion than cropland, they enhance water quality. Further, the natural system that exists on well-managed rangelands makes them increasingly recognized as places for nonconsumptive wildlife associated recreation.

Rangelands also produce intangible products (or nonuse values) that are the result of use. These products include natural beauty, open space, and the mere existence as a natural ecosystem (National Research Council 1994). Others emphasize biological diversity and the associated potential array of products and services as a distinct intangible product (West 1993). In contrast to use values, nonuse values occur almost entirely outside the market system. However, methods are evolving to quantify and assign monetary value to these existence values. As with use values, the costs and/or trade-offs associated with nonuse values can be compared to the estimated benefits (Bishop and Welsh 1992.)

Rangelands possess attributes that give them potential for biodiversity. Since they have not been "put to the plow," rangelands are attributed value as a natural system. Further, rangelands cover vast areas, often contiguously, and thereby possess the scale necessary for biodiversity of communities, ecosystems, and landscapes (West 1993). The biodiversity of rangelands contributes to the intangible products mentioned in the National Research Council (1994) report. Recognition of the importance of biodiversity arises for several reasons: (1) morality, (2) esthetics, (3) economics, and (4) "biological services."

Increasingly land managers are learning of the effects of the impacts of management or lack of management on the ability for various species to survive. Some assert that mankind has a moral obligation to protect fellow creatures. Social awareness has also made managers and others aware of the need to protect spaces, natural systems, and historic sites. In addition to the value of present consumptive and nonconsumptive uses, rangelands also possess esthetic values, and other economic potentials exist. Potentially these natural systems include yet-to-be-identified goods that could be of value to people. Finally, ecosystems are important components of natural cycles affecting the gaseous composition of the atmosphere; genesis, fertility, and stability of soils; disposal of wastes; cycling of nutrients; and natural control of pathogenic and parasitic organisms (West 1993).

A healthy range is recognized as one in which the integrity of the soil and ecological processes of the rangeland ecosystem are sustained (National Research Council 1994). Whenever management intervenes in the natural processes, for whatever reason, the impact of those interventions on the rangeland's ability to sustain commercial as well as intangible products must be considered. Rangeland grasshoppers also can disrupt the natural ecosystem in two ways. First, grasshopper infestations can reach plague proportions. Serious and widespread outbreaks can lead to soil erosion and reductions in water quality and make it difficult—if not impossible—for the range to recover to its original state. Major infestations of grasshoppers destroy cover for ground-nesting birds and mammals and damage the habitat for other wildlife. The desire to protect the range ecosystem and adjacent croplands was an important part of the rationale for initiating the publicly assisted rangeland grasshopper control programs that exist today.

Second, grasshoppers are recognized as an integral and necessary part of a range ecosystem. Grasshoppers and other rangeland insects are an important part of the food chain of some birds and mammals. Some species of grasshoppers are beneficial, feeding on plant forms that are not consumed by other users of the range. Because grasshoppers cut off vegetation as well as consume it, they create litter that becomes an important part of the nutrient cycle on rangelands. The strategy for managing rangeland grasshoppers has to be one of maintaining balance within range ecosystems.

The Grasshopper Integrated Pest Management (GHIPM) Project recognized the potential environmental costs associated with applying grasshopper management programs. One component addressed the safe use of grasshopper management programs around threatened and endangered plant species (Tepedino and Griswold 1993 unpubl.). Another chapter (III.6) in the environmental monitoring and evaluation section of the User Handbook evaluates the effects of grasshopper treatments on wildlife and aquatic species. The economics component of the Project developed procedures to make estimates of the environmental costs of control programs. This valuation recognizes, as the reader shall subsequently see, that fish and wildlife possess a value for recreation that considers both nonconsumptive (bird watching, photography, hiking) and consumptive (fishing, hunting) forms of wildlife-associated recreation.

Grasshopper program managers have been conscious of possible environmental side effects, undesired and beneficial, from these programs. Chemical applications may affect populations of some nontarget insect species as well as grasshoppers. Treatment program managers warn keepers of commercial insects so that those populations are protected. Managers of treatment programs take care to spray chemicals under conditions that minimize drift and to refrain from applying certain chemicals near water.

Evaluating Losses in Wildlife-Associated Recreation

Economists have made estimates of the value of some of the nontraditional outputs from rangelands (Bernardo et al. 1992, Kitts 1992, Loomis et al. 1989, Standiford and Howitt 1993, Young et al. 1987). Most of these studies have focused on consumptive and nonconsumptive forms of wildlife-associated recreation. However, a recent Colorado study estimated the value of open space. It found 80 percent of those spending summer vacations in the Steamboat Springs area indicated that ranch open space added significantly to their willingness to pay for summer visits. Willingness to pay for ranch open space averaged about $20 per day (Walsh et al. 1993).

Many of the biological–physical–management interactions associated with rangeland biodiversity are yet to be understood (West 1993). Consequently, very little has been done to evaluate the contributions of rangelands to biodiversity. Yet, under the Forest Management Act of 1976 and the Surface Mining Control and Reclamation Act of 1977, rangelands must be managed for biodiversity. Intangible values are reflected in policy directives even if quantification of those values has not occurred.

Reported here is an example of how rangeland environmental amenities can be evaluated. Chapter VI.3 of this Handbook discusses the method of estimating the economic loss to ranchers from an uncontrolled grasshopper outbreak. Applying chemical treatments reduces damages for the livestock grazer, and the damage reductions are the benefits of grasshopper controls. Pest managers also can estimate the economic loss if grasshopper control activities deplete wildlife populations. Figure VII.12–1 shows the flow of events.

If grasshopper management programs deplete wildlife populations, a reduction in the wildlife base will result in fewer people participating in wildlife-associated recreation. Because people place an economic value on recreation, less recreation means an economic loss. Investigators link the economic evaluation of wildlife depletion to grasshopper management and take the economic losses from wildlife-associated recreation as a measure of the portion of the environmental costs of the grasshopper treatment programs.

Calculations can start with the net economic values of wildlife-associated recreation estimated by Hay using willingness-to-pay techniques (1988a and b). Using the net economic value estimates for specific regions, it is possible to make estimates of the reduction in consumptive and nonconsumptive forms of wildlife-associated recreation resulting from a decrease in the wildlife resource base.

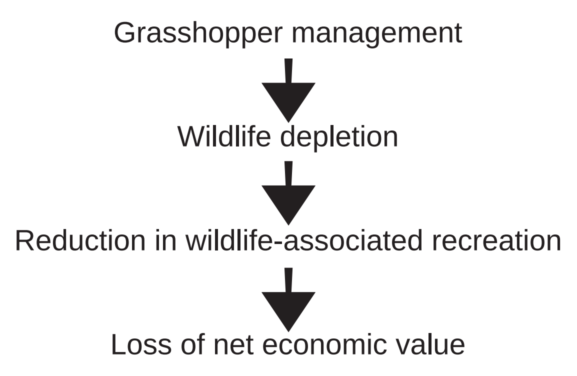

Figure VII.12–1—Sequence of events from grasshopper management to loss of value.

Analyzing the information reveals how participation in wildlife-associated recreation depends on demographic variables, price (cost of participating in recreation) and the wildlife resource base. Managers can use analyses for each type of wildlife-associated recreation (fishing, hunting, and nonconsumptive recreation) in the States for which control of rangeland grasshoppers is a problem.

The economic analysis involves the last two linkages of figure VII.12–1. Potential wildlife depletion results in a reduction in wildlife-associated recreation that, in turn, results in a net economic loss. This loss is a measure of a part of the potential environmental costs associated with grasshopper management programs.

Potential Environmental Costs

Table VII.12–1 shows Hay's net economic values for wildlife-associated recreation. These are the average net economic values for the eight States included in and surrounding the GHIPM demonstration sites. The net economic values are from surveys designed to determine how much participants value a day of recreation in these activities.

The next step to estimating the potential loss in wildlife-associated recreation resulting from grasshopper management programs is to look at the relationship between the wildlife resource base and the amount of participation in wildlife-associated recreation. The U.S. Fish and Wildlife Service conducts periodic surveys of fishing, hunting, and wildlife-associated recreation. The year for which the most recent survey data are available is 1985. Many factors determine the likelihood that an individual will participate in wildlife-associated recreation. For discussion in this chapter, we are primarily interested in one variable—the effects of the wildlife resource base on the probability of participation. If the wildlife resource base declines, we expect that the rate of participation in wildlife-associated recreation also will decline. Since grasshoppers and grasshopper treatments affect the habitat of wildlife, a measure of the wildlife resource base is habitat.

For hunting and nonconsumptive forms of wildlife-associated recreation, the amount of participation was sensitive to changes in the wildlife resource base. Fishing was not responsive to an estimate of changes in the fishing resource base. For hunting, a reduction of 1 percent in the range habitat of wildlife (for example a 1-percent reduction in the capacity of a range to support game wildlife) results in a 3.2-percent reduction in hunting participation. Similarly, a 1-percent reduction in the rangeland wildlife base results in a 2.9-percent reduction in participation in nonconsumptive forms of wildlife recreation.

Table VII.12–1—Net economic values per day of wildlife-associated recreation, by recreational activity in the eight-State region[1]

Activity	Net economic value (dollars/day)
Hunting	
Deer	$35
Elk	$36
Waterfowl	$20
Fishing	$11
Nonconsumptive	$22

[1] Idaho, Montana, Nevada, North Dakota, Oregon, South Dakota, Utah, Wyoming.
Source: Hay (1988 a and b).

The statistical equations give estimates of the number of participants in each wildlife-associated recreation activity. In this chapter, we focus on how wildlife-associated recreation changes in response to changes in the resource base. Table VII.12–2 shows the base level estimate of the number of hunters in the eight-State region, their expenditures, participation days, and the net economic value from hunting in the region.

The table also shows the potential impact of a 1-percent decline in the game wildlife resource base and the associated economic impact. We can interpret the analysis two ways. A 1-percent increase in the wildlife resource base would result in an increase of the same magnitude in participation, expenditures, hunting days, and net economic value, as would a 1-percent decrease. Thus, if the use of a grasshopper treatment program reduces the wildlife resource base, we can measure the cost (loss in net economic value). Conversely, if grasshoppers destroy the habitat for wildlife and a reduction in game wildlife occurs, we also can estimate the potential losses from less hunting on grasshopper-damaged rangeland.

Using the estimated equations for nonconsumptive forms of wildlife recreation, table VII.12–3 shows the base economic activity and potential losses if a grasshopper invasion reduces the wildlife resource base. As with hunting, nonconsumptive wildlife-associated recreation also may suffer if an uncontrolled grasshopper outbreak reduces the wildlife resource base.

Potential Recreation Losses

The economic losses associated with changes in the wildlife resource base are only potential losses. The environmental monitoring component of the GHIPM Project has not found adverse effects on wildlife resulting from use of grasshopper control programs. Approved treatment options are the result of careful evaluation and selection to determine materials and methods which minimize the threat to the environment. When there are grasshopper treatments, these precautions to minimize the environmental damage apparently are successful. So long as the first linkage in figure VII.12–1 remains zero, meaning grasshopper treatments do not result in wildlife depletion, the economic losses from reductions in wildlife-associated recreation are also zero. However, should damages to the wildlife resource base occur, the changes in net economic value due to wildlife-associated recreation can be estimated by applying this procedure.

Conclusions

With increased understanding of the linkages and relationships present in rangeland ecosystems, it will be possible to quantify more of the identified benefits from rangeland biodiversity and other intangible values. Until that time, rangeland management and actions taken to control rangeland pests must proceed with the best available understanding of the results from those management interventions.

Table VII.12–2—Hunting: Effect of reduced wildlife resources on the number of participants and trip-related expenditures and on participation-days and net economic value

Wildlife resource level	Number of participants	Trip-related expenditures	Participation-days	Net economic value
	Thousands	*$ million*	*Thousands*	*$ million*
Base level	790,000	$191.2	11,847	$355.4
1% decline	–25	–6.1	–371	–11.1

Table VII.12–3—Nonconsumptive: Effect of reduced wildlife resources on number of participants and trip-related expenditures and on participation-days and net economic value

Wildlife resource level	Number of participants	Trip-related expenditures	Participation-days	Net economic value
	Thousands	*$ million*	*Thousands*	*$ million*
Base level	1,501	$253.7	15,009	$330.2
1% decline	–43	–7.3	–429	–9.4

References Cited

Bernardo, D. J.; Engle, D. M.; Lochmiller, R. L.; McCollum, F. T. 1992. Optimal vegetation management under multiple use objectives in Goss timbers. Journal of Range Management 45: 452–469.

Bishop, Richard C.; Welsh, Michael P. 1992. Existence values in benefit–cost analysis. Land Economics 68: 405–417.

Hay, M. J. 1988a. Analysis of the 1985 National Survey of Fishing, Hunting, and Wildlife-Associated Recreation—net economic recreation values for deer, elk and waterfowl hunting and bass fishing. Rep. 85-1. Washington, DC: U.S. Department of the Interior, U.S. Fish and Wildlife Service. 23 p.

Hay, M. J. 1988b. Analysis of the 1985 National Survey of Fishing, Hunting, and Wildlife-Associated Recreation—net economic values of nonconsumptive wildlife-related recreation. Rep. 85-2. Washington, DC: U.S. Department of the Interior, U.S. Fish and Wildlife Service. 16 p.

Kitts, Andrew W. 1992. Economic value of some external costs for grasshopper control. M.S. thesis. Ft. Collins, CO: Colorado State University, Department of Agricultural and Resource Economics. 91 p.

Loomis, John; Donnelly, Dennis; Sorg-Swanson, Cindy. 1989. Comparing the economic value of forage on public lands for wildlife and livestock. Journal of Range Management 42: 134–138.

National Research Council. 1994. Rangeland health: new methods to classify, inventory and monitor rangelands. Washington, DC: Committee on Rangeland Classification, Board of Agriculture.

Standiford, Richard B.; Howitt, Richard E. 1993. Multiple use management of California’s hardwood rangelands. Journal of Range Management 46: 176–182.

Walsh, R. G.; McKean, J. R.; Mucklow, C. J. 1993. Recreation value of ranch open space. Report to the Routt County (CO) Board of Commissioners. Ft. Collins, CO: Department of Agricultural and Resource Economics, Colorado State University. 43 p.

West, Neil E. 1993. Biodiversity of Rangelands. Journal of Range Management 46: 2–13.

Young, John S.; Donnelly, Dennis M.; Sorg, Cindy T.; Loomis, John B.; Nelson, Louis J. 1987. Net economic value of upland game hunting in Idaho. Resour. Bull. RM-15. Ft. Collins, CO: U.S. Department of Agriculture, Forest Service, Rocky Mountain Forest and Range Experiment Station. 23 p.

References Cited–Unpublished

Tepedino, Vincent J.; Griswold, Terry L. 1993. Pollination biology of threatened and endangered plants. In: Cooperative Grasshopper Integrated Pest Management Project, 1993 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 181–189.

VII.13 Grasshopper Communities and Methodology

Anthony Joern

Grasshopper populations do not exist in an ecological vacuum. Instead, individual species populations interact with several other species, other individuals, other herbivores, a range of potential host plants and many natural enemies. In western North America, 30 to 50 grasshopper species may coexist, and each may respond individually to environmental change. Although science's interest lies mainly in the ecology and population dynamics of a single or a few species, one species cannot exempt itself from a network of interactions among all species that are present. Consequently, the grasshopper community becomes a central focus in any rational integrated pest management (IPM) project.

Communities are significantly more complex to evaluate and study than single-species populations. Manipulating one small component of the community network (e.g., of one or a few species) may not evoke the desired, long-term control objectives. Consideration of only one or a few species may lead to unnecessarily short-term solutions or even to unexpected problems. Besides problems associated with community complexity, species assemblages vary greatly from year to year at the same site and vary even more dramatically among sites. Scientists require descriptive and analytical methodologies to clearly devise and assess community management practices. Scientists also must simplify the scope of the problem without sacrificing important connections that prescribe creative solutions.

In this section, I summarize simple, standard approaches and methodologies for describing communities and for assessing the importance of key interactions. Some of these methods are best for sporadic evaluation of random sites on a hit-or-miss basis. Others are designed for developing long-term understanding at sites that are regularly monitored for potential grasshopper problems. Government agencies and private organizations that manage the same large tract over many years can expect to develop comprehensive, community-based IPM programs. But individual ranchers with only intermittent grasshopper problems and few resources cannot. As a result, managers must select which of the following approaches to community evaluation meets their situation. Complete annual censuses and evaluations of environmental conditions are the cornerstones of community studies. These require significant effort, and that cost–benefit ratios ultimately determine the value of studying community relationships.

As I list accepted methods to evaluate grasshopper communities, I will stress the difference between merely describing community composition (species identities) and understanding mechanisms driving species interactions and coexistence. IPM measures interrupt dynamic, often subtle, ecological interactions within and among species. Until we work out the impact of these key interactions for many species combinations in detail, species lists alone provide little insight into future system dynamics surrounding IPM efforts.

Community Descriptions: List of Grasshopper Species Present

A list of grasshopper species is the simplest description of a community and is required in any community-level assessment. A good description includes the relative abundance and absolute density of individual species in a community. Density is important because the number of individuals that are available to interact determines, at least in part, what really happens.

Based on past studies, experts can sometimes develop insights regarding community dynamics from such lists—if certain conditions and species are present. Shifts in species composition among years or among sites suggest that different grasshopper species react differently to changing environments. Such variation in the response to different environmental conditions indicates that either the community shifts from one state to another or that the internal dynamic interactions among species shift. Consequently, the same IPM management practice employed under different conditions may produce different long-term responses depending on the state of the community.

Sampling efficiency can vary with habitat type and its three-dimensional structure as well as overall grasshopper densities. Typical methods include sweeping some predetermined number of times or counting grasshoppers at stationary sample sites (e.g., the "ring technique" of Onsager and Henry 1977, Thompson 1987). Berry et al. review appropriate sampling methods and their justification in chapter VI.10 of this handbook. Remember, in obtaining lists of species' relative abundances, the accurate sampling of rare species is the biggest problem. More samples will reduce the chance of missing rare species. To estimate a sampling intensity that will detect most of these species at your site, plot the cumulative number of grasshopper species collected against some measure of sample intensity (number of individuals collected, number of sweeps, number of rings examined, number of transects, area sampled, and number of habitat types sampled). Figure VII.13–1 illustrates a reasonable sampling schedule. In designing sampling plans, be aware that you will probably encounter some unrecorded species if new habitat types are included. Because of this, plan to sample all habitat types found in the area in the proportion that they occur in the environment.

What rules-of-thumb emerge from species lists? Many species thrive only in areas with open bare areas (e.g., *Ageneotettix deorum).* Other species (e.g., *Paropomala wyomingensis)* require significant vertical structure such as that provided by bunchgrasses. Still other species (e.g., *Melanoplus sanguinipes)* occupy a variety of microhabitats, so that little insight can be gained just by knowing what microhabitats exist at a site. Similarly, even among grasshopper species that eat many plants, the range of readily consumed plant species will be similar among sites. Based on use of both food plants (Joern 1979a, 1983) and microhabitat resources (Joern 1982), community level patterns emerge that may help a manager make decisions (Joern 1979a,b, 1986a). The usefulness of such an approach for developing sound grasshopper IPM tactics is idiosyncratic and case-specific at this time.

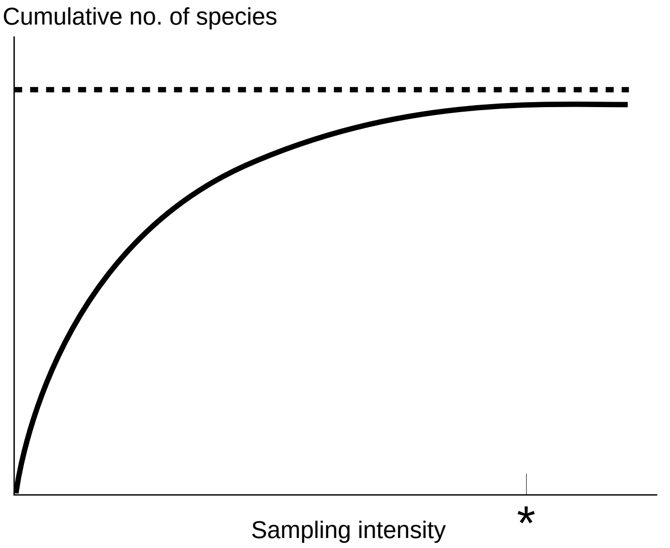

Figure VII.13–1—The number of species sampled is dependent on the sampling intensity. To obtain a good estimate of the number of species at a site, sampling intensity should equal that indicated with an asterisk, near the asymptote for the entire assemblage. If sampling intensity is less than this point, many rare species will likely be missed.

Using Statistics To Estimate Species Replacements and Community Associations

Species replacements and community associations along environmental gradients can be identified using standard multivariate statistical techniques (e.g., discriminant function analysis, principle components analysis, detrended correspondence analysis) or some combination of the statistical techniques developed for ordinating communities (Gauch 1982). As a technique, ordination simplifies multiple species associations by representing the relationships in fewer dimensions using mutivariate descriptive statistics. By using these techniques, you can identify the combinations of species that tend to occur together (and their relative abundances) in association with key attributes of the environment such as vegetation type or soil moisture (fig. VII.13–2). Such community analyses allow you to simplify the community associations along a spatially varying environmental gradient. Be aware of the correlational nature of these results from these analyses. The patterns that you uncover will fully depend on what you include in your initial sampling design. If you add species or sites with different combinations, the ultimate patterns may shift. Ordination provides a refined fit between grasshopper community composition and some environmental gradient, but *you cannot identify dynamic and causal relationships between the two features by using this approach.*

Plotting Against an Environmental Gradient.—You can readily visualize species replacements along gradients by plotting the change in the abundance (or relative abundance) of each species along some environmental gradient (fig VII.13-2a). In this hypothetical analysis, I assess a series of independent sample sites as in number 1 above (a list of grasshopper species). Then, on a species-by-species basis, I plot the abundances (or relative abundances) along the gradient. By comparing these plots among species, you can identify possible environmental conditions at your site best suited and worst suited for each species. In addition, you can compare responses of multiple species along the same gradient.

Multivariate Ordination Techniques.—Species associations can be identified using standard, multivariate ordination techniques (fig. VII.13-2b). While these techniques typically require commercially prepared computer software, the analyses are readily accessible, even on laptop computers. Standard references exist to help the user understand both the statistical guts of the analysis as well as providing insights to interpreting results (Cornell Ecology Programs discussed in Gauch 1982). The computer algorithms help put boundaries around species combinations from each location, largely based on changes in relative abundances rather than in response to massive replacement of individual species. Remember, these boundaries of species composition represent “probability boundaries” and much overlap typically exists in grasshopper species composition among adjoining communities or even when comparing sites some distance away. As a warning: many users of this technology tend to become typological in describing communities and often confuse pattern with a dynamic process. For example, I foresee some managers ordinating grasshoppers from a group of sites and then prescribing specific management options for those assemblages in group A versus group B or C and so on. The assumption that all

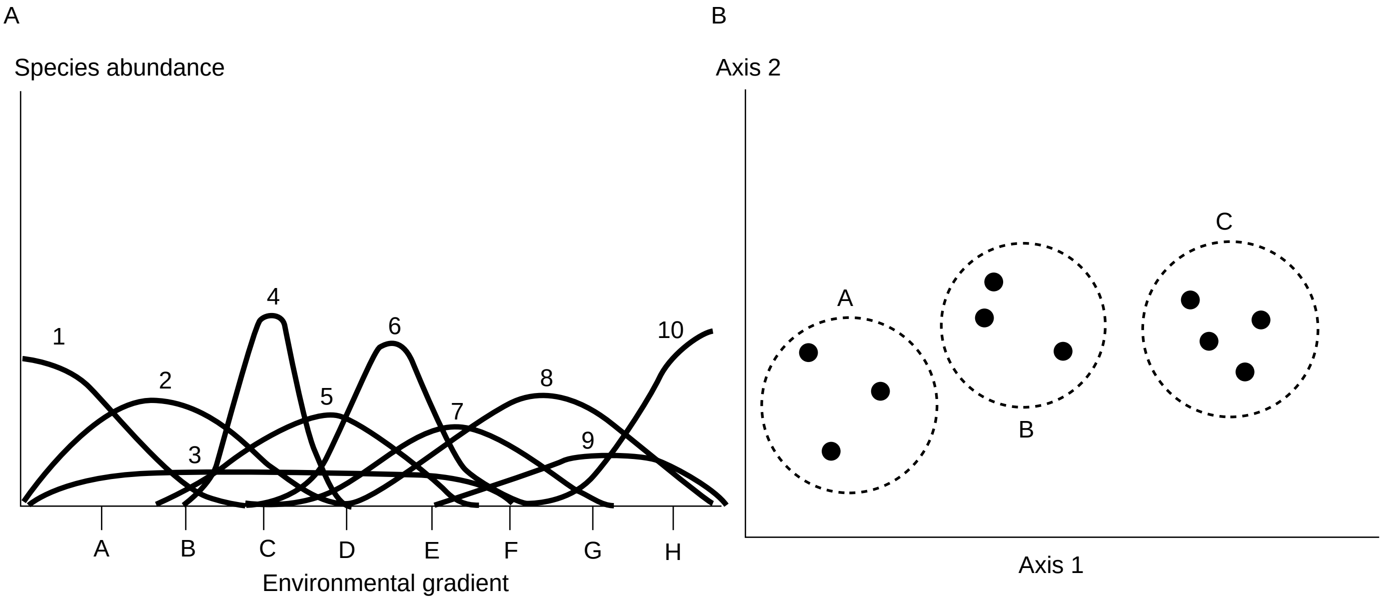

Figure VII.13–2—*A:* Hypothetical distribution of species along some environmental gradient based on sampling at 8 sites (A–H) along a transect. Each curve indicates the distribution along this gradient for a hypothetical grasshopper or plant species. For example, species 4 does best at site C but does not exist at site E while species 3 does not do particularly well at any site but is found along the entire gradient. *B:* This multivariate distribution can be “boiled down” into a simpler relationship using ordination techniques following those outlined in Gauch (1982). Each of these new axes (1 and 2) represent a composite of multivariate data. The points indicated in B represent the average position for each species indicated in A for the two multivariate resource axes developed from a composite of environmental variables. The groupings of species indicated by the dashed lines suggest species that react to environmental conditions in the same fashion. Examples of gradient analyses of grasshopper species along a topographic gradient in Montana are presented in Kemp et al. (1990) and Kemp and O’Neill (1990).

sites exhibiting type A species associations also categorically exhibit the same underlying dynamics is unfounded.

Unless a conceptual framework exists that predicts unique, species-specific relationships, the results will not explain why specific patterns emerge. For example, grasshopper species assemblages often change predictably as the species composition of the plant community changes (see chapter IV.3). What dynamic relationship exists between the two components of this analysis to explain the results? Unfortunately, insufficient information exists to tease apart such relationships, even if the pattern is very strong. Sometimes specific theories exist that predict particular species responses in abundance or in association with specific habitats. In these situations, additional insights regarding dynamic, causal mechanisms might emerge from pattern analysis, but this notion still requires experimental testing to uncover the underlying reasons for the relationships fully. Scientists must base management options on *processes* driving community dynamics, not on easily measured patterns. This fact is unfortunate because scientists can more readily establish measures of pattern than uncover the underlying dynamic mechanisms.

Using Controlled Manipulations To Uncover Site-Specific Dynamics

Experimental manipulation of species interactions can provide powerful community level insights into the dynamic forces that organize communities. However, the effort is great. From an IPM framework, subtle shifts in species composition that changes in the underlying interaction dynamics may provide the key for developing the correct management strategy. After all, those IPM practices that work in concert with naturally occurring dynamic processes will most likely lead to long-term success. However, uncovering the specific nature and strength of interactions among species, including their impact on resulting population densities and community structure, will require experimental manipulations under field conditions. Standard experiments that might uncover these relationships are time consuming and complex. Consequently, an efficient experimental approach requires a strong conceptual framework so that science can simultaneously evaluate key competing possibilities and that investigators can reject alternatives based on experimental results. The conceptual framework identifies alternate hypotheses. By simultaneously testing competing explanations of community pattern and process through experimentation, the manager can rapidly narrow the options. Then it becomes possible to uncover the best explanations upon which to base management options. Despite the difficulties and cost, I strongly believe that the intense effort required to uncover site-specific dynamics using controlled manipulations will pay off, in the long term, for grasshopper IPM managers. Examples of sites that should profit from intensive studies include public lands and large private holdings with constant or predictable land-use practices and a history of grasshopper problems. If managers feel insecure about performing all of the above work by themselves, they should allocate some management funds to contract for research by competent scientists.

A current example illustrates the above process. A conceptual framework that defines alternate views of the problem, combined with experimental manipulation and coupled with appropriate comparisons and descriptive analyses, allows recognition and interpretation of the dynamic interactions that regulate community-level processes. As a general framework, the alternatives include “top-down” versus “bottom-up” processes (Hunter et al. 1992). As herbivores, grasshoppers occupy an intermediate trophic (nutrition) position in the food web, with food plants below them and natural enemies (e.g., parasitoids, invertebrate and vertebrate predators, or fungal, bacterial, or viral pathogens) positioned above them.

What major forces limit grasshopper populations in this food web? From a control standpoint, this information provides the clue to appropriate management planning. Bottom-up forces can arise from insufficient nutrients either when grasshoppers compete for limited food or when time constraints interfere with feeding and digestive capability. Top-down forces can arise from the actions of natural enemies. Other chapters of the Grasshopper Integrated Pest Management User Handbook provide detailed examples of each type of interaction.

Descriptive studies cannot untangle this set of potential interactions, but manipulative experiments can. In fact, under natural conditions, bottom-up (Belovsky and Slade 1995) and top-down (Joern 1986b, 1992) forces operate simultaneously, and either one can drive the interactions and can thus determine the final densities of coexisting grasshoppers (Belovsky and Joern 1995). More importantly, reciprocal indirect effects of species on each other can potentially be more important than the direct interactions. Scientists can see such responses only through experimentation.

The Role of Experimentation in Developing "True" IPM for Grasshoppers

True IPM will require successful description of the above relationships in its development, and perhaps will lead to the development of "ecotechnology" based on a firm conceptual foundation. For example, here are the types of questions that we must address experimentally: How do grasshoppers compete for scarce food resources? Which species are the best competitors for the available food supply? What impacts do such interactions exert on the resulting grasshopper community structure? Will the food resource base change as environmental conditions change and with what consequences? Are competitive interactions altered in response to changing food supplies? How important are natural enemies in deciding which grasshopper species survive and in what relative abundance? How do competition and predation interact to affect grasshopper communities? How do abiotic (weather) and biotic (species-interaction) features of the environment interact to affect grasshopper communities, if they exert any influence at all? Results from experiments to answer these and related questions will allow land managers to define explicitly the key interactions that describe the community relationships a particular grasshopper infestation. Managers can then identify links that will provide the desired IPM results, or those that are susceptible to disruption and will lead to unwanted and unintended results.

Final Comments

Grasshopper IPM must focus on entire grasshopper assemblages, even if only a small proportion of the species are economic targets. Interactions among species may lead to unexpected consequences from control efforts if we ignore rare but otherwise functionally important taxa. Both species lists and more complicated statistical descriptive techniques of grasshopper communities will provide some guidelines, but neither will provide direct insights about dynamic relationships. Because effective control will result in permanent or at least long-lasting alteration of species interactions, scientists would like to understand the dynamics of these interactions. Frankly, much work remains before this approach bears fruit. However, the rich conceptual framework that underlies community dynamics suggests that many important insights will emerge and hopefully will revitalize the basis of control and management planning.

References Cited

Belovsky, G. E.; Joern, A. 1995. The dominance of different regulating factors for rangeland grasshoppers. In: Cappuccino, N.; Price, P. W., eds. Population dynamics: new approaches and syntheses. San Diego, CA: Academic Press: 359–386.

Belovsky, G. E.; Slade, J. B. 1995. Dynamics of two Montana grassland populations: relationships among weather, food abundance and intraspecific competition. Oecologia 101(3): 383–396.

Gauch, H. G., Jr. 1982. Multivariate analysis in community ecology. Cambridge, UK: Cambridge University Press. 298 p.

Hunter, M. D.; Ohgushi, T.; Price, P. W., eds. 1992. Effects of resource distribution on plant-animal interactions. New York: Academic Press. 505 p.

Joern, A. 1979a. Feeding patterns in grasshoppers (Orthoptera: Acrididae): factors influencing diet specialization. Oecologia 38: 325–347.

Joern, A. 1979b. Resource utilization and community structure in assemblages of arid grassland grasshoppers (Orthoptera: Acrididae). Transactions American Entomological Society 150: 253–300.

Joern, A. 1982. Vegetation structure and microhabitat selection in grasshoppers (Orthoptera: Acrididae). Southwestern Naturalist 27: 197–209.

Joern, A. 1983. Host plant utilization by grasshoppers (Orthoptera: Acrididae) from a sandhills prairie. Journal of Range Management 36: 793–797.

Joern, A. 1986a. Resource utilization by a grasshopper assemblage from grassland communities. Triennial Proceedings Pan American Acridological Society 4: 75–100.

Joern, A. 1986b. Experimental study of avian predation on coexisting grasshopper populations (Orthoptera: Acrididae) in a sandhills grassland. Oikos 46: 243–249.

Joern, A. 1992. Variable impact of avian predation on grasshopper assemblies in sandhills grassland. Oikos 64(3): 458-463.

Kemp, W. P.; Harvey, S. J.; O’Neill, K. M. 1990. Patterns of vegetation and grasshopper community composition. Oecologia 83: 299–308.

Kemp, W. P.; O’Neill, K. M. 1990. Habitat and insect biology revisited. American Entomologist 36: 44–49.

Onsager, J. A.; Henry, J. E. 1977. Comparison of five methods for estimating density of rangeland grasshoppers. Journal of Economic Entomology 70: 187–190.

Thompson, D. C. 1987. Sampling rangeland grasshoppers. In: Capinera, J. L., ed. Integrated pest management on rangeland: a shortgrass prairie perspective. Boulder, CO: Westview Press: 219–233.

VII.14 Grasshopper Population Regulation

G. E. Belovsky

Factors controlling the dynamics of a population are often referred to as either limiting or regulating a population (Sinclair 1989). Limiting factors operate to depress a population without regard to its number; limiting factors are density independent. Regulating factors are special depressing factors that tend to bring the population to a specific number; to reach the specific number, the depressing effect must be great when the population is much larger than the specific number and less when the population is below or near the specific number. Regulating factors are density dependent.

Population ecologists have demonstrated that, although there may be a correlation between weather and population numbers, this correlation does not mean that weather is the causal factor determining population dynamics or even the most important factor—even if it is a limiting factor (Horn 1968). In fact, it is well established that the density-independent effects of weather on survival and reproduction cannot regulate populations. The effects can only interact with regulating mechanisms to set population numbers because regulation requires the negative feedback of density dependent processes.

Science's understanding of grasshopper population dynamics has been largely built on long-standing observations that grasshopper numbers in a given year are correlated with temperature and precipitation (Joern and Gaines 1990). While these correlations provide convenient forecasting tools for pest managers, the correlations do not imply that weather is the causal mechanism limiting or regulating populations, nor that scientists understand grasshopper population dynamics. Furthermore, correlations between grasshopper numbers and weather, while statistically significant, are weak and are not consistent between different western rangelands with grasshopper numbers sometimes being greater in hot–dry years and sometimes greater in cool–wet years (see chapter IV.8).

Variability in the response to weather suggests that grasshopper populations may respond to other factors that are correlated with weather and not to the weather directly (for example, the abundance and nutritional value of food, the cover providing protection from predators, diseases, etc.). Consequently, the value of weather as a forecasting tool for particular western regions and the concept of weather as the driving factor in grasshopper population dynamics should not be confused.

A number of general models have been developed to portray insect population dynamics (Southwood and Comins 1976, Berryman 1987). These models are generic and are not based upon specific mechanisms that operate upon the insect's population but attempt to depict the insect's population dynamics in terms of the shape of a Ricker curve. A Ricker curve (fig. VII.14–1) is a plot of a species' number (N) at time t (N_t) against its number at a later time, t+1 (N_{t+1}). This type of population analysis is appropriate for insects that have a single generation each year, which includes nearly all western rangeland grasshoppers (Varley et al. 1973). Ricker curves are depictions of population dynamics because their intersection with a reference line ($N_t = N_{t+1}$) defines the number to which the population is being drawn by regulating factors (fig. VII.14–1).

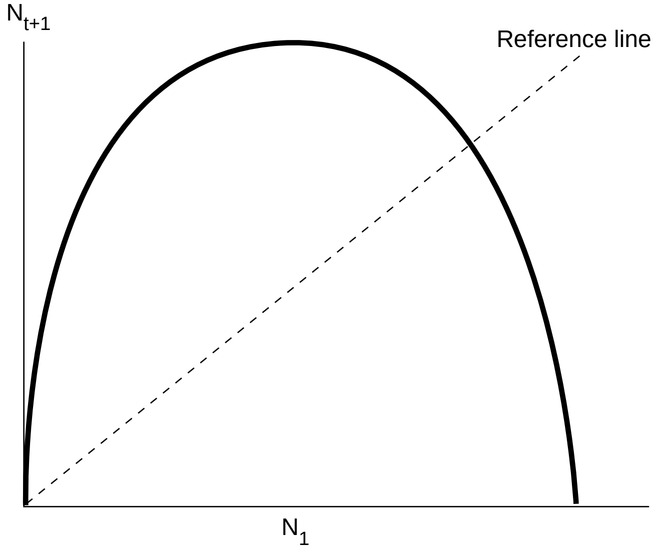

Figure VII.14–1—A simple Ricker curve relating the number of individuals starting the population in generation t (N_t) to the number of individuals produced by them to start the next generation (N_{t+1}). The point where the reference line ($N_t = N_{t+1}$) intersects the Ricker curve is an equilibrium point that the population may approach.

Three Relationships Important in Grasshopper Population Dynamics

The shape of the Ricker curve depends upon the ecological mechanisms that operate on the population and how they change in intensity with density. Three mechanisms may be particularly important for grasshoppers: (1) the relationship between density and the probability of surviving to the adult stage in the absence of natural enemies, (2) the relationship between density and the probability that an individual is killed by a natural enemy, and (3) the relationship between the current year's density and the number of hatchlings produced for the next year by each current female. In each case, density refers to the number of hatchlings per area that initiates the year's population. I will review each of these functions.

Density and Survival.—In the absence of natural enemies, the relationship between initial grasshopper hatchling density and survival determines the density of adult females that can produce hatchlings. First, at low densities, survival should be a constant proportion of the population set by weather and the nutritional value of foods because the individuals consume as much food as they can potentially process. This survival is **density independent** because it does not vary with the density of grasshoppers present. Second, at higher densities, survival becomes **density dependent,** as competition reduces the food available per individual, and the mortality rate increases.

This survival relationship leads to a pattern where the density of adults increases as hatchling density increases and then becomes a constant set by the maximum adult density that the available food can support. This relationship can be seen at a Palouse prairie site in western Montana for *Melanoplus sanguinipes* where the addition of food increases survival to the adult stage (fig. VII.14–2A) (Belovsky and Slade 1995). Weather can increase or decrease food: cool–moist conditions tend to increase plant production, but tend to decrease the nutritional quality of the plants.

Density and Predation.—The relationship between the initial density of hatchling grasshoppers and an individual's probability of being killed by natural enemies depends upon the rate at which an individual enemy can kill grasshoppers (functional response) and the number of enemies present (numerical response). The functional and the numerical responses for a natural enemy frequently increase to constant values as the density of prey increases; this phenomenon is observed in predator–prey systems ranging from insects and spiders to wolves and deer.

The implication is that as density of the grasshoppers increases, the proportion killed (probability of an individual being killed) will first increase with density and then decrease. An example can be seen at a Palouse prairie site in western Montana for the grasshopper *M. sanguinipes* where vertebrate predators, especially birds, are the principal natural enemies (fig. VII.14–2B) (Belovsky and Slade 1993). Weather can modify the effects of these natural enemies. For example, cool–moist conditions can increase plant production, and increased plant biomass enables grasshoppers to conceal themselves from predators. But cool–wet conditions do not always enhance grasshopper survival: they can increase the virulence of some diseases.

Density and Reproduction.—The relationship between the current year's density of hatchlings and the hatchlings produced for the next year's generation by each current female reflects two conditions. First, at low densities, hatchling production per female should be constant because each female has all of the food that she can utilize for egg production. This level of reproduction is **density independent** because it does not vary with the density of hatchlings present. Second, at higher densities, hatchling production per female should decline as the density of current hatchlings increases because each female acquires less and less of the available food. This level of reproduction is **density dependent** because it declines with the current density of hatchlings present. This decline emerges as females acquire less and less food because the increasing number of grasshoppers depletes the available food. The above pattern in reproduction can be seen at a Palouse prairie site in western Montana for *M. sanguinipes* where the addition of food increases reproduction (fig. VII.14–2C) (Belovsky and Slade 1995). Weather can increase or decrease food availability. For example, cool–moist conditions tend to increase plant production but tend to decrease the nutritional quality of the plants.

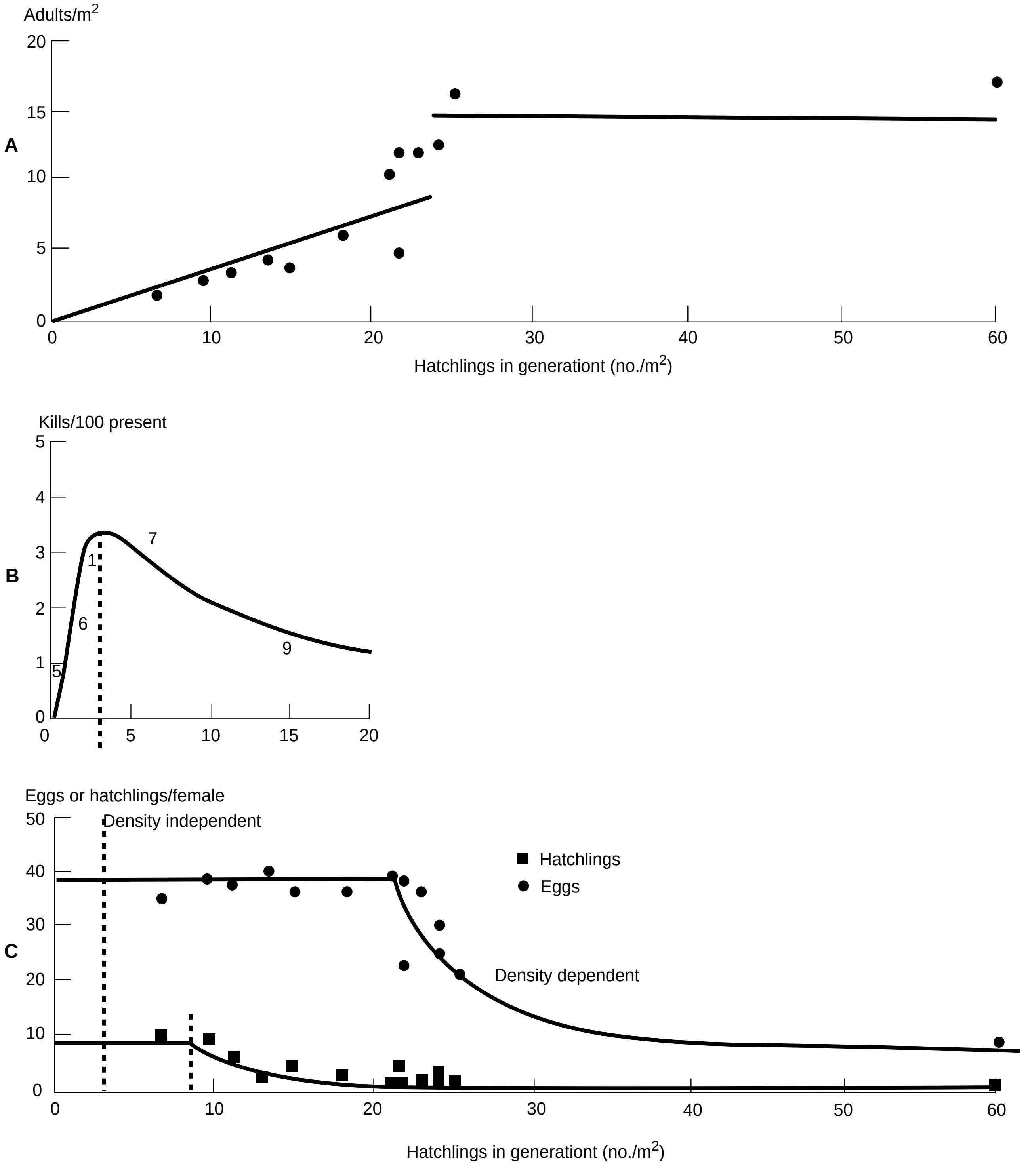

Figure VII.14–2—The relationships between hatchling density of *Melanoplus sanguinipes* and (A) adult density, (B) the probability of an individual being killed by a predator, and (C) the production of eggs and hatchlings per adult female, as observed at a Palouse prairie site in western Montana. The vertical dashed lines relate the points where the probability of predation and reproduction per adult female begin to decline with hatchling density. *(A* and *C* are adapted from Belovsky and Slade [1995]. *B* is adapted from Belovsky and Slade [1993].)

Using the Ricker Curve

The above three relationships can be combined to construct a Ricker curve, which enables scientists to integrate the effects of weather-induced density-independent mortality, natural enemy-caused mortality, and food resources. This integration produces three possible Ricker curve shapes, each reflecting a different dominant form of population regulation.

Population Regulated Only by Natural Enemies.— This type of regulation occurs when the peak of the function relating the probability of being killed by a natural enemy occurs at a grasshopper density that is greater than the density at which hatchling production begins to decline and/or adult densities attain their maximum level. In this case, a Ricker curve emerges with a single peak or two peaks, where the reference line intersects the Ricker curve only on the first peak (fig. VII.14–3A). This case emerges if the actions of the natural enemies (a) are so strong that grasshopper density cannot attain a level at which competition for food occurs or (b) continue to increase as competition for food increases.

Population Regulated Only by Food Availability.— This type of regulation occurs when the peak of the function relating the probability of being killed by a natural enemy occurs at a grasshopper density that is much less than the density at which hatchling production begins to decline and/or adult densities attain their maximum level. The Ricker curve emerges with two peaks, where the reference line intersects the Ricker curve only on the second peak (fig. VII.14–3B). In this case, the population is capable of "escaping" the effects of natural enemies, because (a) the natural enemies are not very effective and/or (b) the impact of the natural enemies rapidly diminishes as grasshopper density increases.

Population Regulated by Either Natural Enemies or Food Availability Depending Upon the Density of Hatchlings Initiating the Population.—This type of regulation occurs when the peak of the function relating the probability of being killed by a natural enemy occurs at a grasshopper density that is less, but not much less, than the density at which hatchling production begins to decline and/or adult densities attain their maximum level. In this case, a Ricker curve emerges with two peaks,

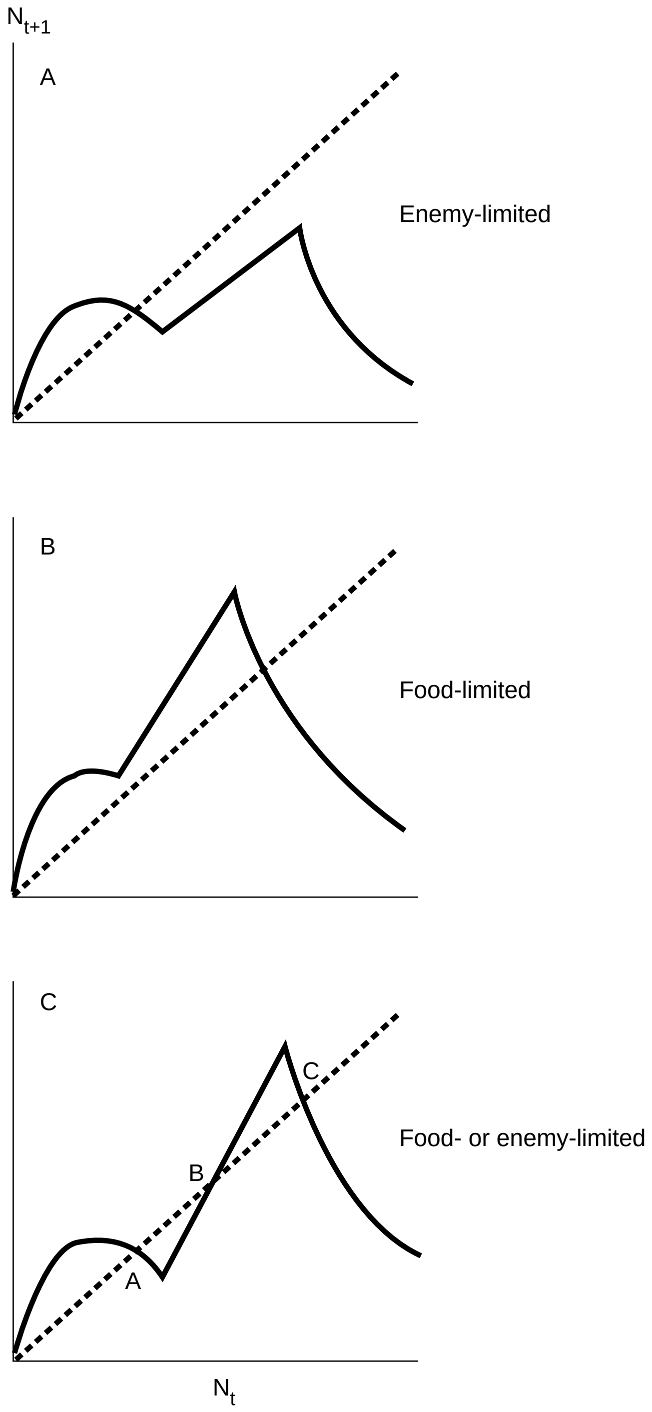

Figure VII.14–3—The three Ricker curve shapes that emerge (see text).

where the reference line intersects the Ricker curve at three points (fig. VII.14–3C).

The intersection with the first peak represents a population state regulated by natural enemies. The intersection with the second peak represents a population state regulated by food availability. The intersection lying between the above two intersections defines the “watershed,” where populations initiated with densities less than this point become limited by natural enemies and with densities greater than this point become limited by food availability. In this case, the population can “jump” from one mode of regulation to the other depending upon the densities of hatchlings initiating a population from year to year.

The picture of grasshopper population regulation described above can be validated experimentally. From experimental (enclosed) populations established at different densities of *M. sanguinipes* at the Palouse prairie site in western Montana, the Ricker curve has been measured (fig. VII.14–4). The curve has two peaks and is intersected by the reference line at three points, indicating a population that can be regulated by either natural enemies or food availability depending on initial hatchling densities.

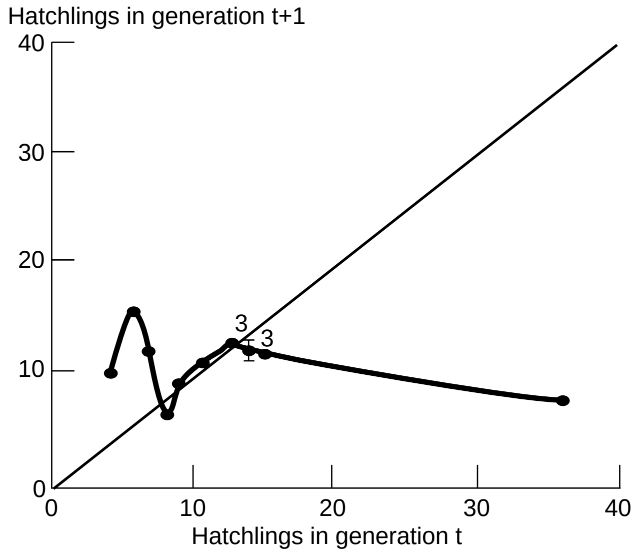

Figure VII.14–4—The Ricker curve for a *M. sanguinipes* population during a single year at a Palouse prairie site in western Montana. Error bars and sample sizes are presented for populations initiated at the same hatchling density.

More than 12 years of observation of this population disclosed that it has consistently been regulated by food availability, not by natural enemies (Belovsky and Slade 1993, 1995). This fact suggests that the population is near the intersection with the second peak of the Ricker curve. Furthermore, this conclusion was expected given the three underlying functions measured at this site and presented in figure VII.14–2.

What Weather Can Do

A new perspective toward weather and grasshopper population regulation can be gained from the Ricker curve model by appreciating that weather can affect both density-independent mortality and food availability.

Weather-induced density-independent mortality can operate in conjunction with natural enemy mortality to prevent populations from attaining levels where food availability becomes regulating. For the density-independent mortality to be important, it would have to accomplish at least one of three things. First, inclement spring weather can kill a high proportion of hatchlings, most likely through cold-induced starvation. Second, weather might be sufficiently severe over the entire life cycle of the grasshoppers so that few individuals can survive to become adults. Third, weather might shorten the period of time that adults have to live so that the number of hatchlings produced is dramatically diminished.

On the other hand, weather exerts a far more pervasive influence by altering food availability from year to year (see chapters IV.4 and IV.5). This variation in food abundance can be as great as sixfold between years and more than twofold within a summer (Belovsky and Slade 1995). The variation in food abundance could easily shift the shape of the Ricker curve from producing a population regulated by natural enemies in years with low food abundance to a population regulated by food abundance in years with high food abundance, and vice versa.

Weather Interacts With Enemies and Food Availability

The weather-induced shifts in food abundance, and perhaps to a lesser extent, changes in density-independent mortality result in domains of attraction (shaded regions in fig. VII.14–5), where the grasshopper population fluctuates with weather, but is regulated by either natural enemies or food availability at any one time. This is the point made by Horn (1968) that weather can create population fluctuations by varying density-independent or density-dependent (such as food availability) factors, but the density-dependent factor(s) must still regulate the population (attract it to particular levels).

In some environments, the points of attraction may be set by population levels created by natural enemies in different years (fig. VII.14–5A). In other environments, the points of attraction may be set by population levels created by food availability in different years (fig. VII.14–5B). In still other environments, the points of attraction may vary between levels set by natural enemies in some years and food availability in other years (fig. VII.14–5C).

Unique spatial relationships for population regulation emerge when several populations are placed in juxtaposition. The above discussion considers that each population is isolated from other populations. The conclusions concerning the regulation of a single population may have to be modified when adjacent populations are considered. For example, consider two adjacent or near populations. One population is regulated by natural enemies (fig. VII.14–3A) and the other population, by food availability (fig. VII.14–3B). It is possible that the food-regulated population will produce individuals that migrate rather than die. Therefore, if the two populations are close enough in relation to the dispersal ability of the grasshopper, the population that would otherwise be regulated by natural enemies may be able to increase in density with the addition of immigrants and, thereby, become food regulated. The immigrants permit the population to escape the effects of natural enemies.

The above simple scenario says that in some situations pest managers need to understand not only how individual populations are regulated but also the juxtaposition (landscape) of populations to determine the potential for population regulation to be complicated by dispersal. For example, the population receiving dispersers and thereby escaping regulation by natural enemies might be causing economic damage, and pest managers might decide to control it. However, control of this population

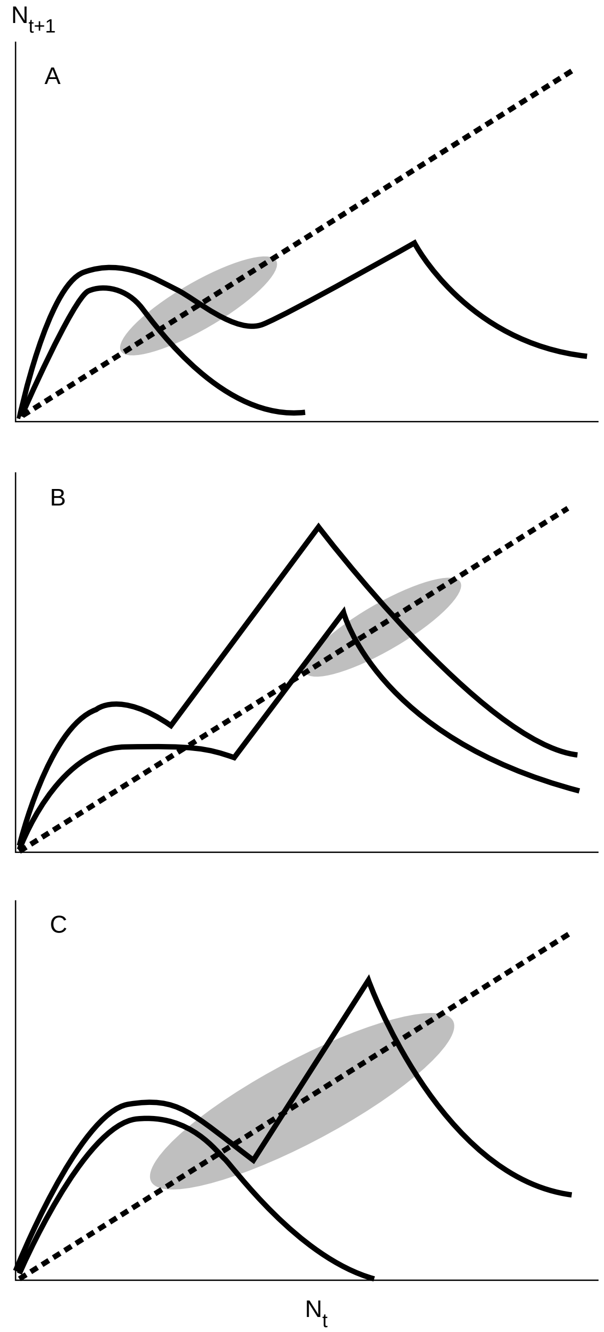

Figure VII.14–5—Domains of attraction might emerge for grasshopper population regulation, where natural enemies along with weather—which primarily affects density-independent survival and reproduction—sets the bounds of population fluctuations *(A);* competition for food along with weather–which primarily affects food abundance–sets the bounds of population fluctuations *(B);* or natural enemies and food competition in different years with weather set the bounds of population fluctuations *(C).*

might be largely ineffective unless the nearby population providing dispersers is controlled, too. In this scenario, the population causing damage is not the population that should be controlled because the dynamics of the former are dependent on the latter.

The implications of population regulation for grasshopper management may seem of little importance to managers entrusted with reducing the economic damage caused by pest grasshoppers. However, understanding how particular populations entrusted to a manager are regulated can provide critical insights that could make monitoring and control more cost effective.

General Conclusions

In terms of monitoring, the following generalizations might be reached:

1. Populations consistently within a domain that is regulated by natural enemies seldom reach densities at which economic damage is sufficient to warrant control; therefore, these populations may not warrant monitoring.

2. Populations consistently within a domain that is regulated by food availability often reach densities that cause economic damage and regularly warrant control; therefore, these populations may not warrant monitoring.

3. Populations in a domain where regulation can frequently “jump” between natural enemy limitation and food limitation will only periodically cause economic damage and warrant control; therefore, these populations may warrant monitoring.

If a manager knows the mode of regulation operating on a specific grasshopper population, monitoring efforts can be more effectively carried out, and that will save time and money.

In terms of control strategies, with the knowledge of how a population is regulated, a manager may be able to enhance efficiency by creating strategies that are tailored to the particular population. For example, I found (1992 unpubl.) that an insecticide application that killed less than 20 percent of the grasshopper nymphs—an application level much less than commonly employed—could shift a population from being regulated by food availability to being regulated by natural enemies. Switching to such a spray regimen would lessen control costs directly and also indirectly, by taking advantage of the more effective actions of natural enemies. Low-mortality spraying also would lead to less future management activity, with further cost reductions, because natural enemies would help to suppress future population increases.

Understanding how grasshopper populations are regulated and how regulation differs between regions of the western rangelands is essential for the development of new control strategies that involve reduced insecticide use, biocontrol agents, and grazing and habitat manipulation.

References Cited

Belovsky, G. E.; Slade, J. B. 1993. The role of vertebrate and invertebrate predators in a grasshopper community. Oikos 68: 193–201.

Belovsky, G. E.; Slade, J. B. 1995. Dynamics of some Montana grasshopper populations: relationships among weather, food abundance and intraspecific competition. Oecologia 101: 383–396.

Berryman, A. A. 1987. The theory and classification of outbreaks. In: Barbosa, P.; Schultz, J. C., eds. Insect outbreaks. New York: Academic Press: 3–30.

Horn, H. S. 1968. Regulation of animal numbers: a model counterexample. Ecology 49: 776–78.

Joern, A.; Gaines, S. B. 1990. Population dynamics and regulation in grasshoppers. In: Chapman, R. F.; Joern, A., eds. Biology of grasshoppers. New York: John Wiley and Sons: 415–82.

Sinclair, A.R.E. 1989. Population regulation in animals. In: Cherett, J. M., ed. Ecological concepts. Oxford, UK: Blackwell Scientific: 197–241.

Southwood, T.R.E.; Comins, H. N. 1976. A synoptic population model. Journal of Animal Ecology 45: 949–65.

Varley, C. G.; Gradwell, G. R.; Hassell, M. P. 1973. Insect population ecology: an analytical approach. Berkeley, CA: University of California Press: 212 p.

Reference Cited–Unpublished

Belovsky, G. E. 1992. Grasshopper control: implications of investigations into population/community ecology. In: Grasshopper Integrated Pest Management Project, 1992 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 57–63.

VII.15 Grasshopper Habitat Manipulation

G. E. Belovsky, M. A. Brusven, D. J. Fielding, and L. Manske

Introduction

Managing grasshopper populations through habitat manipulation (changes) is poorly understood and consequently, seldom considered. However, it may be a very reasonable strategy given the diversity of grasshopper species found in any single habitat (vegetation type) and the large area that pest managers must deal with in the rangelands of the Western United States. In fact, habitat management, such as destruction of prime egg-laying sites, was one of the earliest and most common forms of grasshopper control (Pfadt and Hardy 1987).

Habitat manipulation would seem particularly useful today because many grasshopper outbreaks occur in habitats that have been changed by human activities. Overgrazing, modified fire regimes, and introduction of exotic plants on American rangelands have led in some instances to replacement of relatively grasshopper-resistant native vegetation with vegetation that supports more frequent grasshopper outbreaks. An example may be when the native, perennial sagebrush/bunchgrass of the Intermountain regions are replaced with annual grasses and forbs. Therefore, restoration of the land's productivity can go hand in hand with grasshopper control by habitat manipulation.

The potential use of habitat manipulation as a control strategy is apparent when the following two possibilities are taken into consideration: (1) Most grasshopper species do not reach outbreak levels or cause economic damage (Pfadt 1988). What if managers could replace species that reach outbreak levels and cause economic damage with species that do not? Species substitution on this scale might be possible through habitat manipulation. (2) Even if outbreak species cannot be totally replaced, habitat manipulations may reduce their abundance and lessen the likelihood of outbreaks.

To address these habitat manipulation prospects, we can provide some potential examples but cannot present general strategies because this issue has not been broadly examined. When we refer to habitat manipulation, we are largely concentrating on vegetation changes because both the absolute and relative abundance of grasshoppers are related to vegetation (Kemp et al. 1989, Belovsky and Slade 1995). Vegetation changes can have a variety of impacts.

Fostering Natural Enemy Abundance

If pest managers could change the vegetation, doing so might increase natural enemies of grasshopper species that reach outbreak levels. Such increases could reduce abundance of the pest grasshoppers and the frequency of outbreaks (Belovsky and Slade 1993).

Predators as Grasshopper Population Regulators.— Predators, especially vertebrates such as birds and rodents, are potentially important in regulating grasshopper numbers under certain circumstances (see chapter VII.14). It may be possible by habitat manipulations to extend the circumstances under which predators effectively limit grasshopper numbers. First, greater vegetative cover may increase the numbers of these predators by protecting rodents and bird nests from their predators. Second, less vegetative cover (open *vs.* thick areas) can make grasshoppers more vulnerable to predators (fig. VII.15–1). The figures in this illustration were measured by placing tethered grasshoppers in areas of different vegetative cover and determining how many were killed by predators.

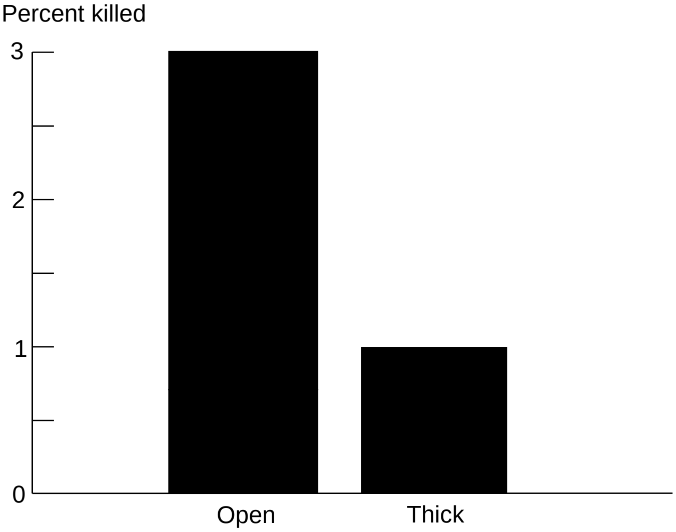

Figure VII.15–1—Comparison of the effectiveness of predators at killing grasshoppers in grasslands with more than 40 percent bare ground (open) versus less than 20 percent bare ground (thick) in western Montana.

The effects of habitat on predation might seem in opposition—on one hand increasing cover for birds and on the other hand decreasing cover for grasshoppers. However, on rangelands, the management trend is to make them more uniform. For example, overgrazing tends to reduce the height of vegetation; while this factor can make the grasshoppers more vulnerable to predation, there are now fewer predators to take advantage of the more open conditions for hunting, so the potential for greater predation on grasshoppers is seldom fully realized.

Manipulation might restore some of the natural variation in the habitat. Changes of that sort might be accomplished by providing small patches of thick cover for protection of the grasshoppers' predators, especially bird-nesting sites. Simultaneously, a pest manager might maintain habitat openness or even reduce cover in the intervening larger areas between patches of thick cover to increase the effectiveness of the predators in capturing grasshoppers. In doing this, a manager might be able to increase the predators' numbers and efficiency and thereby enhance the ability of predators to limit grasshoppers when predators otherwise might not be effective.

Parasitoids and Parasites.—As with predators, parasitoids and parasites might have their numbers and efficiency enhanced by manipulating the vegetation. For example, mites (parasites that attach themselves to a grasshopper's exoskeleton and "suck" the grasshopper's "blood") can dramatically reduce grasshopper survival and egg production, but these parasites generally do not appear to reach high enough densities to limit grasshoppers (see chapter I.9).

The inability of mites to reach high enough densities to limit grasshopper populations appears to be due in many areas to soils that have reduced drainage. Poor drainage should not be confused with moist conditions, a rarity in most western rangelands; poor drainage pertains to soils, such as clays, that tend to hold moisture longer. As with cover for predators, a manager might consider creating patches favorable to mite production that are interspersed throughout the larger area. Changing vegetation composition or cover or even providing small areas of better draining soils in small areas could achieve this end.

Reducing Grasshopper Food Abundance

In many areas of western rangeland, food abundance may be limiting grasshopper populations (see chapter VII.14). It may be possible to diminish food abundance using habitat manipulations in ways that will not negatively affect the forage available to livestock.

Increasing Competitors' Abundance.—If other species compete with the pest grasshoppers for food, then increasing the abundance of these competitors might reduce the abundance of pest grasshoppers. Unfortunately, enhancing the numbers of competitors might simply substitute one pest for another so that the forage available to livestock is not enhanced. However, limiting pest grasshoppers by reducing their available food through consumption by competitors, without simultaneously diminishing the forage available to livestock, might be accomplished under two conditions. First, livestock grazing might be used to reduce grasshopper numbers; this substitutes livestock consumption for grasshopper consumption of the forage. Second, habitat manipulations might be used to replace pest grasshopper species with species that do not reach outbreak levels, especially if these other species do not reduce the forage for livestock to as great a degree as the pest species.

Different studies have disclosed that livestock grazing decreases grasshopper densities (Hutchinson and King 1980; Jepson–Innes and Bock 1989; Capinera and Sechrist 1982; Fielding and Brusven 1995), increases densities (Coyner 1938 unpubl., Nerney 1958, Anderson 1964, Holmes et al. 1979), and has no effect (Miller and Onsager 1991) on grasshopper densities. In cases where grazing reduced grasshopper abundance, it appeared that the grasshoppers encountered a shortage of food. In cases where grazing increased grasshopper abundance, it appeared that the grasshoppers either responded to decreased cover (see thermal cover, below) or increased forb abundance (see vegetation changes, below). All of the above studies found that the grasshopper species composition changed with grazing. Grazing effects are more fully discussed in chapter V.1.

Grasshoppers that compete with the pest species might be encouraged by management to reduce the pests' abundance. This option would be useful if the competitor emerges earlier than the pest, so that survival of the pest species' nymphs is reduced. In addition, it would be particularly useful if the earlier emerging competitor cannot survive later into the season, when the pest would otherwise be most abundant; this scenario would allow the vegetation to regrow after consumption by the competitor.

An example is provided by the nonpest early-season grasshopper *Melanoplus confusus* and the pest late-season grasshopper, *M. sanguinipes,* in the Palouse prairie of western Montana (Belovsky 1990 unpubl). As fourth- and fifth-instar nymphs and adults, *M. confusus* dramatically reduces the survival of *M. sanguinipes* in experimental populations by competing for food plants (fig. VII.15–2). The *M. confusus* adults quickly die off in early July, and the vegetation regrows because rains in most years permit continued growth. The negative effect of *M. confusus* on *M. sanguinipes* is illustrated by *M. sanguinipes* being able to reach the same densities in the experimental mixed populations as in experimental pure populations, when *M. sanguinipes* are placed in the experiments after *M. confusus* dies off (fig. VII.15–2). Unfortunately, under natural conditions, *M. confusus* populations are generally too low to achieve this effect.

Encouraging *M. confusus.*—A straightforward means by which a manager might increase *M. confusus* numbers is not apparent.

Manipulating Plant Species.—The relative abundance of different plant species might be manipulated to reduce the abundance of those species that are more important to the pest grasshoppers than they are to livestock. While grasshoppers and livestock consume many of the same plant species and thereby compete, grasshoppers do not consume identical sets of food plants. A good example of this manipulation might be to reduce the abundance of annual grasses and forbs and to increase the abundance of perennial grasses and shrubs. Many pest grasshoppers, especially in the spurthroated group (Melanoplines), seem to thrive with the annuals, and livestock are capable of foraging on the perennials. But changing vegetative composition can also modify cover and plant abundance. Therefore, habitat manipulations that modify the relative abundances of plants need to be weighed against changes in these other factors and how they affect both pest and livestock.

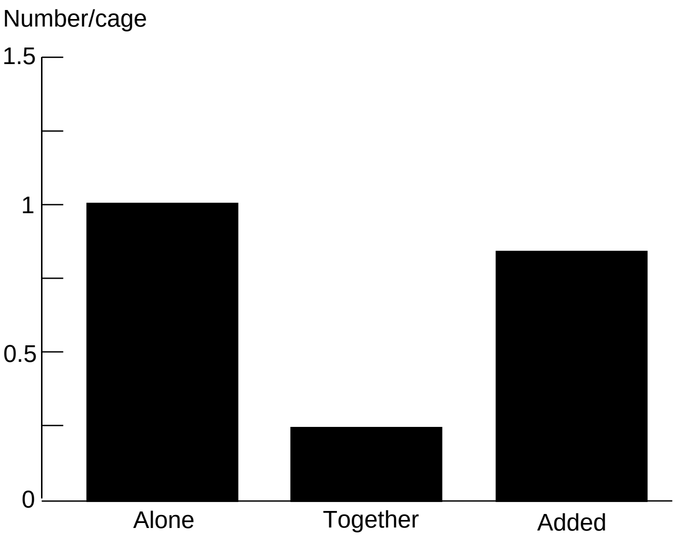

Figure VII.15–2—The densities attained by *Melanoplus sanguinipes* in experimental field populations (cages) when by itself (Alone), when with *M. confusus* (Together), and when it is added after *M. confusus* dies off later in the summer (Added).

Changing Grasshopper Thermal Cover

Vegetation provides more than food—it also provides thermal cover for grasshoppers. Grasshoppers are able to consume a greater quantity of food when they are in favorable thermal conditions. Under favorable conditions, a grasshopper can process more food through its digestive tract and has more time to consume foods. Greater food consumption leads not only to greater immediate losses of forage resources on rangelands but also to larger grasshopper populations by increasing the grasshoppers' survival and reproduction.

Thick vegetative cover for a grasshopper may lead to a thermal environment that is cooler than optimal, reducing grasshopper survival and reproduction. The same effect can be caused when there is too little vegetative cover for a grasshopper and the environment is warmer than optimal. Therefore, land managers might manipulate vegetative cover to diminish grasshopper feeding, and thereby, their survival and reproduction.

Possible Methods for Habitat Manipulation

We have presented a series of ecological processes that habitat management might be able to exploit to reduce pest grasshoppers. However, methods are required to modify the habitat and thereby change the ecological processes.

A number of methods have been investigated without reference to how they changed ecological processes. It has been demonstrated that the use of herbicides on rangelands has little effect on grasshoppers, while furrowing, scalping, and interseeding grazing lands can reduce grasshopper numbers dramatically (Hewitt and Rees 1974). Researchers are not sure if furrowing, scalping, and interseeding change predation cover, thermal cover, plant composition, or all of these factors.

One method that has been investigated at least partially from the perspective of ecological processes operating on pest grasshoppers is fire on rangelands. It primarily operates to change the composition of the vegetation and, thereby, grasshopper food abundance. However, fire can produce different outcomes on pest grasshoppers. Under some conditions, fire enhances grasshopper numbers and in others, decreases them. For example, intense fires destroy sagebrush/native bunchgrasses, enhancing annual plants, which are favored by pest grasshoppers. On the other hand, “cool” fires enhance the abundance of native bunchgrasses and, thereby, decrease pest grasshoppers. Likewise, livestock grazing can be used to manipulate vegetation composition, but as with fire, different grazing intensities result in different outcomes.

Reseeding areas with crested wheatgrass after native bunchgrasses have been destroyed can reduce pest grasshopper abundance but not to the extent that native bunchgrasses can. Therefore, methods for restoring native rangelands may have considerable potential for grasshopper pest management.

A greater variety of these methods needs to be investigated in a range of different habitats. However, these methods may require greater than normal monitoring by managers. For example, grazing and fire both require the manager to assess intensity carefully, and doing that can be difficult as weather conditions dramatically change the vegetation from year to year.

For example, management by grazing might require the manager to manipulate stocking rates much more than ranchers traditionally have undertaken, or in ways that do not maximize the rancher’s income. In addition, habitat manipulations must be evaluated in terms of their impacts on wildlife, recreation activities, and the maintenance and restoration of native vegetation. Habitat manipulations have not been adequately investigated as a viable pest-management strategy for grasshoppers, but manipulations may have great potential to reduce grasshopper-caused damage with fewer negative impacts on the environment.

References Cited

Anderson, N. L. 1964. Some relationships between grasshoppers and vegetation. Annals of Entomological Society of America 57: 736–42.

Belovsky, G. E.; Slade, J. B. 1993. The role of vertebrate and invertebrate predators in a grasshopper community. Oikos 68: 193–201.

Belovsky, G. E.; Slade, J. B. 1995. Dynamics of some Montana grasshopper populations: relationships among weather, food abundance and intraspecific competition. Oecologia 101: 383–396.

Capinera, J. L.; Sechrist, T. S. 1982. Grasshopper (Acrididae)–host plant associations: response of grasshopper populations to cattle grazing intensity. Canadian Entomologist 114: 1055–62.

Fielding, D. J.; Brusven, M. A. 1995. Grasshopper densities on grazed and ungrazed rangeland under drought conditions in southern Idaho. Great Basin Naturalist 55:352–358.

Hewitt, G. B.; Rees, N. E. 1974. Abundance of grasshoppers in relation to rangeland renovation practices. Journal of Range Management 27: 156–60.

Holmes, N. D.; Smith, D. S.; Johnston, A. 1979. Effect of grazing by cattle on the abundance of grasshoppers on fescue grassland. Journal of Range Management 32: 310–11.

Hutchinson, K. L.; King, K. L. 1980. The effects of sheep stocking level on invertebrate abundance, biomass and energy utilization in a temperate, sown grassland. Journal of Applied Ecology 17: 369–87.

Jepson–Innes, K.; Bock, C. E. 1989. Response of grasshoppers (Orthoptera: Acrididae) to livestock grazing in southeastern Arizona: differences between seasons and subfamilies. Oecologia 78: 430–31.

Kemp, W. P.; Kalaris, T. M.; Quimby, W. F. 1989. Rangeland grasshopper (Orthoptera: Acrididae) spatial variability: macroscale population assessment. Journal of Economic Entomology 82: 1270–76.

Miller, R. H.; Onsager, J. A. 1991. Grasshopper (Orthoptera: Acrididae) and plant relationships under different grazing intensities. Environmental Entomology 20: 807–14.

Nerney, N. J. 1958. Grasshopper infestations in relation to range condition. Journal of Range Management 11: 247.

Pfadt, R. E. 1988–1997. Field guide to common western grasshoppers. Bull. 912. Laramie, WY: University of Wyoming and Wyoming Agricultural Experiment Station. 250 p.

Pfadt, R. E.; Hardy, D. M. 1987. A historical look at rangeland grasshoppers and the value of grasshopper control programs. In: Capinera, J. L., ed. Integrated pest management on rangeland: a shortgrass prairie perspective. Boulder CO: Westview Press: 183–95.

References Cited–Unpublished

Belovsky, G. E. 1990. Grasshopper competition and predation: biological control options. In: Grasshopper Integrated Pest Management Project, 1990 annual report. Boise, ID: U.S. Department of Agriculture, Animal and Plant Health Inspection Service: 37–44.

Coyner, W. R. 1938. Insect distribution and seasonal succession in overgrazed and normal grasslands. Unpubl. M.S. thesis. Norman, OK: University of Oklahoma. 78 p.

VII.16 Grasshoppers—Plus and Minus: The Grasshopper Problem on a Regional Basis and a Look at Beneficial Effects of Grasshoppers

G. E. Belovsky, A. Joern, and J. Lockwood

Introduction

From an environmental perspective, grasshopper control in rangelands of the Western United States poses several unique and difficult problems compared to the control of many other insect pests.

- When scientists or land managers speak of grasshoppers, they are not referring to a single pest species but to a group of insects that contains more than 400 species, with as many as 30 to 40 species found in any given area. Some of these species cause economic damage, but most do not; however, current control methods influence all (Lockwood 1993a and b, Carruthers and Onsager 1993).

- None of these insects has been introduced to the West by humans. All are natural elements of a complex ecological system that is highly productive for livestock and wildlife. Therefore, grasshoppers are an important consideration in conservation planning (Lockwood 1993a and b, Carruthers and Onsager 1993).

- While managers often consider rangelands to be uniform grasslands, rangelands can refer to mountain meadows, savannas, forested parklands and shrublands, and steppe grasslands. Rangelands vary dramatically in plant species composition; the amount, frequency, and annual distribution of precipitation; and forage production.

Seeking or expecting a single control strategy for pest grasshoppers may be fruitless. Grasshoppers form a diverse group of species that inhabit a diverse group of habitats. Advocating the elimination or dramatic reduction in grasshopper numbers, even if this action were biologically and economically feasible, could be destructive to the very ecological system whose production we are trying to maintain and exploit (Lockwood 1993a and b, Mitchell and Pfadt 1974). Consequently, control may not be a desirable goal. Management may be the more appropriate perspective.

Grasshopper management should attempt to minimize competition for forage between grasshoppers, livestock, and wildlife in cases when most rangeland production is needed for livestock and wildlife. Of course, all forage that grasshoppers eat cannot be consumed by livestock and wildlife. Grasshoppers have an important role in the ecological processes that make U.S. rangelands so productive. Shifting the management viewpoint from elimination to suppression is a difficult undertaking but places grasshopper management within the larger context of sustainable ecosystem management and the preservation of biodiversity.

Given past concern over grasshopper damage to rangeland production, one would think that the scientific ability to address the central issues would be much more extensive than it is. Most efforts have focused on control, and perhaps in some cases eradication, of grasshoppers. With the development of commercially produced synthetic pesticides in the 1930's, this focus led to a predominance of studies intended to produce better insecticides and means of application. Such a focus also replaced investigating grasshopper biology in ways that might form a basis for alternate approaches.

An integrated pest management approach must be founded upon the biology of the pest species. The Grasshopper Integrated Pest Management Project has helped provide us with more information on grasshopper control and biology. Project-funded investigators have identified many important questions that a pest manager must consider. Considering such questions is the critical first step in fostering the development of management strategies for particular rangeland locations in the future.

Grasshopper Management Over the Variety of Rangelands

One simple observation from grasshopper studies illustrates the enormous task posed by grasshopper management over the range of species and habitats found in the Western United States. In the southern rangelands, increased precipitation and possibly cooler temperatures appear to increase grasshopper numbers. In northern rangelands, the opposite conditions (warm and dry) appear to increase grasshopper numbers (Capinera and Horton 1989). This comparison covers an immense region and glosses over the variability in vegetation among different areas. There also are other ecological factors that lead to variation in grasshopper numbers and species composition (Joern and Gaines 1990). Furthermore, we have little idea of what particular mechanisms are driving the above patterns (including changes in plant production, plant nutritional value, grasshopper developmental rate, predation rate, fungal infection rate, and more), because the weather variables are no more than correlates with grasshopper numbers (Joern and Gaines 1990).

To illustrate further the problems arising from the diversity of rangeland habitats, there are two other major differences that emerge in comparisons of southern and northern rangelands. In the South, warm-season grasses dominate, and the smaller bodied, slantfaced (Gomphocerinae) grasshoppers are most abundant. In northern areas, cool-season grasses dominate, and the larger bodied, spurthroated (Melanoplinae) and bandwinged (Oedopodinae) grasshoppers are most abundant. Warm-season grasses generally are less nutritious for grasshoppers than cool-season grasses. Slantfaced grasshoppers that dominate in areas with warm-season grasses are better at feeding on these plants. Therefore, the weather correlates observed over the rangelands of the Western United States are further complicated by major changes in vegetation and grasshopper species composition.

The above points illustrate the need to better define the environmental conditions that affect grasshoppers in different regions and the ways that grasshopper populations function. Furthermore, some evidence suggests that rapid, human-induced climate changes could make identifying regional patterns worth little to managers. Climate changes may produce new patterns rather than simple latitudinal displacements of existing patterns (southern rangelands may not simply move northward). Similarly, other human-induced changes in the environment (changes in the abundances of native plant species and introductions of exotic plants and animals) could disrupt observed patterns. Therefore, people need to understand the different processes creating the patterns observed in different western U.S. rangelands. By doing so, managers can anticipate and plan responses to the changing environments, policies and values that will confront us in the future.

The Ecological Role of Grasshoppers

Grasshoppers play an important role in the functioning of rangeland ecosystems (Mitchell and Pfadt 1974). First, results from a variety of studies reveal that grasshoppers typically consume at least 10 percent of available plant biomass. Second, grasshoppers often harvest more plant biomass than they consume, influencing the availability and distribution of litter in the environment. This consumption and harvesting could be deemed negative from the perspective of available plant biomass for livestock production. But such “harvesting” processes can serve important functions for the cycling of nutrients.

Microbes can break down the feces produced by grasshoppers more easily than those produced by larger herbivores, such as cattle or sheep. Grasshopper-generated fecal nutrients are therefore more available for plant production. Also grasshoppers have a shorter lifespan and generally decompose where they die. The nutrients in their bodies return more rapidly to the soil for plant use than do nutrients found in the bodies of livestock. Even when grasshoppers create litter, they are enhancing plant production because increased litter increases the water retention of soils and reduces summer soil temperatures. These phenomena, in turn, enhance plant production by making more water and nutrients available in the semiarid and arid conditions of the West. In total, grasshoppers may exert a positive influence on rangeland plant production.

Grasshoppers selectively feed on different plant species and, consequently, influence the plant species composition of the ecosystem. Sometimes, the grasshoppers harvest plants that livestock prefer. In other instances, grasshoppers consume plants that are poisonous or competitively reduce the abundance of plants preferred by livestock. The selective consumption of different plant species by grasshoppers can change the nutrient cycling dynamics in a rangeland. This change happens because the total nutrient content and decomposition rate of the litter depend on the plant species composing the litter (Pastor et al. 1987). Therefore, selective consumption of certain plant species can have a positive or negative effect on primary production for livestock by changing plant species abundances and nutrient cycling.

Grasshoppers are a major food source for other species that inhabit rangelands, especially spiders, reptiles, birds, and small mammals. Consequently, grasshoppers support other biological components of the ecosystem and influence their ability to affect ecosystem functioning. Again, grasshoppers can positively or negatively influence the biological composition of ecosystems and their productivity for livestock.

With the increasing emphasis placed upon ecosystem management by Federal and State agencies, grasshoppers in the rangelands of the Western United States must be considered in terms of their beneficial actions, not just in terms of their potential to reduce the abundance of forage for livestock. Consequently, pest management cannot be considered in isolation from larger ecological issues. This is especially true when the pest is a natural, coevolved component of the ecosystem, as grasshoppers are in western rangelands. Land managers must explicitly acknowledge that in most years, in most places, most grasshopper species do not harm the rangeland resource; rather they may benefit the resource.

Grasshoppers as a Range-Management Tool

Considering the important role grasshoppers serve in ecosystems, these insects deserve consideration as a tool land managers could employ to enhance rangeland productivity for livestock. First, nutrient cycling must be maintained to preserve or enhance rangeland production, and grasshoppers may aid in this goal. Second, the selective foraging of grasshoppers on different plant species might increase the abundance of plants that are more palatable and beneficial to livestock. Therefore, the negative effects of grasshoppers on forage availability for livestock must be compared against their positive effects on maintaining or enhancing rangelands.

Perhaps the greatest potential of grasshoppers as a management tool may be to alleviate the growing problem of weed control (Lockwood 1993a). For example, it appears that the grasshopper *Hesperotettix viridis* may control the abundance and spread of snakeweed *(Gutierrezia* spp.), rabbitbrush *(Chrysothamnus* spp.), ragweed *(Ambrosia* spp.), and locoweeds *(Astralagus* spp.). The grasshopper *Melanoplus occidentalis* may reduce the abundance of prickly pear cacti. Even more important, grasshoppers may prevent or retard the spread of exotic weeds, as with feeding by *Aeoloplides turnbulli* and *Melanoplus lakinus* on Russian thistle *(Salsola iberica).* Scientists need to investigate more fully the potential benefit of weed control through grasshopper feeding. This area of research could become especially important with the difficult problem of controlling the spread of exotic weeds on rangelands. Weeds compete with native flora, and livestock find many weeds especially unpalatable.

Grasshoppers and Conservation

Clearly grasshoppers can provide many benefits that the public frequently has overlooked for the conservation of rangelands. In addition, there is growing social and political concern for the protection of biodiversity. Concern increases because of unrecognized benefits provided by many species and their important role in maintaining healthy ecosystems, and because these species are an important part of our cultural history and they are esthetically pleasing (Wilson 1989). Finally, there is a growing view in U.S. society that people have an ethical obligation to ensure the continued existence of all species and the ecosystems that they inhabit. The view is that each species has the same evolutionary value as the human species, and ecosystems have the same value as human society (Kellert and Wilson 1993).

Grasshoppers usually are abundant enough to be exempt from threats of extinction. Nonetheless, at least one species of grasshopper that was a very abundant pest appears to have become extinct, the Rocky Mountain locust *(Melanoplus spretus).* This species did not die out from control efforts but probably from habitat destruction caused by agriculture and livestock grazing (Lockwood and DeBrey 1990).

Not many years ago, the loss of the Rocky Mountain locust was considered a benefit. Today, many view this loss with apprehension. Few people would wish a return to the state where this species destroyed croplands, but the public can no longer experience, even on a small scale, the swarms that darkened the skies and stopped transcontinental railroads as told as part of America’s national heritage and folklore. More importantly, the loss of the Rocky Mountain locust means that an important element of the Nation's pristine rangelands has been lost, and the loss exemplifies the general assault upon natural environments, especially rangelands, by human actions.

For example, exotic plant species have almost entirely replaced the native annual grasslands of California. Only remnants of tallgrass prairie remain, and the introduction of exotic plants threatens most other western rangelands. What will happen to the native grasshoppers that inhabit these ecosystems? Several species of monkey grasshoppers in native desert grasslands are considered threatened and may eventually be listed for protection under the Endangered Species Act.

The decline of grasshoppers also affects other species, especially those that consume them. Recently, the U.S. Fish and Wildlife Service announced that western rangeland birds have dramatically declined in abundance over the last decade, with the numbers of some species decreasing by as much as 70 percent. Many of these birds feed on grasshoppers as adults, and almost all rely heavily on grasshoppers to provision their nestlings. Therefore, the control of grasshoppers must be considered in a broader conservation perspective than forage production for livestock, protection of threatened grasshopper species, and the maintenance of the ecosystem functions provided by grasshoppers. Grasshopper reduction also might harm declining or threatened species that depend on these insects as food (Belovsky 1993).

Conservation concerns are becoming more pronounced in formulating management plans because of legal and social mandates. Therefore, the scope and scale of grasshopper control programs will no doubt become more restricted in the future and will require consideration of far more than the short-term economic costs of grasshopper consumption of livestock forage.

Questions for the Future

One certainty for the future is that grasshopper management will be changing. There will be little "business as usual."

- The methods of grasshopper control will change as society becomes more concerned with environmental degradation and the protection of all native species. Therefore, new and innovative control methods that are environmentally sound will need to be found and used.

- Grasshoppers, as native components of rangelands, will no longer be considered solely as pests to be suppressed or eradicated, but as important elements for the functioning of our natural ecosystems. Furthermore, society is beginning to view all species that are part of our native biodiversity as having esthetic value, as providing a reflection of our national heritage that deserves some level of protection, and as requiring protection from an ethical perspective. The short-term economic costs/benefits of pest control to livestock production will become less important in decisionmaking and more subject to review by society.

- The general patterns of grasshopper abundance in different regions will change if humans change the global climate as projected by many scientists. Therefore, managers must act in places and ways previously unanticipated. The result is that pest managers need to adopt a broader perspective of their role, become more flexible in their actions, and view the changing environment as an exciting challenge, rather than a hindrance.

References Cited

Belovsky, G. E. 1993. Modelling avian foraging: implications for assessing the ecological effects of pesticides. In: Kendall, R. J.; Lacher, T. E., Jr., eds. Wildlife toxicology and population modeling: integrated studies of agroecosystems. Boca Raton, FL: CRC Press: 123–138.

Capinera, J. L.; Horton, D. R. 1989. Geographic variation in effects of weather on grasshopper populations. Environmental Entomology 18: 8–14.

Carruthers, R. I.; Onsager, J. A. 1993. Perspective on the use of exotic natural enemies for biological control of pest grasshoppers (Orthoptera: Acrididae). Environmental Entomology 22: 885–903.

Joern, A.; Gaines, S. B. 1990. Population dynamics and regulation in grasshoppers. In: Chapman, R. F.; Joern, A., eds. Biology of grasshoppers. New York: John Wiley and Sons: 415–483.

Kellert, S. R.; Wilson, E. O. 1993. The biophilia hypothesis. Washington, DC: Island Press. 484 p.

Lockwood, J. A. 1993a. Environmental issues involved in biological control of rangeland grasshoppers (Orthoptera: Acrididae) with exotic agents. Environmental Entomology 22: 503–518.

Lockwood, J. A. 1993b. Benefits and costs of controlling rangeland grasshoppers (Orthoptera: Acrididae) with exotic organisms: search for a null hypothesis and regulatory compromise. Environmental Entomology 22: 904–914.

Lockwood, J.; DeBrey, L. D. 1990. A solution for the sudden and unexplained extinction of the Rocky Mountain locust, *Melanoplus spretus* (Walsh). Environmental Entomology 19: 1194–1205.

Mitchell, J. E.; Pfadt, R. E. 1974. The role of grasshoppers in a shortgrass prairie ecosystem. Environmental Entomology 3: 358–360.

Pastor, J.; Dewey, B.; Naiman, R. J.; McInnes, P. 1987. Moose, microbes and the boreal forest. BioScience 38: 770–777.

Wilson, E. O. 1990. Threats to biodiversity. Scientific American 9: 108–116.

Appendix 1—Grasshopper Integrated Pest Management Project Cooperators (1987–94) and Authors' Affiliations

Note: This appendix captures the name and address of senior and junior authors of all chapters within the Grasshopper Integrated Pest Management User Handbook. However, some authors may have moved or changed their employment status since the program officially closed on October 1, 1995. Updated listings (current to June 2000) have been included wherever possible.

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