Section N

Integrated Pest Management

Concepts of IPM

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The traditional definitions of IPM are based on the ecological foundation of pest monitoring, exploring the potential of multiple control options prior to the application of pesticides. While this approach was critical for maintaining the ecological balance of the crop production system and protecting environmental and human health, the growth of agriculture as a global enterprise, food security and affordability for growing world populations, and other socio-economic factors warranted a new approach of IPM. The new approach, while continuing to emphasize the ecological balance, presents a more practical approach that is inclusive of science and technology, business management and marketing aspects, communication, and other critical components for an economically viable, socially acceptable, and environmentally sustainable crop production system (Dara 2019).

IPM offers multiple benefits as it explores the potential of several control options, optimizes the use and associated costs of pesticides applications, reduces the risk of pesticide resistance and secondary pest outbreaks, extends the longevity of available options, and increases the overall efficiency of pest management. Depending on the complexity of the management system, an IPM program may target a single pest, a pest category (e.g., insects, weeds, diseases or rodents) or the whole pest complex. While traditional pest control considers each pest exclusively, IPM considers the interactions among pests, beneficial organisms, the environment, and the crop as an ecosystem.

Development of an IPM system requires a thorough understanding of the biology of the crop (or resource), pest complex, mode of action of various control options, and the influence of various factors on each other. The IPM concept was developed from the realization that most pesticide applications affect both pests and beneficial organisms in the crop, sometimes to the disadvantage of the grower.

An IPM system attempts to maintain pest populations below economically damaging levels by using a holistic systems approach that creates synergism by integrating preventative methods that build on agronomic, mechanical, physical, and biological principles, resorting to selective pesticide use when other tools are not effective in addressing the pest situation alone (Barzman et al. 2015). IPM systems are flexible and highly customizable programs that depend on the time of year, location, type of crop, pest problem, available options, and others. Many books, manuals and websites are devoted to discussions of general IPM principles and to the application of IPM to specific agricultural and urban systems. The following components are generally found in IPM programs:

1. **Prevention and suppression** Aims to prevent or suppress any single species from becoming most dominant or damaging in a cropping system by using healthy and infestation free planting material, crop rotation, use of adequate cultivation methods, incorporating resistant and or tolerant cultivars, best management practices for fertilization and irrigation etc.
2. **Management units** Monitoring is conducted with the aim of providing results for the management of a specific management unit – the part of the system that will receive the same pest control decisions. The unit may be part of a field, a single field, or several fields. Chemical control decisions are sometimes based on the area that can be covered by a single spray tank.
3. **Key pests** An IPM program targets specific pests, which may include insects, mites, plant diseases, weeds, or vertebrates. In the development of an IPM program, these pests are identified and monitoring and control programs are designed for each of these pests.
4. **Monitoring** Sampling should accurately assess the pest pressure and the abundance of beneficial organisms in the management unit. Monitoring is conducted so that management actions can take place in a timely and effective manner.
5. **Pest action thresholds** Keeping fields entirely pest free is neither necessary nor desirable. Most crops can tolerate low pest infestation levels without any yield loss and those low pest populations also support their natural enemies. IPM seeks to reduce pest numbers below economically damaging levels rather than eliminate infestations. Pesticides should be applied only when economically justified by the number of pests present.
6. **Selective insecticides or acaricides** Selective or soft insecticide chemistries are designed to target pest species and are life-stage specific while having a minimal impact on the non-target organisms such as beneficial organisms or environment. Use of multiple controls and tactics Control tactics should be employed to make the crop less favorable for pest survival and reproduction, while disturbing the rest of the ecosystem as little as possible. Combining different control tactics into an overall strategy balances the strengths of each against any individual weaknesses. Control tactics should be compatible with beneficial organisms and the environment. Using different techniques (e.g., rotating pesticides with different modes of action) also reduces the probability of the development of pest resistance and application of available anti-resistance strategies to maintain the effectiveness of the products.

Developing or implementing an IPM program for a crop involves a systematic application of knowledge about the crop and the pests involved. The following sources may be useful in acquiring and applying that knowledge:

- Oregon IPM Center—https://agsci.oregonstate.edu/oipmc/
- Washington State University Extension IPM—http://www.ipm.wsu.edu/
- University of Idaho Pest Management Center—http://www.uihome.uidaho.edu/ipm
- US Environmental Protection Agency: Integrated Pest Management (IPM) Principles—http://www.epa.gov/opp00001/factsheets/ipm.htm
- Radcliffe's IPM World Textbook—http://ipmworld.umn.edu
- Flint, M.L, 2012. IPM in Practice, Principles and Methods of Integrated Pest Management. Univ. of Calif. Publ. 3418. 292 pp.
- Barzman, M., P. Bàrberi, A.N.E. Birch, P. Boonekamp, S. Dachbrodt-Saaydeh, B. Graf, B. Hommel, J.E. Jensen, J. Kiss, P. Kudsk, and J.R. Lamichhane. 2015. Eight principles of integrated pest management. Agron. Sustain. Dev. 35: 1199-1215.

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1 Flint, M. L. and R. Van den Bosch. 1981. Introduction to Integrated Pest Management. Plenum Press. 240 pp.

2 Surendra K Dara. 2019. The New Integrated Pest Management Paradigm for the Modern Age. https://doi.org/10.1093/jipm/pmz010

Biological Control

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Introduction

Biological control (or biocontrol) is a key component in establishing an ecological and integrated approach to pest management. We define biological control as the decline in pest density as a result of the presence of natural enemies. The degree of pest decline might be in the form of partial or complete pest suppression. We use the terms "natural enemies," "beneficials," and "biocontrol agents" synonymously to refer to predators, parasites, parasitoids, and diseases of pests. These different groups of natural enemies control pest populations in different ways. Predators and parasites are driven by food-seeking behavior, and control pest populations through predation. Parasitoids are driven by the reproductive need to use another insect as a host for its offspring to develop within, and control pest populations through reproductive pressure. Diseases are driven by infection pressure, and control pest populations through opportunistic outbreaks. How a natural enemy population responds to pest presence is critical to its efficacy.

Biocontrol is generally more compatible with organic and sustainable agricultural approaches than pesticide-dependent agriculture. This is especially evident when non-selective, broad-spectrum chemistries are used because biocontrol agents tend to be highly susceptible to non-selective pesticides. Even short to moderate pesticide exposure time may reduce their populations and allow minor pest insects that would otherwise be held in check to become major pest problems. The term "secondary pest outbreak" is used when this scenario occurs. A reduction in natural enemies can also contribute to dependence on further pesticide usage and result in a cycle of chemical dependency that has been called a "pesticide treadmill."

Ideally, natural enemies reproduce on their own; their populations are self-sustaining, they are not harmful to the ecosystem, and they can be used in combination with other integrated control tactics. Natural enemies used in biological control can target a wide range of pest species (generalist) or a limited range of species (specialist). Generalist natural enemies such as predatory beetles can switch readily among alternative food or host sources. When target pest numbers are low, generalist natural enemies may maintain populations locally by consuming other prey species. Specialist natural enemies such as some parasitoid wasps have more restricted prey choices, and will therefore leave or die out when prey numbers are low. Natural enemy populations may decline or become extinct when habitats are poor or unsuitable, host pest numbers are too low, or non-selective pesticides are applied. Some species may be incapable of suppressing pests below damage thresholds by themselves. In some cases, the benefits of natural enemy presence are often undervalued because many natural enemies are difficult to sample or even detect, and there is a dearth of information on their economic value in most cropping systems. The important role of natural enemies is often not realized until disruptions such as the application of broad-spectrum insecticides precipitates target pest resurgence or secondary pest outbreaks.

Insects are susceptible to entomopathogenic nematodes (roundworms) and a variety of diseases caused by pathogenic microbes, which include viruses, bacteria, fungi, and protozoa. For example, microbial insecticides consist of a pathogen or their toxin product as the active ingredient and may provide a satisfactory alternative to chemical pesticides when used as part of an Integrated Pest Management (IPM) plan (Sarwar 2015, Azizoglu and Karabörkü 2021). The most widely used microbial pesticides are subspecies and strains of *Bacillus thuringiensis* (BT insecticides). Like other natural enemies, pathogens are susceptible to environmental factors, anthropogenic spread, and pest population conditions. High pest density and favorable conditions can lead to disease outbreaks that can control pests, whereas low pest densities may cause pathogens to go dormant. Microbial insecticides may be delivered through many different media including liquids used for root drenches, sprays, or solids including clay-based powders or baits interlaced with microbes. Recent advances in genetic modification technology have led to the development of new engineered microbial insecticides with increased virulence and tolerance to environmental stress, lower insect resistance, and lower spraying requirements (Azizoglu and Karabörkü 2021).

A combination of generalist and specialist natural enemies can be an extremely useful part of IPM programs that recognize and encourage their activity (Lee-Mäder et al. 2014). At the same time, one must keep in mind that, like any pest control method, biological control agents can have unanticipated effects that may include attacking beneficial and native species (Kimberling 2004, van Lenteren et al. 2006).

Types of biological control

There are three principal approaches to biological control:

1. Classical biological control
2. Augmentative biological control
3. Conservation biological control

1. Classical biological control

Classical biological control is the importation of natural enemies for release and permanent establishment in a new region. New classical biocontrol agents increasingly require long-term, stringent evaluations in quarantine to measure their non-target effects and efficacy in controlling the target pest before they may be released. Biocontrol agents that are candidates for introduction may be rejected if host range tests show that they can attack native non-target species. Another risk of introducing new biocontrol agents is that the agent may unexpectedly begin attacking non-target species (host shifting) despite previous efforts to determine its host range. Additionally, new agents may vector pathogens or hyperparasitoids, or compete with other natural enemies that exploit the same resource. Thus, evaluation of risks related to the releases of natural enemies requires integration of many aspects of their biology, as well as information on possible ecological interactions in their recipient environment (van Lenteren et al. 2006).

In the Pacific Northwest (PNW), we have had few cases of highly successful classical biocontrol of insect pests, and there have been many more successful classical weed biocontrol cases using insects (see the PNW Weed Management Handbook). One successful insect biocontrol agent, the filbert aphid parasitoid wasp, *Trioxys pallidus* (Braconidae), was imported from Europe and introduced (in small numbers) by Oregon State University (OSU) scientists in the mid-1980s. Since then, this tiny wasp has spread throughout the growing region and generally maintains the filbert aphid below treatment thresholds, at an estimated economic benefit of about $400,000 per year (AliNiazee 2009). In another case, the spread of and damage caused by the apple ermine moth, *Yponomeuta malinellus* (Yponomeutidae), has been greatly reduced by the successful introduction of a parasitoid wasp, *Ageniaspis fuscicollis* (Encrytidae), in the late 1990s. A cooperative biocontrol program among the US Department of Agriculture, Animal and Plant Health Inspection Service (USDA-APHIS), Oregon Department of Agriculture (ODA), and OSU for cereal leaf beetle began in 2000 and was considered successful by 2010. The establishment of the larval parasitoid wasp, *Tetrastichus julis* (Eulophidae), yielded control of cereal leaf beetle below thresholds in some regions of the PNW, especially when combined with altered cultural practices (tillage, irrigation, crop rotation, etc.) and pesticide applications. In some cases, 100% parasitism was achieved. A small wasp in the family Eulophidae, *Colpoclypeus florus*, a native of Europe, has been credited as a significant biocontrol agent of leafroller pests such as the oblique-banded and pandemis leafrollers in Washington, and has also been collected in western Oregon. An egg-larval parasitoid, *Ascogaster quadridentatus* (Braconidae) was introduced to help manage codling moth, *Cydia pomonella (*Tortricidae), a key pest of apple and pear. The presence of this parasitoid on codling moth has been reported, although the economic success of its introduction is unknown. Previous classical biocontrol efforts in the PNW have also included programs directed at Russian wheat aphid [*Diuraphis noxia* (Aphididae)], larch casebearer [*Coleophora laricella (*Coleophoridae)], and cherry bark tortrix [*Enarmonia formosana* (Tortricidae)].

Searches for biological control agents for two newer invasive pests in the PNW—spotted-wing drosophila [SWD, *Drosophila suzukii* (Drosophilidae)] and brown marmorated stink bug [BMSB, *Hyalomorpha halys* (Pentatomidae)]—were initiated in 2011. Several species of parasitoids, predators and entomopathogens have been evaluated for their use as biological control agents for SWD, including parasitic wasp species that were imported from Asia for quarantine, testing, and potential release (Wang et al. 2020). In 2022, *Ganaspis brasiliensis* (Figitidae) was approved for release across the United States, and an advantageous population of a second parasitic wasp species *Leptopilina japonica* (Figitdae) was discovered in Oregon. The USDA Agricultural Research Service, OSU, and ODA are releasing the tiny parasitoids across Oregon at or adjacent to berry crops impacted by SWD. The samurai wasp, *Trissolcus japonicus* (Scelionidae), is an egg parasitoid of BMSB that was found established outdoors in Vancouver, Washington in 2015 and in Portland, Oregon in 2016. It has also been found in at least 10 states in eastern U.S. and in B.C., Canada. It was reported to result in up to 77% parasitism of BMSB egg masses in Washington (Milnes and Beers 2019). The samurai wasp has become increasingly widespread in Oregon due to ODA's distribution efforts and was detected in Utah and Idaho in 2019 and 2021, respectively. Orchards may benefit from samurai wasp releases in unsprayed areas adjacent to agriculture and in urban sites (Lowenstein et al. 2019). There is good documentation of traits associated with successful introductions of biocontrol agents with regard to life history traits and other attributes, and applications of these "lessons learned" may improve success rates of this strategy in the future (Kimberling 2004, Abram and Moffat 2018, Seehausen et al. 2021). Biological control of emerald ash borer [*Agrilus planipennis (Buprestidae*)] is being implemented as a part of an areawide management strategy to slow the spread of this invasive beetle within Oregon, where it was first detected in 2022.

2. Augmentative biological control

Augmentative or supplemental biological control typically involves the mass-production and repeated release of natural enemies to improve their population sizes, rate of colonization, and effectiveness. This approach is used most often to target slow-moving pests such as mites and aphids, usually in organic agriculture where few disruptive chemicals are applied, including home gardens and enclosed spaces such as greenhouses. The two main types of augmentative releases include 1) inundative, whereby large numbers of a natural enemy, not necessarily native or able to survive the winter, are released with the goal of single-season control (short-term biocontrol); and 2) inoculative, whereby a native or climate-adapted species is released for anticipated control after allowing populations to build up over time (long-term biocontrol). For example, both types of releases have been used to control two-spotted spider mite (*Tetranychus urticae*) in Oregon, which can become a secondary pest of strawberries following pesticide applications for root weevils, *Otiorhynchus ovatus* (Curculionidae). Mites can be controlled with an early fall inoculative release of the PNW-native predatory mite *Neoseiulus fallacis (*Phytoseiidae), which is available from commercial insectaries and can overwinter in the PNW (Croft and Coop 1998). Another commercially available predatory mite*, N. californicus*, is less tolerant of PNW winters but is still capable of providing single-season control when released by inundation (Pratt and Croft 2000).

Since natural enemies are all specialized to some degree, it's important to correctly identify the pest and which agent(s) are appropriate for the given situation. Table 1 lists some target pests commonly found in home garden and agricultural systems, and their associated commercially-available beneficial organisms. Protocols for acquiring and releasing biocontrol agents should be carefully designed and followed to improve chances of success. Release guidelines depend on knowledge of the biology of the pest and its natural enemy, and the host plant's influence on both species. Additionally, decisions on where and when to release the agent should consider the species' dispersal capabilities. For example, many homeowners have wasted money using ladybug adults to control aphids only to see them fly away within minutes, particularly if agents are released during the heat of the day. Conservation efforts (below) can in some cases enhance the outcome of augmented biocontrol agents.

3. Conservation biological control

Conservation biological control refers to the manipulation and/or protection of habitat and resources to support and encourage natural enemies in order to increase their numbers and effectiveness (reviewed in Begg et al. 2017). This includes encouragement of the natural enemies' needs such as nectar and pollen, alternative hosts, and certain types of non-disrupted habitat. Each of these resources may improve pest control because they can potentially enhance the fecundity, longevity, and survival of natural enemies.

Some practices for conservation biological control include:

- Identification skills. Learn about the beneficial insects and other organisms that frequent your crops and gardens and the biological control services they provide. A few resources to get you started include:

    - Natural enemies gallery (University of California IPM): http://ipm.ucanr.edu/natural-enemies/
    - A pocket guide to natural enemies of crop and garden pests in the Pacific Northwest (OSU; revised March 2021): https://catalog.extension.oregonstate.edu/ec1613
    - Natural enemies and beneficial insects in WA tree fruits [Washington State University (WSU)]: https://treefruit.wsu.edu/crop-protection/opm/beneficials/
    - Orchard natural enemies identification guides (WSU): http://enhancedbc.tfrec.wsu.edu/ID_guides.html
- Avoid chemicals that are toxic to your beneficial insects. Careful use of pesticides and tillage will help to avoid disrupting populations of natural enemies, which can keep secondary pests from reaching economically damaging levels. Using less toxic and more selective controls instead of broad-spectrum compounds (such as most organophosphates, carbamates, and pyrethroids) can help prevent secondary pest outbreaks. Online databases and lists of pesticide effects on beneficial organisms include:
    - http://enhancedbc.tfrec.wsu.edu/opened/
    - https://agsci.oregonstate.edu/oipmc/pesticide-risk-reduction-low-risk-pesticide-list
    - http://ipm.ucanr.edu/PMG/r302900111.html
    - http://www.intermountainfruit.org/pesticide-tables/toxicity-pollinators
    - Provide food and shelter. Non-crop plantings in or around the crop field may provide shelter, alternative prey, nectar, and pollen for beneficial species. Table 2 provides some examples of flowering plants that are visited by natural enemies. Also consider:
- Applying food sprays. These can include yeast and sugar sprays that attract parasitoid wasps, lady beetles, lacewings, and hoverflies.
    - Manipulating crop and non-crop architecture. Consider changing your farm design in ways that can improve natural enemy activities. For example, wind-break plantings may be used as a barrier to prevent dry, dusty conditions favorable to pest mite flare-ups. Predatory mites that attack these pests may also be inhibited by such conditions. Shelter and alternate hosts can also be supported through methods such as careful rotation, alternate row harvest, and "beetle banks," which are graded low banks or berms of dense grasses that are placed within a field or in fence row corridors inhabited by appropriate vegetation.
- Providing insectary plants. Insectary plants are grown to attract, feed, and shelter beneficial insects including pollinators and pest natural enemies. They can provide habitat, alternate prey, and floral resources (e.g., pollen, nectar, nectaries), and may include:
    - Planting within the crop field in strips or smaller blocks
    - Using perennial and annual border plantings
    - Planting within hedgerows
    - Establishing cover crops
    - Carefully managing flowering weeds

The above practices make use of beneficial species already present in the landscape, and they can enhance natural enemies released in classical and augmentative biological control programs (Colley 1998). We refer readers to several sources for additional information on practices for conservation biological control (Bugg and Waddington 1994, Long et al. 1998, Bugg 1999, Hogg et al. 2011, Parker et al. 2013, Altieri and Nicholls 2015, and Begg et al. 2017). As with selecting any new crop management method, choosing insectary plantings for conservation biological control should consider numerous biological, agronomic, and economic factors including those listed above. To justify the continued use of an insectary planting, an on-site assessment should consider the same factors as the preliminary selection process and include a sampling of pests and beneficials within and surrounding the crop.

Several studies have measured positive effects of the above practices on biocontrol performance, although efficacy will be case-specific and difficult to quantify due to the complex interactions involved (Wyckhuys et al. 2013, Begg et al. 2017). Two primary factors responsible for the disruption of conservation biological control include spatio-temporal asynchrony in pest and enemy activity and species interactions that result in weakened control of pests (Begg et al. 2017). Limitations to the implementation of effective conservation biological control can be offset to an extent by ensuring that these efforts are part of a comprehensive IPM approach.

Considerations for incorporating insectary plantings to sustain natural enemies

Timing of flowering

1. Will the floral resources be present when needed?
2. Will the flowers attract natural enemies to the target pest at certain times? Or will they draw them away from the pest?

Characteristics of the natural enemies

1. What are the relative preferences that key natural enemy and pest species have for the different flowers?
2. What are the different requirements for nectar, pollen, shelter, and alternate host food among these key species?
3. What are the foraging ranges and dispersal abilities of these key species?

Agronomic considerations

1. How competitive are the plantings with the crop or other weeds?
2. Do the plantings have the potential to harbor weeds or be weeds themselves?
3. Can the plantings serve as an alternate host for crop disease?
4. Are the plants toxic to any livestock or other local animals?

Economic and management considerations

1. Can the planting be harvested as an additional crop?
2. What are the costs of seed, establishment, and maintenance?
3. How do these costs compare to other management options?
4. Are the plantings compatible with the main pest management plan?

Resources for implementation of biological control

The IPM Practitioner's 2015 Directory of Least Toxic Pest Control Products. A comprehensive listing of biological control agents and other "least toxic" pest control products for a variety of agricultural, urban, and domestic uses, and their producers and distributors. Bio-Integral Resource Center—https://www.birc.org/Directory.htm

"Co-managing fresh produce for nature conservation and food safety," An informative 12-minute video on habitat and biological control made in 2015 by Eric Brennan—https://www.youtube.com/watch?v=zLvJLHERYJI

Natural Enemies Handbook: The Illustrated Guide to Biological Pest Control, by M.L. Flint, M. L, S. H. Driestadt, and J.K. Clark. 1998, 2015. University of California Division of Agriculture and Natural Resources. University of California Press, Oakland, California, USA. Publication 3386. 154 pages. Kindle and ebook editions available.

Sandhu, H. S. Wratten, R. Costanza, J. Pretty, J. R. Porter, and J. Reganold. 2015. Significance and value of non-traded ecosystem services on farmland. PeerJ 3:e762; DOI 10.7717/peerj.762—https://peerj.com/articles/762.pdf

Oregon Department of Agriculture provides a list of invertebrates approved for importation into Oregon. Except as otherwise provided in rules of the ODA, invertebrate species listed in this list may be imported, possessed, sold, purchased, exchanged or transported within the state without an ODA permit. A permit for the importation, possession, or intrastate transportation of ODA-approved species may be required by the USDA-APHIS Plant Protection and Quarantine program (Form 526)—
https://www.oregon.gov/ODA/shared/Documents/Publications/IPPM/OregonApprovedInvertebrateList.pdf

USDA SARE (Sustainable Agriculture Research and Education). SARE supports grant programs, strategies and resources that include protection of biocontrol agents and other beneficial insects—https://www.sare.org

- They produced, as the Sustainable Agricultural Network, a 128-page book, "Manage insects on your farm – a guide to ecological strategies" (Altieri and Nicholls 2005), available as a PDF download from: https://www.sare.org/publications/insect/insect.pdf
- A 116-page book, "Biological control of insects and mites," developed largely for the midwestern US but is of interest in the PNW, is available in print and online from: https://www.northcentralsare.org/Educational-Resources/SARE-Project-Products/Biological-Control-of-Insects-and-Mites-An-Introduction-to-Beneficial-Natural-Enemies-and-Their-Use-in-Pest-Management
- A 108-page book, "Greenhouse IPM with an emphasis on biocontrols," is available online from: https://www.sare.org/Learning-Center/SARE-Project-Products/Northeast-SARE-Project-Products/Greenhouse-IPM-with-an-Emphasis-on-Biocontrols

The Xerces Society. A nonprofit organization formed in 1971 which protects wildlife through the conservation of invertebrates and their habitat. Their focus has expanded beyond native pollinators to include other invertebrate species such as native predators and parasitoids. They have programs to document the impacts of pesticides on invertebrates including biocontrol agents. Xerxes has resources to provide education and training on conservation biological control (e.g., Lee-Mäder et al. 2014) and are very active in the Pacific Northwest. 628 NE Broadway Ste 200, Portland OR 97232 USA; tel: 855-232-6639—https://www.xerces.org

References

Abram, P. K., and C. E. Moffat. 2018. Rethinking biological control programs as planned invasions. Current Opinion in Insect Science 27:9–15. https://doi.org/10.1016/j.cois.2018.01.011

AliNiazee, M.T. 2009. Biocontrol of pests a "secret" success story. News posting by Oregon State University. https://today.oregonstate.edu/archives/1997/feb/biocontrol-pests-secret-success-story

Altieri, M. A., and C. I. Nicholls. 2005. Manage Insects on Your Farm: A Guide to Ecological Strategies. Sustainable Agriculture Network handbook series book 7. https://www.sare.org/wp-content/uploads/Manage-Insects-on-Your-Farm.pdf

Azizoglu, U., and S. Karabörklü. 2021. Role of recombinant DNA technology to improve the efficacy of microbial insecticides. Pages 159–182 in Khan, M.A., Ahmad, W. (eds) Microbes for Sustainable Insect Pest Management. Sustainability in Plant and Crop Protection, vol 17. Springer, Cham. https://doi.org/10.1007/978-3-030-67231-7_8

Begg, G. S., S. M. Cook, R. Dye, M. Ferrante, P. Franck, C. Lavigne, G. L. Lövei, A. Mansion-Vaquie, J. K. Pell, S. Petit, N. Quesada, B. Ricci, S. D. Wratten, and A. N. E. Birch. 2017. A functional overview of conservation biological control. Crop Protection 97:145–158. http://dx.doi.org/10.1016/j.cropro.2016.11.008

Bugg, R. L., and C. Waddington. 1994. Using cover crops to manage arthropod pests of orchards: A review. Agriculture, Ecosystems Environment 50:11–28. https://doi.org/10.1016/0167-8809(94)90121-X

Bugg, R. L. 1999. Beneficial insects and their associations with trees, shrubs, cover crops, and weeds. Pages 63–65 in Bring Farm Edges Back to Life! Yolo Country Resource Conservation District, Woodland, California, USA. 105 p.

Colley, M. R. 1998. Enhancement of biological control with beneficial insectary plantings. Master's Thesis. Oregon State University, Corvallis, Oregon, USA. https://ir.library.oregonstate.edu/concern/graduate_thesis_or_dissertations/x633f3882

Croft, B. A., and L. B. Coop. 1998. Heat units, release rate, prey density, and plant age effects on dispersal by *Neoseiulus fallacis* (Acari: Phytoseiidae) after inoculation into strawberry. Journal of Economic Entomology 91:94–100. http://jee.oxfordjournals.org/content/91/1/94

Hogg, B. N., R. L. Bugg, and K. M. Daane. 2011. Attractiveness of common insectary and harvestable floral resources to beneficial insects. Biological Control 56:76-84. https://doi.org/10.1016/j.biocontrol.2010.09.007

Kimberling, D. N. 2004. Lessons from history: predicting successes and risks of intentional introductions for arthropod biological control. Biological Invasions 6:301–318. https://doi.org/10.1023/B:BINV.0000034599.09281.58

Lee-Mäder, E., J. Hopwood, M. Vaughan, S. H. Black, and L. Morandin. 2014. Farming with Native Beneficial Insects: Ecological Pest Control Solutions. Storey Publishing, North Adams, Massachusetts, USA.

van Lenteren, J. C., J. Bale, F. Bigler, H. M. T. Hokkanen, and A. J. M. Loomans. 2006. Assessing risks of releasing exotic biological control agents of arthropod pests. Annual Review of Entomology. 51:609–634. https://doi.org/10.1146/annurev.ento.51.110104.151129

Long, R. F., A. Corbett, L. Lamb, C. R. Horton, J. Chandler, and M. Stimmann. 1998. Beneficial insects move from flowering plants to nearby crops. California Agriculture 52:23–26. http://calag.ucanr.edu/Archive/?article=ca.v052n05p23

Lowenstein, D. M., H. Andrews, A. Mugica, and N. G. Wyman. 2019. Sensitivity of the egg parasitoid *Trissolcus japonicus* (Hymenoptera: Scelionidae) to field and laboratory-applied insecticide residue. Journal of Economic Entomology 112:2077–2084. https://doi.org/10.1093/jee/toz127

Milnes J., and E. Beers. 2019. *Trissolcus japonicus* (Hymenoptera: Scelionidae) causes low levels of parasitism in three North American pentatomids under field conditions. Journal of Insect Science 19:15. https://doi.org/10.1093/jisesa/iez074

Parker, J. E., W. E. Snyder, G. C. Hamilton, and C. Rodriguez-Saona. 2013. Companion planting and insect pest control. In S. Soloneski and M. Larramendy, editors. Weed and Pest Control - Conventional and New Challenges. INTECH Open Access Publisher, Copenhagen, Denmark. https://doi.org/10.5772/55044

Pratt P. D., and Croft B. A. 2000. Screening of predatory mites as potential control agents of pest mites in landscape plant nurseries of the Pacific Northwest. Journal of Environmental Horticulture 18:218–223. https://doi.org/10.24266/0738-2898-18.4.218

Sarwar, M. 2015. Microbial insecticides – an ecofriendly effective line of attack for insect pests management. International Journal of Engineering and Advanced Research Technology 1:4‒9.

Seehausen, M. L., C. Afonso, H. Jactel, and M. Kenis. 2021. Classical biological control against insect pests in Europe, North Africa, and the Middle East: What influences its success? NeoBiota 65:169–191.

Wang, X., J. C. Lee, K. M. Daane, M. L. Buffington, and K. A. Hoelmer. 2020. Biological control of *Drosophila suzukii*. CAB Reviews 15. https://doi.org/10.1079/PAVSNNR202015054

Wyckhuys, K. A. G., Y. Lu, H. Morales, L. L. Vazquez, J. C. Legaspi, P. A. Eliopoulos, and L. M. Hernandez. 2013. Current status and potential of conservation biological control for agriculture in the developing world. Biological Control 65:152–167. https://doi.org/10.1016/j.biocontrol.2012.11.010

Table 1. Target pests and beneficial organisms often used for augmentative biological control releases

Aphid (See also soft-bodied arthropods)	predatory midge	*Aphidoletes aphidimyza*
	parasitoid wasp	*Aphidius ervi, A. matricariae, A. colemani, Lysiphlebus testaceipes, Trioxys pallidus*
	big-eyed bug	*Geocoris pallens*
	lady beetle (“ladybug”)	*Hippodamia convergens*
	lacewing	*Chrysoperla downesi, C. plorabunda, C. rufilabris*
	minute pirate bug	*Orius insidiosus, O. minutus, O. tristicolor*
Armyworm (See also Butterfly and moth)	braconid parasitoid wasp	*Chelonus insularis*
Black fly larvae	bacterial endotoxin (*Bti*)	*Bacillus thuringiensis* var. *israelensis (*e.g., *Bactimos, Teknar, Vectobac)*
Butterfly and moth larvae and eggs of beetle pests in stored grain products, such as almond moth, Indian meal moth, grain weevil	parasitoid wasp	*Bracon hebeter*
Butterfly and moth eggs and young larvae: beet	viral pathogen	Nuclear polyhedrosis virus *(NPV*)

Table 1. Target pests and beneficial organisms often used for augmentative biological control releases

armyworm, cabbage looper, corn earworm, cutworm, diamondback moth, imported cabbageworm, codling moth and other orchard moths, tobacco budworm	bacterial endotoxin (*Btk, Bta*)	*Bacillus thuringiensis* var. *kurstaki* (e.g., Dipel, Javelin, Attack, Thuricide, Bactospeine, Safer's Caterpillar Killer), *Bacillus thuringiensis* var. *aizawai (*e.g., Certan)
	parasitoid wasps of eggs	*Trichogramma minutum, T. pretiosum, T. platneri*
Codling moth larvae	granulosis virus pathogen	*Baculovirus carpocapsae*
Flea	parasitic nematode	*Steinernema carpocapsae, S. feltiae*
Fly (garbage- and manure-breeding)	parasitoids of puparia	*Melittobia digitata, Muscidifurax raptor, Muscidifurax zaraptor, Nasonia vitripennis, Pachcrepoideus vindemiae, Spalangia cameroni, S. endius*
	histerid beetle predator	*Carcinops pumilio*
Fungus gnat (larvae)	predatory mite	*Hypoaspis miles, H. aculeifer*
	parasitic nematode	*Heterorhabditis megidis, Steinernema carpocapsae, S. feltiae*
	bacterial endotoxin (*Bti*)	*Bacillus thuringiensis* var. *israelensis*
Grasshopper (nymphs and adults)	protozoan	*Nosema locustae*
Larvae and grubs that pupate in the soil: cucumber beetles, dampwood termites, flea beetles, root weevils, wireworms	parasitic nematodes of larvae	*Heterorhabditis bacteriophora, H. heliothidis, H. megidis, Steinernema feltia, S. carpocapsae, S. riobravis*
Grubs in soil such as Japanese beetle, June beetle, and white grubs	bacterial endotoxin (*Btg*)	*Bacillus thuringiensis var. galleriae*
Leafminer	braconid parasitoid of larvae	*Dacnusa sibirica*
Mealybug	lady beetle ("mealybug destroyer")	*Cryptolaemus montrouzieri*
Mite: twospotted spider (*Tetranychus urticae*)	predatory mite	*Amblyseius hibisci, A. mckenziei, Galendromus occidentalis, Mesoseiulus longipes, Neoseiulus californicus, N. fallacis, Phytoseiulus persimilis, P. macropilis*
	predatory six-spotted thrips	*Scolothrips sexmaculatus*
	minute pirate bug	*Orius minutus, O. tristicolor*
	big-eyed bug	*Geocoris pallens*
Mosquito larvae	predatory fish	*Gambusia affinis* spp. (only in manmade water bodies or containers that have no

Table 1. Target pests and beneficial organisms often used for augmentative biological control releases

		connection to natural waterways)
	bacterial endotoxin (*Bti*)	*Bacillus thuringiensis* var. *israelensis* (e.g., Dunks, Bactimos, Vectobac, Teknar)
Scale: armored scale, oleander scale, San Jose scale, ivy scale	lady beetle	*Chilocorus fraternus*
Soft scale: citrus black scale, black/brown hemispherical, nigra scale (See also soft-bodied arthropods)	parasitoid wasp	*Metaphycus helvolus*
Soft-bodied arthropods: thrips, scale, aphid, spider mite, whitefly, eggs of harmful pests	lacewing larvae (in larval stage)	*Chrysoperla downesi*, *C. plorabunda*, *C. rufilabris*
	fungal pathogen	*Beauveria bassiana*
	lady beetle	*Chilocorus fraternus*, H*ippodamia convergens*
	pirate bug	*Orius minutis*, O. *tristicolor*
	predatory thrips	*Scolothrips sexmaculatus*
Thrips larvae (See also soft-bodied arthropods)	predatory mite	*Amblyseius cucumeris*, *A. mckenziei, A. barkeri, A. degenerens*
	lacewing	*Chrysoperla downesi, C. plorabunda*, *C. rufilabris*
	minute pirate bug	*Orius minutus*, *O. tristicolor*
Wax moth larvae (in honeycombs)	bacterial endotoxin (Bta)	*Bacillus thuringiensis* var. *aizawai* (e.g. Certan)
Weevil in landscape plants	parasitoid wasps of larvae	*Anisopteromalus calandrae*
	parasitic nematode	*Heterorhabditis heliothidis*, *H. medidis*, *Steinernema carpocapsae*, *S. feltiae*, *S. riobravis*
Whitefly nymph (see also soft-bodied arthropods)	parasitoid wasps of eggs	*Encarsia formosa*, *Eretmocerus californicus*

1 Lady beetles include many species in the family Coccinellidae, order Coleoptera.

2 Lacewings include many species in the families Chrysopidae and Hemerobiidae, order Neuroptera.

3 Parasitoid and predatory wasps include a large number of species in families such as Aphelinidae, Aphidiidae, Braconidae, Chalcididae, Crabronidae, Encyrtidae, Eulophidae, Ichneumonidae, Mymaridae, Pompilidae, Pteromalidae, Scelionidae, Specidae, and Trichogrammatidae, order Hymenoptera.

4 Hoverflies include many species in the family Syrphidae, order Diptera.

5 Predatory bugs include many species in families such as Anthocoridae, Lygaeidae, Nabidae, Pentatomidae, and Reduviidae, order Heteroptera.

6 Minute pirate bugs include many species in the family Anthocoridae, order Heteroptera.

7 Big-eyed bugs include many species in the family Lygaeidae, order Heteroptera.

8 Parasitoid Tachinid flies include many species in the family Tachinidae, order Diptera.

Table 1. Target pests and beneficial organisms often used for augmentative biological control releases
9 Bees include many species in families such as Anthophoridae, Apidae, Halictidae, Andrenidae, Colletidae, and Megachilidae, order Hymenoptera.

Table 2. Flowering plants visited by beneficial insects that can aid biological control conservation efforts

Common name (botanical name)	Beneficial insects
Apiaceae (Carrot family)	
Angelica (*Angelica*)	lady beetle (“ladybugs”), lacewing
Anise (*Pimpinella anisum*)	parasitoid wasp
Blue lace (*Trachymene caerulea*)	parasitoid wasp
Caraway (*Carum caryi*)	hoverfly, minute pirate bug and big-eyed bug, lacewing, parasitoid wasp
Chervil (*Anthriscus cerefolium*)	parasitoid wasp
Coriander (*Coriandrum sativum*)	hoverfly, parasitoid wasp, parasitoid tachinid fly
Dill (*Anethum graveolens*)	hoverfly, lady beetle, parasitoid wasp
Fennel (*Foeniculum vulgare*)	hoverfly, parasitoid wasp, parasitoid tachinid fly
Lovage (*Lovisticum officinale*)	parasitoid wasp
White lace flower (*Ammi majus*)	hoverfly, predatory bug, lady beetle, parasitoid wasp, parasitoid tachinid fly
Wild carrot (*Daucus carota*)	hoverfly, predatory bug, lady beetle, lacewing, parasitoid wasp
Asteraceae (Daisy family)	
Blazing star, gayfeather (*Liatrus* spp.)	minute pirate bug, big-eyed bug, parasitoid wasp
Chamomile (*Anthemis nobilis*)	lady beetle
Cosmos (*Cosmos bipinnatus*)	hoverfly, lacewing, minute pirate bug
Golden marguerite (*Anthemis tinctoria*)	lady beetle, parasitoid wasp, parasitoid tachinid fly
Goldenrod (*Solidago altissima*)	soldier beetle, predatory bug, lady beetle, parasitoid wasp
Marigolds, signet (*Tagetes tenuifolia*)	minute pirate bug, parasitoid wasp

Mexican sunflower (*Tithonia tagetifolia*)	hoverfly, minute pirate bug
Sunflower (*Helianthus annuus* and *H. debilis*)	hoverfly, lady beetle, parasitoid wasp
Tansy (*Tanecetum*)	hoverfly, lady beetle larvae, parasitoid wasp
Yarrow, milfoil (*Achillea millefolium*)	hoverfly, parasitoid wasp
Yarrows (*A. macrophylla*, *A. taygetea*, etc.)	hoverfly, parasitoid wasp
Brassicaceae (Cabbage family)	
Broccoli (*Brassica oleracea*)	hoverfly, parasitoid wasp
Sweet alyssum (*Lobularia maritima*)	hoverfly, parasitoid wasp, parasitoid tachinid fly
Globe candytuft (*Iberis umbellata*)	hoverfly
Mustards (*Brassica hirta* and *B. juncea*)	hoverfly, minute pirate bug, big-eyed bug
Dipsaceae (Scabiosa family)	
Cephalaria (*Cephalaria giganitica*)	hoverfly, parasitoid wasp
Dipsacus (*Dipsacus* spp.)	hoverfly
Pincushion flower (*Scabiosa caucasica*)	hoverfly, parasitoid wasp
Scabiosa (*Scabiosa atropurpurea*)	hoverfly
Fabaceae (Legume family)	
Alfalfa (*Medicago sativa*)	bee, predatory bug, lacewing, lady beetle, parasitoid wasp
Clover (*Trifolium* spp.)	bee, predatory bug, lacewing, lady beetle
Vetch (*Vicia* spp.)	bee, predatory bug, lacewing, lady beetle
Hydrophyllaceae (Waterleaf family)	
Fiddleneck/Phacelia (*Phacelia tanacetifolia*)	bee, predatory bug, hoverfly
Lamiaceae (Mint family)	
Germander (*Teucrium* spp.)	bee, parasitoid wasp
Polygonaceae (Buckwheat family)	
Buckwheat (*Eriogonum* spp. and *Fagopyrum* spp.)	hoverfly
See notes for Table 1.	

Entomopathogenic nematodes

Jana Lee and Amy J. Dreves

Latest revision—March 2024

Insect-pathogenic, or entomopathogenic nematodes, are a group of soil-dwelling roundworms which kill insects that live in, on, or near the soil surface, usually closely associated with plants. These nematodes can occur naturally in soil and are found in most places where plants grow. Research has demonstrated that entomopathogenic nematodes can be mass produced, have a narrow host specificity against pests, and are safe to plants and vertebrates; and, therefore, the U.S. Environmental Protection Agency has exempted them from all registration requirements and related regulation. Entomopathogenic nematodes have been available commercially to agriculturists and used in a variety of cropping systems.

There are two main groups of entomopathogenic nematodes: the steinernematids and the heterorhabditids. Both have similar life cycles, and only the free-living, infective juvenile stage is able to infect the target pest insect. It is the juvenile stage that is found in or on the soil, searching out a host to infect. In fact, the juvenile form is the only form found outside of the host.

Slug-pathogenic, or malacopathogenic, nematodes, are also a group of soil-dwelling roundworms, a novel method that has been used in Europe to kill slugs with some success. One of the most widely established, commercially-available slug biocontrol agents in Europe is the nematode *Phasmarhabditis hermaphrodita* (Schneider) sold as Nemaslug, and mixed with *P. californica* sold as Nemaslug 2.0. This nematode is associated symbiotically with a bacterium that uses an endotoxin that kills slugs. The nematode locates slugs in the soil and enters the slug's mantle cavity. After the slug dies, the nematodes multiply over the decaying slug body and then migrate back into the soil where, if conditions are favorable, they infect more slugs. *Phasmarhabditis hermaphrodita* has also been found in CA and OR, as well as other *Phasmarhabditis* species which have shown promise for control of the grey garden slug. These nematodes must be demonstrated as not harmful to native species such as the banana slug before they can be commercialized. They are currently unavailable for purchase.

This document focuses on insect-pathogenic nematodes.

Nematode selection for Insects

The selected nematode (*Steinernema* spp. or *Heterorhabditis* spp.) depends on the target insect pest. In general, nematodes in the genus *Steinernema* are "sit-and-wait predators" or ambushers and are used against insects whose immature stages (larvae or pupae) spend most of their time at or near the soil surface. Other species are highly mobile and roam through the soil searching for hosts. The host-finding strategy of most *Steinernema* is to wait until the prey bumps into the nematode, and then infects it. In contrast, nematodes in the genus *Heterorhabditis* actively seek out or hunt for their prey, sometimes several inches below the soil surface, and stay in one spot for an extended period of time. Thus, nematodes in the genus *Steinernema* (*S. feltiae*) are the best choice against fungus gnat larvae, often found on the soil surface of potted plants, while the genus *Heterorhabditis,* (*H. megidis, H. marelatus*, or *H. bacteriophora*) are the best choices against the black vine weevil, deeper in the soil. Recently, *H. bacteriophora*, *H. indica*, *Oscheius oniric*, *Steinernema carpocapsae*, *S. feltiae*, and *S. kraussei* appeared promising for targeting spotted-wing drosophila. The nematodes can penetrate the larval and teneral adult stages of spotted-wing drosophila. Given the high dose needed and cost of nematode application, researchers are examining whether nematodes can be effectively delivered by drip irrigation to target flies that pupate in the soil. The fact that infected adult flies can disperse and spread the nematodes to other flies may make this a viable option.

There are over ten entomopathogenic nematodes commercially-produced as a biological insecticide for over 25 insect pests. There is some overlap between the various species with regards to host-finding ability. Consult a nematode manufacturer/supplier for selection of the proper entomopathogenic nematode product.

Life cycle

An infective juvenile may at first move randomly, and then find their insect host via carbon dioxide, host odor or damaged plant odors. Once a juvenile locates an insect, the juvenile enters via a natural opening; or it may penetrate a weak spot in the insect's cuticle. Insect larvae and pupae are more susceptible to nematodes since adult insects are often more mobile. Once inside the host's blood system, the juvenile releases a symbiotic bacterium that it carries. The bacteria are released into the blood of the host, rapidly multiply, and produce compounds that kill the host insect generally within 48 hours. The bacteria protect and preserve (via antibiotics) the dead insect from invasion by unwanted, contaminating soil microbes and the nematodes provide shelter for the bacteria. The infective nematodes complete one to several generations inside the host, feeding on the bacteria and nutrients within the dying host. Only when all the host tissues have been consumed does a new generation of juveniles emerge, all carrying the symbiotic bacteria with them in search of new hosts (see Figure 1).

One generation from egg to egg typically takes from 4 to 7 days. In most instances, there are at least two generations inside a host before the new juveniles emerge seeking a new host, so from the time of first infection by juveniles to the time "new" juveniles emerge may be from 8 to 14 days. The length of time is determined by the temperature of the soil, the size of the host, and which nematode is involved. A large host such as a cutworm will support several generations before conditions become too "crowded" and juveniles emerge, compared to a strawberry root weevil larva, where there may be only one or two generations before juvenile emergence. Similarly, a large nematode such as *S. carpocapsae* has fewer generations than *S. feltiae when infecting similar-size hosts.*

Application methods

Though the adult stage of some insect pests also is susceptible, entomopathogenic nematodes generally are used for controlling the soil-borne larval or pupal stages of a pest. Therefore, entomopathogenic nematodes most often are applied by drench or band application. While broadcast application has been used at times, the immature pest insect usually is not located between the crop rows as there is usually no food source there. If, however, the crop has a closed canopy like cranberries or mint, a broadcast application may be warranted. An adjuvant may help. Select your application method wisely, as it may impact greatly the success of host location, infection, and control by the entomopathogenic nematodes.

Entomopathogenic nematodes come in a variety of formulations: water-dispersible granules, nematodes on gel, micronized vermiculite, nematode wool, and an aqueous suspension of nematodes. These formulations are intended to be mixed with water to release the nematodes through common application equipment such as small pressurized sprayers, mist blowers, electrostatic sprayers, or even helicopters (aerial application). Some more promising methods for applying entomopathogenic nematodes are emerging. One uses irrigation systems in a manner similar to chemigation. Another uses nematode-filled capsules which include attractants or feeding stimulants for the pest; this draws in the pest for infection rather than relying on the nematode to find the pest.

Regardless of the method, nematodes can withstand application pressures of approximately 300 psi and can pass through most spray nozzles without difficulty, though operating pressures between 20 to 60 psi generally are sufficient. Keep in mind that nozzle orifices should not be smaller than 50 microns (0.00019685 inch), and that any screens in the system should have an opening of at least 50 mesh (0.0117 inch) or larger to allow the free passage of nematodes through the system. In any case, follow the manufacturer's directions.

Nematodes require a film of water around soil particles to move through the soil profile in search of a host. Therefore, pre-irrigate the soil in the treatment area with about 0.25 to 0.5 inch of water no later than a few hours before application of the nematodes. Following the application, "water in" the nematodes with an additional 0.5 inch of water to wash them off of foliage and protect them from damaging UV radiation. Further irrigation to maintain adequate soil moisture for at least 7 days following nematode application also is recommended. Be careful not to over-irrigate, because excess water inhibits the movement of oxygen in the soil, and the nematodes will drown. A good rule of thumb is to avoid standing water in your fields.

Key points for success in using entomophathogenic nematodes

1) When applying agrichemicals in the area where entomopathogenic nematodes are to used, be sure that there is enough separation time between applications of toxic compounds and entomopathogenic nematodes (Table 1). Some chemicals have been found to affect nematode efficacy when nematodes are exposed to them. These should be applied with care when used in conjunction with nematodes.
2) Entomopathogenic nematodes require a moist, not saturated, soil environment so they can move around and locate their host.
3) Soil temperature where nematodes are to be applied should be above 55°F and less than 90°F. Nematodes are also affected by suboptimal soil type, thatch depth, and irrigation frequency.
4) Protect nematodes from excessive exposure to ultra violet (UV) rays which can inactivate and kill them.
5) Time application of entomopathogenic nematodes to target the susceptible stage of the pest.
6) Select the proper nematode species to match the most susceptible pest stage.
7) Storage of formulated nematode species varies: Steinernematids at 39 to 46°F; Heterohabditids at 50 to 60°F. Do not leave in a hot vehicle.
8) Select the application rate and method to maximize contact between entomopathogenic nematodes and the target pest.
9) In all cases, refer to the manufacturer's label for recommendations.

Note: We appreciate the contributions of past employees of Oregon State University, Peter Guthro and Ralph Berry, to this document.

ENTOMOPATHOGENIC NEMATODE LIFE CYCLE

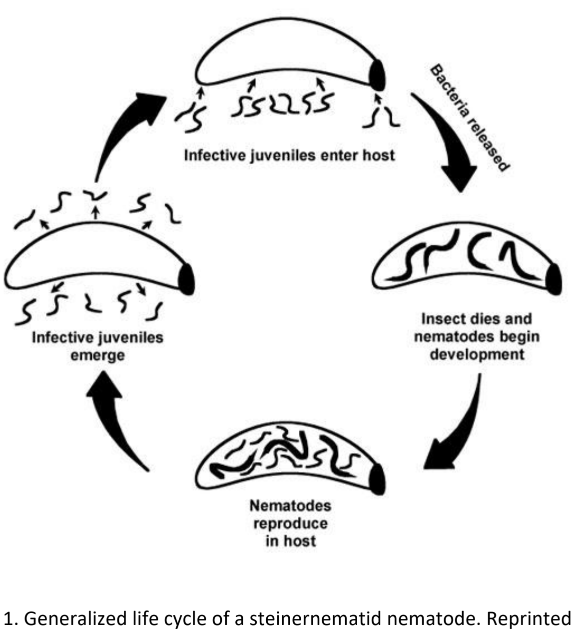

Fig. 1. Generalized life cycle of a steinernematid nematode. Reprinted with permission from Shapiro-Ilan, D.I. and Gaugler, R. (n.d.) Nematodes. In Biological Control: A Guide to Natural Enemies in North America (Anthony Shelton, editor) — http://www.biocontrol.entomology.cornell.edu//pathogens/nematodes.html

References for Table 1

Alumai, A. and P. Grewal 2004. Tank-mix compatibility of the entomopathogenic nematodes, *Heterorhabditis bacteriophora* and *Steinernema carpocapsae*, with selected chemical pesticides used in turfgrass. 2004. Biocontrol Science and Technology, 14(7). DOI: 10.1080/09583150410001724334.

Shetlar, D.J. 1999. "Application Methods in Different Cropping Systems," in Proceedings of Workshop—Optimal Use of Insecticidal Nematodes in Pest Management, Aug. 28–30, 1999. S. Polavarapu, ed.

Smith, K. 1999. "Factors Affecting Efficacy," in Proceedings of Workshop—Optimal Use of Insecticidal Nematodes in Pest Management, Aug. 28–30, 1999. S. Polavarapu, ed.

Additional information on entomopathogenic nematodes and their application can be found in:

Labaude, S., and C.T. Griffin. 2018. Transmission success of entomopathogenic nematodes used in pest control. Insects 9, 72, https://doi.org/10.3390/insects9020072

Mc Donnell, R.J., A.J. Colton, D.K. Howe, and D.R. Denver. 2020. Lethality of four species of *Phasmarhabditis* (Nematoda: Rhabditidae) to the invasive slug, *Deroceras reticulatum* (Gastropoda: Agriolimacidae) in laboratory infectivity trials. Biological Control 150: 104349. https://doi.org/10.1016/j.biocontrol.2020.104349

Miles, C., C. Blethen, R. Gaugler, D. Shapiro-Ilan and T. Murray. 2012. Using entomopathogenic nematodes for crop insect pest control. PNW Extension Publication 544. https://pubs.extension.wsu.edu/using-entomopathogenic-nematodes-for-crop-insect-pest-control

Table 1. Chemical-use patterns with nematodes

May be tank-mixed with nematodes	
Compound	**Trade name**
acephate	Orthene
azadirachtin[2]	Azatin, Neem[2]
Bacillus thuringiensis (Bt)	M-One, Dipel
bifenthrin	Talstar
sodium bromide	Unibrom
carbaryl[2]	Sevin[2]
bifenthrin	Talstar
chlorothalonil	Daconil
dimethyl tetrachloroterephthala	Dacthal
copper hydroxide	Kocide
cyfluthrin	Tempo
malathion	Malathion
diazinon	Diazinon
diflubenzurion	Dimilin
endosulfan	Thiodan
esfenvalerate	Asana
etridiazole	Terrazole

fertilizers	various
fipronil[2]	Chipco Choice[2]
glyphosate	Roundup
insecticidal soap[2]	various
iprodione	Chipco 26019
isofenphos	Oftanol
kinoprene	Enstar
metalaxyl	Subdue
methidathion	Supracide
methomyl[2]	Lannate[2]
methoprene	Apex
oryzalin	Surflan
oxamyl[2]	Vydate[2]
oxazoidinedione	Ornalin
pentachloronitrobenzene (PCNB)	Terrachlor
thiophanate-methyl	Zyban
triademefon	Bayleton

1-wk separation	
Compound	**Trade name**
anilazine	Dyrene
chlorpyrifos	Dursban
dimethyl benzyl ammonium chloride	Physan 20
fenarimol	Rubigan
2,4-D	Various
triclopyr	Turflon, Confront

2-wk separation	
Compound	**Trade name**
ethoprop	Mocap
isazophos	Triumph

Biology and control of the garden symphylan

Alison Willette, Navneet Kaur, Benjamin Diehl and Beverly Gerdeman

Latest revision—March 2024

Introduction

Garden symphylans—*Scutigerella immaculata* (Newport) (GS)—are centipede-like soil arthropods which infest many home gardens and agricultural soils in western Oregon and Washington. Symphylans are omnivores, feeding on germinating seeds, seedlings, roots, plant parts in contact with the soil, and other organic material including decaying plants and fungal hyphae. Poor stand, seedling death, poor growth, stunted plants, reduced vigor, and yield reduction result. Recognizable damage is typically a series of holes chewed in the host plant. Chronic feeding on the roots of both annual and perennial plants reduces a plant's ability to acquire water and nutrients. This results in a poor root system that manifests as general stunting and distortion of plants as well as increased susceptibility to plant pathogens. Sampling and control of GS is complicated by daily and seasonal vertical movement in the soil which is influenced by soil structure, soil moisture, temperature, time of day, season, crop stage, and their feeding cycles.

Conventional growers, organic growers, and small-scale gardeners often approach symphylan management from different perspectives, primarily due to economic and scale-dependent factors. Selection of appropriate tactics to manage GS is largely determined by the cropping system (no-till versus tillage), and use of soil applied insecticides. However, in all systems, effective management results from accurate identification of GS and the damage they cause, a general knowledge of their ecology, as well as appropriate sampling methods and control strategies. Correct diagnosis of a GS problem is sometimes tricky, since damage may be atypical and GS are not always easy to find when damage is observed.

Identification

Symphylans are soil-dwelling myriapods, not insects. They are found worldwide but are poorly described, with only about 160 total species. The class Symphyla contains two families: Scutigerellidae and Scolopendrellidae. Garden symphylans (GS) belong to the family Scutigerellidae, which is characterized by large dorsal tergites that have rounded or slightly lobed posterior margins. Several species occur in Oregon, but the *S. immaculata* is the primary species causing crop damage in the U.S. GS are by far the most common symphyla species found in PNW agricultural systems.

Garden symphylans are white and "centipede-like", measuring about 0.25 inch long when mature. They have 6 to 12 pairs of legs (depending on age), which make them easy to differentiate from common soil insects which only have 3 pairs of legs. Due to their subterranean habitat, symphylans are blind but are able to move quickly though the soil, especially when disturbed. Though their color may vary depending on what they have eaten, they are generally paler and smaller than true centipedes, which are also soil arthropods with many pairs of legs (one pair per body segment). Another similar group of soil arthropods are millipedes, but these myriapods are generally slower moving and possess two pairs of legs on each body segment.

Garden symphylan biology

Symphylans exhibit incomplete metamorphosis. Stages include eggs, immatures, and adults, which can be found together throughout most of the year. Males deposit sperm packets on stalks and on the soil where the female will pick them up and place them in special glands in her mouth. Eggs are found in groups of four to 25 and are pearly white to tan, spherical with hexagonal shaped ridges. Immediately after each egg is laid, she places them in her mouth where they are fertilized before being laid in a cluster, approximately 12 inches deep. Temperature plays a key role in regulating oviposition, and the greatest numbers of eggs are most commonly deposited in the spring and fall. First instars emerge from the egg with only six pairs of legs and fewer body segments than adults. Newly hatched GS may resemble subterranean springtails, but differ in that a pair of legs are attached to most segments, rather than just the thorax, as in the springtails. Symphylans also possess bead-like antennae with numerous segments, compared with the usually 4-segmented antennae of springtails. The GS has an exoskeleton and, like an insect, sheds it (molts) periodically to grow and enlarge body size. Each of the six subsequent molts results in the addition of a body segment with a pair of legs and more antennal segments. Total time from egg to sexually mature adult (seventh instar) is about 2 to 3 months during typical spring soil temperatures in western Oregon. Two complete generations per year can occur.

Occurrence and movement

Garden symphylans are generally a problem in irrigated crops grown on alluvial soils with very good soil structure. Within these soils, GS tend to occur in circular "hotspots" encompassing a few square feet to several acres. Hotspots often remain consistent from year to year with little change in populations and only minor lateral spread.

Within a favorable soil habitat GS can migrate from the soil surface to a depth of over 3 feet. Symphylans are not capable of making their own burrows but instead their movement depends on cracks and crevices in soil and runways created by roots. The soil profile, structure, composition, water table and water holding capacity, determines the depth to which GS migrate. Vertical migration is primarily related to interactions among moisture, temperature, crop stage and endogenous feeding cycles. A general understanding of these interactions is important both for timing and interpreting sampling efforts, and for selecting management tactics.

Garden symphylans tend to aggregate in the top 6 inches of soil when the soil is moist and warm in the spring and fall. They move to deeper soil strata during July and August, though can stay at the surface if sufficient moisture is present and no plants are growing. Garden symphylans migrate to the root zone to feed, then return to the deeper strata to molt, evidenced by the large number of molted skins that may be observed in these strata. Since migration is not entirely synchronized within a population, GS are usually present throughout the habitable portion of the soil profile. Presence of GS in the surface soil may also be influenced by other variables that impede movement, such as tillage and compaction from heavy objects (such as tractor tires).

Sampling

Many of the difficulties in effectively managing GS result from unknowns concerning the density and location of populations in a field. Sampling, although often time-consuming, can provide information critical to managing populations effectively. For annual crops, sampling is commonly conducted in April, May, or June, prior to planting. GS will generally be visible in upper six inches of the soil when air temperatures are above 45°F. In general, the later in the spring that sampling occurs, the more GS will be found in the soil. Samples that include crop or weed roots generally contain more root-feeding GS than those taken in bare soil. The type and extent of sampling may vary depending on the site conditions (e.g., vegetation, size of area, cropping history), and whether populations have been historically problematic in certain areas of a site.

Three main sampling methods are used: baiting methods, soil sampling methods, and indirect sampling methods. Each method has benefits and drawbacks, and the selection of a sampling method will vary depending on the objectives of the sampling (e.g., detection vs. precise population density estimation), time of year, and site conditions.

Part of the difficulty in sampling is a result of the patchy spatial distribution of GS populations. It is important to be aware that an individual sample unit count provides information about a local region within which that sample unit was taken. Counts will often vary from zero to more than 50 GS per sample unit (i.e., soil core or bait). To obtain information about the spatial patterns of the population, sample units are often taken in a grid pattern. Areas with different cropping histories are generally sampled independently.

In most cases, sampling only measures the density of GS in the surface soil. Therefore, sampling should only be conducted when GS are within this region. The best sampling conditions are, generally, when the soil is warm and moist. Sampling within 3 weeks after major tillage, such as disking, plowing, or spading may not reflect the true population because GS often have not had ample time to reestablish in the surface soil.

To detect or identify a GS problem in a crop, bait for GS in suspected areas within 3 weeks of planting. To sample seedlings or established plants, dig them up in the early morning when GS are close to the soil surface. Inspect their roots, along with those of grassy weeds in the area, for evidence of nibbling. Check for presence of neatly chewed round holes in crowns of grasses, as well as the soil around the roots.

Soil sampling is the standard/historic method for estimating how many GS are in a field (i.e., approximate number of GS per unit of soil, or population density estimate). Sample unit sizes vary; the most common soil sample units are 6 x 6 x 12 inches (length, width, depth) or cores of 2.5 inches in diameter by 6- to 12-inch depth. When soil samples are taken, the soil from each sample unit is usually placed on a dark piece of plastic or cloth where the aggregates are broken apart and the GS are counted. Sampling is usually conducted when GS are present in the top 6 to 12 inches of the root zone.

Bait samples are generally much faster to take than soil samples, but they are also more variable and more sensitive to factors such as soil moisture, temperature, and presence of vegetation. To bait a sample, dig into the soil until moist soil is exposed and place one-half of a sliced potato on the soil surface with exposed cut area of potato against the moist soil surface. Shelter with a protective cover (e.g., white pot or 4-inch PVC cap). Baits are generally checked one to three days after placement. Lift the bait, counting the GS on the soil first, then the GS on the potato bait. During warm and/or dry conditions, baits are generally checked one to two days after placement as counts decrease if baits are left to dry out. In cooler conditions, baits are commonly left out for three to five days. Bait sampling works very well for some applications, though it cannot be used during heavy rains, as the bait stations will fill with water. Baiting works best at least two to three weeks after tillage, when the soil has stabilized but before plants are well established. Therefore, both soil sampling and the bait method can be used to determine the presence/absence of GS.

Plant growth can sometimes be a useful, yet indirect measure of GS populations and is often a good starting point for assessing GS populations. Indirect measures, however, should never be used without some direct sampling to confirm the presence of GS.

Determining the number of samples

Sampling requirements will often vary by site, depending on factors such as cropping history and time of year. Sampling involves establishing a balance between the need to be confident about estimates of the number of GS present (implying a large number of samples) and not investing excessive time and energy into the sampling endeavor (implying a small number of samples).

Follow these guidelines for determining the sample size:

1. Sampling for low population densities (e.g., early in the spring or of highly susceptible crops) requires a greater number of sample units (e.g., 100+) than sampling for high population density (e.g., 30 GS/foot) as smaller population clusters are more difficult to detect. Ten samples may be enough to confirm that a high population density exists.
2. As the variability of the sampling method increases, so does the number of sample units required. Since the baiting method is more variable than the soil sampling method, two to three times more bait than soil sample units are required.
3. For estimation of “economic” population densities in moderately susceptible crops, at least 35 soil sample units, or at least 50 bait units, are commonly used. Depending on the size of the field, and the time of year, considerably more sample units are sometimes used.

Action thresholds

Management decisions, such as those regarding pesticide applications and the intensity of tillage, are sometimes made based on pre-plant GS population density estimates. Owing largely to the difficulty in sampling and the numerous crops to which GS are pests, action thresholds for individual crops are not well developed. The relationship between GS population density (estimated by sampling methods) and crop health is often influenced by several factors, including tillage intensity, crop species, planting date, and crop stage.

In the field, noticeable damage has often been observed if populations exceed an average of five to ten GS per cubic foot (or 1 to 2 GS per 6 x 6 x 12-inch sample) in moderately to highly susceptible crops, such as broccoli, squash, spinach, and cabbage. In conventional cropping systems, pesticides are often applied to susceptible crops if populations exceed three GS per cubic foot. In more tolerant crops, such as potato and small grains, GS feeding may not lead to significant damage, even at considerably higher population densities.

Management and control

For management purposes it is important to make a distinction between tactics that may decrease GS population and those that may reduce crop damage but not necessarily reduce pest populations. In most cases, effective GS management involves establishing a balance between these two tactics. It is important to note that in most cases little can be done without replanting after damage is observed. Sampling is, therefore, important in determining the proper course of action.

Tactics for population reduction

No simple, inexpensive, and completely reliable method of controlling GS has been developed. No method will eradicate GS from a site, and the effect of most tactics will not last longer than one to three years. Very little is known about symphylan population dynamics in agroecosystems due to the complexity of their movements up and down in the soil profile. Many control tactics have been successful in some cases but unsuccessful in very similar situations.

Tillage is probably the oldest control tactic used and is still one of the most effective. Tillage can physically crush GS, thus reducing populations. Tillage may also decrease populations of key GS predators such as centipedes and predaceous mites. However, in annual crops, benefits of increased predator populations in reduced tillage systems have not been shown to be as effective as tillage in decreasing GS populations. In general, for most effective control, till when the GS are in the surface soil, and when soil moisture allows preparation of a fine seed bed. Since only a portion of the population is in the surface horizon, tillage never provides complete control; however, surface populations are commonly significantly lower for at least two to three weeks after tillage. Research suggests that symphylans are more often associated with unbroken down organic matter with good soil structure rather than in compact or sandy soils. There is some evidence of reducing populations and injury by packing down the soil surface after planting; and flooding areas for 2 to 3 weeks.

In conjunction with tillage, pesticides are used to manage GS. Plant protection is probably achieved by direct mortality as well as by repelling GS from the root zone. The use of pesticides has been effective to some degree in conventional systems, but many growers still have perennial problems with symphylans. Pesticides are most effective if applied before planting as broadcast and incorporated applications. Banded/incorporated applications may provide acceptable protection for some crops. In some perennial crops, such as hops, post-plant pesticide applications can reduce GS sufficiently to promote plant vigor. Fumigants, organophosphate, and carbamate pesticides have historically been the most effective, but many are no longer registered for GS in many crops. Pyrethroid pesticides generally do not provide as high a level of control. Soil-applied organophosphate insecticides (e.g., Mocap, Lorsban Advanced) have been effective at protecting crops sufficiently from GS, although chemistries containing chlorpyrifos will be banned on food, forage and seed crops after December 31, 2023. Soil fumigation, when properly performed, can reduce symphylan populations enough to allow 3 years or more of crop production with no additional control efforts during that period. Refer to individual crop sections for current registrations.

Insecticide registration is continually changing: always check specific insecticide labels for current registered uses. The following may have registered insecticides for symphylan control: asparagus, snap bean, table beet, blueberry, blackberry, broccoli, Brussels sprout, cabbage, cantaloupe, cauliflower, carrots, celery, chickpea, sweet corn, cucumber, orchard floors, garlic, lettuce, pepper, potato, pumpkin, rhubarb, spinach, sugar beet, hop, mint, strawberry, silage and feed corn, clover, grass seed, wheat, barley, radish seed, sugar beet seed, home garden vegetables, home garden strawberries, and home landscape plants.

Crop rotation may partially explain seemingly sudden shifts in GS populations. While GS feed on a wide range of plants, and can even persist in fallow soil, plants vary greatly in their suitability for GS population development. Populations have been shown to decrease significantly in potato crops, even allowing subsequent cultivation in rotation of susceptible crops. Though at this point no other crops have shown to be nearly as effective as potato, numbers have also been found to be lower after a spring oat ('Monida') winter cover crop than after a mustard ('Martiginia'), barley ('Micah'), or rye ('Wheeler') winter cover crop. Mustard and spinach crops have been shown to be very good hosts and may lead to increasing populations in some cases.

Little information is available on the effect of natural enemies on symphylan populations, which include fungal pathogens, predaceous mites, ground beetles, centipedes, and spiders. Large numbers of small brown centipedes, similar in size to symphylans, have been observed aggressively attacking symphylans and their presence could indicate presence of symphylans.

Tactics for crop damage reduction

Most plants can tolerate some level of GS feeding during all or part of the growing season, and numerous tactics can be used to grow healthy crops successfully in GS-infested soil. These tactics can be classified as those aimed at 1) reducing crop damage under high GS populations and 2) reducing the number of GS on crop roots during establishment, when plants are often most susceptible.

Susceptibility to GS feeding can vary dramatically among different soil types, plant species and crop cultivars. Generally, smaller seeded crops tend to be more susceptible than larger seeded crops. Commonly damaged crops include broccoli and other brassica crops, spinach, beets, onions, carrots, corn, and squash. For some crops (e.g., squash), damage can be reduced by increasing the plant density. This can dilute the number of GS per plant and increase survival of young seedlings during highly sensitive stages. The stand can be thinned after establishment, if needed. Beans and potatoes are rarely damaged even under high GS populations. Perennial crops, such as strawberries, raspberries, blueberries, hops, and bare root trees can also be damaged, particularly during establishment. Within a crop, susceptibility is often related to the stage of the crop planted. For example, direct-seeded tomatoes are generally more susceptible than transplants. Broccoli transplants, conversely, often fail to establish under high GS populations.

Garden symphylans are quite active and surprisingly mobile for their size, moving vertically for soil moisture and temperature fluctuations, and horizontally through the soil profile. They rely on soil pores and channels made by roots and other soil organisms in order to move through the soil. Therefore, access to roots is strongly correlated with soil structure, bulk density, or “fluffiness” of the soil and pore connectivity. Some tactics focus on temporarily reducing the number of GS in the surface soil, then planting, thus allowing these plants to establish while GS densities are low. Sandier soils are less likely to contain harmful quantities of symphylans

Tillage is an important tactic for decreasing populations in the surface soil. Along with directly killing garden symphylans, tillage breaks apart soil aggregates, modifying soil pores and pore connectivity. The effects of tillage may vary with the type of implements used. In general, the more disruptive the tillage the greater effect it will have on GS movement and feeding. Plowing or disking, followed by thorough preparation of a fine seedbed with a rototiller or roterra, often reduces surface feeding GS populations for two to three weeks. Light rolling, with a landscaping roller or similar implement, is used under some conditions to reduce the size and/or number of macropores, thereby restricting GS movement.

Slug Control

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Slugs are among the most common and persistent pests of home gardens and commercial crops in western Oregon and Washington, and if left unmanaged can cause significant damage. Slugs are closely related to snails but generally have no external shell. They are active above ground whenever the relative humidity in their immediate environment approaches 100 percent, the temperature is above 38°F, and the wind speed is negligible (<5 MPH). By day, slugs usually rest in crevices and cracks in the soil, or under surface debris where it is moist. They tend to be active primarily at night, but also feed and reproduce by day during light rain events, foggy periods, or after irrigating. Even in the summer, when air temperatures peak in the Pacific Northwest and soils are dry on the surface, slugs can be active at night in closed canopy crops such as grass seed, legume seed, pasture/hay crops, or certain vegetable crops. This is because as night temperatures decrease, the humidity of the air between the canopy and the soil often increases, if only for a few hours, even in non-irrigated settings. This "extra time" for feeding and reproduction can eventually lead to large slug populations. Slugs are relatively inactive when temperatures drop below 38°F or rise above 88°F. They take cover during windy periods and driving rain. Be aware that no-till, minimum tillage, supplemental irrigation, post-harvest residue buildup, and crop plant structures (e.g., closed canopy) can affect microclimate and promote otherwise unexpected slug activity. Also, some broad-spectrum insecticides can kill slug predators, such as ground beetles, and this reduction in natural enemy pressure can cause a significant increase in slug numbers even in fields and gardens which traditionally have not had slug issues.

Slug damage can be distinguished from that of cutworms, armyworms, and other chewing pests by the presence of slime trails and their small sausage-shaped feces, which are found on or around damaged plants. Underground feeding on roots and tubers is characterized by shallow (0.1 inch) to deep (0.5 inch), smooth-sided pits that are usually less than 0.5 inch in diameter. Leaf damage is typified by removal of plant tissue between veins. Seedling grasses and legumes may disappear when slugs feed in the furrow and destroy the growing points. In cereal crops, slugs favor feeding on newly planted seeds, which they can hollow out. Wheat is most susceptible to slug damage from the time of seeding to plant emergence.

Slug damage to vegetable, cereal, grass seed, and forage crops can be extensive near field margins. Weedy, grassy or wooded borders serve as excellent habitat for slugs. Grass seed, cereal, and vegetable crop plantings that immediately follow a perennial legume or pasture are likely to sustain slug damage. Large populations of the gray field slug and smaller numbers of several less common species (e.g., white-soled slug) build up on most perennial legumes in western Oregon and Washington. Our most economically important species in the Pacific Northwest is the gray field slug, also known as the gray garden slug (*Deroceras reticulatum*). The European black or red slug (*Arion rufus*), the white-soled slug (*Arion circumscriptus*), the garden slug (*Arion hortensis*), the hedgehog slug (*Arion intermedius*), the dusky slug (*Arion subfuscus*), the black greenhouse slug (*Milax gagates)*, the marsh slug (*Deroceras laeve*), and the three banded slug (*Ambigolimax valentianus*) are also important pests. The vast majority of the economically important species in the region are invasive from Europe. A native slug, reticulate taildropper *Prophysaon andersoni*, can also be a minor pest in certain crops such as ornamentals including Christmas trees.

In addition to plant damage, verifying that slugs are present and in damaging numbers in a garden or a crop is usually achieved by putting out a metaldehyde-based slug bait in late afternoon and returning early the next morning to check for dead slugs. Scrape a small area (12 x 12 inches) of the soil surface free of vegetation and debris (making it easier to see small slugs), and scatter four to six pellets of bait inside. You can cover the areas with a scrap piece of wood or an old carpet tile. This prevents other creatures from coming into contact with the bait, and the cover helps to keep slugs poisoned by the bait from moving away. Place bait stations after the first inch or more of rainfall in September or early October when slugs become active on the soil surface after having spent the summer underground. September and October are usually good months to control slugs, however depending on the weather, other control windows may occur when "follow-up" bait applications can be effective. After October, or when weather becomes too cold (< 38°F) and rainy, baits are less effective and slugs tend not to be active or feed above ground. In these cases, bait use is not advised. As day length shortens, eggs are produced and can hatch in fall or spring, when temperatures warm. In late fall and early spring, new juvenile slugs can be difficult to spot in the field but they can cause significant damage to gardens and crops. Slug eggs are also laid during the spring.

Slugs are hermaphrodites, which means that every individual has both male and female reproductive parts and is capable of laying viable eggs without mating. Mating occurs primarily in the fall and spring. Small, round, pearl-like, white or translucent eggs are laid in clusters in sheltered cavities near the soil surface or under debris on the soil surface. They typically hatch within 2 weeks to a month. Occasionally, these eggs overwinter if they are laid in late October or November. The greatest egg-laying activity in non-irrigated environments usually occurs after fall rains and again in the spring. The life expectancy for the gray field slug is approximately 1 year, but other slug species may live longer.

Chemical control

Slug baits (molluscicides) are poisons and therefore can be dangerous to humans, pets, and other wildlife. It is important to use baits properly by following all label instructions and heeding all label warnings. Metaldehyde (e.g., Durham, Deadline M-Ps, Metarex, Slug-Fest), methiocarb (e.g., Mesurol), iron phosphate (e.g., Sluggo, Sluggo Plus, Natria Snail & Slug Killer, Slug Magic, Escar-Go and Worry Free), and sodium ferric EDTA (e.g., IronFist, Ferroxx, Corry's Slug & Snail Killer) are four common active ingredients used to control slugs in the Pacific Northwest. Pellet baits have traditionally been the most commonly used product for homeowners and farmers. Unfortunately, even when "good" control is achieved, typically < 60 percent of the slug population will be removed. This usually suffices for economic crop protection if slug pressure is light, but populations tend to recover over time.

Under favorable conditions, slugs can significantly damage a seedling crop in just 1 or 2 days. As the crop emerges (or in the case of cereals, as the seed swells with moisture soon after planting), slugs begin feeding. Therefore, application timing, the amount of bait used, bait density (number of pellets per square foot), bait distribution and bait quality are crucial for successful treatment.

In cereal crops, the greatest risk comes during the first week after planting. Gray field slugs, for example, enter the seed furrow and begin to hollow out the endosperm shortly after the seed swells with moisture. One slug can destroy 10 to 15 wheat seeds before seedlings emerge. Depending on slug density, baits may be applied prior to planting, at planting (broadcast or band in the furrow), and/or shortly afterward. In broadleaf crops and grasses, slugs do not feed on seeds but instead target small seedlings by feeding upon and destroying the tender growing points. The most effective timing for application in these crops is at planting (if slugs are active) or just before seedlings emerge, as this is the most vulnerable plant stage. Preventive treatments are advisable on fields with a history of slug damage, in no-till or minimal till planting systems, or in situations where post-harvest residue is retained from the previous crop.

The more effective commercially available baits contain cereal bran or flour as an attractant and are formulated into pellets much smaller than the pencil-eraser-size pellets of the past. These mini pellets, or "shorts", are smaller and allow for more pellets per unit area than the larger baits. For instance, some slug bait pellets (e.g., Metarex) are a uniform 2.5 mm long. Look for slug bait in which the pellets are uniform in size, have a high bulk density, are food-based (i.e., smell strongly like cereal to attract slugs from a distance), contain Bitrex to prevent unintentional ingestion by mammals, birds and house pets, and are relatively dust free. The result upon broadcasting these pellets is a very dense and uniform pellet distribution per unit area treated. This is important because slugs tend to encounter these pellets at a greater frequency than the larger, older type. Generally, it is recommended to reapply bait after 10 to 14 days if slug pressure persists, plant damage continues, all bait has been consumed, or the bait has broken down (due to weather). Be sure that the label on the bait product applied will allow for reapplication if needed within this time frame.

The kill rate of a pellet depends on the attractiveness and quality of the carrier, palatability, weather conditions at the time of application, and the toxicant concentration. If the carrier material is not attractive and palatable to the slugs, they may avoid the bait or consume a sublethal dose of toxicant, from which they can recover.

Methaldehyde, iron phosphate, sodium ferric EDTA, and methiocarb

Several chemicals are formulated into slug and snail baits for use on food crops, seed crops, and ornamentals. Metaldehyde has been used since the early 1940s, iron phosphate since 1998, and sodium ferric EDTA since the early 2000s. The most recent generation of molluscicide products has been developed from metal chelates (e.g., sodium ferric EDTA) incorporated into an ingestible bait. These baits (e.g., IronFist and Ferroxx) have been trialed in western Oregon and showed encouraging results in terms of reducing slug populations in grass seed, clover seed, and cereal crops.

Baits that contain methiocarb can be effective but they currently have limited labels and are used primarily in nonfood or ornamental crops. For example, Mesurol 75W is used as a spray in nonfood crops and also has activity on certain insect pests as listed on the label.

Iron phosphate formulations (e.g., Sluggo and Sluggo Plus) are approved for organic production. They are formulated as a uniform and dust-free cereal-based mini pellet. Time to mortality is somewhat slower (5 to 7 days) compared to metaldehyde. The slugs, however, typically cease feeding after having eaten the iron phosphate bait. Trials with this active ingredient have shown it to be as effective in controlling gray field slugs as metaldehyde, although slightly greater rates of the iron phosphate formulations per unit area are usually needed. Slugs that ingest iron phosphate or iron chelate baits usually die underground or under a source of cover, and not above ground as happens when metaldehyde is consumed.

Metaldehyde is available in various formulations for slug and snail control. These include liquids, sand granules as well as traditional cereal-based baits. Meal formulations (for home use, usually a 2% metaldehyde pellet with an insecticide to control other pests) are also available. Liquid metaldehyde and meal formulations may give fast plant protection due to the good coverage, but they do not last more than 2 or 3 days, because UV light and moisture cause metaldehyde to degrade into non-mollusk-killing compounds. Slug-Fest is one such liquid sprayable product and is labeled for use on many food as well as nonfood and ornamental crops. It is often used to control immature slugs prior to canopy closure in establishing a stand and it is best applied at times when slugs are active aboveground.

Large pellets containing metaldehyde need higher application rates for good coverage. They usually provide good control in the first few days, but often degrade quickly and do not persist as long as mini pellets. Cereal-based mini pellets and very small pellets, (e.g., Metarex) have the best performance record in our rainy climate and can last 1 to 2 weeks on wet soil.

Research has shown that metaldehyde has a different mode of action than previously suggested. The toxicant does not dehydrate but rather damages the mucus-producing system unique to slugs and snails, which severely reduces their mobility and consequently promotes their dehydration through exposure to the sun. Wet conditions, therefore, do not reverse the toxic effect of metaldehyde, as was once thought. However, if slugs do not consume a lethal dose of metaldehyde, they may recover, particularly during wet weather, which reduces the likelihood of dehydrating poisoned slugs. Furthermore, under wet conditions, poor control may follow from low-quality baits and low concentrations of active ingredient in the bait. This is usually because of rapid (2 to 3 days) physical degradation or fungal growth on pellets that reduces slug feeding.

Due to metaldehyde's specific mode of action, beneficial organisms (earthworms or predatory insects) are not directly affected by baiting with metaldehyde even when these organisms feed on the bait. However, when applying an insecticide, such as carbaryl, to control certain insect pests like cutworms, armyworms, or wireworms, many beetle predators that feed on slugs, along with earthworms and harvestmen (daddy long-legs), may be killed as well. Be aware, too, that metaldehyde baits are a leading cause of accidental poisoning and deaths of dogs in the Pacific Northwest.

In western Washington and Oregon, slug control is often a year-round necessity in many crops and sites with no-till or conservation tillage practices. Presume damage from slugs in certain crops and fields with a history of problems, but monitor all fields as slugs can suddenly become a problem even in fields which traditionally have not had slug issues. Bait early if slug activity is apparent. In some cases, it may be best to bait for slugs before you work the soil (particularly if tillage is shallow and light). Irrigate before baiting in home gardens in order to bring more slugs to the surface during the night. In vegetables, such as brassicas, baiting must be done before the buttons form or canopy closes, because once the slugs have a chance to enter the head, they are less likely to be attracted to the bait.

Control is seldom, if ever, complete. Around the home garden, removing debris, leaf litter, weeds, and other excess vegetation helps to reduce slug habitat and lower slug numbers.

Alternative control

Barriers

Various materials, such as salt-impregnated plastic strips and copper strips, provide a small-scale barrier that can work for a few days to a few weeks in keeping slugs away from plants. These barriers have been used with varying degrees of success. However, underground slug movement or environmental degradation of the repellent (e.g., copper oxidizes, salt washes away) negatively impacts efficacy over time.

Cultivation

Slug populations can be reduced by tillage. Typically, slug numbers increase when the amount of minimal/zero tillage is increased. Plows, discs, and rototillers crush and bury slugs, disrupt their pathways, expose their eggs to desiccating conditions, dry soil, and remove volunteer-plant food for slugs. Control is more or less proportional to tillage frequency, depth, and efficiency. Plowing followed by disking can be sufficiently effective, so that no further control is needed. A fine seedbed will protect seeds and help prevent slugs accessing seedlings before emergence. In contrast, open, damp and cloddy seedbeds provide ample shelter for slugs and typically promote higher populations. Take steps to ensure that a crop has the best chance to emerge from the ground quickly.

Biological control

Some birds, such as starlings, blackbirds, and killdeer, feed on slugs throughout the fall and winter months. Grazing sheep are also known to inadvertently feed on these pests in grass and clover fields. Many insects including predatory ground beetles and rove beetles feed on slugs. Naturally occurring pathogens, and parasitic nematodes are potential biological control agents of slugs but are not commercially available for use in the United States at this time.

Some nematodes are lethal to slugs and snails, and two species, *Phasmarhabditis hermaphrodita* and *Phasmarhabditis californica* are being used successfully in Europe as commercially available biological control agents (Nemaslug and Nemaslug 2.0 respectively). These nematodes are associated symbiotically with a bacterium that uses an endotoxin to kill a wide range of pest slugs and snails, including many of the species that are economically important in Oregon and Washington. After the slug dies, the nematodes multiply on the decaying slug body and then migrate back into the soil to infect more slugs if conditions are favorable. Both *P. hermaphrodita* and *P. californica* have been found in Oregon and California, and *P. californica* in Washington but no Nemaslug products are available in the United States because of biosecurity reasons. Ongoing research focusing on discovering and testing pathogenic nematodes in the Pacific Northwest will likely prove to be valuable for developing biological control agents for pest slugs and snails.

Novel attractants

In areas where the application of certain baits (e.g., metaldehyde) is not recommended (e.g., around dogs and cats), non-toxic attractants can be used to help manage pest slugs. One cost effective option is bread dough (500 g of all-purpose flour, 500 mL of water, and 0.5 oz of active dry yeast). In home gardens for example, the dough can be deployed in 2-inch diameter balls after sunset, and slugs drawn to the attractant can then be removed. The dough balls will need to be replaced nightly because they become less attractive as they dry out. Alternatively, a more watery dough (e.g., use double the amount of water in the dough recipe) can be placed inside a plastic cup (8 oz) that is buried in the soil with the cup lip level with the soil surface. Slugs entering the cup to feed on the dough typically drown. Fresh cucumber slices are also attractive to many slug species that are pests in Oregon and Washington.

Insecticide Resistance Management

Navneet Kaur

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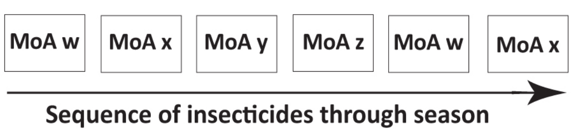
What is insecticide resistance?

According to Insecticide Resistance Action Committee (IRAC), a heritable change in the sensitivity of a pest population that is reflected in the repeated failure of a product to achieve the expected level of control when used according to the label recommendation for that pest species is known as insecticide resistance. Frequent use or overuse/misuse of an insecticide is one of the main reasons for the resistance development in various insect pests and the consequent evolution of insecticide-resistant pest populations. The resistance levels greatly depend on the selection pressure on the pest species affected by its biology, insecticide specificity, and the insecticide application strategy used.

The mechanism or way an insecticide targets an insect is called the mode of action (MoA). Insecticides belonging to a specific chemical group having a common target site are designated by the same number and letter by IRAC. Applicators should incorporate a sequence of diverse MoA classes into their pest management programs to avoid or delay the development of insecticide resistance. The common mode of action creates a significant risk of cross-resistance to all the compounds in the same sub-group. It is advised to use alternations, rotations, or sequences of different insecticide MoA classes to avoid resistance or cross-resistance selection.

Some recent resistance cases have been reported in different insect pests. About 75 *Bt* resistance cases have been reported in *Helicoverpa armigera* in USA to date. Similarly, acetamiprid resistance in melon aphid (*Aphis gossypii*) and spinosad resistance in field populations of melon fly (*Zeugodacus cucurbitae*) have been reported.



Insecticide resistance mechanisms

1. **Metabolic resistance** Metabolic resistance is the most common type of resistance, where an insect can get rid of or clear its body of a toxic compound faster than the other susceptible insects.
2. **Target-site resistance** Target-site resistance occurs when the insecticide cannot connect at the site of action in an insect, minimizing the insecticidal effect of that insecticide.
3. **Penetration resistance** Penetration resistance is when the insect's cuticle creates blockage, eventually slowing down insecticide absorption into their bodies. Penetration resistance is usually present along with other forms of resistance and can aggravate insects' other resistance mechanisms.
4. **Behavioral resistance** When an insect can sense or detect insecticide danger and avoid the treated areas, it is known as behavioral resistance. Insects can simply move away or stop feeding at the application area to avoid an insecticide's toxic effects.

The most effective strategy to manage insecticide resistance is to adopt an integrated approach to prevent its occurrence in the first place.

Insecticide Resistance Management (IRM) components include:

1. Closely monitor pest population and natural enemies. Management tactics should be used when the population exceeds economic thresholds.
2. Integrate multiple control strategies. Incorporate softer chemistries, biological insecticides, beneficial insects (predators/parasites), cultural practices, transgenic plants (where allowed), crop rotation, pest-resistant crop varieties, and chemical attractants or deterrents.

3. Read the product label. Follow the insecticide usage guidelines as well as resistance management recommendations on the label.
4. Refuges. Some unsprayed population refuges can be left adjacent to treated areas to allow the survival of susceptible insect individuals that may outcompete the resistant insects by diluting the potential impacts of any resistance that may have developed previously. This strategy is mainly used in genetically modified crops such as corn and cotton.
5. Assess if the resistance is "true". It is important to realize that resistance isn't always the problem. Poor application techniques i.e., wrong calibration, improper tank mixtures, and failure to use proper adjuvants resulting in inadequate coverage may also result in insufficient control. Proper identification and biology to target the most vulnerable stage of insect pests play a critical role (e.g., for aphids with multiple overlapping generations, rotate the products from different insecticide MoA groups for each spray).

Resources:

UC IPM Pest Management Guidelines: Floriculture and Ornamental Nurseries. UC ANR Publication 3392. https://ipm.ucanr.edu/agriculture/floriculture-and-ornamental-nurseries/managing-pesticide-resistance/

Resistance management for sustainable agriculture and improved public health. https://irac-online.org/about/resistance/management/

Benda N. and A. Dale. Managing Insecticide and Miticide Resistance in Florida Landscapes. https://edis.ifas.ufl.edu/publication/IN714

Arthropod Pesticide Resistance Database | Michigan State University. https://www.pesticideresistance.org/display.php?page=species&arId=572