
April 2007
Perpetual Draft
soybean
commodity based
survey
caps
cooperative agriculture pest survey

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Draft Log cont.

August 2010: Updated hyperlinks

September 2012 – Updated datasheets.

April 2013: Updated *Chrysodeixis chalcites* and *Lissachatina fulica* datasheets, updated the host range and distribution sections for many pests. *Planococcus minor* was removed, because this pest has been deregulated.

August 2016: Removed outdated maps.

Introduction to the Reference

History of Commodity-Based Survey

The Cooperative Agricultural Pest Survey (CAPS) community is made up of a large and varied group of individuals from federal, state, and university organizations who utilize federal and other funding sources to survey for and diagnose exotic and invasive plant pests if possible. By finding pests early, eradication efforts will likely be less expensive and more efficient. For more information on the CAPS and other Plant Protection and Quarantine (PPQ) pest detection programs see: http://www.aphis.usda.gov/plant_health/plant_pest_info/pest_detection/index.shtml.

Traditionally, states have been given a list of pests. Each year, states use this list and choose a number of pests to incorporate in their own specialized surveys. There is certainly value in surveying for plant health threats in terms of discreet pests. However, this approach may not always be the most efficient means of survey. For example, a single pest may occur on a myriad of different hosts, making a comprehensive survey too time consuming and expensive. An alternative method has been suggested. Grouping important pests under the umbrella of a single commodity could be a more efficient way to look for certain pests. The rationale for choosing a commodity survey in certain instances includes the following:

- Survey area will be smaller and targeted.
- Resources can be better utilized with fewer trips to the field.
- Commodities are easy to prioritize in terms of economic and regional (geographic) importance.

The Center for Plant Health Science and Technology (CPHST) has been charged to develop a commodity-based survey strategy in support of the CAPS program. There are two types of end products being developed for each commodity. Each product serves a valuable yet unique purpose. The result is a set of paired documents developed for each commodity. A description of these documents is provided below:

Commodity-based Survey Reference (CSR): This document is composed of a series of pest data sheets, mini-pest risk assessments (PRAs), or early detection PRAs. The data sheets are highly graphic and illustrate the biology, survey, and identification of particular pests in appropriate detail for CAPS surveyors. The pests in this document are numerous. The pests were chosen primarily from the CAPS Analytic Hierarchy Process (AHP) prioritized pest list (Appendices C and D) and the Select Agent list (http://www.selectagents.gov/ or http://www.aphis.usda.gov/programs/ag_selectagent/). Additional pests may be added if they are cited in the literature as being a primary pest of the given commodity and are exotic to the United States, or if specifically requested by the CAPS National Committee. States are not required to survey for all of the pests in this document, but may choose those that are particularly relevant to include in their survey. In general, this document should serve as a desk reference for survey

specialists as they plan their cooperative agreements. It may also be useful for obtaining high quality scientific information quickly during the field season.

Commodity-based Survey Guidelines (CSG): This document is smaller. The list of pests is shorter than those chosen for the CSR. A subgroup of the CAPS National Committee determines which pests from the CSR will be included in the CSG. As such, states that participate in these surveys must survey for all organisms listed in the CSG. The CSG set forth guidelines for survey and identification from a broad scale (site selection, number of acres to survey, number of samples to collect, etc.) and a narrow scale (field methods, survey tools, transporting samples, etc.). States are encouraged to follow the procedure set forth in the CSG. The methods are intended to increase the homogeneity of the national data set and increase the statistical confidence in negative data (*e.g.,* demonstration of "free from" status).

As a pilot project, citrus was undertaken as the first commodity in this initiative. The products were developed for implementation in the 2007 survey season. Citrus was chosen, because it is an economically important commodity that is equally distributed in both PPQ regions but is distributed in few overall states. To date, survey strategies for pests of citrus are also well documented. Shortly after completion of the citrus CSG, several other commodity survey guidelines were initiated, including small grains (wheat, barley, oats, and rye), grape, pine trees, and oak forests.

Soybean Commodity-based Survey Reference

The *Soybean Commodity-based Survey Reference* (CSR) is a companion document to the *Soybean Commodity-based Survey Guidelines* (CSG). Both documents are intended to be tools to help survey professionals develop surveys for exotic pests of Soybean. The *Soybean CSR* is a collection of detailed data sheets on exotic pests of Soybean. Additionally, the authors have identified native pests that may be easily confused with these exotic pests as well as potential vectors of exotic pests. These data sheets contain detailed information on the biology, host range, survey strategy, and identification of these pests. The commonly confused pests and vectors are included in a section of the pest data sheet dealing with the target pest.

By comparison, the *Soybean CSG* companion document is intended to help states focus resources on survey efforts and identification of a smaller group of target pests (usually less than a dozen). The *Soybean CSG* contains little information about biology. Instead, the guideline focuses on survey design, sampling strategies, and methods of identification. There is no single survey that would be wholly applicable to each location in the United States. Environment, personnel, budgets, and resources vary from state to state. Thus, the *Soybean CSG* will provide a template that states can use to increase the uniformity and usability of data across political, geographic, and climatic regions while maintaining flexibility for specificity within individual regions.

Purposes of the Soybean CSR

- To relate scientific information on a group of threatening pests.
- To facilitate collection of pest data at a sub-regional, regional, and national level

versus data collection from a single location.

- To aid in the development of yearly surveys.
- To help CAPS cooperators increase their familiarity with exotic pests and commonly confused pests that are currently found in a given commodity.
- To aid in the identification and screening of pests sampled from the field.
- To collate a large amount of applicable information in a single location.

End Users

As previously noted, this document may be used for many purposes. Likewise, it will be of value to numerous end users. As the document was developed, the authors specifically targeted members of the CAPS community who are actively involved in the development and implementation of CAPS surveys.

State Plant Health Director (SPHD): The SPHD is the responsible PPQ official who administers PPQ regulatory and pest detection activities in his or her state. The SPHD is also responsible for ensuring that the expanded role of CAPS is met in his or her state. In many states, the SPHD provides guidance for the State's ongoing management of pest risk and pest detection. However, SPHD responsibilities will vary according to the extent to which each state carries out the various components of the CAPS program.

State Plant Regulatory Official (SPRO): These individuals are employees of their respective states and generally manage the expanded survey program. The SPRO is the responsible state official who administers state agricultural regulatory programs and activities within his or her respective state.

Pest Survey Specialists (PSS): The PSS, a PPQ employee, is supervised by the SPHD of the state in which he or she is assigned. A PSS may also be responsible for survey activities and may work with the SSC and the survey committee in more than one State.

State Survey Coordinators (SSC): The SSC is a state employee responsible for coordinating each state's CAPS program, participating as a member of the state CAPS committee (SCC), and acting as liaison with the state PPQ office.

Diagnosticians: Diagnostic capabilities vary by state. Some states have advanced networks of diagnosticians, whereas other states access diagnostic support through National Identification Services (NIS) or through contracts with external partners. States are encouraged to utilize qualified diagnosticians in their respective states if expertise is available. PPQ offers diagnostic support for the CAPS program through NIS. A major responsibility for NIS's Domestic Identifiers is to provide diagnostic support to CAPS programs. There are plant pathology and entomology domestic identifiers in each of the regions. A Forest Entomology Domestic Identifier oversees both regions. To learn more

about diagnostic resources available to you, discuss your diagnostic requirements and options with your State Plant Health Director, one of the regional Domestic Identifiers, and/or NIS. Appendix A has a listing of NIS and Domestic Identifier contact information.

Organisms Included in the Soybeans Survey Reference

Organisms included in the soybeans survey reference are organized first by:

1. Pest type, (*e.g.,* arthropods, plant pathogens, nematodes, and mollusks).

2. Organisms are then divided by their pest status on corn [*e.g.,* primary pest (major pest) and secondary (minor pest)]. Primary and secondary is determined by reviewing the literature, host association, yield loss, and etc. associated with the pest on a given commodity

   A. Primary Pests: Full pest datasheets will be developed for primary pests. All pests must be exotic to the conterminous United States.

   - Pests found on the AHP Prioritized Pest List (for the fiscal year of interest) and that are major pests on the commodity will be considered primary pests.

   - Additional **exotic** pests that the author finds in the literature that are major pests on the commodity will be included as primary pests and given the designation of “National threat”.

   B. Secondary Pests: Truncated pest datasheets will be developed for secondary pests.

   - Pests found on the AHP Prioritized Pest List (for the fiscal year of interest) that are **not** identified as major pests of the commodity in the literature.

   C. PPQ Program and Line Item Pests: Plant Protection and Quarantine Program pests and pests with their own line item funding should be listed by scientific name and common name **only**. These pests will **not** receive pest datasheets, unless specifically requested by the National CAPS Committee. If a PPQ website exists for the pest, a link should be provided to that site. CPHST Ft. Collins can assist in determining which program pests and line item pests are relevant to the commodity.

   D. Other Pests Determined by the National CAPS Committee or requested by the CAPS Community: Full pest datasheets will be developed for specific pests requested by the CAPS community.

3. Finally, organisms are arranged alphabetically by their scientific names. Common names are provided as well. Previous manuals have included pests

from the Eastern and Western Region pest lists. The restructuring of the CAPS program and shift from regional guidelines to a single set of national guidelines has made these lists obsolete. Therefore, pests from these lists were not included in this CSR. States now have more flexibility to survey for pests of state concern, and most regional pests were captured in one or more state CAPS pest lists.

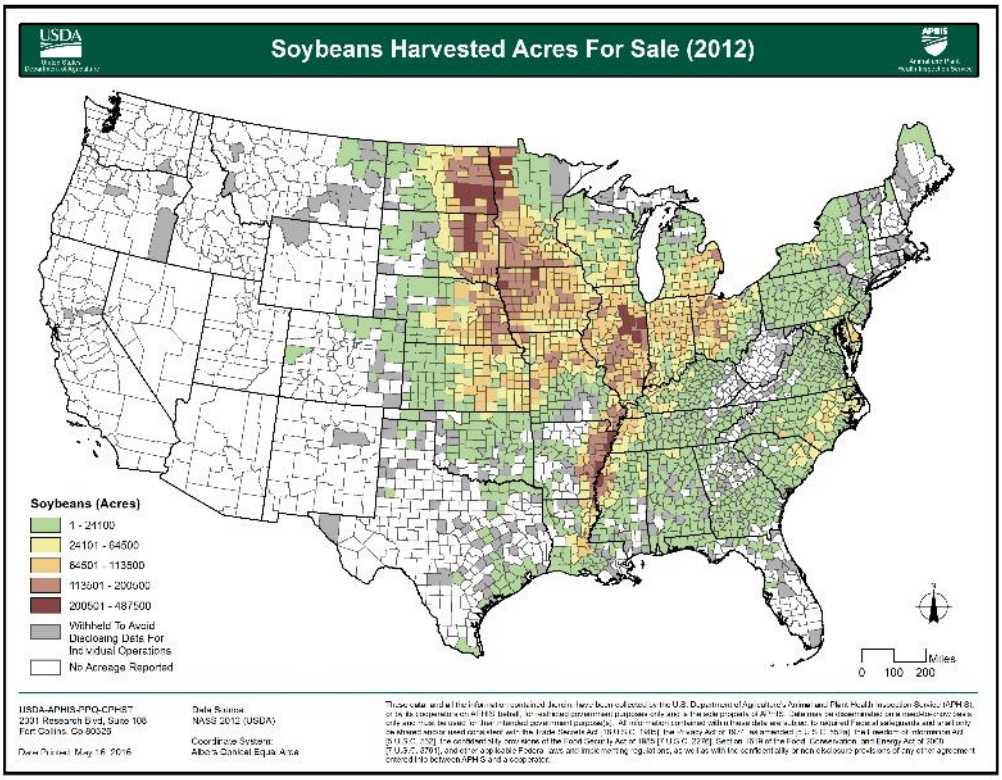
To help provide a rationale for the inclusion of each pest in the reference, the authors have included a section titled, “Reason for Inclusion in Manual”. Pests are either considered to be a CAPS target and are listed in the CAPS prioritized pest list or a national threat. The pests considered as national threats are not known to be present in the United States; however, they are not associated with the CAPS prioritized pest lists but are found on another list or identified through the literature. An additional category, requested by the CAPS community, is present in some manuals if a pest is suggested that is a primary pest, exotic to the United States, or is of regulatory significance.

Appendix M1

The survey methodology presented in Appendix M1 in the 2013 CAPS National Survey Guidelines (http://caps.ceris.purdue.edu/guidelines/2013/apdx_m1; http://pest.ceris.purdue.edu/services/napisquery/query.php?code=approvedmethods2013) lists the most up-to-date, CAPS-approved methods for survey and identification/diagnostics of CAPS target pests from the Priority Pest List, consisting of pests from the 1) commodity- and taxonomic-based surveys and 2) AHP Prioritized Pest List. The information in this table supersedes any survey and identification/ diagnostic information found in any other CAPS document (i.e., Commodity-based Survey References and Guidelines, EWB/BB National Survey Manual, etc.). All other CAPS documents will be revised to include the information contained in this table; however, this table should always be the authoritative source for the most up-to-date, CAPS-approved methods.

Soybean Background

Soybean is a member of the family Leguminoseae, subfamily Papilionaceae. It is an annual, erect bushy plant. The flowers are borne on short axillary or terminal racemes. The flowers are normally self-pollinated and completely self-fertile. Soybean is mainly grown in areas where the summer is hot and humid; however, it does withstand extreme summer and winter temperatures. The optimum temperature for growing soybean is 25 to 30 °C. Well-drained sandy or clay loams and alluviums with good fertility are generally suitable for the cultivation of the crop.



Commodity acreage map of *Glycine max* (soybean). (Map courtesy of USDA-APHIS-PPQ-CPHST.

For many years, soybean acreage increased very slowly. There were only 1.8 million acres in the United States in 1924 when the first official estimates became available. At that time, most of the crop was used for hay. Following World War II, soybean production moved from the southern U.S. into the Corn Belt. The major soybean producing states of Iowa, Illinois, Minnesota, Indiana, Ohio, Missouri, and Nebraska produced 67 percent of the United States total in 2003; the southern and southeastern states of Arkansas, Mississippi, North Carolina, Kentucky, Tennessee, Louisiana, Alabama, and Georgia produced 14 percent. Other states with significant soybean acreage are South Dakota, Kansas, Michigan, Wisconsin, and North Dakota. The USDA estimates the 2005 U.S. soybean acreage at 73.0 million acres.

Vegetative Stages. Vegetative stages are determined by counting the number of nodes on the main stem, beginning with the unifoliolate node, which have or have had a completely unrolled leaf (Fehr et al., 1971). The unifoliate node is the first node on a plant where true leaves develop. A leaf is considered completely unrolled when the leaf at the node immediately above it has unrolled sufficiently so the two edges of each leaflet are no longer touching. At the terminal node on the main stem, the leaf is considered completely unrolled when the leaflets are flat and similar to appearance to older leaves on the plant. Description of vegetative stages is given in Table 1.

Table 1. Vegetative stages and developmental descriptions of soybean.

Stage no.	Description
V1	Completely unrolled leaf at the unifoliolate node.
V2	Completely unrolled leaf at the first node above the unifoliolate node.
V3	Three nodes on the main steam beginning with the unifoliolate node.
V (N)	N nodes on the main stem beginning with the unifoliolate node.

Reproductive Stages. Reproductive stages are determined by examining the flowers and pods at the upper portion of the main stem, which is suitable for genotypes in all environments (Fehr et al., 1971). Description of reproductive stages is given in Table 2.

Table 2. Reproductive stages and developmental descriptions of soybean.

Stage no.	Description
R1	One flower at any node.
R2	Flower at node immediately below the

	uppermost node with a completely unrolled leaf.
R3	Pod 0.5 cm (1/4 inch) long at one of the four uppermost nodes with a completely unrolled leaf.
R4	Pod 2 cm (3/4 inch) long at one of the four uppermost nodes with a completely unrolled leaf.
R5	Beans beginning to develop (can be felt when the pod is squeezed) at one of the four uppermost nodes with a completely unrolled leaf.
R6	Pod containing full size green beans at one of the four uppermost nodes with a completely unrolled leaf.
R7	Pods yellowing; 50% of leaves yellow. Physiological maturity.
R8	95% of pods brown. Harvest maturity.

References

Fehr, W.R., Caviness, C.E., Burmond, D.T., and Pennington, J.S. 1971. Stage development descriptions for soybeans, *Glycine max* (L.) Merrill. Crop Science 11:929-931.

Gibson, L. and Benson, G. 2005. Origin, History, and Uses of Soybean (*Glycine max*). Iowa State University. http://www.agron.iastate.edu/courses/agron212/Readings/Soy_history.htm.

Arthropods

Primary Pests of Soybean (Full Pest Datasheet)

Adoretus sinicus

Scientific Name
Adoretus sinicus Burmeister

Synonyms:
Adoretus tenuimaculatusm, Adoretus tenuimaculatus

Common Name(s)
Rose beetle, Chinese rose beetle

Type of Pest
Beetle

Taxonomic Position
Class: Insecta**, Order:** Coleoptera, **Family:** Scarabaeidae

Reason for Inclusion in Manual
National Threat

Pest Description
Eggs: The small, elliptical eggs of this species are laid in the soil within 1 (2.54 cm) to ½ inch (1.27 cm) from the surface. Eggs are about 1.67 x 1.37 mm, shining white at oviposition and gradually become dull creamy white before hatching in 7 to 16 days (Habeck, 1963). Reddish brown mandibles of first instar larva clearly visible through chorion before hatch.

Larvae: There are three larval stages of this insect that last about a week each. The larval forms of this insect are stout, 'C-shaped', white grubs with a conspicuous head and short legs (typical scarabaeiform) (Fig. 1). Maximum middorsal length about 34 mm. Head capsule smooth, shining, yellowish brown; eyespot black pigmented. Frontal sutures represented by a fine white line extending toward antennal base but fading near eyespot.

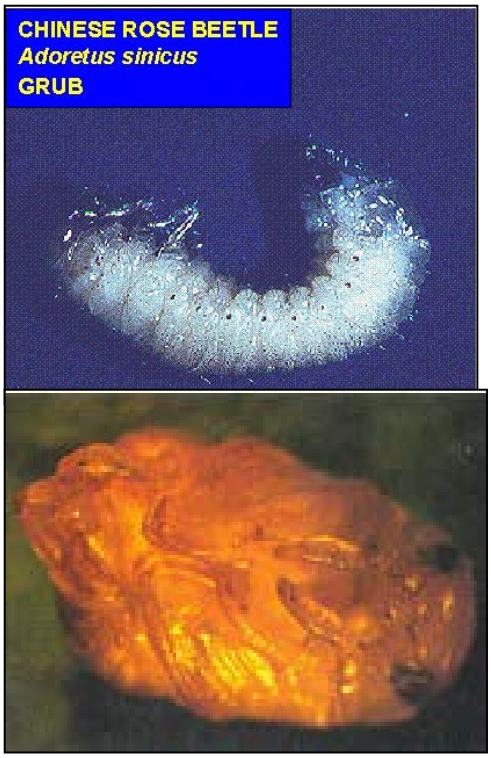


Figure 1. *A. sinicus* larva (top) and pupae (bottom. Photos courtesy of R. Mau and J. Kessing, Department of Entomology, Hawaii. www.extento.hawaii.edu/kbase/crop/Type/adoretus.htm.

Antenna 4-segmented, arising from projection of head capsule near mandibular base. Segment 2 longest, followed in decreasing order by segments 1, 3, and 4. Segment 3 with apical ventral projection with oval transverse sensory area on inner surface of projection. Segment 4 with 3 sensory areas: a single large oval one dorsally; 2 smaller ones ventrally; apical one oval and larger than circular proximal one. Antenna without setae except for few minute ones at segment 4 apex. Haptomerum of epipharynx with dense transverse row of 6-9 heli. Laeotorma well developed with pternotorma laterally and epitorma mesally. Sclerotized plate sharply pointed. Plegmatium composed of 12 to 14 curved setae at each lateral margin. Maxillary stridulatory area with 8 or more sharp, recurved teeth; lacinia of maxilla with 3 unci. Raster lacking palidia; subtriangular teges of hamate setae (Haback, 1963).

Refer to Habeck (1963) for a detailed description of the larvae. Grubs do not attack live plant tissue, but preferably live in loose rich soil, leaf litter, or compost (Williams, 1931).

Pupae: Pupae are yellowish white when initially formed and gradually become brown (Fig. 1). The entire surface of the pupae is densely covered with minute hairs. The pupa is about 13 x 7 mm long. Development is completed in 1 to 2 weeks.

Adults: The adults are sturdy, pale reddish brown beetles, and 7-10.0 mm in length (Fig. 2). The body is elongate, narrow, and nearly parallel-sided. Dorsal surface light to dark brown, covered in short, scale-like hairs. The fine white hairs can give the beetle a grayish appearance. Clypeus with anterior margin semicircular, reflexed; reflexed margin with scale-like hairs very dense. Tarsus slender with last segment expanded and longer than rest of segments combined. Anterior protarsal and mesotarsal claws split apically, much longer than posterior claws. Metatarsal anterior claw not split at apex. Genitalia with lateral lobe slightly shorter than basal piece, apex feebly emarginate (USDA, 1988).

Figure 2. *A. sinicus* adult. Photo courtesy of the Honolulu Rose Society. http://www.honolulurosesociety.org/pests.html.

Biology and Ecology

This beetle is nocturnal in habit and is attracted to lights at night. During the day they remain under leaves, loose bark, or are shallowly buried in the soil, and emerge at dusk to feed (Williams, 1931). Peak feeding and mating activity occurs about 30 minutes after sunset (Tsutsumi, et. al, 1993). The beetle preferentially feeds on leaves and plant species that are relatively high in non-structural carbohydrates (Furtani and Arita, 1990; Arita et al., 1993). It also prefers to feed on leaves with feeding or other types of

damage (Pemberton, 1959). These leaves release ethylene gas, which serve as an attractant to beetles (Arita et al., 1988). The life cycle from egg to adult is completed in 6 to 7 weeks.

Snap bean and strawberry plants sprayed with azadirachtin, an insecticide based on a naturally occurring compound produced by neem (*Azadirachta indica*) had significantly less feeding damage from *A. sinicus* than water treated plants (Arita Tsutsumi et al., 1995). Ginger plants treated with the commercially available hormone etephon, 2-chloroethylphosphonic acid, (an ethylene releasing substance), were preferentially fed upon by the Chinese rose beetle (Arita et al., 1988).

Pest Importance

This polyphagous scarabaeid beetle was introduced into Hawaii sometime before 1896. It is distributed in Southeast and East Asia, including Indonesia, Taiwan, and China; it is also found on Guam. Introduction into Hawaii probably was accomplished by larvae in the soil of plants. Adults feed at night on the leaves of a great variety of plants. At least 255 plant species in 56 families have been recorded as hosts, including rose, grape, cycad, okra, beans, soybean, pigeon pea, sweet potato, eggplant, corn, cucumber, asparagus, taro, banana, and cotton. Plant damage is caused by the adult. Attacked leaves show numerous small holes, or may become entirely skeletonized. Larvae feed on decaying plant matter in the soil, and only rarely attack live roots. Although parasitoids and predators have been introduced into Hawaii, no satisfactory control measures have been developed for *A. sinicus*, and it remains a significant pest.

Symptoms/Signs

On dicotyledonous plants, adults feed on plant foliage at night, creating a lace-like or shot with holes appearance on leaves by feeding on plant tissue between leaf veins. In severe cases, most leaves are skeletonized (Fig. 3). Monocots, such as corn, show both interveinal and veinal feeding (Furutani et al., 1990). The feeding damage can be significant enough to reduce fruit yields (Furutani et al., 1990). Chinese rose beetles leave small, dark colored particles of frass which are excreted on the plant while feeding. Defoliation is also common in many plant species.

Larvae are commonly found in the soil of lawns, gardens, flower beds, and sometimes in cultivated fields, wherever considerable humus is present. The grubs do not attack living vegetable tissue and apparently are humus and detritus feeders.

Known Hosts

Adults are general feeders. The plant host for this species is composed of over 250 plants from a wide variety of ornamental and cultivated crops, representing 56 plant families. Crops attacked include asparagus, beans, broccoli, cabbage, cacao, Chinese broccoli, Chinese cabbage, chiso, corn, cotton, cucumber, eggplant, flowering white cabbage, ginger, grape, green bean, okra, rose, soybeans, strawberry, and sweet potato.

Major hosts
Acalypha (copperleaf), *Alocasia* (elephant ear), *Cajanus cajan* (pigeon pea), *Canna*, *Glycine max* (soybean), *Musa x paradisiaca* (plantain), *Rosa* spp. (rose), and *Vitis vinifera* (grape).

Known Vectors (or associated organisms)
Adoretus sinicus is not a known vector and does not have any associated organisms.

Figure 3. Feeding damage (lace-like appearance) caused by *A. sinicus*. Photos courtesy of R. Mau and J. Kessing, Department of Entomology, Hawaii and B. Villegas http://www.sactorose.org/ipm/84chineserosebeetles.htm.

Known Distribution
Originally from Japan and Taiwan, this beetle currently enjoys a widespread distribution throughout Southeast Asia and many Pacific Islands.

Asia: China, Indonesia, Korea, Laos, Malaysia, Singapore, Thailand, and Vietnam. **Oceania:** Federated States of Micronesia, Guam, and Northern Mariana Islands. **North America:** Hawaii

Potential distribution within the United States
Introduced to Hawaii before 1896, *A. sinicus* is now a common pest on all major islands in the state. This pest is not known to occur in the continental United States.

Survey
CAPS-Approved Method*: Has not been evaluated at this time.

*For the most up-to-date methods for survey and identification, see Approved Methods on the CAPS Resource and Collaboration Site, at http://caps.ceris.purdue.edu/.

Literature-Based Methods:
Visual Survey: Surveys are conducted using a visual survey of symptoms, especially during late May to early September. Look for plants with foliage demonstrating a lace-like or shot with holes appearance caused by adults of *A. sinicus* feeding on plant tissue between leaf veins. In severe cases, most leaves are skeletonized. Look under debris and dig in the first two inches of humusy soil about plants for hairy, light to dark brown adults about 10 mm long. Additionally, you can search loose soil for "C-shaped" scarab larvae about 34 mm long. White or brown pupae 13 mm long will be within the split larval skin in an earthen cell.

Trapping: At night, trap adult *A. sinicus* in blacklight traps.

Sweep Nets: At night, sweep adults from host leaves.

Key Diagnostics/Identification

CAPS-Approved Method*: Has not been evaluated at this time.

*For the most up-to-date methods for survey and identification, see Approved Methods on the CAPS Resource and Collaboration Site, at http://caps.ceris.purdue.edu/.

Literature-Based Methods:
A morphological identification is required for *A. sinicus*. The white, scale-like setae on the dorsal surface, and the semicircular clypeus are completely diagnostic for this species in the New World. The tribe Adoretini, to which *Adoretus* belongs, is Oriental in distribution without any New World representatives (USDA, 1988).

Easily Confused Pests

The adult *A. sinicus* does not resemble any other adult in the subfamily Rutelinae occurring in North America.

A. sinicus larvae (tribe Adoretini) most resemble those of the tribe Anomalini, representatives of which occur in North America (USDA, 1988). The following key (modified from Ritcher. 1948) will separate larvae of these two tribes.

Haptomerum of epipharynx with dense transverse row of 3 (rarely 2 or 4) prominent heli; palidia (paired rows of pointed, recumbent spines) present; maxillary stridulatory area with 4-7 sharp recurved teeth; lacinia or maxilla with 2 unci ……tribe Anomalini.

Haptomerum of epipharynx with dense transverse row of 6-9 heli; raster without palidia but with subtriangular teges of hamate setae; maxillary stridulatory area with 8 or more sharp recurved teeth; lacinia or maxilla with 3 unci ……tribe Adoretini.

References

APPPC.1987. Insect pests of economic significance affecting major crops of the countries in Asia and the Pacific region. Technical Document No. 135. Bangkok, Thailand: Regional FAO Office for Asia and the Pacific (RAPA), 56 pp.

Arita, L.H., Furutani, S.C., and Moniz, J.J. 1988. Preferential Feeding by the Chinese Rose Beetle (Coleoptera: Scarabaeidae) on Ethephon-Treated Plants. J. Econ. Ent. 81: 1373-1376

Arita Tsutsumi, L.H., Furutani, S.C., Yoshimura, E.R., and Sakai, D.T. 1995. Feeding response of the Chinese rose beetle, *Adoretus sinicus* Burmeister, to azadirachtin treated host plants. J. Haw. Pac. Agric. 6: 49-54.

EPPO. 2005. PQR database (version 4.4). Paris, France: European and Mediterranean Plant Protection Organization.

Furtani, S.C., and Arita, L.H. 1990. Effect of light exposure and carbohydrate content of snap bean leaves on Chinese rose beetle (Coleoptera: Scarabaeidae) feeding. J. Econ. Entomol. 83*5): 2022-2025.

Furutani, S.C., Arita, L.H., and Fujii. J.K. 1990. Relationship between simulated Chinese rose beetle (Coleoptera: Scarabaeidae) feeding and photosynthetic rate reduction. Pr. Hawaii Entomol. Soc. 30: 97-104.

Habeck, D.H. 1963. Description of Immature Stages of the Chinese Rose Beetle, *Adoretus sinicus* Burmeister (Coleoptera: Scarabaeidae). Proc. Hawaiian. Entomol. Soc. 18: 251-258.

Pemberton, C.E. 1959. In Report of the rhinocerous beetle technical advisory committee meeting held in Suva, Fiji. February 16-19. South Pacific Commission, New Caledonia. p.11.

Ritcher, P.O. 1948. Descriptions of the larvae of some ruteline beetles with keys to tribes and species (Scarabaeidae). Ann. Entomol. Soc. Am. 41: 206-212.

Tsutsumi, L.H., Furutani, S.C, Nagao, M., Sworts, V., and Vargo, A. 1993. An integrated approach to *Adoretus* control in Hawaii and American Samoa.

USDA. 1988. Pests not known to occur in the United States or of limited distribution No. 94: Chinese Rose Beetle. USDA-APHIS-PPQ.

Waterhouse, D.F. 1993. The major arthropod pests and weeds of agriculture in Southeast Asia. The major arthropod pests and weeds of agriculture in Southeast Asia., v + 141 pp.; [ACIAR Monograph No. 21]; 3 pp. of ref.

Williams, F.X. 1931. Handbook of the Insects and Other Invertebrates of Hawaiian Sugarcane Fields. Honolulu, Hawaii: Hawaiian Sugar Planters' Association. Advertiser Publisher Co., Honolulu, Hawaii. pp. 194-198

Anticarsia irrorata

Scientific Name
Anticarsia irrorata Fabricius, Walker

Synonyms:
Azazia rubricans, Thermesia rubricans

Common Name(s)
Noctuid moth, owl moth

Type of Pest
Moth

Taxonomic Position
Class: Insecta, **Order:** Lepidoptera, **Family:** Noctuidae

Reason for Inclusion in Manual
National Threat

Pest Description
Larva: The larvae are yellowish green with a yellowish line on the lateral sides, a transparent mid-dorsal line and yellowish intersegmental lines. Thin and cylindrical larvae measure 4 to 4.5 cm length on full growth. After about 20 to 25 days, they undergo pupation under leaf debris. Pupal period lasts for 7 to 10 days (Senguttuvan et al., 2008).

Figure 1. *A. irrorata* adult. Photo courtesy of G. McCormick., Cook Islands Natural Heritage Trust, Rarotonga. http://cookislands.bishopmuseum.org/species.asp?id=7003.

Adult: Adult insect (Fig. 1) is medium sized (15 to 17 mm) buff or light brown colored moth with an oblique transverse faint brown line across both wings dorsally. Fore-wings characterized by diagonal line from wing apex to approximately 1/3 in from outer margin; row of black dots between line and wing edge; kidney shaped cell patch approximately half way along wing. Hind wings have continuation of diagonal line and similar subterminal dots. Underside buffish brown with subterminal line not originating in wing apex; brown dots also present and white cell spot. Well marked specimens also have wavy terminal line. Head, thorax, abdomen, legs and antennae similar shade of brown to wing background (McCormack, 2006).

Pest Importance

In the field experiments conducted at National Pulses Research Centre, the defoliation of two varieties of cowpea by this moth ranged from 10.0 to 100% during December and February, 1999-2000 in Pudukkottai, Tamil Nadu, India. The corresponding yield loss was nearly 50% with a severe infestation (Senguttuvan et al., 2008).

Symptoms/Signs

The larval stage of *A. irrorata* feeds on leaves. The damage can be easily recognized on foliage. The leaf margins are eaten away by the caterpillar. The caterpillar can be seen mainly on the leaf under surface. Severely affected plants will look like a mass of veins and stems alone as if grazed by cattle (Senguttuvan et al., 2008).

Known Hosts

Major hosts

Andropogon sorghum (broomcorn), *Cajanus cajan* (pigeon pea), *Canavalia ensiformis* (horsebean), *Cicer arietinum* (chick pea), *Cucumis sativius* (cucumber), Cyamopsis *tetragonoloba* (cluster bean), *Dolichos* spp. (hacinthbean), *Glycine max* (soybean), *Gossypium* spp. (cotton), *Lablab purpureus* (lablab Bean), *Mucuna pruriens* (velvetbean), *Oryza sativa* (rice), *Phaseolus* spp. (bean), *Saccharum officinarum* (sugar cane), *Vigna unguiculata* (cowpea), and *Vigna* spp.

Known Vectors (or associated organisms)

Anticarsia irrorata is not a known vector and does not have any associated organisms.

Known Distribution

The following list is not exhaustive. This species may occur on many other Pacific Islands as a vagrant/migrant species.

Africa: Ghana, Kenya, Malawi, Nigeria, and São Tomé and Príncipe; **Asia:** Hong Kong, India, Indonesia, Malaysia (West), and Sri Lanka; **Oceania:** Australia, Chagos Archipelago[1], Cook Islands, Fiji, French Polynesia, Kermadec Islands[2], Marshall Islands, Norfolk Island[3], Papua New Guinea, Pitcairn Island, and Tonga (EcoPort, n.d.; Smee, 1936; Sugarman, 1972; Dugdale, 1973; Holloway, 1982; Jackai and Daoust, 1986; Smithers, 1998; Barnett et al., 1999; Dall'Asta, 2004; Holloway, 2000; Hong Kong Lepidopterists' Society, 2004; Holloway, 2005; CSIRO Australia, 2006; Gulf of Guinea Conservation Group, 2006; McCormack, 2006; Evenhuis, 2010; Robinson et al., 2010).

[1] Barnett et al. (1999) states that this species is migratory and has been recorded as far east as Rapa Iti (part of Bass Islands in French Polynesia) in the Pacific.

[2] This species is thought to be a vagrant (Dugdale, 1973).

[3] This species is considered to be migrant (Holloway, 1982).

Pathway

Only one interception has been recorded at U.S. ports of entry in the AQAS system. This interception occurred on a wood product originating from the Philippines. No interceptions at the genus level have been recorded (AQAS, 2012; queried June 28, 2012).

The adult is likely a strong flier as it is reported as a vagrant/migratory species of several islands in the Pacific.

Potential Distribution within the United States

There is currently no ost or risk map available for *A. irrorata*. The most commonly grown host plants in the United States are: *Gossypium* spp. (cotton), *Glycine max* (soybean) (Figs 2, 3), and *Vigna unguiculata* (cowpea). Both *Gossypium* (cotton) and *Vigna unguiculata* (cowpea) are grown primarily in the southern portion of the United States, while *Glycine max* (soybean) has high acreages grown in the Midwest.

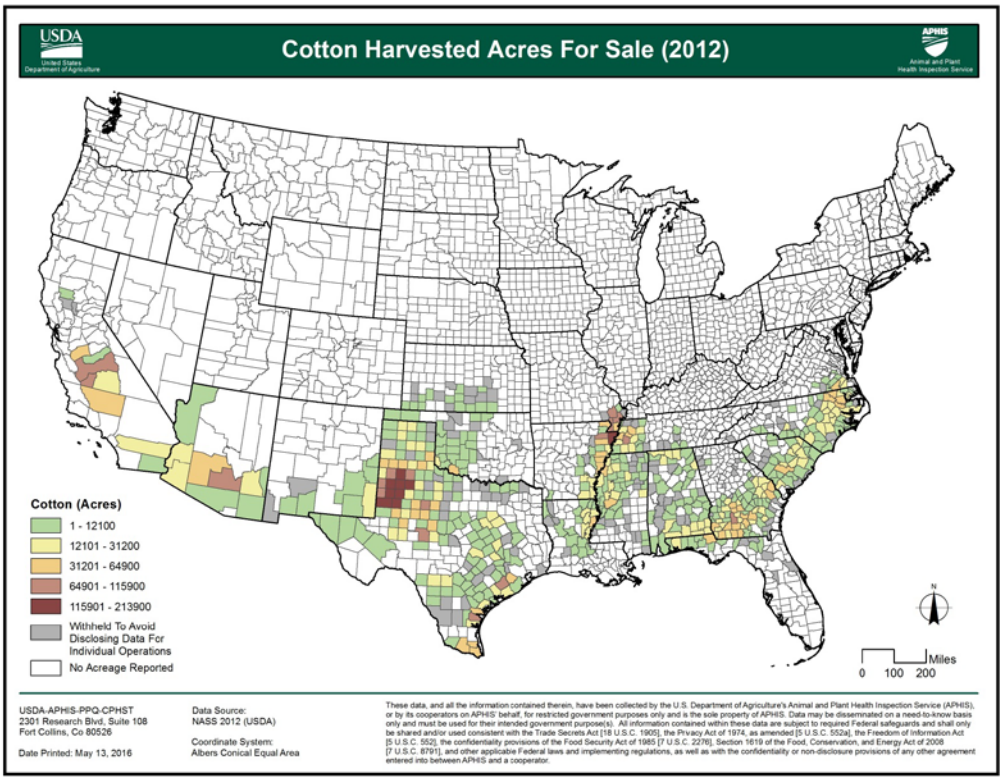


Figure 2. Commodity acreage map of *Gossypium* spp. (cotton). Map courtesy of USDA-APHIS-PPQ-CPHST.

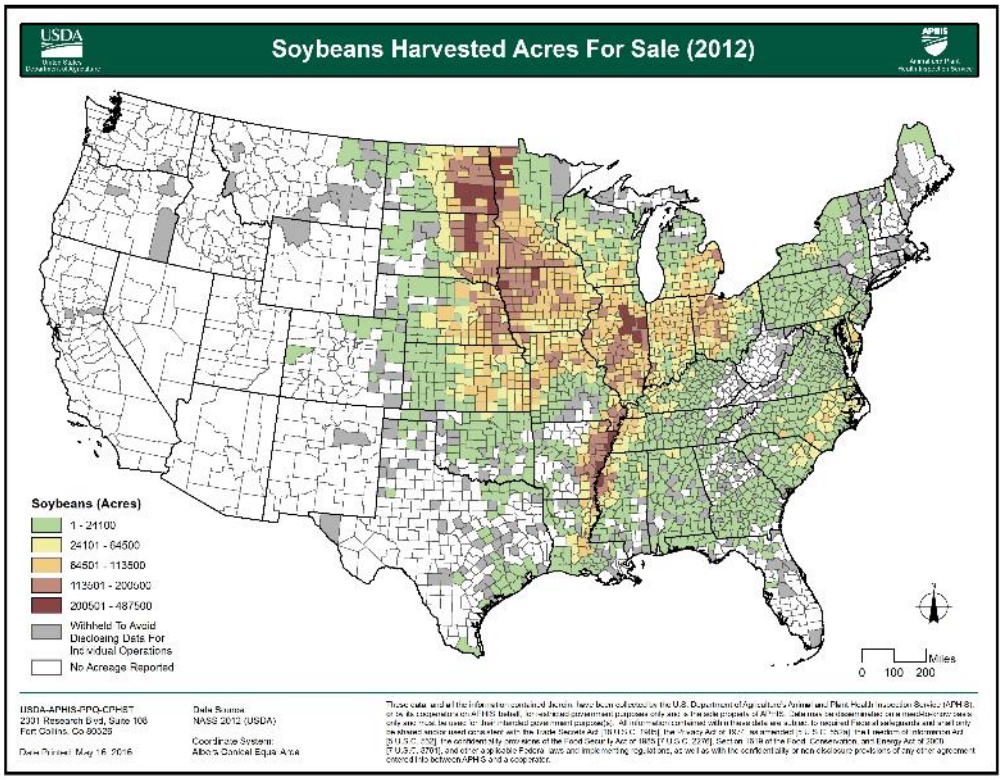


Figure 3. Commodity acreage map of *Glycine max* (soybean). Map courtesy of USDA-APHIS-PPQ-CPHST.

Oryza sativa (rice) and *Saccharum officinarum* (sugarcane) are grown in high densities in parts of the United States, but are rather limited in their distribution.

Survey

CAPS-Approved Method*: Has not been evaluated at this time.

*For the most up-to-date methods for survey and identification, see Approved Methods on the CAPS Resource and Collaboration Site, at http://caps.ceris.purdue.edu/.

Literature-Based Methods: There is limited information available on this pest at this time. Survey appears to be visual based on host symptoms and presence of larvae on the leaf surface.

Key Diagnostics/Identification

CAPS-Approved Method*: Has not been evaluated at this time.

*For the most up-to-date methods for survey and identification, see Approved Methods on the CAPS Resource and Collaboration Site, at http://caps.ceris.purdue.edu/.

Literature-Based Methods: A morphological identification is required for *A. sinicus*. Genitalia preparation is necessary to establish species status.

References

AQAS. 2012. AQAS database, *Anticarsia irrorata* interceptions. Last accessed June 26, 2012 from: https://mokcs14.aphis.usda.gov/aqas/HomePageInit.do.

Barnett, L. K., C. W. Emms, and J. D. Holloway. 1999. The moths of the Chagos Archipelago with notes on their biogeography. Journal of Natural History 33: 1021-1038.

Brou, Jr., V. A. No date. *Epitausa prona* (Moschler, 1880) in Lousiana. 2 pp.

CSIRO Australia. 2006. CntomID-PNG Specimen Database. Accessed July 3, 2012 from: http://anic.ento.csiro.au/entomid-png/index.asp.

Dall'Asta, U. 2004. Moths and conservation values in East Africa: Noctuidae of the Taita Hills, Kenya. Journal of Insect Conservation 8: 191-198.

Dugdale, J. S. 1973. A note on Kermadec Islands Lepidoptera. New Zealand Entomologist 197-203.

EcoPort. No date. Unassigned EcoPort Record: *Anticarsia irrorata*. Accessed July 3, 2012 from: http://ecoport.org/ep?Arthropod=18768&entityType=AR****&entityDisplayCategory=full.

Evenhuis, N. L. 2010. Checklist of Fijian Lepidoptera. Accessed June 27, 2012 from: http://hbs.bishopmuseum.org/fiji/checklists/lepidoptera.html.

Gulf of Guinea Conservation Group. 2006. Gulf of Guinea Islands' Biodiversity Network. Lepidoptera Noctuidae – São Tomé and Príncipe. Accessed July 3, 2012 from: http://www.ggcg.st/Species_Lists/STP%20Lepidoptera%20Noctuidae.htm.

Holloway, J. D. 1982. Further notes on the Lepidoptera of Norfolk Island, with particular reference to migrant species. Journal of Natural History 16(3): 351-365.

Holloway, J. D. 2000. The Lepidoptera of Easter, Pitcairn and Henderson Islands. Journal of Natural History 24(3): 719-729.

Hong Kong Lepidopterists' Society. 2004. Hong Kong Moths Scientific Name List. Accessed June 27, 2012 from: http://www.hkls.org/mot-list.html.

Jackai, L. E. N. and R. A. Daoust. 1986. Insect pests of cowpeas. Annual Review of Entomology 31: 95-119.

McCormack, G. 2006. Cook Islands Biodiversity Database, Version 2006.1. Cook Islands National Heritage Trust, Rarotonga. http://cookislands.bishopmuseum.org.

Robinson, G. S., P. R. Ackery, I. J. Kitching, G. W. Beccaloni, and L. M. Hernández, 2010. HOSTS - A Database of the World's Lepidopteran Hostplants. Natural History Museum, London. Accessed June 27, 2012 from: http://www.nhm.ac.uk/hosts.

Senguttuvan, T., N. Ganapathy, and C. Durairaj. 2000. Green caterpillar menace in cowpea. The Hindu (Online Edition of India's National Newspaper): August 10, 2000. www.hinduonnet.com/2000/08/10/stories/0810000d.htm.

Smee, C. 1936. Report of the Entomologist. Rep. Dep. Agric. Nyasaland pp. 20-24.

Smithers, C. N. 1998. A species list and bibliography of the insects recorded from Norfolk Island. Technical Reports of the Australian Museum 13: 1-55.

Sugarman, B. B. 1972. Insects and other arthropods from Kwajalein Atoll (Marshall Islands). Proceedings of the Hawaiian Entomological Society XXI: 271-286.

Autographa gamma

Scientific Name
Autographa gamma L.

Synonyms:
Phytometra gamma and *Plusia gamma*

Common Name(s)
Silver-Y moth, beet worm

Type of Pest
Moth

Taxonomic Position
Class: Insecta, **Order:** Lepidoptera,
Family: Noctuidae

Figure 1. Eggs of *A. gamma*. Photo courtesy of Jurgen Rodeland. http://www.rodeland.de/fotos/lepidoptera/autographa_gamma.htm.

Reason for Inclusion in Manual
CAPS Target: AHP Prioritized Pest List - 2009

Pest Description
Eggs: Semi-spherical, 0.57 mm in diameter. Eggs are yellowish-white (Fig. 1), later turning yellowish-orange to brown. The number of ribs varies from 28 to 29 (Paulian et al., 1975). The eggs are deposited in bunches or singly on the underside of leaves.

Larvae: The larva is a semi-looper with three pairs of prolegs. It occurs in varying shades of green (Fig. 2), with a dark-green dorsal line and a paler line of whitish-green on each side. The spiracular line is yellowish, edged above with green. There are several white transverse lines between the yellow spiracular line and the dorsal black line. Some larval forms have a number of white spots. The head may have a dark patch below the ocelli or be entirely black. Maximum length 20 to 40 mm (USDA, 1958; Jones and Jones, 1984; Emmett, 1980; Hill, 1987).

Figure 2. Larva of *A. gamma*. Photo courtesy of P. Mazzei. www.invasive.org.

Pupae: Pupation takes place within a translucent, whitish cocoon spun amongst plant foliage (Fig. 3). The leaves may sometimes be folded over. The pupa is brown to black, greenish or even whitish-green on its ventral side, 16 to 21 mm (approx. $^{5}/_{8}$ to $^{13}/_{16}$ in) long, and 4.5 to 6.0 mm (approx. $^{3}/_{16}$ to $^{1}/_{4}$ in) broad. Cremaster globular, with four pairs of hooklets (Paulian et al., 1975; Carter and Hargreaves, 1986).

Figure 3. Cocoon of *A. gamma*. Photo courtesy of Alain Fraval. http://www.inra.fr/hyppz/RAVAGEUR/6autgam.htm.

Adults: The adults are gray-colored and the forewings are marbled in appearance; their color being silvery-gray to reddish-gray to black with a velvety sheen. Wing expanse is 36 to 40 mm (approx. 1 $^{7}/_{16}$ to 1 $^{9}/_{16}$ in). The 'Y' mark on the forewing is distinct and silvery (Fig. 4). The hindwings are brownish with a darker border (USDA, 1958; Jones and Jones, 1984; Hill, 1987).

Figure 4. Adult showing the silver *Y* mark that resembles the Greek letter gamma. Photo courtesy of Jeremy Lee.

Biology and Ecology

Adult moths feed on nectar primarily in the early morning or evening hours, young larvae skeletonize the leaves, and older late-instar larvae eat the entire leaf. The polyphagous larvae damage many agricultural plants and eat the foliage of over 200 plant species (Steudel, 1963).

Females lay from 500 to more than 1,000 whitish eggs (Hill, 1987), singly or in small batches, on the underside of leaves of low-growing plants. *Autographa gamma* passes through five larval instars (Harakly, 1975). Depending on the climate of the region, *A. gamma* can have two to four generations per year (Rashid et al., 1971; Dochkova, 1972; Harakly, 1975; CABI, 2007). *Autographa gamma* does not undergo true diapause (Tyshchenko and Gasanov, 1983), but pupae or late-instar larvae can overwinter in moderate climates (Dochkova, 1972; Tarabrina, 1970; Kaneko, 1993; Saito, 1988). In

areas where *A. gamma* is unable to overwinter, severe infestations occur sporadically. The longevity of females can be almost twice that of males (Harakly, 1975).

High temperatures decrease the life span (Harakly, 1975), and depending on temperature, *A. gamma* has a life span from 28 to 65 days (Rashid et al., 1971; Harakly, 1975). The incubation period lasts for 3 days at 25°C (77°F) (Ugur, 1995), but in temperate regions, egg incubation may take 10 to 12 days (Hill, 1987). Larval development takes from 51 days at 13°C (55.4°F) to 15 to 16 days at 25°C (77°F) and the pupal stage from 32 days at 13°C (55.4°F) to 6 to 8 days at 25°C (77°F) (Hill and Gatehouse, 1992; Ugur, 1995).

The lower threshold temperatures for egg, larvae, pupae and pre-oviposition periods range between 9.3 to 11.0 °C (48.7 to 51.8°F) and depends upon plant species used for feed. Giving an average threshold temperature of 10 °C (50°F), the degree days varied from 177 to 257 for different plant species (Honěk et al., 2002).

Autographa gamma is primarily nocturnal. An average-sized moth that is unaided by wind can fly 16 km/h (approx. 10 mi/h) for 50 km (approx. 31 mi) without feeding; some larger moths can fly over 100 km (approx. 62 mi) (Macaulay, 1974). *Autographa gamma* is migratory and can disperse over distances spanning hundreds of kilometers (Macaulay, 1972, 1974; Harakly, 1975).

Symptoms/Signs

There are no specific symptoms and signs listed in the literature for soybeans. Eggs (singly or in small clusters) may be visible on leaves of low growing plants. Larvae are active at night. During the day, they remain pressed against the underside of the leaf; when disturbed they tend to drop off the plant. Leaves may be skeletonized by larval feeding and may have a brownish appearance. Older leaves are preferred by larvae and consume the leaves completely (Harakly, 1975). The larvae only eat young leaves after destroying the old ones. The petioles or leaf stalks may be cut by the larvae. Frass may or may not be visible. Pupae are found in the folds of the lower leaves of the host plant. Webbing may be present. Adult moths feed on flowers and can often be seen feeding during the day or early evening (USDA, 1985).

Pest Importance

Outbreaks of *A. gamma* occur periodically over wide areas of Europe, Asia and North Africa. The outbreak of 1928, which occurred in most of central Europe, caused widespread defoliation of peas in Poland. Damage from this insect and *Pieris rapae* (cabbage white) in areas of the Netherlands was valued at as much as 320,000 guilders (~$180,000) during some years in the 1800s. It is also very destructive in England and Denmark (CABI, 2007). *A. gamma* causes appreciable damage to agricultural products (lettuce, radish, cowpea, and clover) in Egypt (Harakly, 1975).

Damage to globe artichokes was severe near Bari, Italy in 1982 to 1985, with about 55% of plants being damaged, and *A. gamma* was one of the major pests (Ippolito and Parenzan, 1985).

Studies in Czechoslovakia (Novak, 1975) indicated that damage became of economic significance when 25% of the leaf area of a plant was destroyed.

Known Hosts

This polyphagous pest is found on cereals, grasses, fiber crops, *Brassica* spp. and other vegetables including legumes. *Autographa gamma* can feed on at least 224 plant species, including 100 weeds, from 51 families (Maceljski and Balarin, 1972). Soybean is considered a primary host.

Major hosts

Beta vulgaris (beet), *Beta vulgaris* var. *saccharifera* (sugarbeet), *Borago officinalis* (common borage), *Brassica oleracea* var. *capitata* (cabbage), *Brassica oleracea* var. *gemmifera* (Brussels sprouts), *Brassica rapa* subsp. *chinensis* (Chinese cabbage), *Brassica rapa* subsp. *pekinensis* (Pe-tsai), *Cannabis sativa* (marijuana, hemp), *Capsicum* (peppers), *Chrysanthemum indicum* (chrysanthemum), *Cicer arietinum* (chickpea), *Cichorium intybus* (chicory), *Cynara scolymus* (artichoke), *Daucus carota* (carrot), *Glycine max* (soybean), *Gossypium* spp. (cotton), *Helianthus annuus* (common sunflower), *Hyssopus officinalis* (hyssop), *Lactuca sativa* (lettuce), *Linum usitatissimum* (flax), *Medicago sativa* (alfalfa), *Nicotiana tabacum* (tobacco), *Pelargonium* (geranium) hybrids, *Petroselinum crispum* (parsley), *Pisum sativum* (pea), *Solanum tuberosum* (potato), *Spinacia oleracea* (spinach), *Trifolium pratense* (red clover), *Triticum aestivum* (wheat), *Vitis vinifera* (grape), *Zea mays* (maize), and *Zinnia elegans* (zinnia).

Known Vectors (or associated organisms)

Autographa gamma is not a known vector and does not have any associated organisms.

Known Distribution

Autographa gamma is widely distributed throughout all of Europe and eastward through Asia to India and China; it also occurs in North Africa (USDA, 1958).

Asia: Azerbaijan, China, India, Iran, Iraq, Israel, Japan, Kazakhstan, Korea, Saudi Arabia, Syria, Turkey, and Uzbekistan. **Europe:** Albania, Andorra, Austria, Belarus, Belgium, Bosnia and Herzegovina, Bulgaria, Channel Islands, Croatia, Cyprus, Czech Republic, Denmark (including Faroe Islands), Estonia, Finland, France (including Corsica), Germany, Greece (including Crete, Cyclades, and Dodecanese), Hungary, Iceland, Ireland, Italy (including Sardinia and Sicily), Latvia, Liechtenstein, Lithuania, Luxembourg, Macedonia, Malta, Moldova, Montenegro, the Netherlands, North Aegean Islands, Norway, Poland (including Azores and Madeira), Portugal, Romania, Russian Federation (including Kaliningrad Oblast), Serbia, Slovakia, Slovenia, Spain (including Balearic Islands and Canary Islands), Sweden, Switzerland, Ukraine, and United

Kingdom (including Gibraltar). **Africa:** Algeria, Egypt, Libya, and Morocco (Fibiger and Skule, 2011).

Pathway

Autographa gamma could potentially move through international trade. This species has been intercepted over 500 times at U.S. ports of entry. The top two countries of origin were the Netherlands (271) and Israel (192). Some other countries where infested material originated from include France (7), Italy (7), Germany (3), Zimbabwe (3) and Palestinian Territory (3). Most interceptions were on material for consumption (490), 14 were for non-entry, and 4 were for propagation. Interceptions were most common on *Origanum* sp. (47), *Trachelium* sp. (40), *Thymus* sp. (39), *Veronica* sp. (31), and *Bupleurum* sp. (24). Most interceptions occurred in permit cargo (468), followed by general cargo (13), baggage (13), stores (6), holds (4), miscellaneous (3), and quarters (1) (AQAS, 2012; queried August 6, 2012).

Potential Distribution within the United States

The likelihood and consequences of establishment by *A. gamma* have been evaluated in a pathway-initiated risk assessment. *Autographa gamma* was considered highly likely of becoming established in the United States if introduced. The consequences of its establishment for U.S. agricultural and natural ecosystems were also rated high (*i.e.*, severe) (Lightfield, 1997). A recent risk analysis by USDA-APHIS-PPQ-CPHST indicates that California and the southern United States have the greatest risk for *A. gamma* establishment based on host availability and climate within the continental United States. Establishment is precluded in many areas of the northwestern, north central, and northeastern United States.

Survey

CAPS-Approved Method: **CAPS-Approved Method:**

The CAPS-approved method is a trap and lure combination. The trap is a plastic bucket trap. The lure is effective for 28 days (4 weeks).

The lure is "*Autographa gamma*" Lure.

Trap Spacing: When trapping for more than one species of moth, separate traps for different moth species by at least 20 meters (65 feet).

Method Notes: This trap is also known as the unitrap. The trap has a green canopy, yellow funnel, and white bucket and is used with a dry kill strip. For instructions on using the trap, see Brambila et al. (2010).

Lure Placement: Placing lures for two or more target species in a trap should never be done unless otherwise noted here.

*For the most up-to-date methods for survey and identification, see Approved Methods on the CAPS Resource and Collaboration Site, at http://caps.ceris.purdue.edu/.

Literature-Based Methods:
Due to the migratory nature of this species, adult *A. gamma* can be observed every month from April to November, usually peaking in late summer (CABI, 2007).

Trapping: The sex pheromone, (*Z*)-7-dodecenyl acetate and (*Z*)-7-dodecenol in ratios from 100:1 to 95:5 (19:1) has been used to attract and monitor male flight of *A. gamma*. In field applications, the pheromone may be dispensed from rubber septa at a loading rate of 1 mg. Lures should be replaced every 30 days. Newly-emerged adult males of *A. gamma* are not attracted to the pheromone; 3-day old males are most responsive to the lure. The pheromone of *A. gamma* may also attract other Lepidoptera in the United States including: *Anagrapha ampla*, *Anagrapha falcifera*, *Autographa ampla*, *Autographa biloba*, *Autographa californica*, *Caenurgia* spp., *Epismus argutanus*, *Geina periscelidactyla*, *Helvibotys helvialis*, *Lacinipolia lutura*, *Lacinipolia renigera*, *Ostrinia nubilalis*, *Pieris rapae*, *Polia* spp., *Pseudoplusia includens*, *Rachiplusia ou*, *Spodoptera ornithogalli*, *Syngrapha falcifera,* and *Trichoplusia ni.*

Trapping is suggested in major truck farming areas. Traps should be placed within or on the edge of fields of the host crops. Traps should be suspended from stakes and placed at crop height and raised as the crop matures.

Visual survey: The USDA (1986) provides some considerations for visual inspections of host plants for the presence of eggs, larvae, or pupae. In general, eggs may be found on the lower and upper surfaces of leaves. Larvae are likely to be found, if left undisturbed, on leaves that have been skeletonized or that have holes in the interior. Pupae may be found on the lower leaf surface (USDA, 1986).

Not Recommended: Adult males and females have also been collected using Robinson black-light traps, but these traps attract moths non-discriminately. Such traps, placed 3 meters above the ground, have been used to successfully monitor the dynamics of *A. gamma* and other Noctuid moths. Sticky traps have been used, but are not recommended as pheromone traps are much more effective.

Key Diagnostics/Identification

CAPS-Approved Method: Confirmation of *A. gamma* is by morphological identification.

Screening aids are available at: http://caps.ceris.purdue.edu/webfm_send/548, http://caps.ceris.purdue.edu/webfm_send/633, and http://caps.ceris.purdue.edu/webfm_send/965.

*For the most up-to-date methods for survey and identification, see Approved Methods on the CAPS Resource and Collaboration Site, at http://caps.ceris.purdue.edu/.

Literature-Based Methods: Species are most reliably identified by close examination of the genitalia (Nazmi et al., 1980; USDA, 1986).

Easily Confused Pests

Several life stages of Noctuid pests can be confused with *A. gamma*. Of these, the most important species include: *Trichoplusia ni* (cabbage looper) (Fig. 6)*, Syngrapha celsa* (plain silver-Y or western conifer looper) (Fig. 7), *A. pseudogamma* (delicate silver-Y) (Fig. 8), and *A. californica* (alfalfa looper) (Fig. 9). All are already present in the continental United States. The other easily confused species are *Cornutiplusia circumflexa* (Essex Y), which is distributed in Europe, Asia, and Africa, and *Syngrapha interrogationis* (scarce silver Y), which is established in the United Kingdom (Venette et al., 2003). Adults of *A. gamma* are gray to grayish brown in color with a "Y mark or gamma [γ] on the forewing". See Nazmi et al. (1980) for a comparison of similarities and differences between closely related species.

Figure 6. Adult and larva of *Trichoplusia ni*. Photos courtesy of Keith Naylor and Extension Entomology, Texas A&M University.

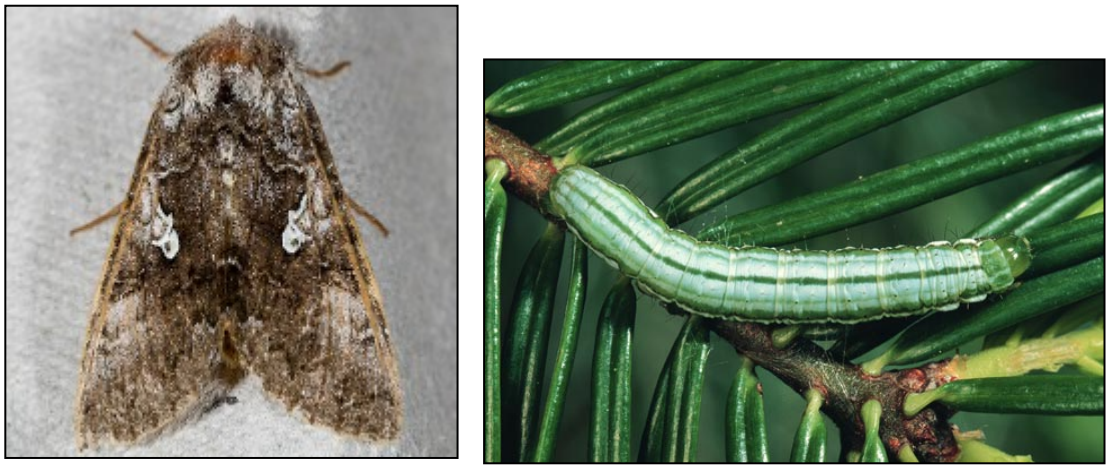


Figure 7. Adult and larva of *Syngrapha celsa*. Photos courtesy of John Cooper and Natural Resources Canada.

Figure 8. Adult of *Autographa pseudogamma.* Photo courtesy of Natural Resources Canada.

References

AQAS. 2012. *Autographa gamma* interceptions. Last accessed August 6, 2012 from: https://mokcs14.aphis.usda.gov/aqas/HomePageInit.do#defaultAnchor.

Brambila, J., L. Jackson, and R. Meagher. 2010. Plastic bucket trap protocol. USDA. 7 pp.

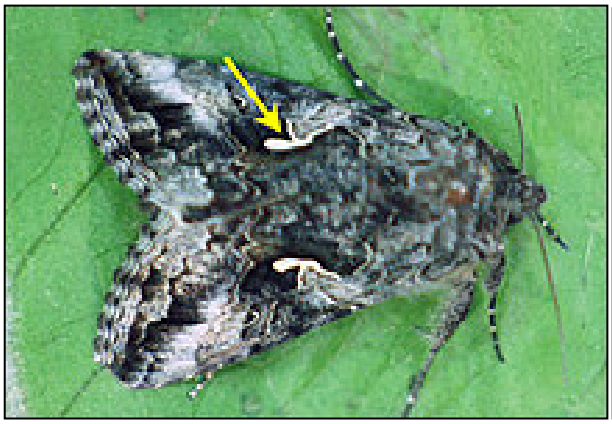


Figure 9. Adult and larva of *Autographa californicum.* Photos courtesy Franklin Dlott, U.C. Cooperative Extension, Monterey County.

Burgio, G., and S. Maini. 1995. Phenylacetaldehyde trapping of *Ostrinia nubilalis* (Hb.), *Autographa gamma* (L.) and hoverflies: trap design efficacy. Bollettino dell'Istituto di Entomologia della Universita degli studi di Bologna 49: 1-14.

CABI. 2007. Crop Protection Compendium Wallingford, UK: CAB International. http://www.cabi.org/compendia/cpc/.

Carter, D. J. and B. Hargreaves. (Illustrator)**.** 1986. A field guide to caterpillars of butterflies and moths in Britain and Europe. London, UK: Collins, 296 pp.

Dochkova, B. 1972. Some biological and ecological studies of the gamma moth. Rasteniev"dni Nauki 9(10):141-149.

Emmett, B. J. 1980. Key for the identification of lepidopterous larvae infesting Brassica crops. Plant Pathology 29(3):122-123.

Fibiger, M. and B. Skule. 2011. Fauna Europaea: *Autographa gamma*. In O. Karsholt and E. J. Nieukerken (eds.) Fauna Europaea: Lepidoptera, Moths, version 2.4. Accessed April 10, 2012 from: http://www.faunaeur.org**.**

Harakly, F. A. 1975. Biological studies on the loopers *Autographa-gamma* and *Cornutiplusia-circumflexa* Lepidoptera Noctuidae infesting truck crops in Egypt. Zeitschrift fuer Angewandte Entomologie 78(3):285-290.

Hill, D. S. 1987. Agricultural insect pests of the tropics and their control. 2nd edition. Cambridge, UK: Cambridge University Press.

Hill, J. K. and A. G. Gatehouse. 1992. Effects of temperature and photoperiod on development and pre-reproductive period of the silver Y moth *Autographa gamma* (Lepidoptera: Noctuidae). Bulletin of Entomological Research 82:335-341.

Honěk, A., V. Jarosik, Z. Martinkova, and I. Novak. 2002. Food induced variation of thermal constants of development and growth of *Autographa gamma* (lepidoptera: Noctuidae) larvae. European Journal of Entomology 99(2):241-252.

Ippolito, R. and P. Parenzan. 1985. *Hoplodrina ambigua* D. & S. (Lepidoptera - Noctuidae) and other noctuids on artichoke in Apulia. Entomologica 20:147-158.

Jones, F. G. W. and M. G. Jones. 1984. Pests of field crops. London, UK: Edward Arnold, 127-127.

Kaneko, J. 1993. Existence ratio of the silver Y moth, *Autographa gamma* (L.) after overwintering in a cabbage field at Sapporo, Japan. Annual Report of the Society of Plant Protection of North Japan No. 44:124-126.

Lightfield, J. 1997. Importation of leaves and stems of parsley, *Petroselinum crispum*, from Israel into the United States: qualitative, pathway-initiated pest risk assessment. Animal and Plant Health Inspection Service, US Department of Agriculture, Riverdale, MD.

Macaulay, E. D. M. 1972. Flight activity of *Plusia gamma i*n the laboratory. Entomologia Experimentalis et Applicata 15:387-391.

Macaulay, E. D. M. 1974. Lipid storage in the pre-imago and young adult *Plusia gamma*. Entomologia Experimentalis et Applicata 17:53-60.

Maceljski, M. and I. Balarin. 1972. On knowledge of polyphagy and its importance for the silver-Y moth (*Autographa gamma* L.). Acta Entomologica Jugoslavica 8:39-54.

Nazmi, N., E. El-Kady, A. Amin, and A. Ahmed. 1980-81. Redescription and classification of subfamily Plusiinae in Egypt. Bulletin de la Societe Entomologique d'Egypte 63: 141-162.

Novak, I. 1975. Critical number of *Autographa gamma* L. caterpillars (Lep., Noctuidae) on sugar-beet. Sbornik UVTI - Ochrana Rostlin 11:295-299.

Paulian, F., M. C. Mateias, T. Sapunaru, and I. Sandru. 1975. Results obtained in the study and control of the pests of lucerne crops in the Socialist Republic of Romania. Probleme de Protectia Plantelor 3:359-385.

Rashid, F. F., S. M. Hammad, and S. M. Hassan. 1971. The biology of *Autographa-gamma* in Alexandria region Lepidoptera Noctuidae. Bulletin de la Societe Entomologique d'Egypte 55:455-459.

Saito, O. 1988. Notes on larval hibernation of the alfalfa looper, *Autographa gamma* (Linnaeus) on cabbage plant in Sapporo, Hokkaido. *Annual Report of the Society of Plant Protection of North Japan*, No. 39: 217.

Steudel, W. 1963. Einige Beoachtungen bei de Aufzucht von Raupen der Gammaeule (*Plusia gamma* L.) im laboratorium. Nachrbl. Dt. Pflschutz Braunschweig 15:152-154.

Tarabrina, A. M. 1970. The silver Y moth in the Voronezh region. Zashchita Rastenii15:19.

Tyshchenko, V. P. and O. G. Gasanov. 1983. Comparative study of photoperiodic regulation of diapause and weight of pupae in several species of Lepidoptera. Zoologicheskii Zhurnal 62:63-68.

Ugur, A. 1995. Investigations on some biological aspects of *Autographa gamma* (L.) (Lepidoptera, Noctuidae). Türkiye Entomoloji Dergisi 19:215-219.

USDA. 1958. Coop. Economic Insect Report 8 (23). USA: USDA.

USDA. 1986. Pests not known to occur in the United States or of limited distribution No. 75: Silver Y Moth, pp. 1-16. APHIS-PPQ, Hyattsville, MD.

Venette, R. C., E. E. Davis, H. Heisler, and M. Larson. 2003. Mini Risk Assessment Silver Y Moth, *Autographa gamma* (L.)[Lepidoptera: Noctuidae]. Cooperative Agricultural Pest Survey, Animal and Plant Health Inspection Service, US Department of Agriculture. Available on line at: http://www.aphis.usda.gov/plant_health/plant_pest_info/pest_detection/downloads/pra/agammapra.pdf.

Chrysodeixis chalcites

Scientific Name
Chrysodeixis chalcites (Esper, 1789)

Synonyms:
Autographa chalcites, Chrysodeixis chalcytes, Noctua chalcites, Noctua chalcytes, Noctua chalsytis, Noctua questionis, Phalaena chalcites, Plusia buchholzi, Plusia chalcites, Plusia chalcytes, Plusia cohaerens, Phytometra chalcites

Note: Zhang (1994) states that Asian and Australian citations referring to *Chrysodeixis chalcites* actually refer to *C. eriosoma*, whereas Murillo et al (2013) state that the relationship between the two species needs further clarification. Lafontaine and Schmidt (2013) state that the only way to distinguish between the two species is by looking at geographic range, DNA, and pheromones.

Common Name(s)
Golden twin-spot moth, green garden looper, green looper, green semi-looper. groundnut semi-looper, tomato leafworm, tomato looper

Type of Pest
Moth

Taxonomic Position
Class: Insecta, **Order:** Lepidoptera, **Family:** Noctuidae

Reason for Inclusion in Manual
National threat; Requested by the CAPS community

Pest Description
Eggs: Eggs are pearly white to pale green and shiny. They are dome-shaped (hemispherical) with 28 to 32 vertical ribs from the micropyle to the base (Bretherton, 1983; Goodey, 1991). The eggs darken shortly before hatching (Harakly and Farag, 1975).

Larvae: Newly hatched larvae are dusky white, with head and thoracic shield blackish. The setae on the body are long and conspicuous, with thin,

Figure 1. Larva of *C. chalcites* (Steve Hatch, Bugwood.org).

longitudinal white lines along the sides; both become less obvious in the last instar. Soon after feeding, the larvae become light green in color (Harakly and Farag, 1975). Mature larvae are 34 to 38 mm (approx. 1 $^{5}/_{16}$ to 1 $^{1}/_{2}$ in) long, pale yellow-green with a glassy green to grey head edged with a black streak (Fig. 1). Above the spiracles on each side of the body is a thin dark green or black line stretching from the head to the seventh abdominal segment; below this is a thicker white line from the head to the tip of the anal proleg. Spiracles are black. The ventral region is speckled with white dots (Haggett, 1980; Bretherton, 1983; Passoa and Gilligan, 1995; Porter, 1997). Larvae have only three pairs of prolegs, instead of the normal five, resulting in the looping gait giving rise to some of the common names. Haggett (1980) provides a detailed description and color illustration of the final larval instar.

Figure 2. Pupa of *C. chalcites* (Paolo Mazzei, Bugwood.org)

Figure 3. Adult of *C. chalcites* (Perry Hampson, Bugwood.org).

Pupae: The pupa is 20 mm (approx. $^{3}/_{4}$ in) long, black in a white cocoon (Fig. 2), which turns brown then black (Harakly and Farag, 1975; Bretherton, 1983).

Adults: The adult wingspan is approximately 40 mm (approx. 1 $^{9}/_{16}$ in). The forewing is 15 to 17 mm (approx. $^{9}/_{16}$ to $^{11}/_{16}$ in), usually gold, although some individuals have more of a bronze color (Fig. 3). There are two oval silver spots on the forewings, although in some individuals these are united. The hindwing is paler than the forewing. There are two prominent crests on the thorax (Pinhey, 1979; Bretherton, 1983; Passoa, 1995a).

Further description of the life stages can be found in Goodey (1991) and Passoa (1995b).

Biology and Ecology

Chrysodeixis chalcites is a polyvoltine species, with up to eight or nine generations per year in Egypt (Rashid et al., 1971). After emergence, females mate then begin oviposition within 2 or 3 days (Gasim and Younis, 1989). Eggs are laid on upper and lower leaf surfaces at night, whilst females are on the wing. Females only briefly touch the leaf to deposit one, two, or a few eggs at a time. Eggs are very widely scattered in the crop (Linden, 1996). At 20°C (68°F) egg incubation lasts between 5 and 26 days (Gaumont and Moreau, 1961).

Reports in the literature show considerable variation in the number of eggs oviposited by *C. chalcites.* Harakly and Farag (1975) reported females lay from 14 to 281 eggs with a mean of 149. In contrast, Gasim and Younis (1989) reported the mean number of eggs laid per female to be much higher with 385, 640, and 405 eggs at 20, 25 and 30°C (68, 77, 86°F), respectively.

Gasim and Younis (1989) studied the development rate of *C. chalcites* eggs at three temperatures, 20, 25 and 30°C (68, 77, 86°F). The mean length of time between oviposition and egg hatch decreased with increasing temperature. At the lower temperature eggs took 4.5 days to hatch, at 25°C (77°F) they took an average of 3.0 days and at the upper temperature they took 2.0 days.

The majority of the larvae pass through six instars. Very few may have four or seven instars. First-instar larvae graze on the underside of leaves feeding on parenchyma. They can be quite difficult to detect. A larva will drop from the leaf and hang on a silken thread if disturbed (Goodey, 1991). During the second and third instars, the larva begins to roll the edges of the leaves together, and silken threads are spun on infested leaves (Rashid et al., 1971). Later instars eat through the leaves making infested leaves appear skeletonized. The last two larval instars are the most voracious feeders and will usually eat the entire leaf but may avoid the midrib, or other large veins. On legumes, they may excavate deep into pods, sometimes cutting them in two. At the optimal temperature of 25°C (77°F), there are six larval instars, each lasting approximately 2.5 to 3.5 days (Rashid et al., 1971; Harakly and Farag, 1975).

The mature larva stops feeding and enters a prepupal stage. It spins a cocoon within which it pupates. The cocoon is usually attached to the underside of a leaf but can be in the soil (Harakly and Farag, 1975). The pupal duration is affected by climatic conditions; being increased by lower temperatures (Harakly and Farag, 1975). Gaumont and Moreau (1961) reported that the pupal period lasted 15 to 26 days, although at the optimal temperature of 25°C (77°F) it averaged 8.8 days (Rashid et al., 1971).

Adults emerge at dusk and throughout the night and soon begin to fly and mate. Males are ready to mate just after emergence, but females usually mate 1 to 4 days after emergence (Harakly and Farag, 1975). Females reach peak activity on the 4th night after emergence between 0.5 to 2 hours into the scotophase. Peak male activity was observed in 5 to 8 day old males, 1 to 4 hours after the onset of scotophase (Snir et al.,

1986). They rest with their wings folded over their back like a tent. Adults are seminocturnal and usually avoid strong sunlight. Generations continually breed through the year with no diapause. There are nine generations per year in Egypt (Harakly and Farag, 1975).

Damage

Leaves may be skeletonized by larval feeding (Taylor and Kunjecku, 1983). Leaves may also be rolled with webbing (CABI, 2007). Frass may or may not be visible. The last two larval instars are the most voracious feeders and will usually eat the entire leaf but may avoid the midrib, or other large veins. It has been reported that on legumes *C. chalcites* may excavate deep into pods, sometimes cutting them in two. In tomato, this species can cause considerable damage to the leaves and vegetative parts of the plant. According to Harakly and Farag (1975), larvae never bore into fruit. However, Daricheva et al. (1983) found that larvae damaged both the leaves and fruits of tomatoes, leading to reduced yield. Napiórkowska-Kowalik and Gawłowska (2006) also recorded this species feeding on both leaves and fruits of tomatoes.

Pest Importance

Chrysodeixis chalcites is a polyphagous polyvoltine species that feeds on the foliage and fruit of vegetable, fruit, and ornamental crops. It is considered one of the most serious lepidopteran pests in many countries, although quantitative data measuring damage is lacking (CABI, 2007). Hill (1983) lists this species as a minor pest of *Brassica* spp., cotton, okra, tobacco, and tomato.

Larvae of *C. chalcites* feed on the leaves of solanaceous plants (EPPO, 2004). *Chrysodeixis chalcites* is the major pest of tomato in Israel during the growing season (Broza and Sneh, 1994) causing considerable damage to the leaves and vegetative parts of the plant, although it does not bore into the fruit (Harakly and Farag, 1975). Daricheva et al. (1983) state that larvae were found to damage leaves and fruits of tomatoes in greenhouses in Turkmenistan. Yield was reduced by 10 – 15%, especially in soft-fruited varieties. Most damage was attributed to mid- and late-instar larvae (Daricheva et al., 1983). It is reported as a serious pest in Bulgaria and Turkey (Loginova, 1992; Uygun and Ozgur, 1980) affecting tomato, cucumber and peppers. This species is also a major pest in Dutch greenhouses on both sweet pepper and tomato (van Oers et al., 2004). It is a serious pest of potato in Mauritius (Anon., 1984).

In Israel, it is also one of the most important noctuid pests of fodder crops, such as alfalfa and clover (Avidov and Harpaz, 1969). This species also feeds on alfalfa, maize, and soybean in Spain (Amate et al., 1998). In northern Italy, *C. chalcites* is one of the principal arthropod pests on soybean (Zandigiacomo, 1990). Yield loss can be seen if larvae feed during the soybean reproductive stages (Taylor and Kunjeku, 1983). It also attacks fields of artichokes (Ippolito and Parenzan, 1985). In Egypt, *C. chalcites* is considered the most serious of all semi-looper pests attacking field fruit and vegetables (Anon., 1984).

In protected cultivation, *C. chalcites* can occur at any time of the year and can reach high levels of infestation on vegetables and ornamental plants. It is reported as a serious pest in Bulgaria and Turkey (Loginova, 1992; Uygun and Ozgur, 1980) affecting tomato, cucumber, and peppers. *Chrysodeixis chalcites* is one of the four main noctuid pests of greenhouse crops in Sicily (Inserra and Calabretta, 1985) and a continual pest in greenhouses in the Netherlands (Vos and Rutten, 1995) and Belgium (Veire, 1983).

Known Hosts

"*Chrysodeixis chalcites* is highly polyphagous, feeding on many fruit, vegetable and ornamental crops, and weeds in many plant families including Acanthaceae, Asteraceae, Bignoniaceae, Boraginaceae, Brassicaceae, Convolvulaceae, Crassulaceae, Lamiaceae, Fabaceae, Malvaceae, Orchidaceae, Rosaceae, Scrophulariaceae, Solanaceae, Verbenaceae and Violaceae. It can be a pest of crops grown outdoors and in protection, including both shade and greenhouses" (CABI, 2007). An abbreviated record of hosts follows (CABI, 2007):

Major hosts

Glycine max (soybean), *Gossypium herbaceum* (short staple cotton), *Nicotiana tabacum* (tobacco), *Phaseolus* spp. (beans), *Phaseolus vulgaris* (common bean), *Solanum lycopersicum* (tomato), and *Solanum tuberosum* (potato).

Minor hosts

Anethum graveolens (dill), *Arachis hypogaea* (peanut), *Aster* spp., *Brassica oleracea* var. *botrytis* (cauliflower), *Brassica oleracea* var. *capitata* (cabbage), *Brassica* spp., *Capsicum annuum* (bell pepper), *Chrysanthemum indicum* (chrysanthemum), *Citrus* spp., *Coffea arabica* (coffee), *Cucumis sativus* (cucumber), *Cucurbita pepo* (zucchini), *Cynara cardunculus* subsp. *cardunculus* (=*C*. *scolymus*) (artichoke), *Dahlia* spp., *Dianthus* spp. (carnation), *Ficus carica* (fig), *Fragaria* spp., *Helianthus tuberosus* (Jerusalem artichoke), *Hippeastrum* hybrids (amaryllis), *Lactuca sativa* (lettuce), *Medicago sativa* (alfalfa), *Musa* spp. (banana), *Pelargonium* spp. (geranium), *Salvia officinalis* (common sage), *Stachytarpheta jamaicensis* (Jamaica vervain), *Trifolium repens* (white clover), *Triticum aestivum* (wheat), and *Zea mays* (corn).

Wild hosts

Echium vulgare (viper's-bugloss), *Marrubium* spp. (horehound), *Teucrium scorodonia* (wood germander), and *Urtica dioica* (stinging nettle).

This species has been found on tomato and green beans in Canada (Murillo et al., 2013).

Pathogens or Associated Organisms Vectored

Chrysodeixis chalcites is not a known vector and does not have any associated organisms.

Known Distribution

Distribution information for *C. chalcites* is difficult to ascertain because older records are now considered to be two species, *C. chalcites* (Africa, Mediterranean, and the Middle East) and *C. eriosoma* (Asian tropics, Australia, and New Zealand).

Chrysodeixis chalcites is primarily distributed between 45°N and 35°S, from southern Europe and the Mediterranean and the Middle East to Africa (CABI, 2007; Murillo et al., 2013)

Africa: Algeria, Angola, Cameroon, Cape Verde, Comoros, Congo, Cote d'Ivoire, Egypt, Gambia, Guinea, Kenya, Libya, Madagascar, Malawi, Mauritius, Morocco, Mozambique, Nigeria, Reunion, Saint Helena, Sao Tome and Principe, Senegal, Seychelles, Sierra Leone, South Africa, Tunisia, Uganda, Zambia, and Zimbabwe; **Europe:** Albania, Austria, Belgium, Bosnia and Herzegovina, Bulgaria, Croatia, Cyprus, Czech Republic, Denmark, France (including Corsica), Germany, Gibraltar, Greece (including Crete, Cyclades Islands, and Dodecanese Islands), Hungary, Italy (including Sardinia and Sicily), Macedonia, Malta, Moldova, Netherlands, Poland, Portugal (including Azores and Madeira), Romania, Serbia, Slovenia, Spain (including Balearic Islands and Canary Islands), Sweden, Switzerland, Ukraine, and the United Kingdom; **Middle East:** Iran, Iraq, Israel, Jordan, Lebanon, Syria, Turkey and Turkmenistan; **North America:** Canada (Daricheva et al., 1983; CABI, 2007; Fibiger and Skule, 2011).

"*Chrysodeixis chalcites* immigrants from North Africa or southern Europe, borne on strong southerly winds, are sometimes recorded in central and northern Europe (Austria, Denmark, Germany, Sweden, Switzerland and the United Kingdom) in the late summer or autumn (Jor, 1973; Bretherton, 1983; Hachler et al., 1998; Palmqvist, 1998, 2002). There are about 50 records of *C. chalcites* as a migrant to the UK between 1943 and 1990 (Bretherton, 1983). Outdoor breeding populations occur in Europe as far north as northern Spain and northern Italy. No successful breeding is reported outdoors in northern Europe" (CABI, 2007).

"Lempke (1982) and Vos and Rutten (1995) noted that *C. chalcites* is present all year round in greenhouses in the Netherlands. Veire (1993) reported populations established in greenhouses in Belgium. However, there is no evidence that *C. chalcites* can overwinter outdoors in the Netherlands (Lempke, 1982) or elsewhere in northern Europe" (CABI, 2007).

In January 2006, a detection of either *C. chalcites* or *C. eriosoma* was made in two vegetable production greenhouses in Delta, British Columbia Canada. The pest has been found in subsequent surveys, delimitation surveys continue, and the area has been put under compliance agreement to prevent further spread. As of May 2007, no further detections have been made and the greenhouses have been released from quarantine. See the following pest alerts for further information:

http://www.pestalert.org/oprDetail.cfm?oprID=187,
http://www.pestalert.org/oprDetail.cfm?oprID=237,

http://www.pestalert.org/oprDetail.cfm?oprID=264.

This species was recently found in the counties of Essex and Chatham-Kent, southwestern Ontario, Canada in tomato and bean crops. It is unknown whether the moth overwinters in southwestern Ontario or if it overwinters further south, migrating to Ontario during the spring. Murillo et al. (2013) suggests that this species is most likely established in surrounding field tomato crops in the United States, specifically in Michigan, Ohio, and New York (Murillo et al., 2013). However, this has not been confirmed by surveys conducted in the United States.

Note: Lafontaine and Schmidt (2013) state that this species is present in Michigan; however, this record cannot be verified. The authors cited (Murillo et al., 2013) state that they believe *C. chalcites* is present in Michigan, but do not provide any records of its occurrence there. There are currently no known records of this species from Michigan.

Pathway

There have been over 300 interceptions of this pest from 1984 to the present. This species has been intercepted on material originating from multiple countries. The most interceptions have occurred on material originating from the Netherlands (229) and Israel (28). The species has been intercepted on many different plant species including: *Lycopersicon* sp. (57), *Capsicum* sp. (22), *Bupleurum* sp. (12), *Ocimum* sp. (11), *Cymbidium* sp. (11), *Celosia* sp. (10), *Gloriosa* sp. (10), *Hydrangea* sp. (12), and *Mentha* sp. (10). *Chrysodeixis chalcites* has also been intercepted on non-host material including: aircraft (3), cargo, containers, and tiles (1 each) (AQAS, 2012; queried June 28, 2012). This species has also been intercepted by California (CDFA, 2005).

This species can easily travel through international trade. It has been found in *Pelargonium* from Germany to Hungary, on *Chrysanthemum morifolium* and *Pelargonium* from the Canary Isles to the UK, on bananas from the Canary Isles to Italy as well as from the Netherlands to the UK, and on *Impatiens* from Israel to the UK (EPPO, 1998; EPPO, 2000; reviewed in CABI, 2007). Larvae have been frequently imported into Britain on chrysanthemum (Carter, 1984) and particular attention should be paid to this commodity.

Passoa (2007) states that interceptions originating from Hawaii are *C. eriosoma*, *Pseudoplusia includens* if originating from the New World, and either *C. chalcites* or *C. eriosoma* (depending on locality) if originating from the Old World.

Potential Distribution within the United States

In 1995, a specimen of *C. chalcites* was found on *Pelargonium* (geraniums) in a greenhouse in Ohio. This pest is not known to be established in the United States (CABI, 2007).

This species is likely to establish in the United States wherever host material and suitable climate are found. Based on its native distribution and migratory behavior

(CABI, 2012), it would likely find suitable climate for establishment in Plant Hardiness Zones 6 to 11 and may migrate farther north during warm temperature seasons. Further investigation is needed to determine where exactly in the continental United States it could overwinter. This species can also establish in areas with unfavorable climates by establishing in greenhouses as it has in parts of northern Europe (CABI, 2007).
A recent risk analysis by USDA-APHIS-PPQ-CPHST shows that permanent populations can occur throughout the southern portion of the United States. Although establishment cannot occur in excluded areas, temporary populations can occur due to migration of the species.

Survey

CAPS-Approved Method*: The CAPS-approved method is a trap and lure combination. The trap is a wing trap. The lure is effective for 28 days (4 weeks). The lure dispenser type is a rubber septum.

Any of the following Trap Product Names in the IPHIS Survey Supply Ordering System may be used for this target:

Wing Trap Kit, Paper
Wing Trap Kit, Plastic

The Lure Product Name is "*Chrysodeixis chalcites* Lure."

IMPORTANT: Do not place lures for two or more target species in a trap unless otherwise recommended.

Trap spacing: When trapping for more than one species of moth, separate traps for different moth species by at least 20 meters (65 feet).

*For the most up-to-date methods for survey and identification, see Approved Methods on the CAPS Resource and Collaboration Site, at http://caps.ceris.purdue.edu/.

Literature-Based Methods:
Trapping: A pheromone for trapping *C. chalcites* is available. The lure (a 5:1:1 mixture of (Z)-7-dodecenyl acetate, (Z)-9-tetradecenyl acetate, and (Z)-9-dodecenyl acetate) (Dunkelblum et al., 1987) is dispensed from a rubber septum and the recommended replacement interval is every 4 weeks.

Dunkelblum and Mazor (1993) captured this species using a lure containing 2,500µg of Z7-12:AC, 50µg of Z9-12:AC, and 2,000µg of Z9-14:Ac.

This species was caught in Ontario, Canada, using universal moth traps baited with pheromone lures for *Trichoplusia ni* (Murillo et al., 2013). The lures for both species have similar components (Dunkelblum and Mazor, 1993).

Survey Site Selection:

Traps should be placed in areas where host material is present. This can include host crop fields and greenhouses with host material, among other places.

Trap placement:
Traps should be placed near the highest point of the plant, about 1 m (approx. 3 1/4 ft) from the ground on supporting posts or higher if the crop is higher.

Time of year to survey:
This species can be present year-round in a greenhouse setting. In warmer climates, this species continually breeds throughout the year with no diapause. In more northern climates, adults are more likely to be found during warmer periods. This species is known to migrate to northern areas in the spring.

Visual survey: Leaves should be examined on upper and lower surfaces for larvae. Damage symptoms, such as skeletonized or rolled leaves with webbing may be easier to detect (CABI, 2007).

Not recommended: This species is sometimes found in light traps (Deans, 2005; Kimber, 2008); however this survey method is not species specific.

Key Diagnostics/Identification

CAPS-Approved Method*: Confirmation of *Chrysodeixis* (*chalcites* or *eriosoma*) is by morphological identification. Identification of adults requires dissection of the male genitalia; use Passoa (1995a) as an aid. Molecular analysis is required for identification at the species level.

Several screening aids are available for this species, including a field screening aid (http://caps.ceris.purdue.edu/webfm_send/2126), a diagnostic aid with non-targets (http://caps.ceris.purdue.edu/webfm_send/2129), and an identification aid with non-targets (http://caps.ceris.purdue.edu/webfm_send/2130).

A screening aid for CAPS target Noctuidae (males), including *C. chalcites*, can be found in Passoa (2009). Adult genitalia are shown in Passoa (1995a) and Passoa and Gilligan (1995).

*For the most up-to-date methods for survey and identification, see Approved Methods on the CAPS Resource and Collaboration Site, at http://caps.ceris.purdue.edu/.

Easily Confused Pests

In Africa and Europe, *C. chalcites* may be confused with *C. acuta*, although *C. acuta* is larger and has a more pointed forewing. The silver spots are also larger (Bretherton, 1983). In the United States, immigrant *C. chalcites* appear similar to *Pseudoplusia includens* (soybean looper), but the male genitalia are quite different (Passoa, 1995a). Larvae should be reared to adulthood to confirm their identity (Passoa, 1995a).

This species is morphologically similar to *C. eriosoma*. EPPO (2001) states that typical "silver Y" wing markings can distinguish *C. eriosoma* from *C. chalcites*, while Murillo et al. (2013) state that this species cannot be reliably distinguished with morphological techniques; the only way to distinguish between the two species is by looking at geographic range, DNA, and pheromones. Murillo et al. (2013) state that these two species could potentially be the same.

A key to genera of Noctuoidea (subfamily Plusiinae) found in the United States and Canada for both adults and larvae can be found in Lafontaine and Poole (1991).

References

Amate, J. P. Barranco, and T. Cabello. 1998. Identification of larvae of the principal noctuid pest species in Spain (Lepidoptera: Noctuidae). Boletin de Sanidad Vegetal, Plagas, 24(1):101-106.

Anon. 1984. Annual report 1983. Mauritius Sugar Industry Research Institute, Reduit, Mauritius, 77 pp.

AQAS. 2012. AQAS database, *Chrysodeixis chalcites* interceptions. Last accessed June 28, 2012 from: https://mokcs14.aphis.usda.gov/aqas/HomePageInit.do.

Avidov, Z. and I. Harpaz. 1969. Plant pests of Israel. Jerusalem, Israel: Israel Universities Press.

Bretherton, R. F. 1983. Noctuidae and Agaristidae. In: J. Heath and E. A. Maitland (eds.) The moths and butterflies of Great Britain and Ireland. Volume 10, Noctuidae (Part ii) and Agaristidae. Colchester, UK: Harley Books.

Broza, M. and B. Sneh. 1994. *Bacillus thuringiensis* spp. *kurstaki* as an effective control agent of lepidopteran pests in tomato fields in Israel. Journal of Economic Entomology, 87:923-928.

CABI. 2007. Crop Protection Compendium. Wallingford, UK: CAB International. www.cabicompendium.org/cpc.

CABI. 2012. Crop Protection Compendium. Wallingford, UK: CAB International. www.cabicompendium.org/cpc.

Carter, D. 1984. Pest Lepidoptera of Europe. Dr. W. Junk Publishers. Boston, MA.

CDFA. 2005. California Plant Pest and Disease Report. 22(1): 78 pp.

Daricheva, M. A., Z. F. Klyuchko, and A. Sakchiev. 1983. A pest of tomatoes (abstract only). Zashchita-Rasteni 7: 44-45.

Deans, 2005. An unprecendented influx of golden twin-spot *Chrysodeixis chalcites* into Suffolk. In T. Prichard (ed.) Suffolk Moth Group Newsletter, Issue 28, Autumn 2005.

Dunkelblum, E. and M. Mazor. 1993. Chemical characterization and species specificity of sex pheromones of Plusiinae moths in Israel. Archives of Insect Biochemistry and Physiology 22: 413-424.

Dunkelblum, E., R. Snir, S. Gothlie, and I. Harpaz. 1987. Identification of sex pheromone components from pheromone gland volatiles of the tomato looper, *Plusia chalcites*. Journal of Chemical Ecology 13(5): 991-1003.

EPPO. 1998. EPPO Reporting Service 1998, No. 4. 98/077.

EPPO. 2000. EPPO Reporting Service 2000, No. 10. 2000/164.

EPPO. 2001. EPPO Reporting Service 2001, No. 10. 2001/182.

EPPO. 2004. Good plant protection practice, solanaceous crops under protected cultivation. EPPO Bulletin 34: 65-77.

Fibiger, M. and B. Skule. 2011. Fauna Europaea: *Chrysodeixis chalcites*. Fauna Europaea version 2.5. Accessed March 27, 2013 from: http://www.faunaeur.org.

Gasim, G. Y. and H. T. Younis. 1989. Biological studies on tomato leafworm *Plusia chalcytes* L. (Noctuidae: Lepidoptera) under effect of constant temperatures. Mesopotamia Journal of Agriculture, 21:325-334.

Gaumont, R. and R. Moreau. 1961. Observations on the bionomics of *Plusia chalcites* Esp. (Lepidoptera, Noctuidae). Revue de Zoologie Agric., 60:31-36 (in French).

Goodey, B. 1991. *Chrysodeixis chalcites* - observations on the life cycle in captivity. The Entomologists Record, 103:111-118.

Hachler, M., M. Jermini, and R. Brunetti. 1998. [Two new harmful noctuids on tomatoes in glasshouse in South and Western Switzerland.] Revue Suisse de Viticulture, d'Arboriculture et d'Horticulture, 30:281-285 (in French).

Haggett, G. M. 1980. Larvae of the British Lepidoptera not figured by Buckler, Part XI. Proceedings and Transactions of the British Entomological and Natural History Society 13(3/4): 95-105.

Harakly, F. A. and S. S. Farag. 1975. Biological studies on the tomato looper *Chrysodeixis chalcytes* (Esper) in Egypt. Bulletin de la Societe Entomologique d'Egypte, 59:295-299.

Hill, D. S. 1983. Agricultural Insect Pests of the Tropics and Their Control. Cambridge University Press, Cambridge.

Inserra, S. and C. Calabretta. 1985. Attack by noctuids: a recurring problem in greenhouse crops of the Ragusa coast. Tecnica Agricola, 37:283-297.

Ippolito, R. and P. Parenzan. 1985. Hoplodrina ambigua D. & S. (Lepidoptera - Noctuidae) and other noctuids on artichoke in Apulia. Entomologica, 20:147-158.

Jor, J. 1973. *Plusia chalcites* Esp., found on Funen (Lep., Noctuidae). Entomologiske Meddelelser, 41:187-188.

Kimber, I. 2008. 2428 Golden Twin-spot *Chrysodeixis chalcites.* UKmoths. Accessed October 6, 2008 from: http://ukmoths.org.uk/show.php?id=1638.

Lafontaine, J. D. and B. C. Schmidt. 2013. Additions and corrections to the check list of the Noctuoidea (Insecta, Lepidoptera) of North America north of Mexico. ZooKeys 264: 227-236.

Lafontaine, J. D. and R. W. Poole. 1991. The Moths of America North of Mexico. Fascicle 25.1. Noctuoidea, Noctuidae (Part). The Wedge Entomological Research Foundation. 182 pp.

Lempke, B. J. 1982. [Interesting observations and catches of Lepidoptera in 1980]. Entomologische Berichten (Amsterdam), 42:116-119.

Linden, A, van der. 1996. [On the caterpillar hunt]. Groenten und Fruit/Glasgroenten.

Loginova, E. 1992. Some new pests of glasshouse crops in Bulgaria and their control by an IPM programme. Bulletin OEPP/EPPO Bulletin, 22:357-361.

Murillo, H., D. W. A. Hunt, and S. L. VanLaerhoven. 2013. First records of *Chrysodeixis chalcites* (Lepidoptera: Noctuidae: Plusiinae) for east-central Canada. Canadian Entomologist 145: 1-5.

Napiórkowska-Kowalik, J. and J. Gawłowska. 2006. *Chrysodeixis chalcites* (Esper 1789) (*Lepidoptera*, *Noctuidae*, *Plusiinae*) a new pest on tomatoes in glasshouses in Poland (abstract only). Progress in Plant Protection 46(1): 295-299.

Palmqvist, G. 1998. Remarkable records of Macrolepidoptera in Sweden 1997. Entomologisk Tidskrift, 119:13-27.

Palmqvist, G. 2002. Remarkable records of Macrolepidoptera in Sweden 2001. Entomologisk Tidskrift, 123:53-63.

Passoa, S. 1995a. Male genitalia of *Chrysodeixis chalcites*. http://caps.ceris.purdue.edu/webfm_send/97.

Passoa, S. 1995b. *Chrysodeixis chalcites* (Esper). PPQ Fact Sheet 35, USDA-APHIS.

Passoa, S. 2007. Quarantine Significant Lepidoptera of Concern to the Southern United States. USDA-APHIS-PPQ. p. 51.

Passoa, S. 2009. Screening key for CAPS target Noctuidae in the Eastern and Midwestern United States (males). Lab Manual for the Lepidopteria Identification Workshop. University of Maryland, March 2009.

Passoa, S. and T. Gilligan. 1995. *Chrysodeixis chalcites* (Esper) (Lepidoptera: Noctuidae). PPQ Alert List Fact Sheet 35, USDA-APHIS. http://caps.ceris.purdue.edu/webfm_send/96.

Pinhey, E. C. G. 1979. Moths of Southern Africa, descriptions and color illustrations of 1183 species. Rotterdam, The Netherlands: AA Balkema.

Porter, J. 1997. The color identification guide to caterpillars of the British Isles: Macrolepidoptera. The color identification guide to caterpillars of the British Isles: Macrolepidoptera., xii + 275 pp

Rashid, F. F., S. M. Hammad, and S. M. Hassan. 1971. The biology of *Autographa chalcites* L. in Alexandria region (Lepidoptera: Noctuidae). Bulletin de la Societe Entomologique d'Egypte, 55:419-426.

Snir, R., E. Dunkelblum, S. Gothilf, and I. Harpaz. 1986. Sexual behavior and pheromone titre in the tomato looper, *Plusia chalcites* (Esp). (Lepidoptera:Noctuidae). Journal of Insect Physiology 32(8): 735-739.

Taylor, D. E., and E. Kunjeku. 1983. Development of an economic threshold for semi-loopers (Lepidoptera: Noctuidae) on soya-beans in Zimbabwe. Zimbabwe J. Agric. Res. 2) 1: 89-100.

Uygun, N. and F. Ozgur. 1980. Identification of pests of greenhouse vegetables in the Icel and Adana regions, and the effects of endosulfan smoke tablets and pirimicarb on *Myzus persicae* (Sulz.). Turkiye Bitki Koruma Dergisi, 4:185-192.

Van Oers, M. M., E. A. Herniou, M. Usmany, G. J. Messelink, J. M. Vlak. 2004. Identification and characterization of a DNA photolyase-containing baculovirus from *Chrysodeixis chalcites.* Virology 330: 460-470.

Veire, M. van de. 1993. First observations in glasshouse sweet peppers in Belgium and laboratory rearing of the parasitic wasp *Eulophus pennicornis* (Hym.: Eulophidae). Entomophaga, 38:61-62.

Vos, R. de, and A. L. M. Rutten. 1995. Migrating Lepidoptera in 1992 (fifty-third report). Entomologische Berichten, 55:37-46.

Zandigiacomo, P. 1990. The principal pests of soybean in north-eastern Italy in 1989. Informatore Fitopatologico, 40:55-58.

Zhang, B. C. 1994. Index of Economically Important Lepidoptera. Wallingford, UK: CAB International

Cydia fabivora

Scientific Name
Cydia fabivora Meyrick

Synonyms:
Eulia prosecta, *Laspeyresia fabivora*, *Laspeyresia leguminis*

Common Name(s)
Tortricid moth

Type of Pest
Moth

Taxonomic Position
Class: Insecta, **Order:** Lepidoptera, **Family:** Tortricidae

Reason for Inclusion in Manual
National Threat

Pest Description
Eggs: Average size 0.89 x 0.66 mm, ventrally flattened, pale yellow initially, covered with a raised hexagonal reticulation. Red spots appear below the chorion within 24 hours of oviposition. Eventually these coalesce, and then the whole egg is red (Stanley and Sanchez, 1990).

Larvae: Neonate larvae light orange in color; later instars have cream colored bodies and prominent prothoracic shields and heart-shaped heads. Fifth-instar larvae are approximately 18 mm long (Stanley and Sanchez, 1990).

Pupae: Pupae have two conspicuous transverse bands of spines on abdominal sterna 3 to 9. Females are larger and heavier than males (Stanley and Sanchez, 1990).

Adults (Fig. 1): Antenna rather stout, somewhat compressed laterally; pubescence very short; scales pale gray to clay color, thicker and more abundant on antennal basal fourth. Labial palpus with second segment extended almost too top of face, ashy gray, scales fuscous or pale brown, white tipped; paler on inside side, sometimes reddish or rust-colored suffusion on upper edge of third segment. Head and throrax cinereous, mid-thorax darker (Heinrich, 1943).

Male: Wing expanse 16-20 mm. Forewing rough scaled, with several small clumps of slightly raised scales between base and outer third, projected fan of scales along inner margin for slight distance from base; general color drab gray, markings, when distinguishable, blackish fuscous (more or less suffused in some

Figure 1. *C. fabivora* adult moth. Photo courtesy of Lynn Meijerman (CABI, 2007).

specimens and completely in few); subtornal spot irregularly shaped, blackish fuscous; subapical bar blackish fuscous, divided at middle, with one arm extended to mid termen, other downward to about M3, in some specimens arms enclosing pale yellowish or orange sport; apical area beyond subapical bar pale, gray, yellowish, or orange; outer third of cell with dark fuscous sport, sometimes extended to costa and inner margin to form, dark, transverse fascia; strongly marked specimens with obscure, pale, smooth sport just beyond cell in area between CuI and R4, edged by slightly raised scales; cilia pale drab gray, in some specimens more or less suffused with reddish ochreous. Hindwing grayish brown to brown; cilia paler. Genetalia: Valva with cucullus elongate triangular, inner (lower) margin densely spined; neck incurvation deep. Aedeagus long, slender, curved; cornuti a cluster of short, thin, flattened spines (Heinrich, 1943).

Female: Essentially like male in color and markings; wing expanse 18-22 mm, antenna more slender, hindwing darker. Genitalia: Ductus bursae sclerotized from about one third of its length from junction with corpus bursa and width small sclerotized collar at middle. Ductus seminalis from ductus bursae just beyond sclerotized part of tube. Bursa weakly granulate, especially toward ductus bursae. Signa slender, sharp, thornlike, with broad bases. Membrane behind and caudad of gential opening with pair of elongate, triangular, sclerotized plates (Henrich, 1943).

Biology and Ecology

Adults are nocturnal and emergence takes place at night or occasionally during the morning. Under laboratory conditions, adults copulate approximately 48 hours after emergence. Females start to oviposit almost immediately afterward, continuing for 2 to 4 days (Stanley and Sanchez, 1990). Eggs are deposited (glued to the substrate) singly, or occasionally in small groups of 2 to 4, on the stems of the host plant, on the abaxial and adaxial sides of the leaves, on the petioles, and on the pods. Mean egg production was 44 per female. Generally, the preferred oviposition site shifts from leaves to reproductive structures over the course of plant development. The eggs hatch 4 to 5 days after they are deposited. Larvae have five instars (Stanley and Sanchez, 1990).

First-instar larvae attacking plants in vegetative stages begin by perforating the stem, often at the axil of the petiole, causing desiccation of the leaf. Otherwise, the neonate larva penetrates the stem directly, leaving a short encircling mine. The larva spins a silken support and remains in the same stem until development is completed. Boring of the main stem kills small plants. Attacked pods can be identified by characteristic short brownish mines indicating where the first-instar larva has passed through to the seed. Silken support webs are also spun inside the pods, and one or two seeds are consumed, depending on seed maturity. Pupation occurs in thin cocoons, at the site of larval development in both stems and pods. The pupal stage lasts 8 to 11 days. The average time from oviposition to emergence of adults is 29.2 days (CABI, 2007).

Pest Importance

Cydia fabivora is an important pest of beans in South America. Larvae cause considerable damage by boring into the stems and pods (Clarke, 1958). The pest causes stunting of host plants and a reduction in yield. Late-maturing and late-planted varieties suffer the greatest damage from this pest. According to Stansly and Sanchez (1990), the pest could potentially build up a large population as it is able to complete three generations per crop cycle.

Symptoms/Signs

Cydia fabivora feeds on stems, shoots, floral buds and pods of host plants. When young plants are attacked by a larva boring into the main stem, the plant may die. Attacked pods can be identified by characteristic short brownish mines, indicating where the first-instar larva has passed through to the seed. Silken support webs are also spun inside the pods, and one or two seeds are often consumed (CABI, 2007).

Cydia fabivora larvae fed on soybean and *Phaseolus vulgaris* in the field. The larvae also damage the terminal shoots, passing from one shoot to another as new shoots are formed, later moving into the flower buds and causing subsequent pod loss. Severely damaged plants may become stunted with few pods produced. Late-planted soybeans seem to withstand less damage than earlier planted crops (Foerster, 1978.)

Known Hosts

Major hosts

Glycine max (soybean), *Phaseolus lunatus* (lima bean), *Phaseolus vulgaris* (common bean), and *Vicia fabia* (broad bean).

Known Vectors (or associated organisms)

Cydia fabivora is not a known vector and does not have any associated organisms.

Known Distribution

Cydia fabivora is widespread throughout Central and South America.

Central America: Costa Rica, El Salvador, Panama. **North America:** Mexico. **South America:** Brazil, Columbia, Ecuador, Peru, and Venezuela (CABI, 2007).

Pathway

Cydia fabivora could potentially move through international trade. There have only been 3 interceptions of this species at U.S. ports of entry. Interceptions originated from Ecuador, Guatemala, and Jamaica (which is not known to have this species). All interceptions occurred on *Phaseolus* spp., two of which were found on baggage and meant for consumption. The other interception was found in stores for non-entry. *Cydia* sp. interceptions have occurred 8,544 times, 12 of which occurred on known host material from areas where *C. fabivora* is known to occur (AQAS, 2012; queried September 11, 2012).

Potential Distribution within the United States

Information is not available at this time. *Cydia fabivora* may, however, enter a country inside the stems, pods, shoots, and buds of its hosts. Specimens identified as *C. fabivora* have been intercepted from *Phaseolus* spp. three times and *Vicia faba* (seeds) once since 1975 from various Central and South American countries (USDA, 1987).

Survey

CAPS-Approved Method*: Has not been evaluated at this time.

*For the most up-to-date methods for survey and identification, see Approved Methods on the CAPS Resource and Collaboration Site, at http://caps.ceris.purdue.edu/.

Literature-Based Methods:

Visual Survey: Inspect leaves, stems and pods for eggs. Cut suspect stems, pods, shoots, and buds and examine for larvae and pupae. Also inspect inside of stems for tunneling by larvae. Short encircling mines where larvae entered the stems might be visible from outside. Inspect pods for presence of larvae, pupae or webbing internally. Attacked pods may often be identified from the outside by short brownish mines where the first-instar larva passed through to the seed**.**

Key Diagnostics/Identification

CAPS-Approved Method*: Has not been evaluated at this time.

*For the most up-to-date methods for survey and identification, see Approved Methods on the CAPS Resource and Collaboration Site, at http://caps.ceris.purdue.edu/.

Literature-Based Methods: Confirmation of *C. fabivora* is by morphological identification. Wing color and/or dissection of genitalia are required to identification of *C. fabivora*. For identification, submit suspect adult specimens, pinned and labeled to a diagnostic authority. Preserve larvae and pupae in alcohol (USDA, 1987).

A key to the adults and larvae can be found in TortAI (Tortricids of Agricultural Importance): http://idtools.org/id/leps/tortai/keys.html.

Easily Confused Pests

Cydia fabivora closely resembles *Crocidosema aporema*. *Crocidosema aporema* attacks young leaflets, while the *C. fabivora* is commonly found on fully developed leaves.

Cydia fabivora and another tortricid moth, *C. torostoma* might easily be confused. *C. torostoma* averages a little smaller than *C. fabivora*. Forewing of *C. torostoma* has a darker ground color and lacks a light gray, yellowish, or orange apical spot. Also hindwing base of *C. torostoma* is light colored, that of *C. fabivora*, wholly dark. In male genetalia, the incurvation of valva is shallow and the aedeagus is straight in *C. torostoma*, but in *C. fabivora*, the incurvation is deep and the aedeagus is bent. Female genitalia of *C. torostoma* has a well-sclerotized antrum and a sclerotized Y-shaped lamella postvaginalis; in *C. fabivora*, the antrum is not sclerotized and the lamella postvaginalis consists of two elongate, sclerotized divergent areas (Clark, 1972; USDA).

References

AQAS. 2012. *Cydia* spp. interceptions. Last accessed September 11, 2012 from: https://mokcs14.aphis.usda.gov/aqas/HomePageInit.do#defaultAnchor.

CABI. 2007. Crop Protection Compendium Wallingford, UK: CAB International. www.cabicompendium.org/cpc.

Clarke, J. F. G. 1958. The correct name for a pest of beans (Lepidoptera, Olethreutidae). Proceeding of the Entomological Society of Washington 60(4): 187.

Clarke, J. F. G. 1972. Two pests of bean from tropical America (Lepidoptera, Olethreutidae). Proceeding of the Entomological Society of Washington 74(4): 467-471.

Foerster, L. A. 1978. Chemical control of soybean pests in Brazil. Van Emden, H. F.: Pests of grain legumes: Ecology and control: 253-258.

Heinrich, C. 1943. A new species of *Laspeyresia*, a bean pest from tropical America (Lepidoptera: Olethreutidae). Proceeding of the Entomological Society of Washington 45(3): 71-73.

Stansly, P. A. and W. Sanchez. 1990. Biology and oviposition behavior of *Cydia fabivora* (Lepidoptera: Tortricidae) in soybean on Ecuador's coastal plain. Florida Entomologist 73: 219-225.

USDA. 1987. Pest not known to occur in the United States or of limited distribution. No. 86: A Tortricid Moth.

Diabrotica speciosa

Scientific name
Diabrotica speciosa Germar

Synonyms:
Diabrotica amabilis, Diabrotica hexaspilota, Diabrotica simoni, Diabrotica simulans, Diabrotica vigens, and *Galeruca speciosa*

Common names
Cucurbit beetle, chrysanthemum beetle, San Antonio beetle, and South American corn rootworm

Type of pest
Beetle

Taxonomic position
Class: Insecta, **Order:** Coleoptera, **Family:** Chrysomelidae

Reason for Inclusion in Manual
CAPS Target: AHP Prioritized Pest List - 2010

Pest Description
Diabrotica speciosa was first described by Germar in 1824, as *Galeruca speciosa*. Two subspecies have been described, *D. speciosa vigens* (Bolivia, Peru and Ecuador), and *D. speciosa amabilis* (Bolivia, Colombia, Venezuela and Panama). These two subspecies differ mainly in the coloring of the head and elytra (Araujo Marques, 1941; Bechyne and Bechyne, 1962).

Eggs: Eggs are ovoid, about 0.74 x 0.36 mm, clear white to pale yellow. They exhibit fine reticulation that under the microscope appears like a pattern of polygonal ridges that enclose a variable number of pits (12 to 30) (Krysan, 1986). Eggs are laid in the soil near the base of a host plant in clusters, lightly agglutinated by a colorless secretion. The mandibles and anal plate of the developing larvae can be seen in mature eggs.

Larvae: Defago (1991) published a detailed description of the third instar of *D. speciosa*. First instars are about 1.2 mm long, and mature third instars are about 8.5 mm long. They are subcylindrical; chalky white; head capsule dirty yellow to light brown, epicraneal and frontal sutures lighter, with long light-brown setae; mandibles reddish dark brown; antennae and palpi pale yellow. Body covered by sparse, short, dark setae; light brown irregular prothoracic plate; dark brown anal plate on the ninth segment, with a pair of small urogomphi. A pygopod is formed by the tenth segment, which serves as a locomotion and adherence organ.

Pupae: Pupae are 5.8 to 7.1 mm long and white. Females with a pair of tubercles near the apex. Mature third instars build an 8 x 4 mm oval cell in the soil in which they pupate, and tenerals remain for about 3 days.

Adults: Full descriptions of *D. speciosa* are given by Baly (1886), Araujo Marques (1941), and Christensen (1943). Adults are 5.5 to 7.3 mm long; antennae 4 to 5 mm (Fig. 1). General color grass-green (USDA, 1957); antennae filiform and dark (reddish-brown to black) and nearly equal to the body in length, first three basal segments lighter; head ranging from reddish brown to black; labrum, scutellum, metathorax, tibiae and tarsi black; elytra each with three large oval transverse spots, basal spots larger and usually reddish toward the humeral callus, the rest yellow. Ventrally, head and metathorax dark brown, prothorax green, mesothorax and abdomen light brown or yellow-green. Pronotum bi-foveate, convex, smooth, shiny, ¼ wider than long. Male antennae proportionally longer than female antennae. Males with an extra sclerite on the apex of the abdomen that makes it look blunt, compared with the rather pointed female apex.

Figure 1. Adult *Diabrotica speciosa*. Photo courtesy of Hernan Tolosa.

Biology and Ecology

Eggs are laid on the soil near a larval host plant. An approximately 92% success rate at 27°C is takes place after about 8 days. *Diabrotica speciosa* undergoes three larval instars, which are easily differentiated by the size of the head capsule (see larval description above). In laboratory tests, maize was included in the grouping of most suitable hosts (along with wheat and peanuts), in terms of survival from egg to adult (Cabrera Walsh, 2003). First instars are normally scattered throughout the host's root system, but mature larvae tend to congregate in the upper 10 cm of the root under the crown. The larval stage lasts 23 to 25 days (~12 days in laboratory conditions at 25°C), including an inactive prepupal period of 2 to 3 days. At 25°C, the pupal stage lasts 6 days, and is followed by a period of 3 to 5 days during which the recently molted adults remain in the pupal cell, presumably for the cuticle to tan (USDA, 1957).

Young beetles have a yellowish or pale brown color, which turns green with bright yellow spots in 3 days if fresh food is provided. Under laboratory conditions, mating has been observed between 4 and 6 days after emergence, and some females were observed mating again at day 35. Each female laid an average of 1164 eggs during her lifetime, starting on day 8 and extending for a maximum of 77 days. Peak oviposition was observed on days 16 through 56. In a laboratory environment, oviposition on

maize was preferred over pumpkin, potato and bean seedlings, and maize was as attractive as peanuts in choice tests (Cabrera Walsh, 2003). The number of overlapping generations is conditioned by latitude and climate, being continuous in tropical areas. In Buenos Aires, Argentina, observations indicate there are about three generations per year; the number and timing depends on latitude and climate. Overwintering occurs as an adult (USDA, 1957). These adults can be found concealed in the rosette and crown of winter-growing plants, and they are fairly cold-tolerant (EPPO, 2005).

Pest Importance

Diabrotica speciosa is considered to be an important pest throughout southern South America (except Chile), but, being highly polyphagous, qualitative reports of its impact on different crops vary in different regions. It is considered an important pest of maize, cucurbits, and orchard crops throughout its distribution (CABI, 2007). Although it migrates as an adult, no information on observed distances has been found. Redistributing soil via farm machinery that is contaminated with eggs and-or pupae is also a concern.

Adults of this chrysomelid feed on foliage, pollen, flowers and fruits of many plants. The larvae are pests of roots, especially maize. It is the most harmful species of *Diabrotica* in Argentina, mainly affecting peanuts in the center of the country. It causes considerable damage to watermelon, squash and tomatoes in Brazil, and potatoes and wheat in southeast Brazil. Young squash plantings and immature tomato fruits are severely damaged in Brazil. Populations are so heavy in some years in Paraguay that vegetable crops are almost completely destroyed. Severe injury also occurs on flowers of various ornamentals such as dahlias and chrysanthemums (USDA, 1957). Economic thresholds of two insects per plant for *Phaseolus vulgaris* were determined by Pereira et al. (1997).

IPM programs to combat *D. speciosa* in South America recommend no-till cultural practices, insecticides when reaching economically damaging levels and a rotation of maize, wheat, and soybeans. In South America, insecticides (carbamates, organophosphates and, more recently, tefluthrin and chlorethoxyfos) to control larvae and baits (along with broad-spectrum insecticides) to control adults are widely used. These baits are sliced roots of several different wild cucurbits laced with insecticides.

Although there is research into using parasitoids (brachonids and tachinids) and pathogens (*Beauveria* spp. and *Metarhizium anisopliae*) to combat this pest, no successful biological control programs have been mentioned.

Symptoms/Signs

The larval damage resulting from root feeding can cause host death when the host is small, but the larvae will usually only induce stunted growth in larger host plants, due to a reduction in nutrient uptake. In corn, attack on young plants by larvae produces a typical condition known as 'goose neck', in which the plant exhibits stunted growth, reduced vigor, and the first few internodes of the plant grow bent, sometimes to such an extent that the plant actually lies on the ground (Figure 2). In the case of peanuts and

potatoes, the larvae cause external damage or short bores, similar to those of several other pests such as wireworms and other chrysomelids.

Figure 2. 'Gooseneck' growth form of corn. Photo courtesy of The Ohio State University.

On corn, the most economically important stage is the adult, which feeds on the tassels, preventing pollination and kernel number. Adults also cause defoliation and general feeding damage to leaves, flowers and fruit (EPPO, 2005). Like other *Diabrotica* spp., they are especially associated with Cucurbitaceae and are tolerant of cucubitacins and generally feed on pollen-rich plant structures of over 70 plant species. When flowers are scarce, beetles may feed on the tender green parts of other hosts, such as alfalfa, potatoes, corn, bean, soybean, lettuce, and cabbage causing plant damage, stunting, and possibly plant death (EPPO, 2005).

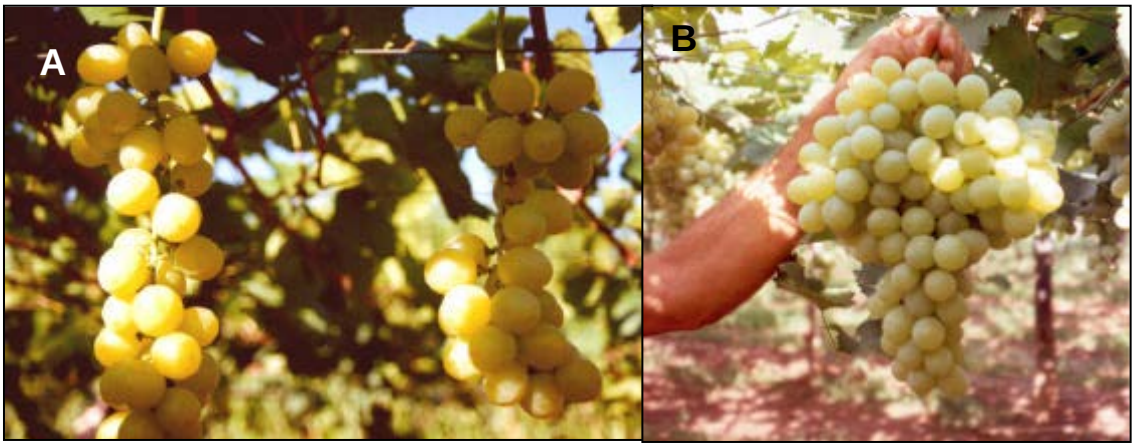


Figure 3. Grape cluster after a severe outbreak of *D. speciosa* during the bloom period (A) and normal cluster (B). Photos courtesy of Roberto et al. (2001).

In grape, adult beetles eat young leaf edges during budding, which usually does not seriously damage the host (Roberto et al., 2001). During the blooming period, however, beetles have been observed on flowers eating the style, stigma, and eventually the ovary. Beetle stigma feeding determines flower aborting and, as a consequence, clusters show low numbers of flowers and fruits (Fig. 3). Weedy hosts need to be

controlled as beetles can also be observed feeding on and moving into grape from surrounding weeds.

Known Hosts

Root-feeding larvae of *D. speciosa* are polyphagous, but the known host range includes corn, wheat, peanut, soybean, and potato. Cabrera Walsh (2003) found that larvae developed well on corn, peanut, and soybean roots, but not so well on pumpkin, beans, and potato. Oviposition preferences roughly parallel larval suitability, but there was a clear preference for cucurbits as adult food, when available; pigweed, sunflower, and alfalfa are secondary hosts. As an adult, *D. speciosa* has been reported feeding on more than 70 host species (Christensen, 1943; Heineck-Leon and Salles, 1997).

Major hosts

Arachis spp. (peanut), *Capsicum* spp. (pepper), *Cucurbita maxima* (winter squash), *Cucurbita pepo* (ornamental gourd), *Glycine max* (soybean), *Solanum tuberosum* (potato), *Triticum* spp. (wheat), *Vitis vinifera* (grape), and *Zea* spp. (corn).

Minor hosts

Allium spp. (onion, leek), *Alternanthera philoxeroides* (alligatorweed), *Amaranthus* spp. (pigweeds), *Apium graveolens* (celery), *Arachis hypogaea* (peanut), *Artemisia* spp. (absinthium, tarragon), *Asparagus* spp. (asparagus), *Avena* spp. (oats), *Baccharis articulata*, *Beta vulgaris* (beet), *Brassica* spp. (mustards), *Bromus catharticus* (prairie grass), *Camellia sinensis* var. *sinensis* (tea), *Capsicum annuum* (pepper), *Capsicum frutescens* (pepper), *Carica papaya* (papaya), *Cattleya* spp. (orchid), *Cayaponia* spp., *Chenopodium* spp., *Chrysanthemum* spp., *Cichorium* spp. (chicory, endive), *Citrullus lanatus* (watermelon), *Citrus* spp., *Coriandrum sativum* (coriander), *Cucumis* spp. (melons, cucumbers, gerkins), *Cucurbita* spp. (cucurbits), *Cucurbitella asperata, Cynara* spp. (artichoke), *Cynodon dactylon* (Bahama crass),, *Dahlia pinnata* (pinnate dahlia), *Datura* spp., *Daucus carota* (carrot), *Fragaria vesca* (wild strawberry), *Gossypium* spp. (cotton), *Helianthus annuus*(sunflower), *Helianthus tuberosus* (Jerusalem artichoke), *Hibiscus* spp., *Ilex paraguayensis* (Paraguay tea), *Ipomoea* spp. (sweet potato, morning glory), *Ipomoea purpurea* (common morning glory), *Lactuca sativa* (lettuce), *Lagenaria siceraria* (bottle gourd), *Lavandula angustifolia* subsp. *angustifolia* (English lavender), *Lepidium didymum* (twin cress), *Lilium maculatum* (sukash-yuri), *Linum usitatissimum* (flax), *Lolium perenne* (rye grass), *Luffa* spp. (loofah), *Malus* spp. (apple), *Malva* spp. (mallow), *Matricaria chamomilla* (chamomile), *Medicago sativa* (alfalfa), *Melilotus albus* (yellow sweet clover), *Mentha* spp. (mint), *Morrenia odorata* (latexplant), *Musa* spp. (banana), *Nasturtium officinale* (watercress), *Nicotiana tabacum* (tobacco), *Ocimum basilicum* (basil), *Orginanum vulgare* (oregano), *Oryza sativa* (rice), *Passiflora coerulea* (passion flower), *Petroselinum crispum* (parsley), *Phaseolus* spp. (beans), *Physalis viscose* (starhair groundcherry), *Pimpinella anisum* (anise), *Pisum sativum* (pea), *Prunus* spp. (stone fruit), *Raphanus sativus* (radish), *Rosa* spp. (rose), *Sechium edule* (chayote), *Sicyos polyacanthus, Solanum* spp., *Solanum betaceum* (tree tomato), *Solanum lycopersicum* (tomato), *Solidago chilensis* (goldenrod), *Sorghum* spp., *Spinacia oleracea* (spinach), *Taraxicum officinale* (dandelion), *Trifolium* spp. (clover), *Tropaeolum majus* (Nasturtium), *Vaccinium virgatum* (smallflower blueberry), *Vaccinium*

corymbosum (highbrush blueberry), and *Zingiber officinale* (ginger) (Cabrera Walsh, 2003; Ministerio de Granaderia, Agricultura y Pesca, 2005; Anonymous, 2009).

Known Vectors (or associated organisms)

There is evidence that *D. speciosa* is a viral vector for comoviruses, southern bean mosaic virus, mimosa mosaic virus, tymoviruses (such as passionfruit yellow mosaic virus), carmoviruses, and purple granadilla mosaic virus (Ribeiro et al., 1996; Germain, 2000). Lin et al. (1984) showed that *D. speciosa* transmitted cowpea severe mosaic virus (CPSMV – comovirus) to bean. Ribeiro et al. (1996) showed that eggplant mosaic virus (EMV – tymovirus) was transmitted to tobacco by *D. speciosa*. Carbrera Walsh (2003) mention that *D. speciosa* may also transmit bacterial wilt, caused by *Erwinia tracheiphila*, in cucurbits.

Known Distribution

Central America: Costa Rica and Panama. **South America:** Argentina, Bolivia, Brazil, Columbia, Ecuador, French Guiana, Paraguay, Peru, Uruguay, and Venezuela. There is a record of *D. speciosa* from Mexico, but according to Krysan (1986), it is almost certainly an error.

Pathway

Diabrotica speciosa could potentially move through international trade. This species has only been intercepted at U.S. ports of entry 2 times. Once from *Solanum lycopersicum* (tomato) originating from Argentina and once from *Lactuca* sp. originating from Peru. However, *Diabrotica* sp. have been intercepted over 1,000 times at U.S. ports of entry. Of these, 84 interceptions originated from countries where *D. speciosa* is known to occur (32 from Columbia, 21 from Ecuador, 16 from Costa Rica, 4 from Brazil, Peru, and Venezuela, 2 from Panama, and 1 from Argentina). These interceptions occurred in permit cargo (72), general cargo (6), holds (3), baggage (2), and miscellaneous (1). Interceptions occurred most commonly on *Chryssanthemum* sp. (6), *Aster* sp. (5), at large (5), *Musa* sp. (5), *Delphinium* sp. (5), and *Ananas comosus* (4) (AQAS, 2012; queried August 6, 2012).

Potential Distribution within the United States

According to a recent risk analysis by USDA-APHIS-PPQ-CPHST, the greatest risk for establishment of *D. speciosa* based on the presence of hosts and climate suitability occurs in much of the Midwest and portions of the South. The pest occurs from temperate Argentina to tropical Brazil. The polyphagous nature of *D. speciosa* increases the likelihood of finding hosts and suitable environment if it were introduced into the United States, and is thought to be able to adapt to more temperate climates.

Survey

CAPS-Approved Method*:
Visual.

*For the most up-to-date methods for survey and identification, see Approved Methods on the CAPS Resource and Collaboration Site, at http://caps.ceris.purdue.edu/.

Literature-Based Methods:
Visual survey: Visual detection of adults is easy, as their feeding period spans from dawn until dusk. Detection of larval damage, on the other hand, is more

Figure 4. Adult banded cucumber beetle, *Diabrotica balteata. Photo courtesy of* John L. Capinera, University of Florida

difficult. First instars are very difficult to sample, and even large infestations can go undetected until the damage caused to the host is extensive. Larger larvae can sometimes be observed feeding on the roots of plants immediately after pulling out of the soil, but methodical sampling and counting methods have not been developed, as they have been for the North American pest species (Fisher and Bergman, 1986).

Figure 5. Western corn rootworm, *Diabrotica virgifera.* Courtesy of USDA-ARS.

Trapping: Adults of *D. speciosa* appear to be universally attracted to aromatic compounds from squash blossoms, though the specific compound(s) that attract the beetles varies from species to species. Often, simple blends of two or three compounds are much more potent attractants than any single compound. In addition, female-produced sex attractant pheromones are used for mate location in this genus. In a preliminary trapping test in Brazil, a number of squash volatiles were screened for potential attraction, and 1,4-dimethoxybenzene showed promise as an attractant for *D. speciosa* (Ventura et al., 2000). Traps baited with 1,4-dimethoxybenzene, a volatile substance of *Cucurbita maxima* blossoms captured 29.4 times and 9.4 times more beetles than controls in soybean and common bean fields, respectively (Ventura et al., 2000).

Figure 6. Southern corn rootworm, *Diabrotica undecimpunctata*. Courtesy of Clemson University - USDA Cooperative Extension Slide Series, http://www.bugwood.org/.

The USDA-CPHST laboratory in Otis, MA has applied for funding to manufacture and test potential lures for *D. speciosa*, but has yet to begin work toward this goal.

Key Diagnostics/Identification

CAPS-Approved Method*: Confirmation of *D. speciosa* is by morphological identification. *Diabrotica speciosa* is almost identical to *D. balteata* (Fig. 4), which is widely present in the southern United States. Confirmation by a chrysomelid specialist is required. *Diabrotica speciosa* can also be confused with *Diabrotica viridula* (not present in the United States) and other pestiferous *Diabrotica* species in South America.

*For the most up-to-date methods for survey and identification, see Approved Methods on the CAPS Resource and Collaboration Site, at http://caps.ceris.purdue.edu/.

Literature-Based Methods:
Diabrotica speciosa somewhat resembles the other main pestiferous *Diabrotica* in South America, *D. viridula*, in coloring, size, biology and host range; but *D. viridula* has dark brown areas toward the cephalic edge of the elytral spots, and distinct humeral plicae. Also, the larvae of *D. viridula* lack urogomphi on the anal plate.

Diabrotica ID is available at http://idtools.org/id/beetles/diabrotica/. *Diabrotica ID* is designed to allow identification of *Diabrotica* species that originate from North and Central America to users lacking an expertise in taxonomy of *Diabrotica*. The tool treats and provides identification support for all 112 North and Central American species of the genus recognized by the authors. Each species is fully illustrated, treated with a fact sheet, and included in the key.

Easily Confused Pests

Survey and detection based on visual detection of symptoms is quite difficult and many other pests can be easily confused. Symptoms, such as dead heart in wheat, goose neck in maize, or stunted growth in most of the larval hosts of *D. speciosa*, could be attributed to several other root feeders, such as wireworms (*Conoderus* spp.; Elateridae), white grubs, (*Phytalus* spp., *Cyclocephala* spp., *Diloboderus abderus*; Melolonthidae), *Pantomorus spp.* and *Listronotus bonariensis* (Curculionidae), and several chrysomelids (*Caeporis* spp., *Colaspis* spp., *Maecolaspis* spp., *Diphaulaca* spp. and *Cerotoma arcuata*) (Gassen, 1984, 1989).

Other rootworms (western corn rootworm, southern corn rootworm) are easily distinguished from *D. speciosa* as adults by the markings on elytra (compare Figs. 1, 5 and 6).

References

Anonymous. 2009. *Diabrotica speciosa*. Orchids online. Accessed April 10, 2012 from: http://www.orchidsonline.com.au/node/7074.

AQAS. 2012. *Diabrotica* sp. interceptions. Last accessed August 6, 2012 from: https://mokcs14.aphis.usda.gov/aqas/HomePageInit.do#defaultAnchor.

Araujo Marques, M. 1941. Contributio ao estudo dos crisomeledeos do gunero *Diabrotica*. Bol. Escola Nac. Agron., 2:61-143. (In Portuguese)

Baly, J. S. 1886. The Colombian species of the genus *Diabrotica*, with descriptions of those hitherto uncharacterized. Part I. Zoological Journal of the Linnean Society, 19:213-229.

Bechyne, J. and B. Bechyne. 1962. Liste der bisher in Rio Grande do Sul gefundenen Galeruciden. Pesquisas (Zool.), 6:1-63.

CABI. 2007. Crop Protection Compendium. Wallingford, UK: CAB International. www.cabicompendium.org/cpc.

Cabrera Walsh, G. 2003. Host range and reproductive traits of *Diabrotica speciosa* (Germar) and *Diabrotica viridula* (F.) (Coleoptera: Chrysomelidae) two species of South American pest rootworms, with notes on other species of Diabroticina. Environmental Entomology 32(2): 276-285.

Christensen, J. R. 1943. Estudio sobre el g'nero *Diabrotica* Chev. en la Argentina (In Spanish). Rev. Facultad de Agronomia y Veterinaria, 10:464-516.

Defago, M. T. 1991. Caracterizacion del tercer estadio larval de *Diabrotica speciosa*. Rev. Peruana de Ent., 33:102-104. (In Spanish)

EPPO. 2005. *Diabrotica speciosa*. OEPP/EPPO Bulletin 35: 374-376.

Fisher, J. R. and M. K. Bergman. 1986. In: [Krysan JL, Miller TA, eds.] Methods for the Study of Pest Diabrotica. New York, USA: Springer.

Gassen, D. N. 1984. Insetos associados a cultura do trigo no Brasil. Circular Tecnica, 3. Passo Fundo, RS, Brazil: EMRAPA-CPNT.

Gassen, D. N. 1989. Insetos subterraneos prejudiciais as culturas no sul do Brasil. EMBRAPA-CNPT. Documentos, 13. Passo Fundo, RS, Brazil: EMBRAPA-CNPT.

Germain, J. F. 2000. *Diabrotica speciosa* (Germar), PRA Area: Europe (01/8504). 1-3. European and Mediterranean Plant Protection Organization.

Heineck-Leonel, M. A. and L. A. B. Salles. 1997. Incidence of parasitoids and pathogens in adults of *Diabrotica speciosa* (Germ.) (Coleoptera: Chysomelidae) in Pelotas, RS. Anais da Sociedade Entomológica do Brasil, 26(1):81-85.

Krysan, J. L. 1986. Introduction: biology, distribution, and identification of pest *Diabrotica*. In: [Krysan JL, Miller TA, eds.] Methods for the Study of Pest *Diabrotica*. New York, USA: Springer.

Lin, M. T., J. H. Hill, E. W. Kitajima, and C. L. Costa. 1984. Two new serotypes of cowpea severe mosaic virus. Phytopathology 74: 581-585.

Ministerio de Granaderia, Agricultura y Pesca. 2005. Information Requested for Pest Risk Analysis to Export Blueberries Fruit (*Vaccinium corymbosum* and *V. ashei*) from Uruguay to United States. Ministerio de Granaderia, Agricultural y Pesca, Direccion General de Servicios Agricolas, Division Proteccion Agricola. 9 pp.

Pereira, M. F. A., L. G. Delfini, M. R. Antoniacomi, and M. H. Calafiori. 1997. Damage caused by the leaf beetle, *Diabrotica speciosa* (Germar, 1824), on beans (*Phaseolus vulgaris* L.), with integrated management. Ecossistema, 22:17-20.

Ribeiro, S. G., E. W. Kitajima, C. R. B. Oliveira, and R. Koenig. 1996. A strain of eggplant mosaic virus isolated from naturally infected tobacco plants in Brazil. Plant Dis. 80: 446-449.

Roberto, S. R., W. Genta, and M. U. Ventura. 2001. *Diabrotica speciosa* (Ger.) (Coleoptera: Chrysomelida): New Pest in Table Grape Orchards. Neotropical Entomology 30(4): 721-722.

USDA, 1957. Cooperative Economic Insect Report, 7(2):5-6.

Ventura, M. U., M. C. Martins, and A. Pasini. 2000. Response of *Diabrotica speciosa* and *Cerotoma acuata tingomariana* (Coleoptera: Chrysomelidae) to volatile attractants. Florida Entomologist 83(4): 403-410.

Helicoverpa armigera

Scientific Name
Helicoverpa armigera Hübner

Synonyms:
Bombyx obsoleta, Chloridea armigera, Chloridea obsoleta, Helicoverpa commoni, Helicoverpa obsoleta, Heliothis armigera, *Heliothis conferta, Heliothis fusca, Heliothis obsoleta*, *Heliothis pulverosa*, *Heliothis rama*, *Heliothis uniformis*, *Noctua armigera*, and *Noctua barbara*

Common Name(s)
Old world bollworm, scarce bordered straw worm, corn earworm, African cotton bollworm, American bollworm, tomato worm, gram pod borer

Type of Pest
Moth

Taxonomic Position
Class: Insecta, **Order:** Lepidoptera, **Family:** Noctuidae

Reason for Inclusion in Manual
CAPS Target: AHP Prioritized Pest List – 2009 through 2013

Pest Description
Eggs: Freshly laid eggs (Fig. 1A) are hemispherical to spherical in shape, 0.4 to 0.6 mm in diameter with a flat base, and yellowish-white in color; changing to a deep yellow after a day. The eggs then change to dark or gray black a day before hatching (Bhatt and Patel, 2001; CABI, 2007). The eggs are sculpted with vertical ridges of alternating length, which surround a smooth apical area that contains the micropile (King, 1994).

Larvae: Larval color darkens with successive molts for the six instars typically observed for *H. armigera*. Coloration can vary considerably due to diet content. Coloration ranges from bluish green to brownish red (Fowler and Lakin, 2001). Freshly emerged first instar larvae are translucent and yellowish-white in color with a black to brown head capsule and have a spotted appearance (Fig. 1B) due to sclerotized setae, tubercle bases and spiracles (King, 1994; Bhatt and Patel, 2001). Second instar larvae are yellowish green in color with black thoracic legs. The full grown larvae are brownish or pale green with brown lateral stripes and distinct dorsal stripe; long and ventrally flattened but convex dorsally. Larval size in the final instar ranges from 3.5 to 4.2 cm (approx. 1 $^{3}/_{8}$ to 1 $^{5}/_{8}$ in) in length (King, 1994).

Pupae: Dark-brown, 14 to 22 mm (approx. $^{9}/_{16}$ to $^{7}/_{8}$ in) long and 4.5 to 6.5 mm (approx. $^{3}/_{16}$ to $^{1}/_{4}$ in) in width, with a smooth surface, rounded both anteriorly and posteriorly, with two tapering parallel spines at posterior tip.

Adults: Stout-bodied moth of typical noctuid appearance (Fig. 1C), with 3.5 to 4 cm (1 $^{3}/_{8}$ to 1 $^{9}/_{16}$ in) wing span; broad across the thorax and then tapering, 18 to 19 mm (approx. $^{3}/_{4}$ in) long. The coloration varies from dull greenish yellow to olive gray or light brown and females are darker than males (King, 1994).

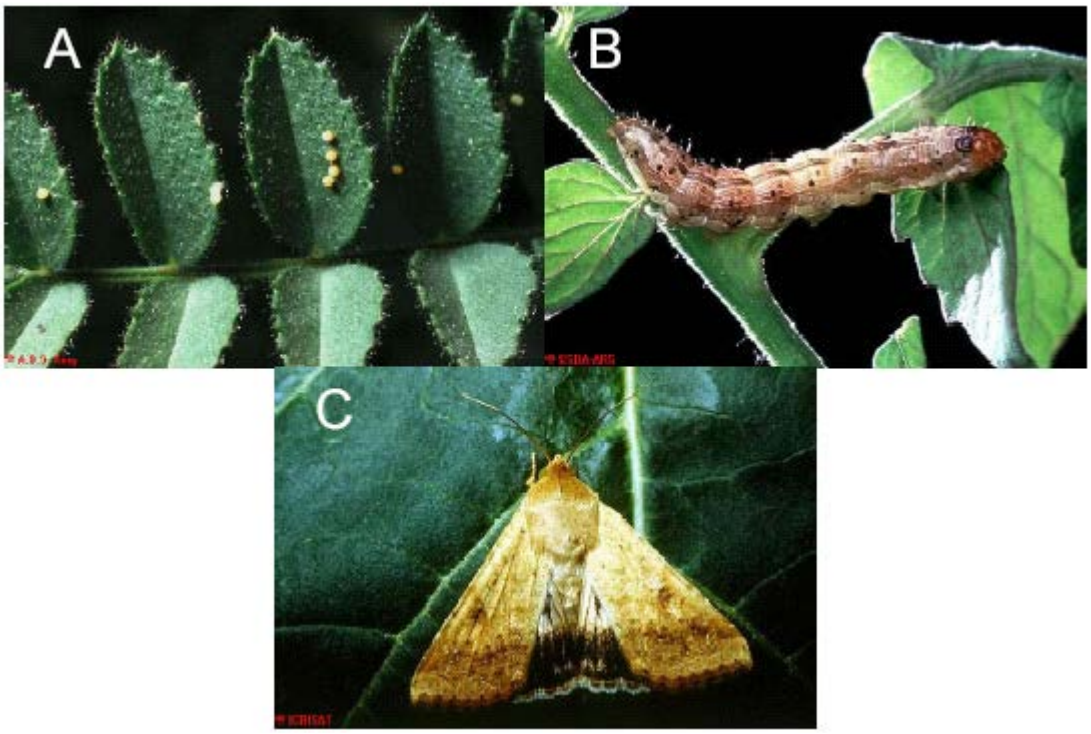


Figure 1. Life stages of *Helicoverpa armigera* (images not to scale): (A) eggs, (B) larva, and (C) adult. Photos courtesy of CABI, 2007.

For more information on descriptions see Common (1953), Kirkpatrick (1961), Hardwick (1965, 1970), King (1994).

Biology and Ecology (From Venette et al., 2003)
Because *H. armigera* exhibits overlapping generations, it can be difficult to determine the number of completed generations. Typically 2-5 generations are achieved in subtropical and temperate regions and up to 11 generations can occur under optimal conditions, particularly in tropical areas (Tripathi and Singh, 1991; King, 1994, Fowler and Lakin, 2001). Temperature and availability of suitable host plants are the most important factors influencing the seasonality, number of generations, and the size of *H. armigera* populations (King, 1994).

Adults emerge from the ground in the spring between dusk and midnight, climb vertical structures, and dry their wings for a period of 2 or more hours (King, 1994; CABI, 2007). In order to mate and lay eggs, adults typically must feed on nectar. About 2-5 days after emergence, females release a pheromone during early morning hours before dawn to attract mates (King, 1994). Mating occurs 1-4 days after emergence and is strongly

influenced by humidity and temperature (King, 1994; Saito, 1999; Fowler and Lakin, 2001).

Helicoverpa armigera lays eggs prolifically (Tripathi and Singh, 1991). A female may produce a maximum of 4394 eggs, but on average a female will produce 730-1,702 eggs (King, 1994; Fowler and Lakin, 2001; CABI, 2007). Eggs can be laid over 10 to 23 days (King, 1994). Oviposition begins 2-6 days after emergence, and egg-laying often occurs at night (Kyi and Zalucki, 1991; Akashe et al., 1997; Fowler and Lakin, 2001; CABI, 2007). Moths tend to lay eggs singly, on or near floral structures. Peak egg-laying typically occurs prior to or during host flower production (King, 1994). Depending on the quality of the host, *H. armigera* may also lay eggs on leaf surfaces. Female moths tend to choose pubescent (hairy) surfaces for oviposition rather than smooth leaf surfaces (King, 1994). In India, heavy egg-laying is a normal occurrence after rainfall (Das et al., 2001). In the Sudan, there is a significant rise in egg numbers after 6 days had elapsed following a rainfall event (Madden et al., 1993). King (1994) reviewed several adult longevity studies and reports a range in adult life span of 5 to 36 days.

Under adverse conditions, moths can migrate long distances (King, 1994; Zhou et al. 2000, Casimero et al., 2001; Shimizu and Fujisaki, 2002; CABI, 2007). Adults can disperse distances of 10 km during “non-migratory flights” and hundreds of kilometers (up to 250 km) when making “migratory flights”, which probably occur when host quality or availability declines (Saito, 1999; Zhou et al.; 2000, Casimero et al., 2001; Fowler and Lakin, 2001).

Eggs hatch in about 3 days at 25°C, but at lower temperatures, hatch may take up to 11 days. Larvae may complete up to 7 instars, though generally there are between 5 and 7 instars (Twine; 1978; King, 1994; Fowler and Lakin, 2001). In laboratory studies, the complete larval period (all instars combined) lasted between 12-36 days (Kirkpatrick, 1962; Bhatt and Patel, 2001; Fowler and Lakin, 2001). During summer, larval development is completed in 14-18 days, while it may take up to 21 days in fall (CABI, 2007). The prepupal stage lasts 1-4 days, and during this time larval activity decreases (King, 1994).

Molting often occurs in full sun on leaf surfaces (King, 1994). Before feeding on their host plant, newly hatched larvae typically consume all or part of their egg shells; larvae may then feed on leaf surfaces or floral structures, moving about the plant for a short distance before selecting a preferred feeding spot (King, 1994). Small, young larvae have the ability to feed inside floral structures, detectable only by a small hole with spun silk at the entrance and visible frass; larger larvae feed with a portion of their body outside the floral or fruiting structure (King, 1994).

Helicoverpa armigera is damaging to crops because larvae can move from plant to plant, particularly when food is scarce (King, 1994). Late-instar larvae are more damaging to the host plant due to their attraction to “full buds” (Mabbett et al., 1980).

Once feeding is completed, larvae move 2.5 to 17.5 cm (approx. 1 to 6 $^{7}/_{8}$ in) below the soil surface to pupate (King, 1994). Less frequently, pupation occurs within a spun web

on the host plant (*e.g.*, in a corn cob) or on the soil surface (King, 1994). Depending on temperature, the pupal stage lasts between 6 to 33 days, unless the insect goes into diapause, in which case pupation may require several months. *Helicoverpa armigera* overwinter as pupae (King, 1994; Akashe et al., 1997; Maelzer and Zalucki, 1999, Bhatt and Patel, 2001; Fowler and Lakin, 2001; CABI, 2007).

Diapause is facultative and occurs during the pupal stage (King, 1994). Diapause induction begins when larvae are exposed to day lengths between 11.5 to 12.5 hours, and low temperatures (19 to 23°C, 66.2 to 73.4°F), or when larvae are exposed to lengthy periods of extremely hot and dry weather (≥ 35°C, 95°F) (King, 1994; Zhou et al., 2000). Little to no diapause occurs in tropical areas (King, 1994). Total longevity (from egg to adult death) is 30 to 40 days with females generally living 2 to 3 days longer than males (King, 1994; Akashe et al., 1997). Bhatt and Patel (2001) recorded a slightly longer life span of about 51 days for males and 54 days for females. Rochester et al. (2002) reported a span 35 to 75 days from egg to adult.

The optimum temperature for development from 1st instar larva to adult was 33.9°C (93°F) (Twine, 1978) when reared on artificial diet. However, Twine (1978) reported optimal survival temperatures of 27° C (80.6°F) for pupae and 24° C (75.2°F) for larvae. In a laboratory study, high temperatures (above 37°C, 98.6°F) caused pupal dormancy (Nibouche, 1998). A standard threshold for development of *H. armigera* was determined to be 11°C (51.8°F) (Twine, 1978; Maelzer and Zalucki, 1999).

Symptoms/Signs

In most host species, fruit, leaves, shoots, and flower buds may be consumed by larvae. Bore holes are visible at the base of flower buds, the latter being hollowed out. Larger larvae bore into maturing flowers, fruit, and seed. It may be necessary to cut open the plant organs to detect the pest. Frass may be evident. Fruit drop and defoliation are possible. Secondary infections by other organisms are common and lead to rotting.

In sorghum and other grains, the larvae feed on the head when the grains are in the milky stage. They are especially damaging to sorghum varieties with tight compact heads. Varieties with loose open panicles are rarely damaged (Bijlmakers, 1989). Yield loss is caused by *H. armigera* feeding directly on the grain.

Pest Importance

Heliothine moths of the genus *Helicoverpa* are considered to be among the most damaging insect pests in Australian agriculture, costing approximately $225.2 million per year to control (Clearly et al., 2006). *Helicoverpa armigera* is a major insect pest of both field and horticultural crops in many parts of the world (Fitt, 1989). The pest status of *H. armigera* is due in part to the highly polyphagous nature of its larvae, its high fecundity, its high mobility, and its ability to enter facultative diapause (Cleary et al., 2006). These characteristics make *H. armigera* particularly well adapted to exploit transient habitats such as man-made ecosystems.

Worldwide, *H. armigera* has been reported on over 180 cultivated hosts and wild species in at least 45 plant families (Venette et al., 2003). The larvae feed mainly on the flowers and fruit of high value crops and thus high economic damage can be caused at low population densities (Cameron, 1989; CABI, 2007). In pigeonpea, an important grain legume in south Asia, east Africa and Latin America, this single pest causes yield losses of up to 100% in some years and locations and worldwide losses to pigeonpea of more than $300 million per year (Thomas et al., 1997).

Helicoverpa armigera is capable of long-distance migratory flights (King, 1994; Zhou et al., 2000; Casimero et al., 2001; Shimizu and Fujisaki, 2002; CABI, 2007).

Management of *Helicoverpa* spp. in the past has relied heavily on the use of insecticides, and this has led to resistance problems in cotton (Fitt, 1994). Resistance to pyrethroids amongst *H. armigera* is a serious problem (McCaffrey et al., 1989; Trowell et al., 1993).

Known Hosts

Note: Not all host plants are equally preferred for oviposition but can be utilized in the absence of a preferred host. There have been several studies within the laboratory setting on host preference. Jallow and Zalucki (1996) found that most females ranked corn, sorghum, and tobacco highest, followed by cotton varieties. The least preferred were cowpea and alfalfa. Cotton and corn were more suitable for development and reproduction of the cotton bollworm than peanut (Hou and Sheng, 2000). Pigeonpea and corn are considered to be the most suitable host for this insect, when compared to sorghum, red ambadi (*Hibiscus subdariffa*), marigold, and artificial diet (Bantewad and Sarode, 2000). Tobacco, corn, and sunflower were categorized as the most preferred hosts; soybean, cotton, and alfalfa were categorized as intermediate hosts; and cabbage, pigweed, and linseed were the least preferred in an additional study (Firempong and Zalucki, 1990).

Major hosts

Abelmoschus esculentus (okra), *Allium* spp. (onions, garlic, leek, etc.), *Arachis hypogaea* (peanut), *Avena sativa* (oats), Brassicaeae (cruciferous crops), *Cajanus cajan* (pigeon pea), *Capsicum annuum* (bell pepper), *Carthamus tinctorius* (safflower), *Cicer arietinum* (chickpea, gram), *Citrus*, Cucurbitaceae (cucurbits), *Dianthus caryophyllus* (carnation), *Eleusine coracana* (finger millet), *Glycine max* (soybean), *Gossypium* spp. (cotton), *Helianthus annuus* (common sunflower), *Hordeum vulgare* (barley), *Lablab purpureus* (hyacinth bean), *Linum usitatissimum* (flax), *Malus* spp. (apple), *Mangifera indica* (mango), *Medicago sativa* (alfalfa), *Nicotiana tabacum* (tobacco), *Pennisetum glaucum* (pearl millet), *Phaseolus* spp. (beans), *Phaseolus vulgaris* (common bean), *Pinus* spp. (pines), *Pisum sativum* (pea), *Prunus* spp. (stone fruit), *Solanum melongena* (eggplant), *Solanum lycopersicum* (tomato), *Solanum tuberosum* (potato), *Sorghum bicolor* (sorghum), *Triticum* spp. (wheat), *Triticum aestivum* (wheat), *Vigna unguiculata* (cowpea), and *Zea mays* (corn) (CABI, 2007).

Wheat, oats, and barley are considered primary hosts for *H. armigera.*

Poor hosts
Vitis vinifera (grape) (Vorus, 1996).

Wild hosts
Acalypha spp. (copperleaf), *Amaranthus* spp. (pigweed, amaranth), *Datura* spp., *Datura metel* (datura), *Gomphrena*, *Hyoscyamus niger* (black henbane), *Sonchus oleraceus* (annual sowthislte) (Gu and Walter, 1999; CABI, 2007).

For a complete listing of hosts see Venette (2003).

Known Vectors (or associated organisms)
Helicoverpa armigera is not a known vector and does not have any associated organisms.

Known Distribution
Helicoverpa armigera is found in the Palearctic, Oriental, Ethiopian, and Australian zoogeographic provinces, south of a line at approximately 52°N. The range occupied by the species includes tropical, dry, and temperate climates (CABI, 2007).

Asia: Afghanistan, Armenia, Azerbaijan, Bangladesh, Bhutan, Brunei Darussalam, Cambodia, China, Cocos Islands, Republic of Georgia, Hong Kong, India, Indonesia, Iran, Iraq, Israel, Japan, Jordan, Kazakhstan, Korea, Kuwait, Kyrgyzstan, Laos, Lebanon, Malaysia, Myanmar, Nepal, Pakistan, Philippines, Saudi Arabia, Singapore, Sri Lanka, Syria, Taiwan, Tajikistan, Thailand, Turkey, Turkmenistan, United Arab Emirates, Uzbekistan, Vietnam, and Yemen. **Europe:** Albania, Andorra, Austria, Bosnia and Herzegovina, Bulgaria, Cyprus, Finland, France, Germany, Gibraltar, Greece, Hungary, Ireland, Italy, Lithuania, Macedonia, Malta, Moldova, Portugal, Romania, Russia, Serbia and Montenegro, Slovenia, Spain, Sweden, Switzerland, and Ukraine. **Africa:** Algeria, Angola, Benin, Botswana, Burkina Faso, Burundi, Cameroon, Cape Verde, Central African Republic, Chad, Congo, Cote d'Ivoire, Democratic Republic of Congo, Egypt, Eritrea, Ethiopia, Gabon, Gambia, Ghana, Guinea, Kenya, Lesotho, Libya, Madagascar, Malawi, Mali, Mauritania, Mauritius, Mayotte, Morocco, Mozambique, Namibia, Niger, Nigeria, Reunion, Rwanda, Saint Helena, Senegal, Seychelles, Sierra Leone, Somalia, South Africa, Sudan, Swaziland, Tanzania, Togo, Tunisia, Uganda, Zambia, and Zimbabwe. **Oceania:** American Samoa, Australia (including Christmas Island), Belau, Cook Islands, Federated States of Micronesia, Fiji, Guam, Kiribati, Marshall Islands, New Caledonia, New Zealand, Norfolk Island, Northern Mariana Islands, Papua New Guinea, Samoa, Solomon Islands, Tonga, Tuvalu, and Vanuatu. **South America:** Brazil (Fibiger and Skule, 2011; EPPO, 2012; Spect et al., 2013)

Pathway
Helicoverpa armigera could potentially move through international trade. This species has been intercepted 860 times at U.S. ports of entry. Interceptions have occurred in

permit cargo (655), baggage (131), general cargo (47), stores (13), holds (7), miscellaneous (3), quarters (3), and mail (1). Most interceptions originated from the Netherlands (275), Israel (209), India (63), Italy (27), Kenya (27), and Spain (25). This species is mostly intercepted on plant material, including *Bupleurum* sp. (73), *Ornithogalum* sp. (59), *Leucospermum* sp. (46), *Veronica* sp. (38), *Tagetes* sp. (32), and *Capsicum* sp. (25) (AQAS, 2012; queried August 6, 2012).

Potential Distribution within the United States

According to Fowler and Lakin (2001), it is probable that *H. armigera* could establish in every state in the continental United States based on habitat and host suitability and would probably pose the greatest economic threat to the following states: Alabama, Arizona, Arkansas, California, Georgia, Illinois, Iowa, Kansas, Louisiana, Michigan, Minnesota, Mississippi, Nebraska, New Mexico, North Carolina, Ohio, Pennsylvania, South Carolina, South Dakota, Tennessee, Texas, Virginia, and Wisconsin. A recent risk analysis by USDA-APHIS-PPQ-CPHST, however, indicates that areas of Alabama, Arizona, California, Florida, Idaho, Louisiana, Mississippi, Nevada, New Mexico, Oregon, South Carolina, Texas, and Washington have the greatest risk for *H. armigera* establishment based on host availability, climate, and pathway within the continental United States. Areas of most states, however, have moderate risk for *H. armigera* establishment.

Survey

CAPS-Approved Method*: The CAPS-approved method is a trap and lure combination. The lure is effective for 28 days (4 weeks). The lure dispenser type is a rubber septum.

Any of the following Trap Product Names in the IPHIS Survey Supply Ordering System may be used for this target:

Plastic Bucket Trap (Unitrap)
Heliothis Trap (Plastic mesh cone trap)
Texas (Hartstack) Trap

The Lure Product Name is "*Helicoverpa armigera* Lure."

IMPORTANT: Do not place lures for two or more target species in a trap unless otherwise recommended.

Trap spacing: When trapping for more than one species of moth, separate traps for different moth species by at least 20 meters (65 feet).

Method Notes: The Plastic Bucket Trap is also known as the unitrap. The trap has a green canopy, yellow funnel, and white bucket and is used with a dry kill strip. See Brambila et al. (2010) for for instructions on using the plastic bucket trap.

The Texas (Hartstack) trap is not available commercially. See Hartstack et al. (1979) or Johnson and McNeil (no date) for images and trap design.

Lure Notes: May 24, 2012: The length of effectiveness of this lure may be reduced in hot and dry climates. In these environments, lures may need to be changed every two weeks instead of every four weeks.

Survey site and selection:
Helicoverpa armigera is a major insect pest of both field and horticultural crops in many parts of the world (Fitt, 1989). *H. armigera* has been reported causing serious losses throughout its range, in particular to cotton, tomatoes, and corn.

Surveys should be focused on areas that have abundant host material like agricultural crops and nurseries.

Time of year to survey:
Moths emerge in May to June depending on latitude. In Australia, *H. armigera* begin emerging in the spring (Duffield and Steer, 2006). In Pakistan, moths are active in June and July when ambient temperatures fall (Mustafa, 2004). Adults can be present at different times of the year as this pest can have multiple generations per year.

Trap Placement:
Traps should be hung at or slightly above crop height. This can range from 1.5 to 1.8 m (5 to 6 ft) in height (Gauthier et al., 1991; Greg and Wilson, 1991), depending on the crop. The Texas trap should be placed directly above the crop canopy Greg and Wilson, 1991).

*For the most up-to-date methods for survey and identification, see Approved Methods on the CAPS Resource and Collaboration Site, at http://caps.ceris.purdue.edu/.

Literature-Based Methods:
Trapping: (From Venette et al., 2003). Pheromone traps using (*Z*)-11-hexadecenal and (*Z*)-9-hexadecenal in a 97:3 ratio have been used to monitor populations of *H. armigera* (Pawar et al., 1988; Loganathan and Uthamasamy, 1998; Loganathan et al., 1999; Visalakshmi et al., 2000; Zhou et al., 2000). Of three pheromone doses tested in the field (0.75, 1.0, and 1.25 mg/septum), 1 mg attracted the most males (Loganathan and Uthamasamy, 1998); the trap type was not specified. Rubber septa impregnated with these sex pheromone components (1 mg/septum) were equally effective in capturing males for 11 days in the laboratory (Loganathan et al., 1999). Captures of *H. armigera* in the field were significantly lower with 15-day-old lures than with fresh lures, and the authors recommend replacing lures every 13 days (Loganathan et al., 1999). Similar observations were reported by Pawar et al. (1988). Males responded to the pheromone during dark hours only, commencing at 6:00 PM and terminating at 6:00 AM. The highest response was between 11:00 PM and 4:00 AM (Kant et al., 1999).

Trap design has a significant impact on the number of male *H. armigera* moths that will be captured with pheromone lures. Funnel traps and Texas traps are substantially more effective than sticky traps (Kant et al., 1999). Hartstack *(i.e.,* hollow cone) traps

have also been used to effectively monitor densities of adults (Walker and Cameron, 1990). Cone traps are significantly more effective than water-pan traps (Sheng et al., 2002). Traps have been placed approximately 6 feet (1.8 meter) above the ground (Kant et al., 1999; Zhou et al., 2000), and have been separated by a distance of at least 160 feet (50 meters) (Kant et al., 1999). Aheer et al. (2009), however, installed traps at a height of 4.9 feet (1.5 meters) and were separated by a distance of about 33 feet (10 meters). For routine monitoring of pests, pheromone traps are deployed at a density of 5 traps per hectare (Sidde Gowda et al., 2002).

Adults of both sexes can be captured in black light traps.

Visual survey: Visual inspections of plants for eggs and/or larvae are frequently used to monitor and assess population sizes for *H. armigera*. Females lay several hundred eggs on the leaves (top 20 cm), flowers and fruits (Duffield and Chapple, 2000). The lower leaf surface is a preferred oviposition site. Eggs may hatch in less than 3 days at an optimum temperature of 27 to 28°C (81 to 82°F). The feeding larvae can be seen on the surface of plants but they are often hidden within plant organs (flowers, fruits, etc.). Bore holes and heaps of frass (excrement) may be visible, but otherwise it is necessary to cut open the plant organs, especially damaged fruit, to detect the pest (Bouchard et al., 1992). In temperate regions, *H. armigera* overwinters as a pupa buried several cm in the soil. Adults appear in April to May and can be observed until October, because of the long migration period.

In vegetative Australian cotton and irrigated soybean, a minimum of 60 whole plants per 100 hectare commercial field are examined for the presence of *H. armigera* eggs or larvae; when plants begin to produce squares, only the upper terminal (approximately 20 cm) of a plant is inspected (Brown, 1984; Dillon and Fitt, 1995; Duffield and Chapple, 2000). In experimental plots, visual inspections for *H. armigera* in pigeon pea were restricted to the upper third of whole plants (4 sets of five plants in a 30 x 30 meter plot) (Sigsgaard and Ersbøll, 1999).

Leaves of tomato plants are more attractive than flowers or fruits as *H. armigera* oviposition sites, but use of a single-leaf sample unit (with a sample size of 30 plants per field) has proven ineffective in detecting low densities of *H. armigera* (Cameron et al., 2001). On some tomato cultivars, leaves in the upper half of the plant are preferentially selected for oviposition (Saour and Causse, 1993).

Key Diagnostics/Identification

CAPS-Approved Method*:

Confirmation of *Helicoverpa armigera* is by morphological identification. *Helicoverpa armigera* can be visually screened to some degree, but definitive screening and identification requires dissection. *Helicoverpa armigera* and the native, abundant species, *Helicoverpa zeae* are very similar looking. Final identification is by dissection of (adult) male genitalic structures.

Screening aids and instructions for dissecting *H. armigera* are available at:

Brambila, J. 2009. *Helicoverpa armigera* Screening Aids: http://caps.ceris.purdue.edu/webfm_send/552.

Brambila, J. 2009. Dissection instructions for identifying male *Heliocoverpa amigera* and *H. zea*: http://caps.ceris.purdue.edu/webfm_send/551.

Passoa, S. 2007. Identification guide to larval Heliothinae (Lepidoptera: Noctuidae) of quarantine significance: http://caps.ceris.purdue.edu/webfm_send/109.

*For the most up-to-date methods for survey and identification, see Approved Methods on the CAPS Resource and Collaboration Site, at http://caps.ceris.purdue.edu/.

Literature-Based Methods:

Helicoverpa armigera belong to a complex of similar species. Adults may be identified by distinct differences in genitalia (Common, 1953; Kirkpatrick, 1961; Hardwick, 1965; EPPO, 2003). Differentiation between *H. armigera* and *H. zea*, which is present in the United States, is very difficult; identification is by dissection of internal structures of adult males (Pogue, 2004). A morphological study of *H. assulta, H. punctigera,* and *Heliothis virescens* (formerly *H. rubrescens*) compares similarities and differences between species; a key is provided for identifying adults (Kirkpatrick, 1961). Immunological tests are available to differentiate *H. punctigera* and *Heliothis virescens* in egg or larval stages (Ng et al., 1998).

A diagnostic protocol for *H. armigera* has been developed by the EPPO (2003). This protocol includes identification sections on the last instar larvae as well as the adult. It also includes a comparison between the male genitalia of *H. armigera* and similar species, including *H. zea* and *H. punctigera* (EPPO, 2003).

The LepTon test, an Enzyme Linked Immunosorbent Assay (ELISA) based approach, has been developed to distinguish between *H. armigera* and *H. punctigera* in the egg and larval stages (Trowell et al., 1993). Cahill et al. (1984) provide morphological information to distinguish third/fourth and sixth instar larvae of *H. armigera* and *H. punctigera*.

Agusti et al., (1999) developed sequence amplified characterized region (SCAR) markers to detect *H. armigera* eggs in the gut of predators. It may be possible to adapt this procedure to detect *H. armigera in planta*.

Easily Confused Pests

Helicoverpa zea is native to the United States and is attracted to the same lure as *H. armigera. Helicoverpa zea* is very similar looking to *H. armigera* and is encountered frequently in *H. armigera* traps. Additional noctuid species that can be confused easily with *H. armigera* include *H. assulta* (not known in the United States), *H. punctigera (*not known in the United States), and *Heliothis virescens* (present in the United States) (Kirkpatrick, 1961; CABI, 2007).

References

Agusti, N., M. C. de Vicente, and R. Gabarra. 1999. Development of sequence amplified characterized region (SCAR) markers of *Helicoverpa armigera*: a new polymerase chain reaction-based technique for predator gut analysis. Molecular Ecology 8: 1467-1474.

Aheer, G. M., A. Ali, and M. Akram. 2009. Effect of weather factors on populations of *Helicoverpa armigera* moths at cotton-based agro-ecological sites. Entomological Research 29: 36-42.

Akashe, V., S. Mehtre, B. Koli, and D. Veer. 1997. Biology of *Helicoverpa armigera* (Hb.) on safflower. The P.K.V. Research Journal 21: 230-231.

AQAS. 2012. *Helicoverpa* sp. interceptions. Last accessed August 6, 2012 from: https://mokcs14.aphis.usda.gov/aqas/HomePageInit.do#defaultAnchor.

Bantewad, S. D. and S. V. Sarode. 2000. Influence of different hosts on the biology of *Helicoverpa armigera* (Hübner). Shashpa 7(2): 133-136.

Bhatt, N. J. and R. K. Patel. 2001. Biology of chickpea pod borer, *Helicoverpa armigera*. Indian Journal of Entomology 63(3): 255-259.

Bijlmakers, H. 1989. Insect pests of cereals in Ethiopia identification and control methods. FAO/UNDP Project ETH/86/029 Crop Protection Phase II, Addis Ababa, October 1989.

Bouchard, D., A. Oudraogo, and G. Boivins. 1992. Vertical distribution, spatial dispersion and sequential sampling plan for fruit damage by *Helicoverpa armigera* (Hübner) (Lepidoptera: Noctuidae) on tomato crop in Burkina Faso. Tropical Pest Management 38(3): 250-253.

Brambila, J., L. Jackson, and R. Meagher. 2010. Plastic bucket trap protocol. USDA. 7 pp.

Brown, G. 1984. Field experience in cotton pest management in north western New South Wales, pp. 128-134. *In* P. Bailey and D. Swincer [eds.], Proceedings of the fourth Australian Applied Entomological Research Conference, Adelaide, Australia.

CABI. 2007. Crop Protection Compendium. Wallingford, UK: CAB International. www.cabicompendium.org/cpc

Cameron, P. J. 1989. *Helicoverpa armigera* (Hübner), a tomato fruitworm (Lepidoptera: Noctuidae). Tech. Commun. Commw. Inst. Biol. Control. 10: 87-91.

Cameron, P., G. Walker, T. Herman, and A. Wallace. 2001. Development of economic thresholds and monitoring systems for *Helicoverpa armigera* (Lepidoptera: Noctuidae) in tomatoes. Journal of Economic Entomology 94: 1104-1112.

Casimero, V., F. Nakasuji, and K. Fujisaki. 2001. The influence of larval and adult food quality on the calling rate and pre-calling period of females of the cotton bollworm, *Helicoverpa armigera* Hübner (Lepidoptera : Noctuidae). Appl. Entomol. Zool. 36(1): 33-40.

Cleary, A. J., B. W. Cribb, and D. A. H. Murray. 2006. *Helicoverpa armigera* (Hübner): can wheat stubble protect cotton from attack. Australian Journal of Entomology 45: 10-15.

Common, I. F. B. 1953. The Australian species of *Heliothis* (Lepidoptera: Noctuidae) and their pest status. Australian Journal of Zoology 1: 319-344.

Das, D. K., T. P. Trivedi, and C. P. Srivastava. 2001. Simple rule to predict attack of *Helicoverpa armigera* on crops growing in Andhra Pradesh. Indian Journal of Agricultural Sciences 71(6): 421-423.

Dillon, G. and G. Fitt. 1995**.** Reassessment of sampling relationships for *Helicoverpa* spp. (Lepidoptera: Noctuidae) in Australian cotton. Bulletin of Entomological Research 85: 321-329.

Duffield, S. J., and D. G. Chapple. 2000. Within-plant distribution of *Helicoverpa armigera* (Hubner) and *Helicoverpa punctigera* (Wallengren) (Lepidoptera: Noctuidae) eggs on irrigated soybean. Australian Journal of Entomology 40:151-157.

EPPO. 2003. Diagnostic protocols for regulated pests, *Helicoverpa armigera.* EPPO Bulletin 33, 289-295.

EPPO. 2012. EPPO Plant Quarantine Information Retrieval System (PQR), version 5.0.5540. European and Mediterranean Plant Protection Organization.

Fibiger, M. and B. Skule. 2011. Fauna Europaea: *Helicoverpa armigera*. In O. Karsholt and E. J. Nieukerken (eds.) Fauna Europaea: Lepidoptera, Moths, version 2.4. Accessed April 10, 2012 from: http://www.faunaeur.org**.**

Firempong, S., and M. Zalucki. 1990. Host plant preferences of populations of *Helicoverpa armigera* (Hübner) (Lepidoptera: Noctuidae) from different geographic locations. Australian Journal of Zoology 37: 665-673.

Fitt, G. P. 1989. The ecology of *Heliothis spp.* in relation to agroecosystems. Annual Review of Entomology 34: 17-52.

Fitt, G. P. 1994. Cotton pest management: Part 3. An Australian perspective. Annual Review of Entomology 39: 543-562.

Fowler, G. A., and K. R. Lakin. 2001. Risk Assessment: The old world bollworm, Helicoverpa armigera (Hübner), (Lepidoptera: Noctuidae). USDA-APHIS-PPQ-CPHST-PERAL.

Gu, H., and Walter, G.H. 1999. Is the common sowthistle (*Sonchus oleraceus)* a primary host plant of the cotton bollworm, *Helicoverpa armigera* (Lep. Noctuidae)? Oviposition and larval performance. J. Appl. Ent. 123: 99-105.

Hardwick, D. F. 1965. The corn earworm complex. Memoirs of the Entomological Society of Canada, 40: 1-247.

Hardwick, D. F. 1970. A generic revision of North American Heliothidinae (Lepidoptera: Noctuidae). Memoirs of the Entomological Society of Canada, 73. 59 pp.

Hartstack, A. W., J. A. Witz, and D. R. Buck. 1979. Moth traps for the tobacco budworm. J. Econ. Entomol. 72: 519-522.

Hou, M., and C. Sheng. 2000. Effects of different foods on growth, development, and reproduction of cotton bollworm, *Helicoverpa armigera* (Hubner) (Lepidoptera: Noctuidae). Acta Entomologica Sinica 43(2): 168-175. English summary pg. 174-175.

Jallow, M. F. A. and M. P. Zalucki. 1996. Within- and between- population variation in host-plant preference and specificity in Australian *Helicoverpa armigera* (Hübner) (Lepidoptera: Noctuidae). Australian journal of zoology 44: 503-519.

Johnson, D. No date. Plans and Parts List: “Texas” Style Cone Trap for Monitoring Certain Insect Pests. ENTFACT-010. 2 pp.

Kant, K., K. R. Kanaujia, and S. Kanaujia. 1999. Rhythmicity and orientation of *Helicoverpa armigera* (Hübner) to pheromone and influence of trap design and distance on moth trapping. Journal of Insect Science, 12: 6-8.

King, A. B. S. 1994. *Heliothis*/*Heliocoverpa* (Lepidoptera: Noctuidae). *In* Insect Pests of Cotton, G.A. Mathews and J.P. Turnstall (eds.). CAB International. Wallingford, U.K. pp. 39-106.

Kirkpatrick, T. H. 1961. Comparative morphological studies of *Heliothis* species (Lepidoptera: Noctuidae) in Queensland. Queensland Journal of Agricultural Science, 18: 179-194.

Kirkpatrick, T. H. 1962. Notes on the life-histories of species of *Heliothis* (Lepidoptera: Noctuidae) from Queensland. Queensland Journal of Agricultural and Animal Sciences 19: 567-570.

Kyi, A., and M. Zalucki. 1991. An experimental study of early stage survival of *Helicoverpa armigera* (Lepidoptera: Noctuidae) on cotton. Bulletin of Entomological Research 81: 263-271.

Loganathan, M. and S. Uthamasamy. 1998. Efficacy of a sex pheromone formulation for monitoring *Heliothis arrmigera* Hübner moths on cotton. Journal of Entomological Research, 22: 35-38.

Loganathan, M., M. Sasikumar, and S. Uthamasamy. 1999. Assessment of duration of pheromone dispersion for monitoring *Heliothis armigera* (Hüb.) on cotton. Journal of Entomological Research, 23: 61-64.

Mabbett, T., P. Dareepat, and M. Nachapong. 1980. Behaviour studies on *Heliothis armigera* and their application to scouting techniques for cotton in Thailand. Tropical Pest Management 26: 268-273.

Madden, A. D., M. J. Haggis, and J. Holt. 1993. *Helicoverpa armigera* oviposition on cotton in the Sudan Gezira associated with rainfall. Crop Protection 12 (1): 51-54.

Maelzer, D. and M. Zaluck. 1999. Analysis of long-term light-trap data for *Helicoverpa* spp. (Lepidoptera: Noctiuidae) in Australia: the effect of climate and crop host plants. Bulletin of Entomological Research 89: 455-463.

McCaffery, A. R., A. B. S. King, A. J. Walker, and H. El-Nayir. 1989. Resistance to synthetic pyrethroids in the bollworm, *Heliothis armigera* from Andhra Pradesh, India. Pesticide Science 27: 65-76.

Ng, S., R. Cibulsky, and S. Trowell. 1998. LepTon HTK - a heliothine diagnostic test kit: an update. In: Dugger P, Richter D, Proceedings of the Beltwide Cotton Conferences, Volume 2, pp. 1040-1043., pp. 1040-1043, Beltwide Cotton Conferences. National Cotton Council, Memphis, USA.

Nibouche, S. 1998. High temperature induced diapause in the cotton bollworm *Helicoverpa armigera*. Entomologia Experimentalis et Applicata 87: 271-274.

Pawar, C., S. Sithanantham, V. Bhatnagar, C. Srivastava, and W. Reed. 1988. The development of sex pheromone trapping of *Heliothis armigera* at ICRISAT, India. Tropical Pest Management, 34: 39-43.

Pogue, M. G. 2004. A new synonym of *Helicoverpa zea* (Boddie) and differentiation of adult males of *H. zea* and *f* (Hubner) (Lepidoptera:Nocutidae:Heliothinae). Ann. Entomol. Soc. Am. 97(6): 1222-1226.

Rochester, W., M. Zalucki, A. Ward, M. Miles, and D. Murray. 2002. Testing insect movement theory: empiciral analysis of pest data routinely collected from agricultural crops. Computers and Electronics in Agriculture 35: 139-149.

Saito, O. 1999. Flight activity changes of the cotton bollworm, *Helicoverpa armigera* (Hübner) (Lepidoptera: Noctuidae), by aging and copulation as measured by flight actograph. Applied Entomology and Zoology 35: 53-61.

Saour, G. and R. Causse. 1993. Oviposition behaviour of *Helicoverpa armigera* Hübner (Lepidoptera: Noctuidae) on tomato. Journal of Applied Entomology/Zeitschrift für Angewandte Entomologie, 115: 203-209. (English Abstract)

Sheng, C. F., J. W. Su, H. T. Wang, W. M. Fan, and W. J. Xuan. 2002. An efficiency comparison of cone and water tray traps baited with pheromone for capturing male moths of *Helicoverpa armigera*. Acta Entomologica Sinica, 45: 271-274. (English abstract).

Shimizu, K. and K. Fujisaki. 2002. Sexual differences in diapause induction of the cotton bollworm, *Helicoverpa armigera* (Hb.) (Lepidoptera: Noctuidae). Applied Entomology and Zoology 37: 527-533.

Sidde Gowda, D. K., S. Yelshetty, Y. K. Kotikal, B. V. Patil, and V. I. Benagi. 2002. Validation of integrated pest management of pigeonpea pod borer *Helicoverpa armigera*. International Chickpea and Pigeonpea Newsletter: 46-47.

Sigsgaard, L. and A. Ersbøll. 1999. Effects of cowpea intersowing and insecticide application on *Helicoverpa armigera* Hübner (Lepidoptera: Noctuidae) and its natural enemies in pigeonpea intercropped with sorghum. International Journal of Pest Management 45: 61-67.

Specht, A., D. R. S. Gomez, and S. Paula-Moraes. 2013. Resultado do trabalho inicial de levantamento da ocorrência da lagarta do gênero *Helicoverpa* - detecção da espécie *Helicoverpa armigera* no Brasil. Nota Técnica. Empresa Brasileira de Pesquisa Agropecuária (Embrapa). March 22, 2013.

Thomas, C. S., M. Yoshida, and J. Peter. 1997. Survival, growth, fecundity, and behavior of *Helicoverpa armigera* (Lepidoptera: Nocutidae) on pigeonpea and two wild *Cajanus* species. Journal of Economic Entomology 90(3): 837-841.

Tripathi, S., and R. Singh. 1991. Population dynamics of *Heliocoverpa armigera* (Hübner) (Lepidoptera: Noctuidae). Insect Science Applications 12: 367-374.

Trowell, S. C., G. A. Lang, and K. A. Garsia. 1993. A *Heliothis* identification kit, pp. 176-179. *In* S. A. Corey, D. J. Dall and W. M. Milne [eds.], Pest Control and Sustainable Agriculture. CSIRO Publishing, Collingwood, Australia.

Twine, P. H. 1978. Effect of temperature on the development of larvae and pupae of the corn earworm, *Heliothis armigera* (Hübner) (Lepidoptera: Noctuidae) . Queensland Journal of Agricultural and Animal Sciences 35(1): 23-28.

Venette, R. C., E. E. Davis, J. Zaspel, H. Heisler, and M. Larson. 2003. Mini Risk Assessment Old World bollworm, *Helicoverpa armigera* Hübner [Lepidoptera: Noctuidae]. Cooperative Agricultural Pest Survey, Animal and Plant Health Inspection Service, US Department of Agriculture. Available online at: http://www.aphis.usda.gov/plant_health/plant_pest_info/pest_detection/downloads/pra/harmigerapra.pdf

Visalakshmi, V., P. Arjuna Rao, and P. Krishnayya. 2000. Utility of sex pheromone for monitoring *Heliothis armigera* (Hüb.) infesting sunflower. Journal of Entomological Research, 24: 255-258.

Voros, G. 1996. Damage of cotton bollworm (*Helicoverpa armigera* Hubner) in grapevine. Novenyvedelem 32(5): 229-234. (English Summary).

Walker, G. and P. Cameron. 1990. Pheromone trapping and field scouting for tomato fruitworm in tomatoes and sweet corn. pp. 17-20, Proceedings of the 43rd New Zealand Weed and Pest Control Conference. New Zealand Weed and Pest Control Society, Inc.

Zhou, X., S. Applebaum, and M. Coll. 2000. Overwintering and spring migration in the bollworm *Helicoverpa armigera* (Lepidoptera: Noctuidae) in Israel. Environmental Entomology, 29: 1289-1294.

Leguminivora glycinivorella

Scientific names
Leguminivora glycinivorella Matsumura

Synonyms:
Cydia glycinivorella, *Eucosma glycinivorella*, *Grapholita glycinivorella*, *Laspeyresia glycinivorella*

Common Name(s)
Soybean pod borer, soybean moth, soybean pod moth

Type of Pest
Moth

Taxonomic Position
Class: Insecta, **Order:** Lepidoptera, **Family:** Tortricidae

Reason for Inclusion in Manual
National threat

Pest Description
The Tortricidae are among the largest of the families of the so-called micro-lepidoptera, with over 5000 species described worldwide. In North America, there are approximately 1,200 described species (Triplehorn and Johnson, 2005). Tortricidae members are more commonly found in temperate and tropical upland regions than in the lowland tropics (Meijerman and Ulenberg, 2000). Many members of this family are leafrollers. Moths in this family are small, and gray, tan or brown in color with dark bands or mottled wing areas. Front wings are often square-tipped. Wings are held roof-like over the body when at rest (Triplehorn and Johnson, 2005).

Figure 1. *L. glycinivorella* adult male. Photo courtesy of R. De Vos http://ip30.eti.uva.nl/BIS/tortricidae.php?selected=beschrijving&menuentry=soorten&id=229.

Leguminivora glycinivorella was first described by Matsumura in 1900. In Asia, *L. glycinivorella* is associated with late season soybean plants and is found on pods and seeds (Sinclair et al*.,* 1997). Adults are small dark-colored moths (Fig. 1, 2). *Leguminivora glycinivorella* larvae are responsible for the

majority of soybean crop loss.

Eggs: *Leguminivora glycinivorella* eggs are flat and oval (Fig. 2), measuring 0.48 x 0.35 mm. They are pearly-white when freshly laid. During development, a red spot appears that may later fuse to a pink streak (Meijerman and Ulenberg, 2000).

Larvae: The larval stage of the non-hibernating generation lasts 18 to 25 days, during which the larva undergoes five instars. When young, larvae are orange-yellow in color, changing to milky white or greenish in the third instar and turning orange or pink in the final instar. Heads are black and prothoracic shield is brownish (Meijerman and Ulenberg, 2000).

Pupae: Brown in color, pupae are 6 to 7mm (approx. $^{1}/_{4}$ in) in length (Meijerman and Ulenberg, 2000).

Adults: Adult (Fig. 1) wingspans measure 13 to 17 mm (approx. $^{1}/_{2}$ to $^{11}/_{16}$ in). The forewing is gray with weak purplish blue hue, becoming more yellowish near the termen. Fasciae are fuscous, irregular and narrow. Dorsal spot is well developed, giving rise to dark colored stria. Ocellus has three small black dashes. Hindwing is fuscous, and paler in color basally (Meijerman and Ulenberg, 2000).

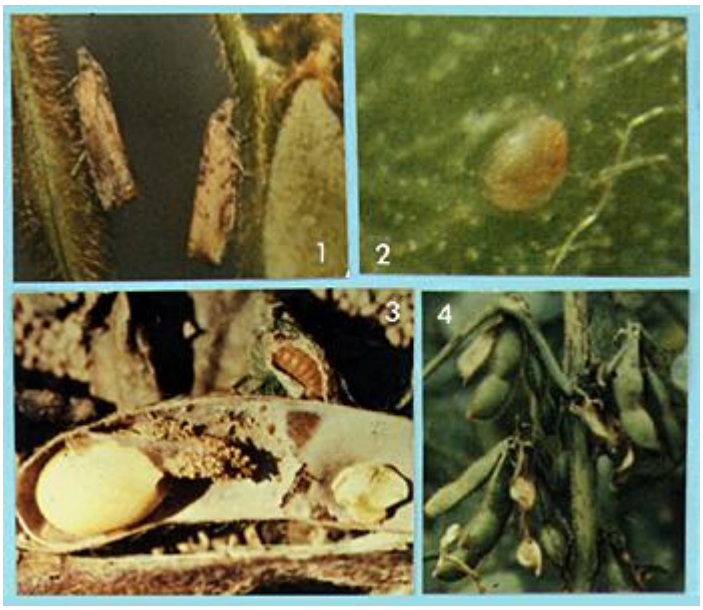


Figure 2: Adults (1) and egg (2) of *L. glycinivorella*. Damage to seeds (3) and to pods (4). Photos courtesy of WJATC, Korea.

Adult male (external characters): 13 to 17 mm mm (approx. $^{1}/_{2}$ to $^{11}/_{16}$ in) wingspan; head and thorax ochreous-brown, abdomen fuscous. Forewing grey with weak purplish blue hue, more yellowish near termen, the latter with a slight notch. Costal strigulae brown, some giving rise to bluish striae reaching termen. Interspaces between costal strigulae yellowish. Basal, subbasal and median fasciae fuscous, irregular, narrow, angulate near costa. Interspaces between these with dark colored irregular spots. Dorsal spot well developed, fuscous, triangular, giving rise to dark colored stria; this stria connecting to stria arising from costal strigula, forming a 'T-shaped' marking. Ocellus ochreous with three small black dashes. Cilia dark yellowish. Hindwing with anal fold, fuscous, paler basally; cilia yellowish grey (Meijerman and Ulenberg, 2000).

Male genitalia: Tegumen long, broad terminally, proximal portion expanding dorsally, with long-haired patches situated laterally before apex of tegumen. Cucullus broad, somewhat expanding posteriorly; notch in ventral margin of valva rather small. Aedeagus long, curved (Meijerman and Ulenberg 2000).

Female adult (external characters): Similar forewing to male; hindwing without anal fold.

Female genitalia: Ovipositor fairly long, papillae analis small. Sterigma in form of a weakly sclerotized, indistinct lamella postvaginalis marked with some terminal hairs; ostium with short sclerite; ductus bursae long, membranous; corpus bursae with well developed signa and posterior diverticulum.

Biology and Ecology

Leguminivora glycinivorella is univoltine in northern Japan (Sakagami et al., 1985); however, a second generation has been noted in other locations. Adults emerge in late-July to early August, and females oviposit on young bean pods. Larvae, once hatched, enter pods and eat immature beans. In mid- to late-October, full grown larvae leave the pods, enter the soil and spin cocoons usually at a depth of down to 3 cm (approx. 1 $^{3}/_{16}$ in) and overwinters in the soil (Shimada et al., 1984). The larvae spend eight or more months in cocoons until pupation the following year, which occurs in July, approximately. The cocoon does not protect the larva from ultra-low temperatures, but it is thought to prevent inoculative freezing, which takes place at approximately -4.0°C (24.8°F). Cocoons also provide protection from submergence during early spring flooding and pre-emergence (Sakagami et al., 1985).

The adult female lives for 8 to 30 days and is active in the morning and evening. Females selectively lay about 160 to 300 eggs each just after completion of pod elongation (Kobayashi, 1976; Meijerman and Ulenberg, 2000). Over 80% of the eggs are deposited on young pods. Before young pods are available, petioles and stipules are common sites. After 7 to 9 days, the eggs hatch. The larva spins a loose silken covering, probably for support when gouging out pod tissue (Meijerman and Ulenberg, 2000).

Pest Importance

L. glycinivorella is considered one of the most serious soybean pests in Northeast Asia (Sakagami et al., 1985). It is a severe pest in the plains of north east China. Seed damage in soybean fields usually is 10% to 20% and may be greater than 40% for some susceptible cultivars when severely attacked (Zhang and Fu, 1983).

Symptoms/Signs

Larvae feed on the seeds inside the pod (Fig. 2). The entrance hole in the pod created by *L. glycinivorella* is very small, and the callus tissue formed over it resembles the feeding punctures made by pod sucking bugs. Inside the pod, the larva feeds on the seeds. The number of larvae per pod varies with pod size and host variety (Meijerman and Ulenberg, 2000). Soybean seeds, incompletely consumed by the developing larvae,

can be found within pods (Kobayashi, 1976). Minute, ellipsoidal fecal pellets can also be observed.

Late and widely spaced planting tends to result in heavier pod-borer damage than does early and dense planting. The date of pod setting and the duration of pod ripening also appear to be related to the damage-rate (Meijerman and Ulenberg, 2000).

Known Hosts

Major Host
Glycine max (soybean) and *Phaseolus* spp. (beans

Wild Hosts
Lupinus spp. (lupine), and *Pueraria lobata* (kudzu)

Known Vectors (or associated organisms)

Leguminivora glycinivorella is not a known vector and does not have any associated organisms.

Known Distribution

Leguminivora glycinivorella is known to occur in China, Japan, Korea, and areas of the former Soviet Union (Meijerman and Ulenberg, 2000).

Pathway

Neither this species nor genus has been intercepted at U.S. ports of entry. This species could potentially move through infested host material.

Potential distribution within the United States

Specific information is not available at this time. Areas growing soybean or common bean are at risk.

Survey

CAPS-Approved Method*: Has not been examined at this time.

*For the most up-to-date methods for survey and identification, see Approved Methods on the CAPS Resource and Collaboration Site, at http://caps.ceris.purdue.edu/.

Literature-Based Methods:
Visual: Other moths in this family are surveyed using visual observation of symptoms, larvae, pupae, webbing, and frass.

Trapping: A pheromone has recently been extracted from *L. glycinivorella* by Vang et al. (2006). The extract contained dodecyl acetate, (8E,10E)-8,10 dodecadienyl acetae (E8,E10-12:OAc) and its (8E,10Z)-isomer in a ratio of 10:100:25. In a soybean field, synthetic E8,E10-12:OAc successfully attracted male moths of *L. glycinivorella*. The

role of the two minor components identified in the extract is unclear. The commercial availability of this pheromone, however, is currently unknown.

Key Diagnostics/Identification

CAPS-Approved Method*: Has not been examined at this time.

*For the most up-to-date methods for survey and identification, see Approved Methods on the CAPS Resource and Collaboration Site, at http://caps.ceris.purdue.edu/.

Literature-Based Methods: A morphological identification is required for *L. glycinivorella*. *L. glycinivorella* are small, dark colored moths that must generally be distinguished using dissection of genetalia. Small entrance holes on pods can be observed. Larvae may be found inside pods.

Easily Confused Pests

The tortricid pod-borer *Fulcrifera orientis,* collected from *Sophora flavescens* (a medicinal plant) in Japan, has been confused with *L. glycinivorella* (Meijerman and Ulenberg, 2000). *F. orientis* can be differentiated based on male genitalia. The aedeagus of *F. orientis* is armed with a long process, originating from the anellus above the base of the coecum penis.

References

Kobayashi, T. 1976. Pod borers and the seed pest complex in Asian soybeans. Intsoy. Int Soybean Prog. Ser. 10: 171-173.

Meijerman, L. and Ulenberg, S.A..2000. Arthropods of Economic Importance: Eurasian Tortricidae. http://ip30.eti.uva.nl/bis/tortricidae.php?menuentry=inleiding.

Sakagami, S.F., Tanno, K., Tsutsui, H., and Honma, K. 1985. The role of cocoons in overwintering of the soybean pod borer *Leguminivora glycinivorella* (Lepidoptera: Tortricidae). Journal of the Kansas Entomological Society 58: 240-247.

Shimada, K., Sakagami, S.F., Honma, K., and Tsutsui, H. 1984. Seasonal changes of glycogen/trehalose contents, supercooling points and survival rate in mature larvae of the overwintering soybean pod borer *Leguminivora glycinivorella*. Journal of Insect Physiology 30(5): 369-373.

Sinclair, J.B., Kogan, M., and McGlamery, M.D. 1997. Guidelines for the Integrated Management of Soybean Pests. National Soybean Research Laboratory Publication Number 2, College of Agricultural, Consumer and Environmental Sciences, University of Illinois at Urbana-Champaign, Urbana, IL. http://www.nsrl.uiuc.edu/news/nsrl_pubs/.

Triplehorn, C.A. and Johnson, N.F. 2005. Borror and DeLong's Introduction to the Study of Insects, 7th Ed. Thompson Learning.

Vang, L.V., Ishitani, M., Komai, F., Yamamoto, M., and Ando, T. 2006. Sex pheromone of the soybean pod borer, Leguminivora glycinivorella (Lepidoptera:Totricidae): Identification and field evaluation. Appl. Entomol. Zool. 41(3):507-513.

Zhang, Z.J., and Fu, C.Q. 1983. Inheritance of resistance to soybean pod borer in soybean. Intsoy. Int Soybean Prog. Ser. 25: 74-79.

Mamestra brassicae

Scientific Name
Mamestra brassicae Linnaeus

Synonyms:
Barathra brassicae, Hypobarathra unicolor, *Mamestra brassicae* var. *andalusica*, *M. brassica* var. *decolorata*, *Noctua albidilinea, Phalaena noctua brassicae*, *Phalaena omicron*

Common Name(s)
Cabbage moth, cabbage armyworm

Type of Pest
Moth

Taxonomic Position
Class: Insecta, **Order:** Lepidoptera, **Family:** Noctuidae

Reason for Inclusion in Manual
National Threat

Pest Description
Eggs: The eggs are relatively small, hemispherical, ribbed and reticulate. They are whitish in color when newly laid, but turn gradually to purplish-brown with a brown to purple micropyle and basal ring. A few hours before hatching, they darken to grayish-black. The eggs are laid singly in regular batches of up to 20 to 30 eggs, mainly on the undersides of leaves.

Figure 1. Eggs of *M. brassicae*. Photo courtesy of R. Coutin /OPIE.

Larvae: There are six instars. First- and second-instar larvae are about 3 to 10 mm (approx. $^{1}/_{8}$ to $^{5}/_{16}$ in) long, greenish and more or less translucent with black hairs on black warts. First-instar larvae have a black head capsule, but after the first molting it turns light-brown (Fig. 1). The prolegs on the third and fourth abdominal segments are poorly developed in the first two or three instars. From the third instar, the larvae are pale-green with yellowish intersegmental bands. The dorsal

region turns gradually darker with each molt, and in the last instar the majority of the larvae are brownish-green or blackish-green.

Heath and Emmet (1979) described full-grown larvae. The body is about 50 mm (approx. 2 in) long, elongate, and with a slight dorsal hump on abdominal segment 8. The head capsule is light-brown, and the dorsal region of the body is from fairly bright-green, through brownish-green to almost black. The dorsal line is fine and black. On each side, there is one sub-dorsal line of blackish bars. The spiracular line is broad and pale-green or pale-ochreous. The spiracles are white. The ventral region is yellowish-green.

Figure 2. Larvae of *M. brassicae*. Photos courtesy of R. Coutin /OPIE and D. Griffin (http://ukmoths.org.uk).

Pupae: The pupae are elongate, 17 to 22 mm (approx. $^{11}/_{16}$ to $^{7}/_{8}$ in) long, and reddish-brown and glossy. The wing- and limb-cases are finely sculptured. The abdominal segments are darker brown and evenly tapered, and there is a finely pitted anterior band on each segment. Segment 8 is sharply excavated to a narrow conical cremaster with two short apically hooked spines. Pupation takes place within flimsy cocoons in the soil (Heath and Emmet, 1979).

Adults: The adult moths have a wingspan of 35 to 50 mm (approx. 1 $^{3}/_{8}$ to 2 in). The forewings are mottled and may appear grey-brown, brown or blackish-brown, with variable reddish-brown scaling. Sub-basal, antemedian and postmedian lines are inconspicuous and slightly paler than the background color, and have a fine dark edge. A kidney-shaped stigmata outlined in black with a whitish distal margin and a less clearly defined proximal margin, is placed near the center of each forewing. The subterminal line is very variable. When present it is whitish and irregular, with two

angular projections (like a W). The hindwings are fuscous and generally paler than the forewings. They are light-grayish towards the base, and have a darker terminal shade. The fringe has a grayish central line. The eyes are hairy and the forelegs have a characteristic brown, slightly curved, apically pointed tibial spur. Like other species in Hadeninae, the eyes are hairy.

Biology and Ecology

Mamestra brassicae usually exhibits two generations per year but there may be only 1 in colder regions (Johnasen, 1997; Fowler and Lakin, 2000). The life cycle is as follows: 1) Adult moths mate and oviposit from May to June. 2) Up to 200 eggs are laid in groups of 20-30 on the under surface of leaves. The eggs hatch in 10-15 days. 3) Larvae undergo 5 molts with the first 4 instars feeding on the underside of the outermost leaves. The last instars feed in the center or crown of the plants (Omino et al., 1973). Larval development requires approximately 2 months and culminates in late July. 4) Mature larvae then burrow into the soil surface and pupate. 5) From late July to September, the second generation emerges and repeats the life cycle. 6) From late August through October, the mature larvae burrow into the soil and enter diapause (Fowler and Lakin, 2000). The species is nocturnal in habit and emergence from pupae, flight, mating activity, egg deposition, and feeding mostly take place during the dark period.

Figure 3. Adult of *M. brassicae*. Photo courtesy of I. Kimber (http://ukmoths.org.uk).

Rojas (1999) showed that female *M. brassicae* are more attracted/landed significantly more on damaged (mechanical or locust damage) cabbage plants than undamaged cabbage plants. This behavior was not observed in chrysanthemum or tomato.

According to Kimber (2008), the species shows a rather complex life-history with two or three overlapping generations and moths on the wing from May to September in the United Kingdom.

Diapause is facultative and occurs in the pupal stage. Under harsh environmental conditions (*e.g.* high density, cold and low humidity) *M. brassicae* larvae exhibit phase polymorphism (Goulson, 1994; Goulson and Cory, 1995; Kazimirova, 1992). The larvae will become melanized. In this state, they exhibit rapid development, size reduction, and elevated body temperatures, which promote larval survival (Goulson, 1994; Goulson and Cory, 1995). In addition, *M. brassicae* can modify its pupal duration depending

upon environmental conditions (Sauer and Gruner, 1988). This allows *M. brassicae* to optimally exploit a given habitat while reducing the risk of mortality (Fowler and Lakin, 2000).

Pest Importance From CABI (2007)

Mamestra brassicae is a serious pest, mainly on *Brassica* spp., beets and legumes, but also on other vegetable crops (Heath and Emmet, 1979; Filippov, 1982; Poitout and Bues, 1982; Hommes 1983; Øgaard, 1983; Kahrer, 1984; Injac and Krnjajic, 1989; Finch and Thomson, 1992; Van de Steene, 1994). In these areas, the greatest damage is usually caused by the larvae of the second generation, which are often more numerous than the first generation (Kahrer, 1984; Injac and Krnjajic, 1989). In the northern areas (Scandinavia and Finland), the occurrence *of M. brassicae* as a serious pest is more sporadic (Skou, 1991; Johansen, 1997).

In cabbage crops in Germany, *M. brassicae* is a main pest with regular occurrence. In field experiments, 27 to 98% of the plants in different cabbage crops were infested (Hommes, 1983). According to Filippov (1982) larval infestation of cabbage in Moldavia led to harvest losses of 8 to 80%. In a study of white cabbage in Norway, weight losses due to larval damage were 10 to 13% (Rygg and Kjos, 1975). In Belgium, insecticides are often applied to Brussels sprouts every 2 to 3 weeks to control *M. brassicae* larvae (Van de Steene, 1994). Because of the tunneling action of the larvae, they are often sheltered from any insecticide application.

Symptoms/Signs

Small larvae feed on the underside of the external leaves, where they make small perforations. As the larvae grow older, the feeding holes become larger. Severe infestations of small larvae may rapidly skeletonize the leaves, and can sometimes destroy small plants. Older larvae tunnel into the heart of the plants. They leave considerable amounts of feces, which favor growth of decaying bacteria and fungi. Most crop losses caused by the larvae occur as a result of boring and fouling rather from the amount of plant tissue eaten. Even slight infestations of older larvae can be damaging, particularly in crops such as heading cabbage, where the larvae destroy the marketable product (Heath and Emmet, 1979; Finch and Thomson, 1992).

In cauliflower and broccoli, the larvae also feed on the inflorescence, where they chew more or less deep holes. Small larvae live well hidden between the flower stems and may pass sorting procedures, contaminating processed products.

Soybean leaves may be completely skeletonized. The feeding may destroy young buds, leading to distorted growth. The larvae bore into the pods and feed on the seeds (Lihnell, 1940).

The larvae feed on leaves, buds and petals in ornamentals such as *Dahlia*, *Chrysanthemum* and *Rosa* spp., and they may bore into the fruits in fruiting crops, such as tomato.

Known Hosts

Mamestra brassicae are extremely polyphagous (70 species in 22 plant families), although they prefer species in the Brassicaceae and Chenopodiaceae (Heath and Emmet, 1979; Skou, 1991; Finch and Thomson, 1992; Popova, 1993). Beets, legumes, lettuces, onions and potatoes are also frequently reported to be infested (Øgaard, 1983; Injac and Krnjajic, 1989; Finch and Thomson, 1992; Zhang, 1994).

Major hosts

Allium cepa (onion), *Allium sativum* (garlic), *Beta vulgaris* var. *vulgaris* (sugarbeet), *Brassica* spp., *Brassica oleracea* (cabbages, cauliflowers), *Brassica oleracea* var. *botrytis* (cauliflower), *Brassica oleracea* var. *capitata* (cabbage), *Brassica oleracea* var. *gemmifera* (Brussels sprouts), *Brassica rapa* subsp. *pekinensis* (Pe-tsai), *Glycine max* (soybean), *Lactuca sativa* (lettuce), *Nicotiana* spp., *Nicotiana tabacum* (tobacco), *Phaseolus* spp. (beans), *Phaseolus vulgaris* (common bean), *Pisum sativum* (pea), *Solanum lycopersicum* (tomato), *Solanum tuberosum* (potato), and *Zea mays* (maize).

Minor hosts

Amaranthus retroflexus (redroot), *Aquilegia vulgaris* (European columbine), *Betula pendula* (European white birch), *Bryonia alba* (white bryony), *Calendula* spp., *Callistephus chinensis* (China aster), *Cannabis sativa* (hemp), *Capsella bursa-pastoris* (shepherd's purse), *Capsicum* spp. (peppers), *Capsicum annuum* (bell pepper), *Chenopodium album* (lambsquarters), *Chenopodium giganteum*, *Chrysanthemum* spp. (daisy), *Cucurbita pepo* (squash), *Dahlia* spp., *Daucus carota* (carrot), *Dianthus caryophyllus* (carnation), *Epilobium* spp. (fireweed), *Fagus* spp. (beech), *Fallopia convolvulus* (=*Polygonum convolvulus*) (wild buckwheat), *Fragaria* spp., *Geum rivale* (purple avens), *Gladiolus* spp., *Helianthus annuus* (sunflower), *Humulus lupulus* (hop), *Hyoscyamus niger* (black henbane), *Hyssopus officinalis* (hyssop), *Ipomoea batatas* (sweet potato), *Lamprocapnos spectabilis* (=*Dielytra spectabilis*) (seal-flower), *Larix* spp. (larch), *Linum usitatissimum* (flax), *Lupinus* spp. (lupine), *Malus domestica* (apple), *Malus sylvestris* (apple), *Medicago sativa* (lucerne), *Papaver somniferum* (poppy), *Pelargonium* spp. (geranium), *Potentilla anserina* (silverweed), *Prunus padus* (European bird cherry), *Prunus persica* (peach), *Prunus salicina* (Japanese plum), *Quercus* spp. (oak), *Quercus cerris* (European turkey oak), *Quercus robur* (English oak), *Rhaphanus sativus* (radish), *Rheum* x *rhabarbarum* (rhubarb), *Rosa* spp. (roses), *Rubus idaeus* (raspberry), *Rudbeckia* spp. (coneflower), *Salix* spp. (willow), *Salix caprea* (goat willow), *Sambucus racemosa* (red elderberry), *Senecio vulgaris* (groundsel), *Silene chalcedonica* (=*Lychnis chalcedonica*) (maltesecross), *Silene latifolia* subsp. *alba* (=*Melandrium album*), *Solanum melongena* (eggplant), *Spinacia oleracea* (spinach), *Trifolium repens* (white clover), *Vicia faba* (broad bean), *Vicia sativa* (vetch), and *Vitis vinifera* (grape) (USDA, 1986; Savela, 2001, CABI, 2008).

Intercepted on

Aconitum spp. (monkshood), *Alstroemeria* spp., *Apium graveolens* (celery) (USDA, 1986).

Known Vectors (or associated organisms)

Mamestra brassicae is not a known vector and does not have any associated organisms.

Known Distribution

Mamestra brassicae is present throughout the Palaearctic region from Europe to Japan and subtropical Asia. According to Finch and Thomson (1992), *M. brassicae* is abundant throughout Central Europe and temperate Asia. Øgaard (1983) states that the species is present mainly between 30°N and 70°N. The species is abundant all over Denmark and in southern Scandinavia and Finland (Skou, 1991). In Norway, *M. brassicae* occurs as a pest up to 62°N (Johansen, 1997). The species is not found on Iceland.

Africa: Canary Islands, Libya. **Asia:** Armenia, Azerbaijan, China, Georgia (Republic), India, Iran, Japan, Kazakhstan, Korea, Kyrgyzstan, Lebanon, Mongolia, Pakistan, Syria, Turkey, and Uzbekistan. **Europe:** Austria, Belarus, Belgium, Bulgaria, Czech Republic, Denmark, Finland, France, Germany, Hungary, Ireland, Italy, Latvia, Lithuania, Malta, Moldova, Netherlands, Norway, Poland, Portugal, Romania, Russia, Serbia and Montenegro, Slovakia, Spain, Sweden, Switzerland, Taiwan, Ukraine, and United Kingdom (EPPO, 2012).

Pathway

Mamestra brassicae could potentially move through international trade. This species has been intercepted at U.S. ports of entry over 1,300 times. Most of these interceptions originated on material from the Netherlands (1,207), as well as France (25) and Italy (25). Many interceptions occurred on host material including *Delphinium* sp. (364), *Aconitum* sp. (163), *Brassica* sp. (114), *Amaranthus* sp. (79), *Alstroemeria* sp. (43), and *Anemone* sp. (41). Infested material was found in permit cargo (1,154), general cargo (89), baggage (52), and stores (32). Most material was for consumption (1,294) while the rest was for non-entry and propagation (33 and 7) (AQAS, 2012; September 11, 2012).

Potential Distribution within the United States

The species is not present in America or Oceania (APPPC, 1987; Zhang, 1994). The predicted range for *M. brassicae* based on habitat suitability and host availability includes 8 USDA plant hardiness zones (3-10). According to Fowler and Lakin (2000), if introduced it is probable that *M. brassicae* could establish in following states based on habitat and host suitability (conservative estimate): Alabama, Arizona, Arkansas, California, Colorado, Delaware, Florida, Georgia, Idaho, Illinois, Indiana, Iowa, Kentucky, Louisiana, Maine, Maryland, Massachusetts, Michigan, Minnesota, Missouri, Montana, Nebraska, Nevada, New Jersey, New Mexico, New York, North Carolina, Ohio, Oklahoma, Oregon, Pennsylvania, Rhode Island, South Carolina, Tennessee, Texas, Utah, Virginia, West Virginia, and Wisconsin.

Survey

CAPS-Approved Method*: Has not been evaluated at this time.

*For the most up-to-date methods for survey and identification, see Approved Methods on the CAPS Resource and Collaboration Site, at http://caps.ceris.purdue.edu/.

Literature-Based Methods:

Trapping: Adults can be detected with pheromone or light traps. Multiple lures for *M. brassicae* have been described. Z11-hexadecenyl is the main component of many of these lures. A lure is available from the CPHST- Otis lab. Z11-hexadecenyl acetate (1 mg/lure) is dispensed using a polyethylene “beem” capsule and should be replaced every 12 weeks. A listing of lures and key references is given at: http://www.pherobase.com/database/species/species-Mamestra-brassicae.php.

Visual survey: Egg batches and small larvae (less than about 1.5 cm) are found mostly on the undersides of the larger external leaves. Look for eggs on the underside of leaves, petioles, or stems. Feeding perforations from the smallest larvae are difficult to detect. Large larvae are found between the internal leaves in the heart of plants, in tunnels or cavities in cabbage heads, flowers, buds or fruits. Look for feeding holes, entrance holes and feces. Larvae are active at night and often curl up when disturbed. Larvae can easily be observed when cabbage are cut open.

Crop scouting should be done on a number of plants per field at least weekly, and should start 1 to 2 weeks after the first adults are caught in the traps. Scouting methods have been developed and are recommended when the moth is known to be present (Kahrer, 1984; Freuler, 1992; Planteforsk and ITAS, 1997).

Key Diagnostics/Identification

CAPS-Approved Method*: Has not been evaluated at this time.

*For the most up-to-date methods for survey and identification, see Approved Methods on the CAPS Resource and Collaboration Site, at http://caps.ceris.purdue.edu/.

Literature-Based Methods: A morphological identification is required for *M. brassicae*. Identification of adult noctuids is often based on characteristics of male genitalia. *Mamestra brassicae* can be identified by the presence of a curved ‘spur’ on the tibia of the foreleg.

Easily Confused Pests

It is difficult to distinguish between larvae from different noctuid species, especially in the youngest instars. See Heath and Emmet (1979) or Skinner (1998) for full description. Larvae of *M. brassica* can be confused with the domestic *Pieris rapae* (small white butterfly) and the exotic *P. brassicae* (large white butterfly). Color varies, but *M. brassicae* larvae have smooth skin and few hairs; while *P. rapa* and *P. brassicae* appear velvety (USDA, 1986).

The adults resemble many other dull-colored members of the Noctuidae. *Mythimna pallens, Discestra trifolii,* and *Lacanobia w-latinum c*an be distinguished from *M. brassicae* by no tibial spur on foreleg. *Manilkara zapota* and *Apamea* spp. can be distinguished from *M. brassicae* by no tibial spur on foreleg, and glabrous eyes.

References

APPPC. 1987. Insect pests of economic significance affecting major crops of the countries in Asia and the Pacific region. Technical Document No. 135. Bangkok, Thailand: Regional FAO Office for Asia and the Pacific (RAPA), 56 pp.

AQAS. 2012. AQAS database, *Mamestra brassicae* interceptions. Last accessed September 11, 2012 from: https://mokcs14.aphis.usda.gov/aqas/HomePageInit.do.

Bonnemaison, L. 1965. Insect pests of crucifers and their control. Annual Review of Entomology, 10:233-356.

CABI. 2007. Crop Protection Compendium Wallingford, UK: CAB International. www.cabicompendium.org/cpc.

CABI. 2008. Crop Protection Compendium Wallingford, UK: CAB International. www.cabicompendium.org/cpc.

EPPO. 2012. EPPO Plant Quarantine Information Retrieval System, (version 5.0.5540). European and Mediterranean Plant Protection Organization.

Filippov, N. 1982. Integrated control of pests of vegetable crops grown in the open in Moldavia. Acta Entomologica Fennica, 40:6-9.

Finch, S. and A. R. Thompson. 1992. Pests of cruciferous crops. In: McKinlay RG ed. Vegetable Crop Pests. Basingstoke, UK: Macmillan, 87-138.

Fowler, G. A., and K. R. Lakin. 2000. Risk Assessment: The cabbage army moth, *Mamestra brassicae* (L.), (Lepidoptera:Noctuidae). USDA-APHIS

Freuler, J. 1992. Guidelines and scouting methods in integrated production of field vegetable crops; the situation in Switzerland. Bulletin OILB/SROP, 15:122-126.

Goulson, D. 1994. Determination of larval melanisation in the moth, *Mamestra brassicae*, and the role of melanin in thermo-regulation. Heredity 73: 471-479.

Goulson, D., and J. S. Cory. 1995. Responses of *Mamestra brassicae* (Lepidoptera: Noctuidae) to crowding interactions with disease resistance, colour phase and growth. Oecologia 104: 416-423.

Heath, J. and A. M. Emmet. (eds). 1979. The Moths and Butterflies of Great Britain and Ireland. Vol. 9. Colchester, UK: Harley Books. Pages 214-215.

Hommes, M. 1983. Studies on the population dynamics and integrated control of cabbage pests. Mitteilungen aus der Biologischen Bundesanstalt fur Land- und Forstwirtschaft, No. 213:210 pp.

Injac, M. and S. Krnjajic. 1989. Phenology of the cabbage moth (*Mamestra brassicae* L.) in the Belgrade region. Zastita Bilja, 40:423-431.

Johansen, N. S. 1997. Mortality of eggs, larvae and pupae and larval dispersal of the cabbage moth, *Mamestra brassicae*, in white cabbage in south-eastern Norway. Entomologia Experimentalis et Applicata, 83(3):347-360.

Kahrer, A. 1984. The cabbage moth (*Mamestra brassicae* L.). Pflanzenarzt, 37:132-133.

Kazimirova, M. 1992. The role of physical contact in the induction of phase polymorphism of *Mamestra brassicae* (Lepidoptera: Noctuidae). Acta Entomol Bohemoslov 89: 87-95.
Kimber, I. 2008. 2154 Cabbage Moth *Mamestra brassicae*. http://ukmoths.org.uk/show.php?bf=2154

Lihnell, D. 1940. Sjukdomar og Insektsangrepp i srets soyaodlingar. [Diseases and insect infestations in soybeans]. VSxtskyddnotiser, 4:90-92.

Øgaard, L. 1983. The cabbage moth (*Mamestra brassicae* (L.), Lepidoptera, Noctuidae). Development in Denmark illustrated by a simple simulation. Entomologiske Meddelelser, 50:36-46.

Planteforsk and ITAS. 1997. Brukerhsndbok TELEVIS. Rsd om plantedyrking. [Guidelines for the use of TELEVIS. Advice on crop growing]. The Norwegian Crop
Research Institute, Plant Protection Centre and the Instrument Service AS, Norway. In Norwegian.

Poitout, S. and R. Bues. 1982. The principal injurious noctuids. Phytoma, No. 337:39-43.

Popova, T. 1993. A study of antibiotic effects on cabbage cultivars on the cabbage moth *Mamestra brassicae* (Lepidoptera: Noctuidae). Entomol. Rev. 72 125-.

Rojas, J. C. 1999. Influence of host plant damage on the host-finding behavior of *Mamestra brassicae* (Lepidoptera: Noctuidae). Physiological and Chemical Ecology 28(4): 588-593.

Rygg, T. and ¥. Kjos. 1975. Kslfly, (*Mamestra brassicae* (L.)). Noen unders¢kelser over dets biologi og bekjempelse. [Biology and control of the cabbage moth (*Mamestra brassicae* (L.)).] Gartneryrket, 65:286-290.

Sauer, K. P. and C. Gruner. 1988. Aestival dormancy in the cabbage moth *Mamestra brassicae* L. (Lepidoptera:Noctuidae) 2. Geographical variation in two traits: day length thresholds triggering aestival dormancy and duration of aestival dormancy. Oecologia 76: 89-96.

Savela, M. 2001. Lepidoptera and Other Life Forms: Mamestra brassicae. Accessed September 11, 2012 from:
http://www.nic.funet.fi/pub/sci/bio/life/insecta/lepidoptera/ditrysia/noctuoidea/noctuidae/hadeninae/mamestra/index.html#brassicae.

Skinner, B. 1998. Colour Identification Guide to the Moths of the British Isles (Macrolepidoptera). 2nd edition. London, UK: Viking.

Skou, P. 1991. Nordens Ugler. Hsndbog over de i Danmark, Norge, Sverige, Finland og Island forekommende arter av Herminiidae og Noctuidae (Lepidoptera). [The Hermidiidae and Noctuidae of Denmark and Fennoscandinavia.] Stenstrup, Denmark. Apollo Books.

USDA. 1986. Pests not known to occur in the United States of limited distribution No. 74: Cabbage Moth. USDA-APHIS-PPQ.

Van de Steene, F.. 1994. Laboratory studies of the effectiveness of insecticides against caterpillars of the cabbage moth, *Mamestra brassicae* (L.). Bulletin OILB SROP, 17:194-201.

Zhang, B. C. (Compiler). 1994. Index of economically important Lepidoptera. Wallingford, UK: CAB International, 599 pp.

Spodoptera littoralis

Scientific Name
Spodoptera littoralis Boisduval

Synonyms:
Hadena littoralis, Noctua gossypii, Prodenia littoralis, Prodenia litura, Prodenia retina, Spodoptera retina, Spodoptera testaceoides

The two Old World cotton leafworm species *S. littoralis* and *S. litura* are allopatric, their ranges covering Africa and Asia, respectively. Many authors have regarded them as the same species, but they have been differentiated based on adult genitalia differences (Mochida, 1973; CABI, 2007).

Common Name(s)
Egyptian cotton leafworm, cotton leafworm, Mediterranean climbing cutworm, tobacco caterpillar, tomato caterpillar, Egyptian cotton worm, Mediterranean brocade moth, Mediterranean climbing cutworm

Type of Pest
Moth

Taxonomic Position
Class: Insecta, **Order:** Lepidoptera, **Family:** Noctuidae

Reason for Inclusion in Manual
CAPS Target: AHP Prioritized Pest List – 2009 through 2013

Pest Description
Eggs: Spherical, somewhat flattened, 0.6 mm in diameter, laid in clusters arranged in more or less regular rows in one to three layers, with hair scales derived from the tip of the abdomen of the female moth (Fig. 1). The hair scales give the eggs a “felt-like appearance”. Usually whitish-yellow in color, changing to black just prior to hatching, due to the big head of the larva showing through the transparent shell (Pinhey, 1975).

Figure 1. Eggs and neonates. Eggs are laid in batches covered with orange-brown hair scales. Photo courtesy of http://www.defra.gov.uk/planth/pestnote/spod.htm.

Larvae: Upon hatching, larvae are 2 to 3 mm (approx. $^{1}/_{16}$ to $^{1}/_{8}$ in) long with white bodies and black heads and are very

difficult to detect visually. Larvae grow to 40 to 45 mm (approx. 1 $^{9}/_{16}$ to 1 $^{3}/_{4}$ in) and are hairless, cylindrical, tapering towards the posterior and variable in color (blackish-gray to dark green, becoming reddish-brown or whitish-yellow) (Fig. 2). The sides of the body have dark and light longitudinal bands; dorsal side with two dark semilunar spots laterally on each segment, except for the prothorax; spots on the first and eighth abdominal segments larger than the others, interrupting the lateral lines on the first segment. The larva of *S. littoralis* is figured by Bishari (1934) and Brown and Dewhurst (1975).Larvae are nocturnal and during the day can be found at the base of the plants

Figure 2. Larva of *S. littoralis*. Photos courtesy of CABI, 2007.

or under pots.
Pupae: When newly formed, pupae are green with a reddish color on the abdomen, turning dark reddish-brown after a few hours (Fig. 3A). The general shape is cylindrical, 14 to 20 x 5 mm (approx. $^{9}/_{16}$ to $^{13}/_{16}$ x $^{3}/_{16}$ in), tapering towards the posterior segments of the abdomen. The last segment ends in two strong straight hooks (Pinhey, 1975).

Adults: Moth with gray-brown body (Fig. 3B), 15 to 20 mm (approx. $^{9}/_{16}$ to $^{13}/_{16}$ in) long; wingspan 30 to 38 mm (approx. 1 $^{3}/_{16}$ to 1 $^{1}/_{2}$ in); forewings gray to reddish brown with paler lines along the veins (in males, bluish areas occur on the wing base and tip); the ocellus is marked by two or three oblique whitish stripes. Hindwings are grayish white, iridescent with gray margins and usually lack darker veins (EPPO, 1997).

Biology and Ecology
Spodoptera littoralis larvae damage many agricultural plants, particularly cotton (Venturini, 1975). Adult moths feed on nectar, and females oviposit eggs on the leaves of crop plants.

Depending on the climate of the region, *S. littoralis* can have from two to seven generations per year and does not undergo diapause (Salem and Salama, 1985). Egg masses consist of hundreds of eggs and are most abundant on young leaves on the upper parts of the plant (Khalifa et al., 1982), the undersurface of leaves (Nasr and Nassif, 1970; Gawaad and El Gayar, 1974), and the younger leaves (Khalifa et al., 1982).

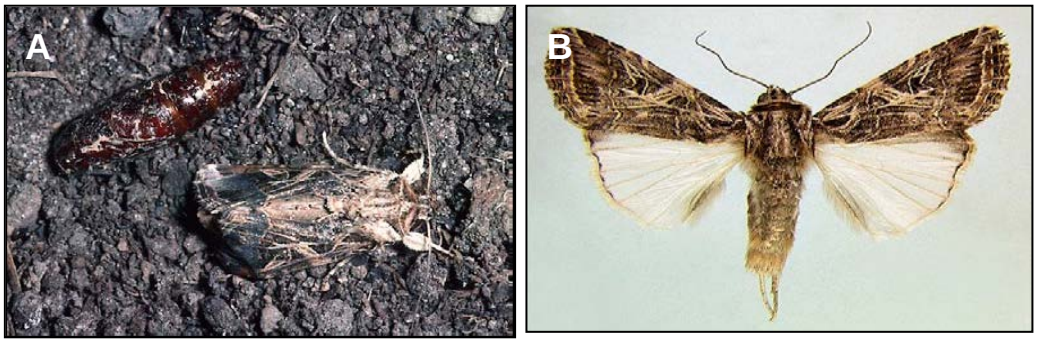


Figure 3. Pupa and adult of *S. littoralis* on soil (A). Adult moth of *S. littoralis* (museum set specimen) (B). Photos courtesy of CABI, 2007 and Entopix.

As the moth develops, it completes six larval instars. First through third instars do not move about the plant, hence 80% of early instar larvae inhabit the same location where the eggs were deposited (i.e., the upper parts of plants and the lower leaf surfaces) (Hoeny et al., 1982). Late instar larvae move about the plant and appear to prefer the upper parts of plants during the early morning hours and the lower plant areas or the soil during the afternoon hours. Both the early and late instars larvae avoid the mid-regions of the plants (Abdel Megeed and Iss Hak, 1975). Fourth through sixth instars move to the ground during the hot hours of the day, and late sixth instars bury themselves in the soil to pupate (Gawaad and El Gayar, 1974). Adult moths emerge at night and can live for 5 to 10 days (Salama and Shoukry, 1972). About half of females will lay their eggs before sunrise the same night of mating (Hassan et al., 1960).

Egg masses have shown to have variable distribution in fields. One study reported that the distribution egg masses where increased towards the center of the cotton field (Iss Hak and Abdel Megeed, 1975), whereas another study reported that in some years *S. littoralis* was more abundant at the edges and in other years it was more abundant in the center of the field (Khalifa et al., 1982).

The lower threshold temperatures for egg, larvae, pupae, and pre-oviposition periods was 11.86, 7.69, 12.34 and 10.66°C (53.3, 45.8, 54.2, and 51.2°F), respectively (Dahi, 2005). The upper temperature threshold for complete development of *S. littoralis* is 37°C (98.6°F) (El-Malki, 2000). *Spodoptera littoralis* requires 53.2, 314.7, 155.8 and 27.5 degree-days for egg, larvae, pupae and pre- oviposition period, respectively (Dahi, 2005). At temperatures of 18°C (64.4°F) and 36°C (96.8°F), eggs hatched within 2 and 9 days, larval stage lasted 10 and 35 days, and pupal stage took 8 and 27 days, respectively (Ocete Rubio, 1984). The optimal temperature for maximum weight gain in larvae is 20°C (68°F) (Bhatt, 1976), egg production is 25°C (77°F) (Nasr, 1974), larval survivorship to adult is 25°C (77°F) (Sidibea and Lauge, 1977; Hegazi and Schopf, 1984; Ocete Rubio, 1984), and pupation is 30°C (86°F) (Nasr and Nassif ,1977). In general, cold resistance is the lowest in the egg stage, increases with in maturing larvae, and is greatest in the pupal stage (Miller, 1977).

In one study, most moths flew from up to 250 m to 500 m (0.15 to 0.31 mi) from release point, and the farthest recapture obtained was at 1,500 m (0.93 mi) (Salama and Shoukry, 1972). In another study, most males were captured within 100 m (.06 mi) and most were recaptured the same night as they were released (Kehat et al., 1976).

Pest Importance

Spodoptera littoralis is one of the most destructive agricultural lepidopterous pests within its subtropical and tropical range. The pest causes a variety of damage as a leaf feeder and sometimes as a cut worm on seedlings. It can attack numerous economically important crops throughout the year (EPPO, 1997). On cotton, the pest may cause considerable damage by feeding on the leaves, fruiting points, flower buds and occasionally on bolls. When peanuts are infested, larvae first select young folded leaves for feeding, but in severe attacks, leaves of any age are stripped off. Sometimes, even the ripening kernels in the pods in the soil may be attacked. Pods of cowpeas and the seeds they contain are also often badly damaged. In tomatoes, larvae bore into the fruit, rendering them unsuitable for consumption. Numerous other crops are attacked, mainly on their leaves.

In Europe, damage caused by *S. littoralis* was minimal until about 1937. In 1949, there was a catastrophic population explosion in southern Spain, which affected alfalfa, potatoes, and other vegetable crops. At present, this noctuid pest is of great economic importance in Cyprus, Israel, Malta, Morocco, and Spain (except the north). In Italy, it is especially important on protected crops of ornamentals and vegetables (Inserra and Calabretta, 1985; Nucifora, 1985). In Greece, *S. littoralis* causes slight damage in Crete on alfalfa and clover only. In North Africa, tomato, *Capsicum* spp., cotton, corn, and other vegetables are affected. In Egypt, it is one of the most serious cotton pests.

Many populations of *S. littoralis* are extremely resistant to pesticides, and if they become well established, can be exceptionally difficult to control (USDA, 1982).

Symptoms/Signs

On most crops, damage arises from extensive feeding by larvae, leading to complete stripping of the plants. Larvae prefer to feed on young, tender leaves. They may also feed on growing points, young shoots, stalks, bolls, buds, and fruits, often gnawing boles which allow disease or rot to enter the host.

On newly infested hosts, young larvae feed at numerous small feeding points that eventually spread over the entire leaf. Older instars chew large holes or wholly consume leaves, or mine their way into young shoots or bare sections on young stalks, bolls, and buds. They may destroy fruit such as tomatoes and peppers. If larvae feed on a young plant heavily, the plant's development is retarded and it may only produce small or late fruit.

In deciduous orchards, larvae may cause severe damage to trees by feeding on leaves and terminal growing points. Young orchards suffer great damage. Larvae can completely defoliate ornamental plants and fruit trees in nurseries. If food supply is in

short supply, large numbers of larvae may migrate en masse to new cropland. On pasture, some *Spodoptera* spp. prefer to feed on legumes over grasses. On grape, larvae gnaw holes in the leaves until sometimes only the veins remain. The damage caused by larvae to grapevines is not merely temporary; vines may suffer so severely from exposure to intense sunlight during the summer that their development in the following year will be retarded. Larvae also gnaw at grape bunch stalks, which as a result, dry up, and the larvae feed on the grape berries (USDA, 1982).

Known Hosts

The host range of *S. littoralis* covers over 40 families, containing at least 87 species of economic importance (Salama et al., 1970).

Major Hosts

Abelmoschus esculentus (okra), *Allium* spp. (onion), *Amaranthus* spp., *Apios* spp. (groundnut), *Arachis hypogea* (peanut), *Beta vulgaris* (beet), *Brassica oleracea* (cabbage, broccoli), *Brassica rapa* (turnip), *Brassica* spp. (mustards), *Camellia sinensis* (tea), *Capsicum annuum* (pepper), *Chrysanthemum* spp., *Citrullus lanatus* (watermelon), *Citrus* spp., *Coffea arabica* (coffee), *Colocasia esculenta* (taro), *Corchorus* spp. (jute), *Cucumis* spp. (squash, pumpkin), *Cynara scolymus* (artichoke), *Daucus carota* (carrot), *Dianthus caryophyllus* (carnation), *Ficus* spp. (fig), *Glycine max* (soybean), *Gossypium* spp. (cotton), *Helianthus annus* (sunflower), *Ipomoea batatas* (sweet potato), *Lactuca sativa* (lettuce), *Linum* spp. (flax), *Lycopersicon esculentum* (tomato), *Medicago sativa* (alfalfa), *Morus* spp. (mulberry), *Musa* spp. (banana, plantain), *Nicotiana tabacum* (tobacco), *Oryza sativa* (rice), *Pennisetum glaucum* (pearl millet), *Persea americana* (avocado), *Phaseolus* spp. (bean), *Pisum sativum* (pea), *Prunus domestica* (plum), *Psidium guajava* (guava), *Punica granatum* (pomegranate), *Raphanus sativus* (radish), Rosa spp. (rose), *Saccharum officinarum* (sugarcane), *Solanum melongena* (eggplant), *Solanum tuberosum* (potato), *Sorghum bicolor* (sorghum), *Spinacia* spp. (spinach), *Theobroma cacao* (cacao), *Trifolium* spp. (clover), *Triticum aestivum* (wheat), *Vicia faba* (broad bean), Vigna spp. (cowpea, black-eyed pea), *Vitis vinifera* (grape), and *Zea mays* (corn).

Minor Hosts

Acacia spp. (wattles), *Actinidia arguta* (tara vine), *Alcea rosea* (hollyhock), *Anacardium occidentale* (cashew), *Anemone* spp. (anemone), *Antirrhinum* spp., *Apium graveolens* (celery), *Asparagus officinalis* (asparagus), *Caladium* spp. (caladium), *Canna* spp. (canna), *Casuarina equisetifolia* (she-oak), *Convolvulus* spp. (morning glory, bindweeds), *Cryptomeria* spp. (Japanese cedar), *Cupressus* spp. (cypress), Datura spp. (jimsonweed), *Eichhornia* spp. (water hyacinth), *Eucalyptus* spp. (eucalyptus), *Geranium* spp. (geranium), *Gladiolus* spp. (gladiolus), *Malus domesticus* (apple), *Mentha* spp. (mint), *Phoenix dacylifera* (date palm), *Pinus* spp. (pine), and *Zinia* spp. (zinnia).

Known Vectors (or associated organisms)

Spodoptera littoralis is not a known vector and does not have any associated organisms.

Known Distribution

The northerly distribution limit of *S. littoralis* in Europe corresponds to the climatic zone in which winter frosts are infrequent. It occurs throughout Africa and extends eastwards into Turkey and north into eastern Spain, southern France and northern Italy. However, this boundary is probably the extent of migrant activity only; although the pest overwinters in southern Spain, it does not do so in northern Italy or France. In southern Greece, pupae have been observed in the soil after November and the species overwinters in this stage in Crete. Low winter temperatures are, therefore, an important limiting factor affecting the northerly distribution, especially in a species with no known diapause (Miller, 1976; Sidibe and Lauge, 1977).

Africa: Algeria, Angola, Ascension Island, Benin, Botswana, Burkina Faso, Burundi, Cameroon, Cape Verde, Central African Republic, Chad, Comoros, Congo, Democratic Republic of the Congo, Cote d'Ivoire, Egypt, Equatorial Guinea (including Bioko), Eritrea, Ethiopia, Gabon, Gambia, Ghana, Guinea, Kenya, Liberia, Libya, Madagascar, Malawi, Mali, Mauritania, Mauritius (including Rodrigues), Morocco, Mozambique, Namibia, Niger, Nigeria, Reunion, Rwanda, Saint Helena, Sao Tome and Principe, Senegal, Seychelles, Sierra Leone, Somalia, South Africa, Sudan, Swaziland, Tanzania, Togo, Tunisia, Uganda, Zaire, Zambia, and Zimbabwe. **Asia:** Bangladesh, Brunei, India, and Turkey. **Europe:** Albania,Greece (including Crete and Dodecanese), Italy (including Sardinia and Sicily), Malta, Portugal (including Azores and Madeira), and Spain (including Balearic Islands and Canary Islands) **Middle East:** Afghanistan, Bahrain, Cyprus, Iran, Iraq, Israel, Jordan, Lebanon, Oman, Saudi Arabia, Syria, United Arab Emirates, and Yemen. **Oceania:** American Samoa and Fiji (CIE, 1964; Evenhuis, 2010; Fibiger and Skule, 2011; EPPO, 2012).

Pathway

Spodoptera littoralis could potentially move through international trade. This species has been intercepted over 170 times at U.S. ports of entry. Interceptions have occurred in permit cargo (164), baggage (5), stores (5), and general cargo (1). Most interceptions originated from Israel (121), the Netherlands (22), Spain (6) and Kenya (5). This species is mostly intercepted on plant material, including *Eustoma* spp. (18), *Anemone* spp. (16), *Gerbera* spp. (15), *Origanum* spp. (12), and *Thymus* spp. (9) (AQAS, 2012; queried August 6, 2012).

Potential Distribution within the United States

The pest has been intercepted at U.S. ports on plant parts, leaves, and flowers. The potential U.S. range of most *S. littoralis* may be limited to the west coast through the lower southwestern and southeastern United States, reaching only as far north as Maryland (USDA, 1982). Migratory moths may be capable of periodic spread into northern states and even Canada by late summer or early fall. Venette et al. (2003) suggest that approximately 49% of the continental United States would be suitable for *S. littoralis.* A recent risk analysis by USDA-APHIS-PPQ-CPHST shows that much of the United States has a moderate to high risk based on host availability, climate, and pathway.

Survey

CAPS-Approved Method*: The CAPS-approved method is a trap and lure combination. The trap is a plastic bucket trap. The lure is effective for 84 days (12 weeks).

The lure is "*Spodoptera littoralis*" Lure.

Trap Spacing: When trapping for more than one species of moth, separate traps for different moth species by at least 20 meters (65 feet).

Method Notes: This trap is also known as the unitrap. The trap has a green canopy, yellow funnel, and white bucket and is used with a dry kill strip. For instructions on using the trap, see Brambila et al. (2010).

Lure Placement: Placing lures for two or more target species in a trap should never be done unless otherwise noted here.

Lure Notes: Place *S. litura* and *S. littoralis* lures in different traps and separate at least 20 meters (65 feet).

Though the lures for *Spodoptera littoralis* and *S. litura* are composed of the same two compounds (Z,E,9,11-14:AC and Z,E,9,12-14:AC), the compounds are loaded into the lure dispensers in different amounts depending on the target species. Therefore, it is necessary to use the specific lure for each of the two targets.

*For the most up-to-date methods for survey and identification, see Approved Methods on the CAPS Resource and Collaboration Site, at http://caps.ceris.purdue.edu/.

Literature-Based Methods: (From Venette et al., 2003; CABI, 2007)
Trapping: Pheromone traps can be used to monitor the incidence of *S. littoralis* (Rizk et al., 1990). The synthetic sex pheromone (*Z*,*E*)-(9,11)-tetradecadienyl acetate has proven highly effective at trapping male moths of *S. littoralis* (Salem and Salama, 1985). Kehat and Dunkelblum (1993) found that the minor sex pheromone component, (9*Z*,12*Z*)-9,12-tetradecadienyl acetate in addition to the major component (9*Z*,11*Z*)-9,11-tetradecadienyl acetate was required to attract males.

Sex-pheromone baited delta traps remained attractive for approximately 2 weeks, but effectiveness declined after 3 to 4 weeks of use (Ahmad, 1988). To monitor male flight activity in vegetable production areas, delta traps were placed 1.7 m above the ground at a rate of 2 traps/ha (approximately 1 trap/acre) (Ahmad, 1988). Pheromone lures impregnated with 2 mg of the pheromone blend (blend not specified) were replaced after 4 weeks of use (Ahmad, 1988). Traps are deployed at a similar height (1.5 m) to monitor male flight in cotton (Salem and Salama, 1985). Catches in pheromone traps did not correlate as well with densities of egg-masses in cotton fields as did catches in a black-light trap (Rizk et al., 1990). The attractiveness of traps baited with (*Z*,*E*)-(9,11)-tetradecadienyl acetate is governed primarily by minimum air temperature, relative

humidity, adult abundance, and wind velocity. Densities of female *S. littoralis* also affect the number of males that are captured at different times of the year (Rizk et al., 1990). Lures for *S. littoralis* may also attract *Erastria* spp. (established in the United States) (PPQ, 1993).

Visual survey: Visual surveys for this pest can take place any time during the growing season while plants are actively growing (usually spring through fall in temperate areas). Early instars (<3rd) are likely to be on lower leaf surfaces during the day. The larvae will skeletonize leaves by feeding on this surface and such damage to the leaf provides evidence of the presence of larvae. Sweep net sampling may be effective at dawn or dusk. Specimen identification should be confirmed by a trained taxonomist (USDA, 1982). However, not all sampling methods are equally effective for all life-stages of the insect. Eggs are only likely to be found by visual inspection of leaves. First through third instars may be detected by sweep net sampling; nearly all instars can be detected by visual inspection of plants; and, later instars (4th-6th) and pupae may be found by sieving soil samples (Abul-Nasr and Naguib, 1968; Abul-Nasr et al., 1971).

Not recommended: Light traps using a 125 W mercury-vapor bulb have been used to nondiscriminately capture multiple *Spodoptera* spp. (Blair, 1974) and most assuredly other insects as well. A modified light trap using six 20-W fluorescent lights also proved effective for monitoring flight activity of *S. littoralis* (El-Mezayyen et al., 1997).

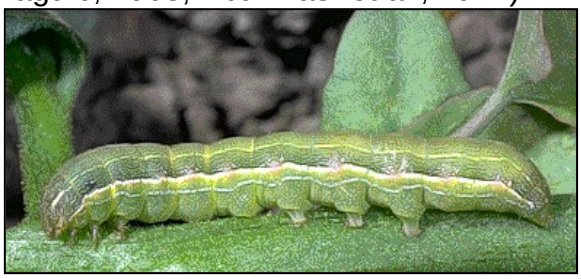


Figure 4. Larva of *S. exigua*. Photo courtesy of Oklahoma State University.

For additional survey information see: http://www.aphis.usda.gov/import_export/plants/manuals/emergency/downloads/nprg_spodoptera.pdf.

Key Diagnostics/Identification

CAPS-Approved Method*:

Confirmation of *S. littoralis* is by morphological identification. *Spodoptera littoralis* is difficult to distinguish from *S. litura* without close examination of the genitalia. *Spodoptera littoralis* is also confused with *S. dolichos*, *S. ornithogalli*, *S. latifascia* and other *Spodoptera* species (present in the United States).

*For the most up-to-date methods for survey and identification, see Approved Methods on the CAPS Resource and Collaboration Site, at http://caps.ceris.purdue.edu/.

Figure 5. Adult of *S. ornithogalli*. Photo courtesy of Mississippi Entomological Museum. http://mothphotographersgroup.msstate.edu/Files/JV/JV50.7.shtml

Literature-Based Methods:
Observation of adult genitalia is often the only certain method to separate species. Screening aids to help identify *S. littoralis* in the field and by using wing diagnostics are available http://caps.ceris.purdue.edu/webfm_send/553 and http://caps.ceris.purdue.edu/webfm_send/554

Easily Confused Pests

Spodoptera littoralis is often confused with *S. litura,* and the variability and similarity of the two species makes correct identification difficult; examination of adult genitalia is often the only certain method to separate the two species. For more information on morphological discrimination between the adult, pupal, and larval stages of the two species, refer to Schmutterer (1969), Cayrol (1972), Mochida (1973), and Brown and Dewhurst (1975).

Although markings on larvae are variable, a bright-yellow stripe along the length of the dorsal surface is characteristic of *S. litura*. On dissection of the genitalia, the ductus and ostium bursae are the same length in female *S. littoralis*, whereas they are different lengths in *S. litura*. The shape of the juxta in males in both species is very characteristic, and the ornamentation of the aedeagus vesica is also diagnostic. The genitalia must be removed, cleaned in alkali, and examined microscopically. *Spodoptera litura* is not established in the continental United States, but has been reported in Hawaii.

Larvae of *S. littoralis* can be confused with *S. exigua*, the beet armyworm, (established in the United States) (Fig. 4), but *S. littoralis* larvae are light or dark brown, while *S. exigua* are brown or green. *Spodoptera littoralis* is also larger than *S. exigua* (Venette et al., 2003).

Adults of *S. littoralis* are almost nearly identical in appearance to *S. ornithogalli* (Fig. 5), the yellow striped armyworm, a common pest in the United States. The hind wings of female *S. littoralis* are darker than those of *S. ornithogalli* (USDA, 1982).

References

Abdel-Megeed, M. I. and R. R. Iss-Hak. 1975. Field observations on the vertical distribution of the cotton leafworm, *Spodoptera littoralis* (Boisd.). Z. Angew. Entomol. 78(1): 59-62.

Abul-Nasr, S. and M. A. Naguib. 1968. The population density of larvae and pupae of *Spodoptera littoralis* (Boisd.) in clover fields in Egypt (Lepid.: Agrotidae). Bulletin De La Societe Entomologique D'Egypte 52: 297-312.

Abul-Nasr, S., S. I. El-Sherif, and M. A. Naguib. 1971. Relative efficiency of certain sampling methods for the assessment of the larval and pupal populations of the cotton leafworm *Spodoptera littoralis* (Boisd.) (Lepid.: Agrotidae) in clover fields. Journal of Applied Entomology/Zeitschrift für Angewandte Entomologie 69: 98-101.

Ahmad, T. R. 1988. Field studies on sex pheromone trapping of cotton leafworm *Spodoptera littoralis* (Boisd.) (Lep.: Noctuidae). Journal of Applied Entomology/Zeitschrift für Angewandte Entomologie 105: 212-215.

AQAS. 2012. *Spodoptera littoralis* interceptions. Last accessed August 6, 2012 from: https://mokcs14.aphis.usda.gov/aqas/HomePageInit.do#defaultAnchor.

Bhatt, N. S. 1976. Development of S*podoptera littoralis* (Boisd.) (Lepidoptera, Noctuidae) at constant temperatures on two host plants. Z Angew Entomol 80(2):201-206.

Bishara, I. 1934. The cotton worm *Prodenia litura* F. in Egypt. Bulletin de la Société Entomologique d'Egypte, 18:223-404.

Blair, B. W. 1974. Identification of economically important *Spodoptera* larvae (Lepidoptera: Noctuidae). In: Scientific Note. Journal of the Entomological Society of Southern Africa 37: 195-196.

Brambila, J., L. Jackson, and R. Meagher. 2010. Plastic bucket trap protocol. USDA. 7 pp.

Brown, E. S. and C. F. Dewhurst. 1975. The genus *Spodoptera* (Lepidoptera, Noctuidae) in Africa and the Near East. Bulletin of Entomological Research, 65(2):221-262.

CABI. 2007. Crop protection compendium: global module. Commonwealth Agricultural Bureau International, Wallingford, UK. http://www.cabi.org/compendia/cpc/.

Cayrol, R. A. 1972. Famille des Noctuidae. In: Balachowsky AS, ed. Entomologie appliquée à l'agriculture. Vol. 2. Paris, France: Masson, 1411-1423.
CIE. 1964. *Spodoptera littoralis* (Boisd.). Distribution Maps of Pests, Series A (Agricultural). Map No. 232. Commonwealth Institute of Entomology.

Dahi, H. F. 2005. Egyptian cotton leafworm *Spodoptera littoralis* development on artificial diet in relation to heat unit requirements The Third International Conference on IPM Role in Integrated Crop management and Impacts on Environment and Agricultural Products. Plant Protection Research Institute, ARC, Dokki, Giza, Egypt, Giza, Egypt.

El-Malki, K. G. 2000. Thermal requirements and prediction models of cotton leafworm *Spodoptera littoralis* (Boisd). Pages 1019-1021 2000 Beltwide Cotton Conferences. Cotton Insect Research and Control Conference, San Antonio, Texas.

El-Mezayyen, G. A., A. A. El-Dahan, G. M. Moawad, and M. S. Tadros. 1997. A modified light trap as a tool for insects survey in relation to the main weather factors. Egyptian Journal of Agricultural Research 75: 995-1005.

EPPO. 1997. *Spodoptera littoralis* and *Spodoptera litura*. In: Smith IM, McNamara DG, Scott PR, Holderness M, eds. Quarantine pests for Europe. 2nd edition. Wallingford, UK: CAB International, 518-525.

EPPO. 2012. EPPO Plant Quarantine Information Retrieval System (PQR), version 5.0.5540. European and Mediterranean Plant Protection Organization.

Evenhuis, N. L. 2010. Checklist of Fijian Lepidoptera. Accessed April 12, 2012 from: http://hbs.bishopmuseum.org/fiji/checklists/lepidoptera.html.

Fibiger, M. and B. Skule. 2011. Fauna Europaea: *Spodoptera littoralis*. In O. Karsholt and E. J. Nieukerken (eds.) Fauna Europaea: Lepidoptera, Moths, version 2.4. Accessed April 10, 2012 from: http://www.faunaeur.org**.**

Gawaad, A. A. A. and F. H. El Gayar. 1974. Habits and behavior of *Spodoptera-littoralis* Lepidoptera Noctuidae and their importance for control measures. Zeitschrift fuer Angewandte Entomologie 75(3):295-300.

Hassan, A. S., M. A. Moussa, and E. A. Nasr. 1960. Behavior of larvae and adults of the cotton leaf worm, *Prodenia litura*. Bulletin de la Société Entomologique d'Egypte 44:337-343.

Hegazi, E. M. and R. Schopf. 1984. The influence of temperature on consumption and utilization of artificial diet by *Spodoptera-littoralis* lepidoptera noctuidae. Zeitschrift fuer Angewandte Entomologie 97(4):321-326.

Hoeny, M. M., R. R. Iss Hak, and M. E. Foda. 1982. Daily movement and time of activity of Egyptian cotton leafworm larvae *Spodoptera-littoralis* on cotton plants. Ain Shams University Faculty of Agriculture Research Bulletin (1750):1-11.

Inserra, S. and C. Calabretta. 1985. Attack by noctuids: a recurring problem in greenhouse crops of the Ragusa coast. Tecnica Agricola, 37(3-4):283-297.

Iss Hak, R. R. and M. I. Abdel Megeed. 1975. Horizontal distribution of egg masses of the cotton leafworm *Spodoptera-littoralis* in cotton fields. Zeitschrift fuer Angewandte Entomologie 78(1):56-59.

Kehat, M. and E. Dunkelblum. 1993. Sex pheromones: achievements in monitoring and mating disruption of cotton pests in Israel. Archives of Insect Biochemistry and Physiology, 22(3-4):425-431.

Kehat, M., A. Navon, and S. Greenberg. 1976. Captures of marked *Spodoptera-littoralis* male moths in virgin female traps effects of wild male population distance of traps from release point and wind. Phytoparasitica 4(2):77-84.

Khalifa, A., R. R. Iss Hak, and M. E. Foda. 1982. Vertical and horizontal distribution of the Egyptian cotton leafworm *Spodoptera-littoralis* egg masses in cotton fields in Egypt. Ain Shams University Faculty of Agriculture Research Bulletin (1749):1-13.

Miller, G. W. 1976. Cold storage as a quarantine treatment to prevent the introduction of Spodoptera littoralis (Boisd.) into glasshouses in the UK. Plant Pathology, 25(4):193-196.

Miller, G. W. 1977. Mortality of *Spodoptera-littoralis* Lepidoptera Noctuidae at nonfreezing temperatures. Bulletin of entomological research 67(1):143-152.

Mochida, O. 1973. Two important insect pests, *Spodoptera litura* (F.) and *S. littoralis* (Boisd.) (Lepidoptera:Noctuidae), on various crops - morphological discrimination of the adult, pupal and larval stages. Applied Entomology and Zoology, 8(4):205-214.

Nasr, E. S. A. 1974. Response of various stages of the cotton leafworm, *Spodoptera littoralis* (boisd.) to temperature and relative humidity (lepidoptera: Noctuidae). Bull Soc Entomol Egypte 58:123-131.

Nasr, E. S. A. and F. Nassif. 1970. Behavior of egg laying and population density of the cotton leafworm *Spodoptera-littoralis* on different host plants. Bulletin de la Société Entomologique d'Egypte 54: 541-544.

Nasr, E. S. A. and F. M. Nassif. 1977. Effect of soil moisture content on the vitality of the pupal stage of the cotton leafworm *Spodoptera-littoralis* in relation to temperature. Bulletin de la Société Entomologique d'Egypte(61): 15-20.

Nucifora, A. 1985. Successive cultivation and systems of integrated control in protected crops of the Mediterranean area. Tecnica Agricola, 37(3-4):223-241.

Ocete Rubio, E. 1984. Study of the biological cycle of *Spodoptera-littoralis* at different temperatures Lepidoptera Noctuidae. Graellsia 40:195-206.

Pinhey, E. C. G. 1975. Moths of Southern Africa. Descriptions and colour illustrations of 1183 species. Moths of Southern Africa. Descriptions and colour illustrations of 1183 species., [7+]273pp.; [col. frontis., 63 col. pl., 18 fig., 290 X 220 mm]; many ref.

PPQ. 1993. Fact sheet for exotic pest detection survey recommendations. Cooperative Agricultural Pest Survey (CAPS) and Plant Protection and Quarantine, US Department of Agriculture.

Rizk, G. A., M. A. Soliman, and H. M. Ismael. 1990. Efficiency of sex pheromone and U. V. light traps attracting male moths of the cotton leafworm *Spodoptera littoralis* (Boisd.). Assiut Journal of Agricultural Sciences, 21(3):86-102.

Salama, H. S. and A. Shoukry. 1972. Flight range of the moth of the cotton leafworm *Spodoptera littoralis*. Zeitschrift fuer Angewandte Entomologie 71(2):181-184.

Salama, H. S., N. Z. Dimetry, and S. A. Salem. 1971. On the host preference and biology of the cotton leaf worm *Spodoptera littoralis.* Zeitung für Angewandte Entomologie, 67:261-266.

Salem, S. and H. S. Salama. 1985. Sex pheromones for mass trapping of *Spodoptera littoralis* (Boisd.) in Egypt. Journal of Applied Entomology/Zeitschrift fur Angewandte Entomologie 100: 316-319.

Schmutterer, H. 1969. Pests of crops in Northeast and Central Africa with particular reference to the Sudan. Stuttgart, Germany: Gustav Fischer Verlag.

Sidibe, B. and G. Lauge. 1977. Effect of warm periods and of constant temperatures on some biological criteria in *Spodoptera littoralis* Boisduval (Lepidoptera Noctuidae). Annales de la Societe Entomologique de France, 13(2):369-379.

USDA. 1982. Pests not known to occur in the United States or of limited distribution, No. 25: Egyptian cottonworm., pp. 1-14. APHIS-PPQ, Hyattsville, MD.

Venette, R. C., E. E. Davis, J. Zaspel, H. Heisler, and M. Larson. 2003. Mini Risk Assessment Egyptian cotton leafworm, *Spodoptera littoralis* Boisduval [Lepidoptera: Noctuidae]. Cooperative Agricultural Pest Survey, Animal and Plant Health Inspection Service, US Department of Agriculture. Available on line at: http://www.aphis.usda.gov/plant_health/plant_pest_info/pest_detection/downloads/pra/slittoralispra.pdf

Venturini, V. 1975. New hosts of *Spodoptera-littoralis* Lepidoptera Noctuidae in Italy. Bollettino di Zoologia Agraria e di Bachicoltura 13167-172.

Spodoptera litura

Scientific Name
Spodoptera litura Fabricius

Synonyms:
Mamestra albisparsa, Noctua elata, Noctua histrionica, Noctua litura, Prodenia ciligera, Prodenia declinata, Prodenia evanescens, Prodenia glaucistriga, Prodenia litura, Prodenia subterminalis, *Prodenia tasmanica, Prodenia testaceoides*, *Prodenia littoralis, Spodoptera littoralis*

Common Name(s)
Rice cutworm, armyworm, taro caterpillar, tobacco budworm, cotton leafworm, cluster caterpillar, cotton worm, Egyptian cotton leafworm, tobacco caterpillar, tobacco cutworm, tobacco leaf caterpillar, common cutworm

Type of Pest
Moth

Taxonomic Position
Class: Insecta, **Order:** Lepidoptera, **Family:** Noctuidae

Reason for Inclusion in Manual
CAPS Target: AHP Prioritized Pest List – 2009 through 2013

Pest Description
The two Old World cotton leafworm species, *Spodoptera litura* and *S. littoralis*, are allopatric, their ranges covering Asia and Africa, Europe and the Middle East, respectively. Many authors have regarded them as the same species, but they have been differentiated based on adult genitalia differences (Mochida, 1973; CABI, 2007).

Eggs: Spherical, somewhat flattened, sculpted with approximately 40 longitudinal ribs, 0.4 - 0.7 mm in diameter; pearly green, turning black with time, laid in batches covered with pale orange-brown or pink hair-like scales from the females body (Pearson, 1958; CABI, 2007).

Larva: Newly hatched larvae are tiny, blackish green (Fig. 1) with a distinct black band on the first abdominal segment. Fully grown larvae are stout and smooth with scattered short setae. Head is shiny black, and has conspicuous black tubercules each with a long hair on each segment. Color of fully grown larvae not constant, but varies from dark gray to dark brown, or black, sometimes marked with yellow dorsal and lateral stripes of unequal width. The lateral yellow stripe is bordered dorsally with series of semilunar black marks (Fig 2). Mature larvae are 40 to 50 mm (approx. 1 $^{9}/_{16}$ to 2 in) with two large black spots on first and eighth abdominal segments (Hill, 1975; USDA,

1982; CABI, 2007). When disturbed, the larvae curl into a tight spiral with the head protected in the center.

Figure 1. Egg mass (left), larva (center), and adult (right). Photos courtesy of CABI, 2007.

Pupa: Reddish brown in color, enclosed inside rough earthen cases in the soil, 18 to 22 mm (approx. $^{11}/_{16}$ to $^{7}/_{8}$ in) long, last abdominal segment terminates in two hooks (USDA, 1982; CABI, 2007).

Adult: Body whitish to yellowish, suffused with pale red. Forewings dark brown with lighter shaded lines and stripes (Fig. 3). Hind wings whitish with violet sheen, margin dark brown and venation brown. Thorax and abdomen orange to light brown with hair-like tufts on dorsal surface. Head clothed with tufts of light and dark brown scales. Body length 14 to 18 mm (approx. $^{9}/_{16}$ to $^{11}/_{16}$ in), wing span 28 to 38 mm (approx. 1 $^{1}/_{8}$ to 1 $^{1}/_{2}$ in) (Hill, 1975; USDA, 1982).

See Schmutterer (1969), Cayrol (1972), and Brown and Dewhurst (1975) for additional information.

Biology and Ecology

The eggs of *S. litura* are laid in bunches of 50 to 300 on the under surface of leaves (preferred) by female moths (Chari and Patel, 1983). They hatch in 3 to 4 days. A single female lays 1500 to 2500 eggs in about 6 to 8 days. Castor bean is the most preferred host for ovipositing females (Chari and Patel, 1983). Newly irrigated fields are also very attractive to ovipositing females. Three peak periods of egg laying have been observed in the third weeks of June and July and in mid-August. Newly hatched larvae feed gregariously on the epidermis of the leaf. If the population density is high or the host is not suitable, the young larvae will hang on silken threads and migrate to other leaves or preferred hosts. There are generally six instars. The general habit of the larva is that the 1st, 2nd, and 3rd instars remain on the lower surface of leaves. The 4th, 5th, and 6th instars escape from sunshine, push to loosen the surface of the soil, and bite out soil particles to form a clay cell or cocoon in which to pupate (Chari and Patel, 1983).

Ahmed et al. (1979) showed that *S. litura* adults developed from first instar larvae in 23.4 days at 28°C (82.4°F). Mean female longevity was 8.3 days and mean fecundity was 2673 eggs. Mean male longevity was 10.4 days. No mating took place on the night

of emergence and maximum mating response occurred on the second night after emergence (Yamanaka et al., 1975; Ahmed et al., 1979). According to Yamanaka et al. (1975) the female continues to lay eggs in egg masses over a period of 5 days at 25°C (77°F).

Maximum fecundity for *S. litura* was observed at 27°C (81°F) under 12 hours per 24 hours of light (100 foot candle light) (Hasmat and Khan, 1977, 1978). Temperatures between 24 and 30°C (75 to 86°F) were also favorable for fecundity and fertility. At 33 and 39°C (91 and 102°F), both fecundity and fertility were decreased, and in the latter, fertility was completely inhibited (Hasmat and Khan, 1977). Twenty four hours exposure to light markedly reduced both fecundity and fertility. Hatching was highest in dark conditions (Hashmat and Khan, 1978). Parasuraman and Jayaraj (1983a) noted that 25°C (77°F) and 75% relative humidity were favorable for development of *S. litura* with a shorter larval period, 100% pupation, a shortened pupal period, and 100% adult emergence.

Ranga Rao et al. (1989) reported that an average of 64 degree-days (DD) above a threshold of 8°C (46°F) was required for oviposition to egg hatch. The larval period required 303 DD, and the pupal stage 155 DD above a 10°C (50°F) threshold. Females needed 29 DD above a 10.8°C (51°F) threshold from emergence to oviposition. The upper developmental threshold temperature of all stages was 37°C (99°F); 40°C (104°F) was lethal.

Maheswara Reddy (1983) showed that the majority of mating occurred between 23.30 and 00.30 hrs under controlled conditions. The duration of matings ranged between 82.5 and 90 minutes. Although males are capable of insemination throughout their lifecycle, no males inseminated more than one female in one night. Some males failed to inseminate even one female on some nights. The mean number of mating per males was 10.3 and per female was 3.1 (Ahmed et al., 1979). Ohbayashi et al. (1973) showed two peaks in mating behavior at 23.00 (3 hours after initiation of a dark period) and a minor peak at 3:00 (1 hour before the end of the dark period).

Spodoptera litura spends its pre-pupal and pupal period inside soil. In India, Parasuraman and Jayaraj (1983b) found pupation was maximal under fallen leaves, especially in wet, sandy loam soil. Although the depth of pupation varied, no pupation was observed beyond 12 cm (approx. 4 $^{3}/_{4}$ in) deep. Across soil types, most larvae pupated at a 4 cm (approx. 1 $^{9}/_{16}$ in) depth.

Symptoms/Signs

On most crops, including soybean, damage arises from extensive feeding by larvae, leading to complete stripping of the plants. Larvae are leaf eaters but sometimes act as a cutworm with crop seedlings. Frass is often visible.

Spodoptera litura feeds on the underside of leaves causing feeding scars and skeletonization of leaves. Early larval stages remain together radiating out from the egg mass. However, later stages are solitary. Initially there are numerous small feeding

points, which eventually spread over the entire leaf. Because of this pest's feeding activities, holes and bare sections are later found on leaves, young stalks, bolls, and buds. Larvae mine into young shoots. In certain cases, whole shoot tips wilt above a hole and eventually die (Hill, 1975; USDA, 1982).

On grape, larvae scrape the leaf tissue and cause 'drying of the leaves' (Balasubramaniam et al., 1978). The larvae damage the growing berries and cause defoliation. Balikai et al. (1999) also showed that later instar larvae cut the rachis of grape bunches and petioles of individual berries during the night hours leading to fruit drop. During the day, the larvae move to the lower portion of the leaf vines and the crevices of the soil. The larvae use the main stem for climbing from the soil level during dusk.

On cotton, leaves are heavily attacked and bolls have large holes in them from which yellowish-green to dark-green larval excrement protrudes. In tobacco, leaves develop irregular, brownish-red patches and the stem base may be gnawed off. The stems of corn are often mined and young grains in the ear may be injured (CABI, 2007).

Pest Importance

Spodoptera litura larvae are polyphagous defoliators, seasonally common in annual and perennial agricultural systems in tropical and temperate Asia. This noctuid is often found as part of a complex of lepidopteran and non-lepidopteran foliar feeders but may also damage tubers and roots. Hosts include field crops grown for food and fiber, plantation and forestry crops, as well as certain weed species (CABI, 2007).

Most work on the economic impact of *S. litura* has been conducted in India, where it is a serious pest of a range of field crops. It has caused 12 to 23% loss to tomatoes in the monsoon season, and 9 to 24% loss in the winter (Patnaik, 1998). In a 40- to 45-day-old potato crop, damage ranged from 20 to 100% in different parts of the field depending on moisture availability. Larvae also attacked exposed tubers when young succulent leaves were unavailable (CABI, 2007). *S. litura* is also a pest of sugarbeet, with infestations commencing in March and peaking in late March and April (Chatterjee and Nayak, 1987). Severe infestations led to the skeletonization of leaves, as well as feeding holes in roots that rendered the crop 'virtually unfit for marketing'. Late harvested crops were most severely affected and, in extreme cases, 100% of the roots were damaged, leading to considerable yield reduction. Aroid tuber crops (including taro (*Colocasia esculenta*)) suffered yield losses of up to 29% as a result of infestation by *S. litura*, *Aphis gossypii* and spider mites (Pillai et al., 1993).

Spodoptera litura causes damage to many species of forest and plantation trees and shrubs (Roychoudhury et al., 1995). It is responsible for brown flag syndrome in banana (Ranjith et al., 1997) and 5 to 10% fruit damage in grapes (Balikai et al., 1999). *Spodoptera litura* is also a member of a complex that causes extensive defoliation of soybean (Bhattacharjee and Ghude, 1985). Defoliation as severe as 48.7% during the pre-bloom stage of growth caused no 'marked' difference from a control treatment in which defoliation was prevented by repeated insecticide application. Number and weight

of pods and grains per plant were, however, reduced when defoliation occurred at, or after, blooming.

Insecticide resistance has been reported in India (Armes et al., 1997; Kranthi et al., 2001) and Pakistan (Ahmad et al, 2007).

Known Hosts

Both *S. litura* and *S. littoralis* are widely polyphagous (Brown and Dewhurst, 1975; Holloway, 1989). The host range of *S. litura* covers at least 120 species (Venette et al., 2003). Among the main crop species attacked by *S. litura* in the tropics are taro, cotton, flax, peanuts, jute, alfalfa, corn, rice, soybeans, tea, tobacco, vegetables, eggplant, *Brassica* spp., *Capsicum* spp., cucurbits, beans, potatoes, sweet potatoes, grape, and cowpea. Other hosts include ornamentals, wild plants, weeds and shade trees (for example, *Leucaena leucocephala,* the shade tree of cocoa plantations in Indonesia). Balasubramanian et al. (1984) showed better larval growth and higher adult fecundity when reared on castor bean compared to tomato, sweet potato, okra, cotton, sunflower, eggplant and alfalfa.

Major Hosts

Abelmoschus esculentus (okra), *Acacia mangium* (brown salwood), *Allium cepa* (onion), *Amaranthus* (grain amaranth), *Arachis hypogaea* (peanut), *Beta vulgaris* var. *saccharifera* (sugarbeet), *Boehmeria nivea* (ramie), *Brassica*, *Brassica oleracea* var. *botrytis* (cauliflower), *Brassica oleracea* var. *capitata* (cabbage), *Camellia sinensis* (tea), *Capsicum frutescens* (chili), *Castilla elastica elastica* (castilloa rubber), *Cicer arietinum* (chickpea), *Citrus*, *Coffea* (coffee), *Colocasia esculenta* (taro), *Corchorus* (jutes), *Corchorus olitorius* (jute), *Coriandrum sativum* (coriander), *Crotalaria juncea* (sunn hemp), *Cynara scolymus* (artichoke), *Erythroxylum coca* (coca), *Fabaceae* (leguminous plants), *Foeniculum vulgare* (fennel), *Fragaria ananassa* (strawberry), *Gladiolus* hybrids (gladiola), *Glycine max* (soybean), *Gossypium* spp.(cotton), *Helianthus annuus* (sunflower), *Hevea brasiliensis* (rubber), *Ipomoea batatas* (sweet potato), *Jatropha curcas* (Barbados nut), *Lathyrus odoratus* (sweet pea), *Lilium* spp. (lily), *Linum usitatissimum* (flax), *Lycopersicon esculentum* (tomato), *Malus domestica* (apple), *Manihot esculenta* (cassava), *Medicago sativa* (alfalfa), *Morus alba* (mora), *Musa* spp. (banana), *Nicotiana tabacum* (tobacco), *Oryza sativa* (rice), *Papaver* (poppies), *Paulownia tomentosa* (paulownia), *Phaseolus* (beans), *Piper nigrum* (black pepper), *Poaceae* (grasses), *Psophocarpus tetragonolobus* (winged bean), *Raphanus sativus* (radish), *Ricinus communis* (castor bean), *Rosa* (roses), *Sesbania grandiflora* (agati), *Solanum melongena* (aubergine, eggplant), *Solanum tuberosum* (potato), *Sorghum bicolor* (sorghum), *Syzygium aromaticum* (clove), *Tectona grandis* (teak), *Theobroma cacao* (cocoa), *Trigonella foenum-graecum* (fenugreek), *Vigna mungo* (black gram), *Vigna radiata* (mung bean), *Vigna unguiculata* (cowpea), *Vitis vinifera* (grape), *Zea mays* (corn), and *Zinnia elegans* (zinnia).

For a complete listing of hosts see Venette et al. (2003).

Known Vectors (or associated insects)

Spodoptera litura is not a known vector and does not have any associated organisms

Known Distribution

The rice cutworm, *S. litura*, is one of the most important insect pests of agricultural crops in the Asian tropics. This species is widely distributed throughout tropical and temperate Asia, Australasia and the Pacific Islands (Kranz et al., 1977).

Asia: Afghanistan, Andaman Islands, Bangladesh, Bonin Islands, Brunei, Cambodia, China, Christmas Island, Cocos Islands, India, Indonesia, Iran, Japan, Korea, Laos, Lebanon, Malaysia, Maldives, Myanmar, Nepal, Nicobar Islands, Oman, Pakistan, Philippines, Singapore, Sri Lanka, Syria, Taiwan, Thailand, and Vietnam. **Europe:** France. Russia. **Africa:** Ghana* and Reunion. **North America:** United States (Hawaii). **Oceania:** American Samoa, Australia (including Tasmania), Austral Islands, Belau, Caroline Islands, Cook Islands, Federated states of Micronesia, Fiji (including Rotuma), French Polynesia (including Marquesas Islands and Tuamotus), Guam, Kiribati (including Line Islands), Mariana Islands, Marshall Islands, New Caledonia (including Loyalty Islands), New Zealand (including Kermadec Islands), Niue, Norfolk Island, Northern Mariana Islands, Papua New Guinea, Phoenix Islands, Pitcairn Islands (including Henderson Island), Samoa, Society Islands, Solomon Islands, Tonga, Tuvalu, Vanuatu, Wake Island, and the Wallis and Futuna Islands (CABI, 1993; Obeng-Ofori and Sackey, 2003; EPPO, 2012).

This species was found in 2010 in the United Kingdom and is considered transient and under eradication (EPPO, 2012).

*Listed as *Prodentia litura* (Obeng-Ofori and Sackey, 2003).

Pathway

This species can move readily through international trade. This species has been intercepted at U.S. ports of entry over 700 times. Most interceptions originated on material from Thailand (595), Singapore (24), and Malaysia (21). *Spodoptera litura* was most commonly intercepted on the following material: *Oncidium* sp. (355), *Dendrobium* sp. (193), and Orchidaceae (46). Interceptions occurred mostly on permit cargo (650), baggage (32), and general cargo (25) (AQAS, 2012; queried August 31, 2012).

Potential Distribution within the United States

The pest has been present in Hawaii since 1964 (CABI, 2007). *Spodoptera litura* was identified in a sample from a Miami-Dade County, Florida nursery in April 2007. Pheromone traps have been placed over a nine square mile area and have yielded no additional finds.

A recent risk analysis by USDA-APHIS-PPQ-CPHST shows that the lower portion of the United States has the greatest risk of *S. litura* establishment. Portions of Alabama, Arkansas, Florida, Georgia, Louisiana, Mississippi, North Carolina, Oklahoma, South Carolina, and Texas are at the greatest risk from *S. litura*. Establishment of *S. litura* is

unlikely in the northern portion of the United States, except for a small part of the West Coast.

Survey

CAPS-Approved Method*:

The CAPS-approved method is a trap and lure combination. The trap is a plastic bucket trap. The lure is effective for 84 days (12 weeks).

The lure is "*Spodoptera litura*" Lure.

Trap Spacing: When trapping for more than one species of moth, separate traps for different moth species by at least 20 meters (65 feet).

Method Notes: This trap is also known as the unitrap. The trap has a green canopy, yellow funnel, and white bucket and is used with a dry kill strip. For instructions on using the trap, see Brambila et al. (2010).

Lure Placement: Placing lures for two or more target species in a trap should never be done unless otherwise noted here.

Lure Notes: Place *S. litura* and *S. littoralis* lures in different traps and separate at least 20 meters (65 feet).

Though the lures for *Spodoptera littoralis* and *S. litura* are composed of the same two compounds (Z,E,9,11-14:AC and Z,E,9,12-14:AC), the compounds are loaded into the lure dispensers in different amounts depending on the target species. Therefore, it is necessary to use the specific lure for each of the two targets.

*For the most up-to-date methods for survey and identification, see Approved Methods on the CAPS Resource and Collaboration Site, at http://caps.ceris.purdue.edu/.

Literature-Based Methods:

Trapping: The identification of a male sex pheromone of *S. litura*, (Z,E)-(9,11)-tetradecadienyl acetate and (Z,E)-(9,12)-tetradecadienyl acetate by Tamaki (1973) has enabled effective monitoring of this species for several years. One milligram of a 10:1 mixture of these two compounds in a rubber septum attracted a comparable number of males as 10 caged virgin females in the field (Yushima et al., 1974). The compounds are most effective in a ratio (A:B) between 4:1 to 39:1 (Yushima et al., 1974). The two components in a ratio of 9:1 are available commercially as Litlure in Japan (Yushima et al., 1974). For early detection sampling, traps should be placed in open areas with short vegetation (Hirano, 1976). Krishnananda and Satyanarayana (1985) found that trap catches at 2.0 m above the ground level caught significantly more male *S. litura* than those placed at higher of lower heights (ranging from 0.5 m to 4.0 m). Ranga Rao et al. (1991) suggest trap placement at 1 m.

A standard sex pheromone trap (plastic dry funnel trap and pheromone septa) has been developed at the International Crop Research Institute for the Semi-Arid Tropics (ICRISAT) (Pawar et al., 1988; Ranga Rao et al., 1990, 1991; Singh and Sachan, 1993). Water traps baited with synthetic pheromone, box traps with rectangular windows, and cylindrical traps equipped with a blowing fan (to suck the males into a bag attached to bottom of the cylinder) have been used in Japan (Yushima et al., 1974; Hirano, 1976; Hirano, 1977; Oyama, 1977; Nakamura, 1977). Kamano et al. (1976) also mention a trap composed of two cylindrical parts and four cones made of wire screen that opened to the outside. Krishnanda and Satyanarayana (1985) used a dry trap that incorporated a tin sheet for the trap head, to which a polythene sleeve (45 x 10 cm) was attached. A small cylindrical polythene vial with 2.5 mg of pheromone was fastened to a small hook inside the dome. Rango Rao et al. (1991), however, found that at night many moths escaped from 'sleeve' traps and recommended either single or double funnel traps.

Visual survey: Visual survey can be used to determine the presence of *S. litura*. The presence of newly hatched larvae can be detected by the 'scratch' marks they make on the leaf surface. Particular attention should be given to leaves in the upper and middle portion of the plants (Parasuraman, 1983). The older larvae are night-feeders, feeding primarily between midnight and 3:00 am and are usually found in the soil around the base of plants during the day. They chew large areas of the leaf, and can, at high population densities, strip a crop of its leaves. In such cases, larvae migrate in large groups from one field to another in search of food. *Spodoptera litura* may be detected any time the hosts are in an actively growing stage with foliage available, usually spring and fall. Check for 1st and 2nd instar larvae during the day on the undersurface of leaves and host plants. Watch for skeletonized foliage and perforated leaves. If no larvae are obvious, look in nearby hiding places. Third instar larvae rest in upper soil layers during the day. Sweep net for adults and larvae at dawn or dusk. Watch for external feeding damage to fruits. Watch near lights and light trap collections for adult specimens. Submit similar noctuid moths in any stage for identification (USDA, 1982).

Not recommended: Light traps have been used to monitor *S. litura* populations (Vaishampayan and Verma, 1983). Capture of *S. litura* moths was affected by the stage of the moon, with the traps being least effective during the full moon and most effective during the new moon (Parasuraman and Jayaraj, 1982).

Figure 2. Adult of *S. ornithogalli*. Photo courtesy of Mississippi Entomological Museum. http://mothphotographersgroup.msstate.edu/Files/JV/JV50.7.shtml

Key Diagnostics/Identification

CAPS-Approved Method*:

Confirmation of *S. litura* is by morphological identification. It is difficult to distinguish *S. litura* from *S. littoralis* without close examination of the

genitalia; consult appropriate keys by Todd and Poole (1980) and Pogue (2002). To separate from other noctuids, use the key developed by Todd and Poole (1980). *Spodoptera litura* can also be confused with *S. dolichos*, *S. ornithogalli*, *S. pulcella* and other *Spodoptera* species (present in the United States).

*For the most up-to-date methods for survey and identification, see Approved Methods on the CAPS Resource and Collaboration Site, at http://caps.ceris.purdue.edu/.

Literature-Based Methods: Wing coloration has been used to separate the sexes of *S. litura* (Singh et al., 1975). *Spodoptera litura* can be easily confused with *S. littoralis*. Adults are similar, and they can be distinguished only through examination of genitalia. On dissection of the genitalia, ductus and ostium bursae are the same length in female *S. littoralis,* different lengths in *S. litura*. The shape of the juxta in males is very characteristic, and the ornamentation of the aedeagus vesica is also diagnostic. The larvae of the two species are not easily separable, but some distinguishing criteria are used for the 6th instar. Mochida (1973) provides information on morphological discrimination between the adult, pupal and larval stages of the two species. Screening aids to help identify *S. litura* in the field http://caps.ceris.purdue.edu/webfm_send/555 and using wing diagnostics are available http://caps.ceris.purdue.edu/webfm_send/556.

Easily Confused Pests

Adult *S. litura* closely resemble *S. ornithogali* (yellowstriped armyworm), a pest in the United States (Fig. 2). However, the hindwings of female *S. litura* are darker than those of *S. ornithogalli*.

References

Ahmad, M., M. Iqbal Arif, and M. Ahmad. 2007. Occurrence of insecticide resistance in field populations of *Spodoptera litura* (Lepidoptera: Noctuidae) in Pakistan. Crop Protection 26: 809-817.

Ahmed, A. M., M. Etman, and G. H. S. Hooper. 1979. Developmental and reproductive biology of *Spodoptera litura* (F.) (Lepidoptera: Noctuidae). J. Aust. Ent. Soc. 18: 363-372.

AQAS. 2012. *Spodoptera litura* interceptions. Last accessed August 31, 2012 from: https://mokcs14.aphis.usda.gov/aqas/HomePageInit.do#defaultAnchor.

Armes, N. J., J. A. Wightman, D. R. Jadhav, and G. V. Ranga Rao. 1997. Status of insecticide resistance in *Spodoptera litura* in Andhra Prades, India. Pesticide Science 50: 240-248.

Balasubramanian, G., S. Chellia, and M. Balasubramanian. 1984. Effect of host plants on the biology of *Spodoptera litura* Fabricius. Indian J. Agric. Soc. 54(12): 1075-1080.

Balasubramanian, M., S. Murugesan, and S. Parameswaran. 1978. Beware of cutworm on grapes. Kisan World 5(6): 51-52.

Balikai, R. A., A. N. Bagali, and Y. H. Ryagi. 1999. Incidence of *Spodoptera litura* Fab. on grapevine, *Vitis vinifera* L. Insect Environment, 5:32.

Bhattacharjee, N. S. and D. B. Ghude. 1985. Effect of artificial and natural defoliation on the yield of soybean. Indian Journal of Agricultural Sciences, 55:427-429.

Brambila, J., L. Jackson, and R. Meagher. 2010. Plastic bucket trap protocol. USDA. 7 pp.

Brown, E. S. and C. F. Dewhurst. 1975. The genus *Spodoptera* (Lepidoptera, Noctuidae) in Africa and the Near East. Bulletin of Entomological Research, 65:221-262.

CABI. 2003. Distribution maps of pests, *Spodoptera litura* (Fabricius) Lepidoptera: Noctuidae. Series A: Map No. 61.

CABI. 2007. Crop protection compendium: global module. Commonwealth Agricultural Bureau International, Wallingford, UK. http://www.cabi.org/compendia/cpc/

Cayrol, R. A. 1972. Famille des Noctuidae. In: Balachowsky AS, ed. Entomologie appliquée à l'agriculture. Vol. 2. Paris, France: Masson, 1411-1423.

Chari, M. S. and S. N. Patel. 1983. Cotton leaf worm *Spodoptera litura* Fabricius its biology and integrated control measures. Cotton Dev. 13(1): 7-8.

Chatterje, P. B. and D. K. Nayak. 1987. Occurrence of Spodoptera litura (Fabr.) as a new pest of sugar-beet in West Bengal. Pesticides, 21:21-22.

EPPO. 2012. EPPO Plant Quarantine Information Retrieval System (PQR), version 5.0.5540. European and Mediterranean Plant Protection Organization.
Hashmat, M. and M. A. Khan. 1977. The effect of temperature on the fecundity and fertility of *Spodoptera litura* (Fabr.) (Lepidoptera: Noctuidae). J. Anim. Morphol. Physiol. 24(2): 203-210.

Hashmat, M. and M. A. Khan. 1978. The fecundity and the fertility of *Spodoptera litura* (Fabr.) in relation to photoperiod. Z. Angew. Entomol. 85(2): 215-219.

Hill, D. S. 1975. Agricultural insect pests of the tropics and their control. Cambridge Univ. Press, London.

Hirano, C. 1976. Effect of trap location on catches of males of *Spodoptera litura* (Lepidoptera: Noctuidae) in pheromone baited traps. Appl. Entomol. Zool. 11(4): 335-339.

Holloway, J. D. 1989. The moths of Borneo: family Noctuidae, trifine subfamilies: Noctuinae, Heliothinae, Hadeninae, Acronictinae, Amphipyrinae, Agaristinae. Malayan Nature Journal, 42:57-228.

Kranthi, K. R., D. R. Jadhav, R. R. Wajari, S. S. Ali, and D. Russell. 2001. Carbamate and organophosphate resistance in cotton pests in India, 1995 to 1999. Bull. Ent. Res. 91: 37-46.

Kranz, J., H. Schumutterer, and W. Koch (eds.). 1977. Diseases Pests and Weeds in Tropical Crops. Berlin and Hamburg, Germany: Verlag Paul Parley.

Krishnananda, N. and S. V. V. Satyanarayana. 1985. Effect of height of pheromone trap on the catch of *Spodoptera litura* moths in tobacco nurseries. Phytoparasitica 13(1): 59-62.

Maheswara Reddy, A. 1983. Observations on the mating behaviour of tobacco cut worm, *Spodoptera litura* (F.). Madras Agric. J. 70(2):137-138.

Mochida, O. 1973. Two important insect pests, *Spodoptera litura* (F.) and *S. littoralis* (Boisd.) (Lepidoptera: Noctuidae), on various crops - morphological discrimination of the adult, pupal and larval stages. Applied Entomology and Zoology, 8: 205-214.

Obeng-Ofori, D. and J. Sackey. 2003. Field evaluation of non-synthetic insecticides for the management of insect pests of okra *Abelmoschus esculentus* (L.) Moench in Ghana. Ethiop. J. Sci. 26(2): 145-150.

Ohbayashi, N., T. Yushima, H. Noguchi, and Y. Tamaki. 1973. Time of mating and sex pheromone production and release of *Spodoptera litura* (F.) (Lepidoptera: Noctuidae). Kontyu: 41(4): 389-395.

Parasuraman, S. 1983. Larval behaviour of *Spodoptera litura* (F.) in cotton agro-ecosystem. Madras Agric. J. 70(2): 131-134.

Parasuraman, S. and S. Jayaraj. 1982. Studies on light trap catches of tobacco caterpillar *Spodoptera litura* (F.). Cotton Dev. 13(1): 15-16.

Parasuraman, S. and S. Jayaraj. 1983a. Effect of temperature and relative humidity on the development and adult longevity of the polyphagous *Spodoptera litura* (Fabr.) (Lepidoptera: Noctuidae). Indian J. Agric. Soc. 53(7): 582-584.

Parasuraman, S. and S. Jayaraj. 1983b.Pupation behaviour of *Spodoptera litura* (F). Madras Agric. J. 69(12): 830-831.

Patnaik, H. P. 1998. Pheromone trap catches of *Spodoptera litura* F. and extent of damage on hybrid tomato in Orissa. Advances in IPM for horticultural crops. Proceedings of the First National Symposium on Pest Management in Horticultural Crops: environmental implications and thrusts, Bangalore, India, 15-17 October 1997, 68-72.

Pearson, E. O. 1958. The insect pests of cotton in tropical Africa. Commonwealth Inst. Entomol., London.

Pillai, K. S., M. S. Palaniswami, P. Rajamma, C. Mohandas, and C. A. Jayaprakas. 1993. Pest management in tuber crops. Indian Horticulture, 38:20-23.

Pogue, M. G. 2002. A world revision of the genus *Spodoptera* guenee (Lepidoptera: Noctuidae). Mem. American Entomol. Soc. 43: 1-202.

Ranga Rao, G. V., J. A. Wightman, and D. V. Ranga Rao. 1989. Threshold temperatures and thermal requirements for the development of Spodoptera litura (Lepidoptera: Noctuidae). Environ. Entomol. 18(4): 548-551.

Ranga Rao, G. V., J. A. Wightman, and D. V. Ranga Rao. 1991. Monitoring Spodoptera litura (F.) (Lepidoptera: Noctuidae) using sex attractant traps: Effect of trap height and time of the night on moth catch. Insect. Sci. Applic. 12(4): 443-447.

Ranjith, A. M., B. Haseena, and S. S. Nambiar. 1997. *Spodoptera litura* causes brown flag syndrome in banana. Insect Environment, 3:44.

Roychoudhury, N., K. Shamila, and K. C. Joshi. 1995. Pest status and larval feeding preference of *Spodoptera litura* (Fabricius) Boursin (Lepidoptera: Noctuidae) on teak. Indian Forester, 121:581-583.

Schmutterer H. 1969. Pests of crops in Northeast and Central Africa. Stuttgart, Germany: Gustav Fischer Verlag, 186-188.

Singh, O. P., S. M. Matkar, and G. M. Gangrade. 1975. Sex identification of *Spodoptera* (*Prodenia*) *litura* (Fabricius) by wing coloration. Curr. Sci. 44(18): 673.

Tamaki, Y. 1973. Sex pheromone of *Spodoptera litura* (F.) (Lepidoptera:Nocutidae): isolation, identification, and synthesis, Appl. Entomol. Zool. 8(3): 200-203.

Todd, E. L. and R. W. Poole. 1980. Keys and illustrations for the armyworm moths of the Noctuidae genus Spodoptera *guenee* from the Western Hemisphere. Ann. Entomol. Soc. Am. 73: 722-738.

USDA. 1982. Pests not known to occur in the United States or of limited distribution: Rice cutworm. USDA-APHIS-PPQ.

Vaishampayan, S. Y. and R. Verma. 1983. Comparative efficiency of various light sources in trapping adults of *Heliothis armigera* (Hubn.), *Spodoptera litura* (Bois.) and *Agrotis ipsilon* (Hufn.) (Lepidoptera: Noctuidae). Indian J. Agric. Sci. 53(3): 163-167.

Venette, R. C., E. E. Davis, J. Zaspel, H. Heisler, and M. Larson. 2003. Mini-risk assessment: Rice cutworm, *Spodoptera litura* Fabricius [Lepidoptera: Noctuidae].

Yamanaka, H., R. Nakasuji, and K. Kiritani. 1975. Development of the tobacco cutworm *Spodoptera litura* in special reference to the density of larvae. Bull. Koch. Inst. Agric. For Sci. 7: 1-7. (English summary).

Yushima, T., Y. Tamaki, S. Kamano, and Oyama. 1974. Field evaluation of synthetic sex pheromone 'Litlure' as an attractant for males of *Spodoptera litura* (F.) (Lepidoptera: Noctuidae). Applied Entomology and Zoology 9:147-152.

Secondary Pests of Soybean (Truncated Pest Datasheet)

Adoxophyes orana

Scientific Name
Adoxophyes orana Fischer von Roeslerstamm

Synonyms:
Adoxophyes reticulana, Capua reticulana, Cacoecia reticulana, Capua orana, Tortrix ornana, Tortrix reticulana, Capua congruana, Adoxopjues tripsiana, Adoxophyes fasciata, Adoxophyes congruana, Acleris reticulana.

Common Name
Summer fruit tortrix, reticulated tortrix, apple peel tortricid

Type of Pest
Moth

Taxonomic Position
Class: Insecta, **Order:** Lepidoptera, **Family:** Tortricidae

Reason for Inclusion in Manual
CAPS Target: AHP Prioritized Pest List – 2009 through 2013

Pest Description
Eggs: Yellowish and deposited in masses (Fig. 1). After hatching, the transparent egg shells remain present.

Larvae: (Fig. 1) Greenish with light hairs and warts. The head is light brown to yellow (sometimes somewhat spotted) as is the thoracic shield and the anal shield. The anal comb is very fine and long with light colored teeth. The thoracal legs are brown to black. The head is long and wide. Abdominal and anal prolegs are greenish.

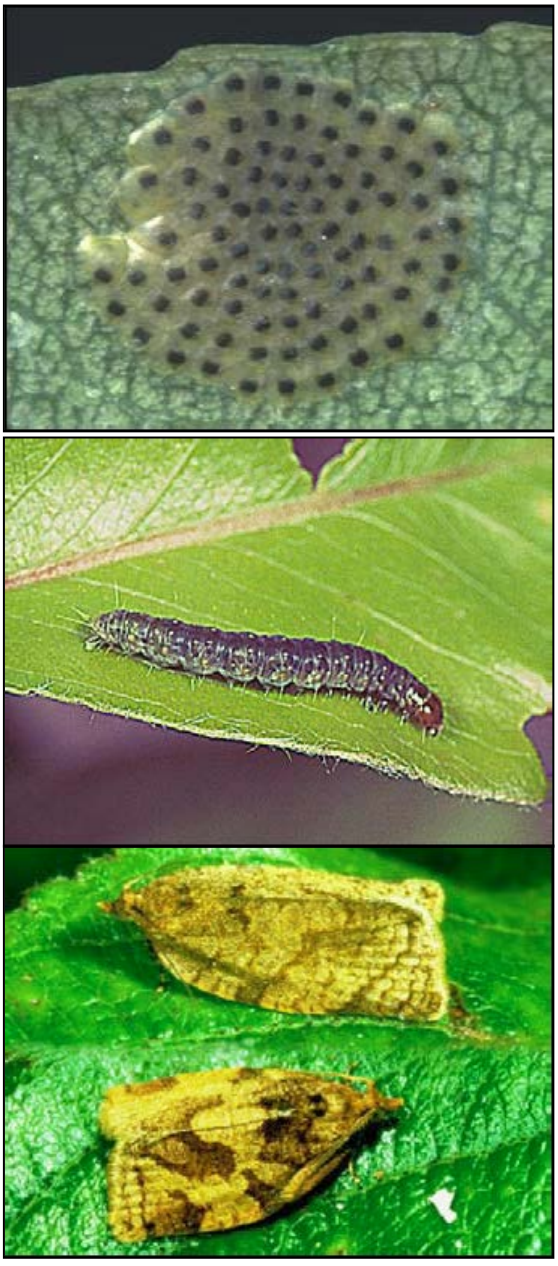


Figure 1. Eggs, larva, and adult *A. orana* (female top, male bottom). Photos courtesy of R. Coutin/OPIE.

Pupae: The pupae of *A. orana* are initially light brown, but become dark brown towards the time of emergence of the adult moth. The length is between 8 and 11 mm (approx. $^{5}/_{16}$ to $^{7}/_{16}$ in). The posterior margin of abdominal segments 2 to 8 of the pupae contains very small bristles. These bristles cannot be distinguished with a regular magnifying glass and are hence visible as a line. The specific fork-shape of wing veins 7 and 8 is already visible in the pupal stage.

Adults: A very specific characteristic of *A. orana* is the fork-shaped structure of the wing veins 7 and 8. The forewing of the female is rather dull grayish brown, while in the male the coloration is brighter and is a yellowish brown (Fig. 1). Male wingspan 15 to 19 mm (approx. $^{5}/_{8}$ to $^{3}/_{4}$ in), female 18 to 22 mm (approx. $^{11}/_{16}$ to $^{7}/_{8}$ in). Sexual dimorphism pronounced; antenna of male shortly ciliate, forewing with broad costal fold from base to about one-third, markings usually conspicuous, contrasting with paler ground color; female usually larger, antenna minutely ciliate, forewing without costal fold, with darker general coloration and less contrasting markings (Bradley et al., 1973).

Symptoms/Signs

External feeding will be visible on leaves and fresh growth of twigs. Feeding will deform leaves and create areas with necrosis (dead tissue). Leaves may appear wilted, yellow, shredded, or dead. Leaves are likely to be rolled or folded and held together with silk webbing. Feeding on new growth of twigs will leave lesions. If the insect is feeding in flowers, external feeding damage and silk webbing will be evident. In all areas where the insect has fed, frass should also be visible.

Summer generation larvae feed extensively and severely damage fruit (Fig. 2). Feeding on fruits or pods causes scabs or pitting, and frass may be present. On fruit crops, larvae prefer to feed sheltered under a leaf bound to fruit and silk.

Figure 2. Damage to apple epidermis showing “gnawed” appearance (top) and damage to pear foliage and fruit (bottom). Photos courtesy of R. Coutin/OPIE.

Survey

CAPS-Approved Method*:

The CAPS-approved method is a trap and lure combination. The trap is a paper delta trap. The lure is effective for 84 days (12 weeks). The lure dispenser type is a rubber septum.

Any of the following Trap Product Names in the IPHIS Survey Supply Ordering System may be used for this target:

Paper Delta Trap, 2 sticky sides, Brown
Paper Delta Trap, 2 sticky sides, Green
Paper Delta Trap, 2 sticky sides, Orange

The Lure Product Name is "*Adoxophyes orana* Lure."

IMPORTANT: Do not place lures for two or more target species in a trap unless otherwise recommended.

Trap spacing: When trapping for more than one species of moth, separate traps for different moth species by at least 20 meters (65 feet).

Notes: Trap should be used with ends open. Trap color is up to the state and does not affect trap efficacy.

*For the most up-to-date methods for survey and identification, see Approved Methods on the CAPS Resource and Collaboration Site, at http://caps.ceris.purdue.edu/.

Literature-Based Methods:

Trapping: Several monitoring techniques have been developed and applied to *A. orana*. The most effective approach involves sex pheromone- baited traps. The sex pheromone is a blend of (*Z*)-9-tetradecenyl acetate and (*Z*)-11-tetradecenyl acetate. These two compounds are most attractive to males in a 9:1 blend of (Z)-9:(Z)-11 isomers; *E*-isomers of either compound had a strong inhibitory effect (Davis et al., 2005). The 9:1 pheromone blend is available commercially as Adoxomone (Murphy Pherocon™ Summer Fruit Tortrix Moth Attractant) for use with Pherocon 1C traps [Zoecon Corp].

Visual survey: Visual sampling and beat sampling may also be used to inspect plants for eggs and larvae. Eggs may be observed on the stems and leaves; late instars may be found in the crown on new shoot growth; and pupal cocoons may be found in leaves, on stems, or in mummified pods/seeds. Both methods are time consuming. Visual sampling or beat sampling are not commonly recommended (Davis et al., 2005).

Not recommended: As an alternative to pheromone traps, Robinson light traps with 125W mercury vapor bulbs, 125 W black light bulbs, or 100W flood lights can be used. While sex pheromone traps attract males of a targeted species, light traps non-selectively draw in many flying insects.

Surveys should be focused where the greatest risk for establishment occurs. A recent risk analysis by USDA-APHIS-PPQ-CPHST indicates that most states in the United States have a low to moderate risk rating for *A. orana* establishment based on host availability, climate, and pathway within the continental United States. Areas of the northeastern and southeastern United States, as well as Arizona, California,

Kansas, Minnesota, Nebraska, New Mexico, Oklahoma, Texas, and Wisconsin have the highest risk of *A. orana* establishment.

Key Diagnostics/Identification

CAPS-Approved Method*: Confirmation requires a morphological identification. *Adoxophyes orana* may occur in mixed populations with other morphologically similar species, including other *Adoxophyes* species. Final identification is by dissection of male genitalic structures.

*For the most up-to-date methods for survey and identification, see Approved Methods on the CAPS Resource and Collaboration Site, at http://caps.ceris.purdue.edu/.

Literature-Based Methods: Because of their very secretive nature, leafrollers are difficult to detect. Distinguishing between males and females of adult *Adoxophyes* is difficult in general (Balachowsky 1966). According to Yasuda (1998), the extensive color and pattern variation of the forewing and morphological resemblance among *Adoxophyes* species have created difficulties in the identification of the species. *A. orana* very closely resembles two U.S. species, *Adoxophyes furcatana* and *A. negundana*, but there are slight differences in male genitalia. Any identification should be confirmed by an appropriately trained entomologist.

References

Bradley, J.D., Tremewan, W.G., and Smith, A. 1973. British Tortricoid moths. Cochylidae and Tortricidae: Tortricinae. Ray Society, London.

Davis, E.E., French, S., and Venette, R.C. 2005. Mini Risk Assessment – Summer Fruit Tortrix Moth, *Adoxophyes orana* (Fischer von Roslerstamm, 1834) [Lepidoptera: Tortricidae]. http://www.aphis.usda.gov/plant_health/plant_pest_info/pest_detection/downloads/pra/aoranapra.pdf.

Eutetranychus orientalis

Scientific Name
Eutetranychus orientalis Klein

Synonyms:
Anychus orientalis, Anychus ricini, Eutetranychus monodi, Eutetranychus sudanicaus, Eutetranychus anneckei, Anychus latus, Eutetranychus latus

Common Name(s)
Citrus brown mite, oriental mite, oriental red mite, oriental spider mite

Type of Pest
Mite

Taxonomic Position
Class: Arachnida, **Order:** Acarina, **Family:** Tetranychidae

Reason for Inclusion in Manual
CAPS Target: AHP Prioritized Pest List - 2009

Pest Description
The genus *Eutetranychus* is characterized by its empodium, which is reduced to a small protuberance (Avidov and Harper, 1969). The life cycle of *E. orientalis* is completed in four active (larva, protonymph, deutonymph, and adult) and three quiescent stages (nymphochrysalis, deutochrysalis, and teleochrysalis) (Lal, 1977).

Eggs: The eggs of *E. orientalis* are oval or circular (Fig. 1) and flattened, coming to a point dorsally, but lacking the long dorsal stalk of other spider mites. Newly laid, the eggs are bright and hyaline, but later they take on a yellow, parchment-like color (Smith-Meyer, 1981). Diameter of the eggs is 0.14 mm (Avidov and Harper, 1969).

Figure 1. Eggs (left) and adult (right) of *E. orientalis*. Photos courtesy of Pedro Torrent Chocarro.

Larvae: Average size of the larva of *E. orientalis* is 190 x 120 µm. The abdomen of female larvae and nymphs is greenish brown, while the abdomen of male larvae is reddish brown. The protonymph is pale-brown to light-green, with legs shorter than the

body, average size 240 x 140 µm. The deutonymph is pale-brown to light-green, average size 300 x 220 µm.

Adults: Adult female *E. orientalis* are broad, oval and flattened. They vary in color from pale brown through brownish-green to dark green with darker spots within the body. The legs are about as long as the body and are yellow-brown (Fig. 1). Average size is 410 x 280 µm. Adult male *E. orientalis* are much smaller than the females. They are elongate and triangular in shape with long legs (leg about 1.5 x body length). The body setae are short and cannot be seen with a 10x lens (Dhooria and Butani, 1984; Smith-Meyer, 1981).

Technical description: Empodia lacking on all tarsi; true claws slender, padlike, each with pair of tenent hairs; duplex setae of tarsi loosely associated, not paired as in other spider mites; 2 pairs of anal setae; 3 pairs of dorsal propodosomal setae, and 10 pairs of dorsal hysterosomal setae, all setae stout, serrate; dorsal striae of hysterosoma form V-pattern between setae D1 and E1, and setal bases E1 and F1 form a square; setal cout (solenidia or sensory rodlike setae in parentheses) of legs (Meyer, 1974). L coxa 2-1-1-1, trochanter 1-1-1-1, femur (8-6-3/4-1/2), genu (5-5-2-2), tibia 9(1/4)-6(0/2)-6(0/1)-7, and tarsus 15(3)-13(1/2)-10(1)-10(1).

Adult male *E. orientalis* are much smaller than the females. They are elongate and triangular in shape with long legs (leg about 1.5 x body length). Usually males have a higher solenidia count.

Short setae are found on legs and body of both sexes at all stages. The body setae are short, however, and cannot be seen with a 10x lens (Smith-Meyer, 1981; Dhooria and Butani, 1984).

The outstanding characteristic in the adult is that the legs are equal to, or longer than, the body length (Avidov and Harper, 1969).

Symptoms/Signs

Eutetranychus orientalis begins feeding on the upper side of the leaf along the midrib and then spreads to the lateral veins, causing the leaves to become chlorotic. Pale yellow streaks develop along the midrib and veins (Fig. 2) initially, which later progress to a grayish or silvery appearance of the leaves. At times, the leaves appear to be

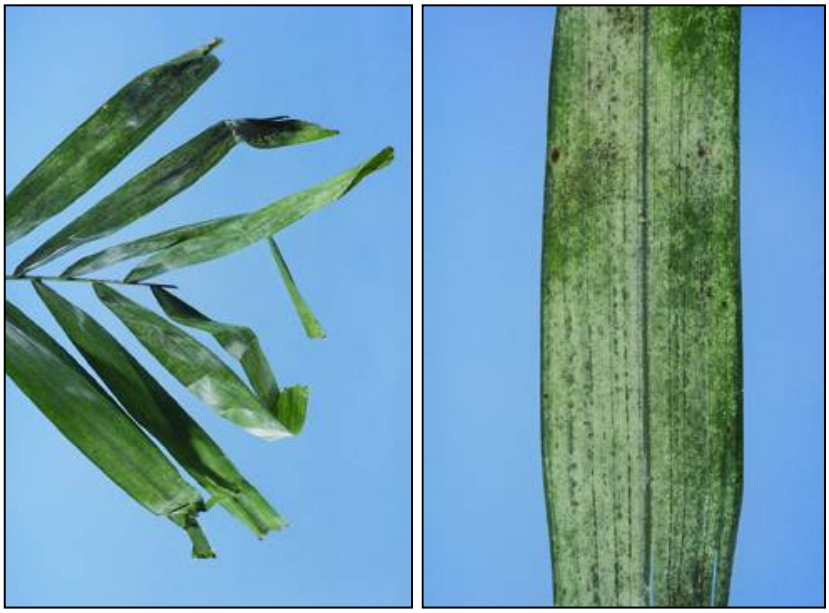


Figure 2. *Eutetranychus* feeding damage on *Ptychosperma* palm. Photos courtesy of http://www.pestalert.org/viewArchPestAlert.cfm?rid=62.

covered in a layer of fine dust. When damaged, the younger, tender leaves show margins that are twisted upwards. Usually, little webbing is produced but can occur. In heavier infestations, the mites feed and oviposit over the whole upper surface of the leaf. Very heavy infestations on citrus cause leaf fall and die-back of branches, which may result in defoliated trees. Lower populations in dry areas can produce the same effect.

Survey

CAPS-Approved Method*: Visual survey is the method to survey for *E. orientalis.*

*For the most up-to-date methods for survey and identification, see Approved Methods on the CAPS Resource and Collaboration Site, at http://caps.ceris.purdue.edu/.

Literature-Based Methods:

Visual survey: The presence of *E. orientalis* can be detected by discoloration of the host leaves and pale-yellow streaks along the midribs and veins. Eggs, immature stages, and adults may be observed visually on the upper leaf surface. Adult females are larger than the males. They are oval and flattened and are often pale brown through brownish-green to dark green. Webbing is possible (often dust colored), providing protection for the eggs. The spread of the mite is windborne, and new infestations commonly occur at the field perimeters. Field perimeters should, therefore, be scouted, especially field perimeters facing prevailing winds. Studies indicate that alfalfa plays a role in dispersing tetranychid mites to other crops (Osman, 1976). Fields near alfalfa should be targeted for survey. Shake leaves above white paper or cloth, and use a hand lens to observe mites.

Surveys should be focused where the greatest risk for establishment occurs. A recent risk analysis by USDA-APHIS-PPQ-CPHST indicates that most states in the United States are at low to moderate risk for *E. orientalis* establishment based on climate and host availability. Florida, however, has a moderate to high risk for establishment of this mite. Establishment of *E. orientalis* is unlikely in portions of Colorado, Idaho, Indiana, Iowa, Kansas, Louisiana, Minnesota, Montana, Nebraska, New Mexico, North Dakota, Oklahoma, Oregon, South Dakota, Texas, Utah, Washington, and Wyoming.

Hall (1992) discusses sampling strategies for spider mites in orange groves. The author's sampling method consisted of examining 16 leaves per tree, 5 trees within a small area of trees, and 3 areas per block. The leaves were collected by gently pulling four leaves from each of the north, east, south, and west sides of a tree. The leaves from each side of the tree were placed into separate plastic bags. The bags were placed in a cold ice chest, taken to the laboratory, and examined under a microscope to count the number of spider mites present per leaf (both surfaces).

Gilstrap and Browing (1983) recommend using a liquid sampling procedure for leaf collecting of mites, where leaves are placed in a jar filled with 0.5% liquid dishwashing soap and 0.5% standard bleach (5% NaCl) (each % by volume) in a solvent of distilled water. The liquid soap is used to break up surface tension; while the bleach is used to

dissolve any webbing. The author showed that the liquid sampling procedure collected more mites than more mites than the 'normal procedure'. In the 'normal procedure', leaves are placed in a paper bag and a mite brushing machine is used to dislodge mites from the samples when processed the next day. Dhorria et al. (1982) collected forty random leaves (10 leaves/tree) from each almond variety at different heights and all sides of the plants to assess mite resistance. A mite brushing machine was used to dislodge the mites from the leaves on to counting disks.

Key Diagnostics/Identification

CAPS-Approved Method*: Confirmation of *E. orientalis* is by morphological identification. The mite can only be identified by examination of the adult male.

*For the most up-to-date methods for survey and identification, see Approved Methods on the CAPS Resource and Collaboration Site, at http://caps.ceris.purdue.edu/.

Literature-Based Methods: According to a NAPPO pest alert, the only form of *E. orientalis* that can be identified is the adult male. Conflicting information states that identification of *E. orientalis* requires examination of cleared and mounted female specimens by transmitted light microscopy.

Mite experts agree that though it may be possible to identify a specimen with a slide mounted female, one can never be 100% sure without a male for confirmation. *E. orientalis* can be easily mistaken for the Texas citrus mite (*E. banksii*). Similarity of the female *E. orientalis* with other tetranychid mites such as the two-spotted mite *(Tetranychus urticae)* can make identification difficult.

References

Avidov, Z. and Harper, I. 1969. Plant Pests of Israel. Israel Universities, Jerusalem.

Dhorria, M.S., Sandhu, G.S., Khangura, J.S., and Dhatt, A.S. 1982. Incidence of citrus mite, *Eutetranychus orientalis* (Klein) on different varieties of almond in Ludhiana. Punjab Hortic. J. 22(1/2): 110-112.

Dhooria, M.S. and Butani, D.K. 1984. Citrus mite, *Eutetranychus orientalis* (Klein) and its control. Pesticides 18(10): 35-38.

Gilstrap, F.E. and Browing, H.W. 1983. Sampling predaceous mites associated with citrus. PR Tex. Agric. Exp. Stn. 4149.

Hall, D.G. 1992. Sampling citrus and red mites and Texas citrus mites in young orange trees. Proc. Annu. Meet. Fla. State Hort. Soc. 105: 42-46.

Lal, L. 1977. Studies on the biology of the mite *Eutetranychus orientalis* (Klein) (Tetranychidae: Acarina). Entomol. 2(1): 53-57.

Meyer, M.K.P.S. 1974. A revision of the Tetranychidae of Africa (Acari) with a key to genera of the world. Entomology Memoir, Department of Agricultural Technical Services, Republic of South Africa No. 36.

Osman, A.A. 1976 (publ. 1980). The role of alfalfa in dispersing tetranychid mites to other crops. Bull. Soc. Ent. Egypte 60: 279-283.

Smith-Meyer, M.K.P. 1981. Mite pests of crops in southern Africa. Science Bulletin, Department of Agriculture and Fisheries, Republic of South Africa, (No. 397): 65.

Requested by CAPS Community (Full Pest Datasheet)

Crocidosema aporema

Scientific Name
Crocidosema aporema Walsingham

Synonyms:
Epinotia aporema, Epinotia opposita, Eucosma opposita, Eucosma aporema

Common Name(s)
Bud borer, bean shoot moth, budworm

Type of Pest
Moth

Taxonomic position:
Class: Insecta, **Order:** Lepidoptera, **Family:** Tortricidae

Reason for Inclusion in Manual:
Requested by the CAPS community

Pest Description
Crocidosema aporema was first described by Walsingham (1914) as *Eucosma aporema* from Costa Rica. Heinrich (1931) described it as *Epinotia opposite*. Peru and Clarke (1954) considered both species synonymous, and named it *Epinotia aporema*. Powell et al. (1995) transferred it to the genus *Crocidosema.*

Morey (1972) described and illustrated the larval, pupal and adult morphology of *C. aporema* (Fig. 1).

Eggs: Oval, 0.47-0.6 in length x 0.31 mm in width, pale yellow or light green soon after oviposition. The chorion is translucent and dotted (Morey, 1972; Sanchez and Pereyra, 2004).

Larvae: There are five larval instars; the first to the fourth are light yellow or green with a black head. These instars are morphologically similar, except for the size of the larva (about 10 mm (approx. $^{3}/_{8}$ in) long when fully developed). The fifth instar has a reddish head and a yellow body.

Pupae: Brown, 6.2 to 7.8 mm (approx. $^{1}/_{4}$ to $^{5}/_{16}$ in) long and 1.8 to 2.2 mm (approx. $^{1}/_{16}$) wide (Morey, 1972).

Adults: Small, dark moths; forewings with a brown patterning, hindwings grey. The adult measures 10 to 17mm (approx. $^{3}/_{8}$ to $^{11}/_{16}$ in) in width when wings are expanded. Sexes can be identified by the distribution patterns of black, bronze, and light gray scales of the first pair of wings (Morey, 1972; Sanchez and Pereyra, 2004). Males are laterally dark and dorsally light in color, while females are exactly the opposite (Ferreira, 1980).

Biology and Ecology

Little work has been completed on *C. aporema*. Many aspects of its biology are not known, and the literature on this species is very limited (Ferreira, 1980). The life cycle is multivoltine; there are four to five generations per year. At least two generations occur on soybean and other legumes. Adult activity starts at the end of the spring and lasts until the beginning of winter. Females start to lay eggs (a maximum of 119 eggs) about 2 to 4 days after emergence and first larvae are detected approximately two weeks after adult emergence (Sanchez and Pereyra, 2004). There are five larval instars that vary in size. A winter diapause occurs during the last larval instar. Before pupation, they turn a reddish color and spin a cocoon.

Mean development time at 25 °C (77°F), for the different stages is 7, 18, 11, and about 20 days for egg, larvae, pupae, and adult stages, respectively. The maximal adult longevity is 23 days for the male and 21 days for the female (Sanchez and Pereyra, 2004).

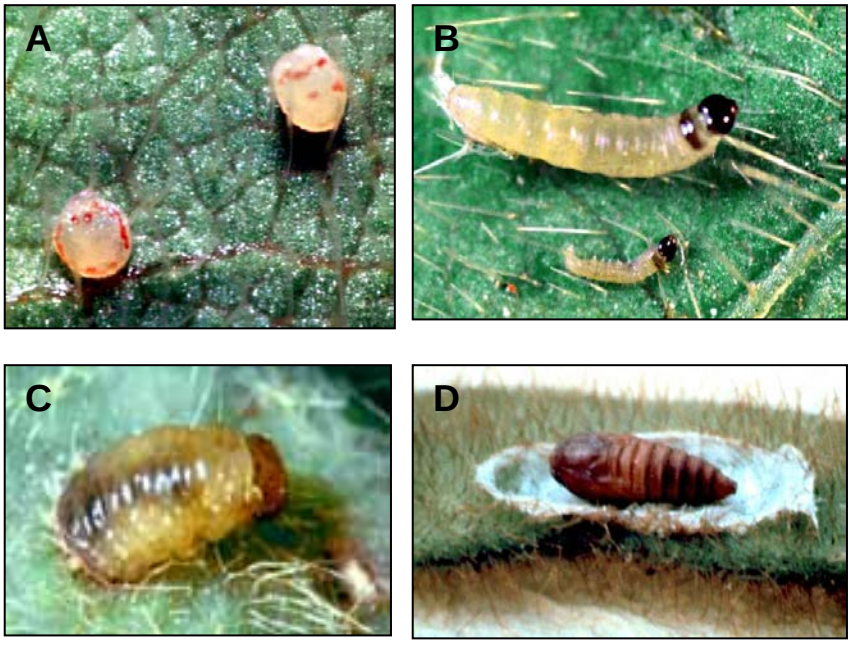


Figure 1. (A) Eggs, (B) first (smaller) and third instar larvae, (C) fifth instar larva and (D) pupa. Photos courtesy of CABI, 2007.

Pest Importance

C. aporema is a frequent species attacking soybeans and other Fabaceous plant species in southern Brazil, Uruguay, Chile and Argentina. Higher incidence occurs during the vegetative stages of soybeans. Plant height and insertion of the lower pods are significantly reduced as a result of its attack on terminal buds. The larval habits as a stem borer and leaf roller make control procedures difficult (Ferreira, 1980).

Symptoms/Signs

Larvae feed mainly on vegetative buds and can also bore into stems, floral buds, and pods. The damage caused to the buds is easily recognized. Newly hatched larvae move into the most tender buds, where they feed on one of the youngest furled leaflets. As the larvae develop, the damaged apical leaflets remain attached to one another by a silken web forming a sort of a cartridge that characterizes the attack of these lepidopterous larvae (Ferreira, 1980). Larvae persist inside the rolled leaves until they pupate in the soil or in the same leaflets. Attacked plants may be recognized by the rolled young leaflets, which contain the larvae. The larvae also tunnel along the main and secondary stems of soybean plants, drying out the terminal shoots (Pereyra and Sanchez, 1998; Sanchez and Pereyra, 2004). Webbing is possible.

Plant height and insertion of the lower pods can be severely reduced due to the death of the apical buds. Yield reductions are possible if infestations occur during flowering. Adults and larvae feed on reproductive structures of flowers of other host crops (e.g. *Lotus spp.*) causing flower parts to stick together (Alzugaray, 2003).

Known Host

Host records for *C. aporema* are restricted to the Fabaceae (leguminous hosts) (Biezanko et al., 1974; King and Saunders, 1984). *Crocidosema aporema* feeds on the buds on many native and cultivated legumes such as peanut, clover, alfalfa, lotus, pea, melilotus, lupine, broad bean, common bean, and others (Sanchez and Pereyra, 2004).

Major hosts

Glycine max (soybean)

Minor hosts

Arachis hypogaea (peanut), *Lotus* spp.(trefoils), *Lupinus* (lupins), *Medicago sativa* (alfalfa), *Melilotus* (melilots), *Phaseolus vulgaris* (common bean), *Pisum sativum* (pea), *Trifolium* spp. (clover), and *Vicia faba* (broad bean)

Known Vectors (or associated insects)

Crocidosema aporema is not a known vector and does not have any associated organisms.

Known Distribution
Crocidosema aporema is distributed throughout the Neotropical region, including Mexico and southern United States. Its southern range includes: Chile, Uruguay, Argentina and southern Brazil (Sanchez and Pereyra, 2004).

Pathway
Crocidosema aporema could potentially move through international trade. This species has been intercepted at U.S. ports of entry 980 times. Most interceptions originated on material from Guatemala (640), Peru (115), Mexico (51), and Equador (46). Interceptions occurred mostly on plant material including *Phaseolus vulgaris* (469), *Phaseolus* sp. (348), and *Pisum sativum* (41). These were found mainly in permit cargo (742), baggage (164), and general cargo (34) (AQAS, 2012; queried September 12, 2012).

Potential Distribution within the United States
According to Clarke (1954), *C. aporema* is currently present in Texas. The NAPIS database, however, does not have any records of *C. aporema* within the United States. A recent risk analysis by USDA-APHIS-PPQ-CPHST indicates areas from North Dakota east to Ohio and south to Mississippi are at moderate risk *C. aporema* establishment based on strictly on host availability within the continental United States. Portions of the east coast are also at moderate risk.

Survey
CAPS-Approved Method*: Blacklight trapping.

*For the most up-to-date methods for survey and identification, see Approved Methods on the CAPS Resource and Collaboration Site, at http://caps.ceris.purdue.edu/.

Literature-Based Methods:
Visual Survey: Damage by *C. aporema* larvae is easily detected visually by rolled leaflets during the vegetative stage, and by the presence of frass at the larval entrance hole in stems. Although the preferred feeding sites of *C. aporema* larvae in soybean are the buds (the most pubescent part), a greater proportion of its eggs are laid mainly on nodes and secondarily on expanded leaves (Sanchez and Pereyra, 2004). Since the eggs of this pest are laid normally on the most tender leaflets of the buds and since they are very small, observation of the eggs in the field is practically impossible. In Brazil, high populations of *C. aporema* are detected at the end of the soybean vegetative stage through the flowering stage (R2), and these populations are considered to be economically important (Ferreira, 1980).

Trapping: Black light trap have been used to monitor adult activity in Uruguay. Results have shown that approximately two weeks after adult captures in light traps, high larvae populations are observed in the field (Alzugaray, 2003). In Brazil, adults have been monitored using direct observation after collection and examination of plants and by sweep netting at dusk. Additionally adults have been sampled on alfalfa by counting the

number of moths that flew each 25 steps within a field (flushing method) (Ferreiro, 1980).

Note: Although the pheromone (Z, Z)-7,9-dodecadienyl acetate has been reported to attract males of up to four *Epinotia* spp. (the previously assigned genus for the soybean bud borer), there have been no reports of a pheromone for this species (Reed and Chisholm, 1985; Priesner et al., 1989).

Soil Sampling: Sampling methods for pupae have not been investigated. As the pupae are normally located in the soil (1 or 2 cm below the soil surface) near the stems, measurements of pupal populations may be obtained by sampling a given area of soil (Ferreira, 1980).

Key Diagnostics/Identification

CAPS-Approved Method*: Confirmation of *C. aporema* is via morphological identification.

*For the most up-to-date methods for survey and identification, see Approved Methods on the CAPS Resource and Collaboration Site, at http://caps.ceris.purdue.edu/.

Literature-Based Methods: Wing color and/or dissection of genitalia are required to identification of *C. aporema*.

Easily Confused Pests

The leaf rolling behavior of *C. aporema* can be mistaken for that of *Omiodes indicata* and *Cydia fabivora*. However, *C. aporema* attacks young leaflets, while the two other species are commonly found on fully developed leaves.

References

Alzugaray, R. 2003. Insect pests damaging *Lotus corniculatus* L. flowers and seeds in Uruguay. Lotus Newsletter 33: 11-18.

AQAS. 2012. AQAS database, *Crocidosema aporema* interceptions. Last accessed September 12, 2012 from: https://mokcs14.aphis.usda.gov/aqas/HomePageInit.do.

Biezanko, C. M., A. Ruffinelli, and D. Link. 1974. Plantas y otras sustancias alimenticias de las orugas de los lepidopteros uruguayos. Revista do Centro de Cioncias Rurais, 4:107-148.

CABI. 2007. Crop Protection Compendium. Wallingford, UK: CAB International. www.cabicompendium.org/cpc

Clarke, J. F. G. 1954. The correct name for a pest of legumes. Proceeding of the Entomological Society of Washington 56: 309-310.

Ferreira, B. S. C. 1980. Sampling *Epinotia aporema* on soybean. *In:* M. Kogan and D.C. Herzog (eds.) Sampling methods in soybean entomology. Springer Verlag, New York. Pages 372-382.

Heinrich, C. 1931. Notes on and description of some American moths. Proceedings of the United States National Museum, 79:1-16.

King, A. B. S. and J. L. Saunders. 1984. The invertebrate pests of annual food crops in Central America. A guide to their recognition and control. London, UK: Overseas Development Administration.

Liljesthröm, G. and G. Rojas-Fajardo. 2005. Parasitismo larval de *Crocidosema* (=*Epinotia*) *aporema* (Lepidoptera: Tortricidea) en el noreste de la provincial de Buenos Aires (Argentina). Rev. Soc. Entomol. Argent. 64(1-2): 37-44.

Morey, C. S. 1972. Biologia y morfologia larval de *Epinotia aporema* (Wals.) (Lepidoptera: Olethreutidae). Facultad de Agronomia Montevideu, Boletin 123.

Pereyra, P. C. and N. E. Sánchez. 1998. Effects of different host-plant species on growth, development and feeding of the bud borer, *Epinotia aporema* (Lepidoptera: Tortricidae) in La Plata, Argentina. Revista Chilena de Historia Natural, 71:269-275.

Powell, J. A., J. Razowski, J. W. Brown, and R. L. Brown. 1995. Tortricidae. In: Heppner JB, (Ed.), Atlas of Neotropical Lepidoptera - Checklist: Part 2 Hyblaeoidea - Pyraloidea - Tortricoidea. Gainesville, USA: Scientific Publishers, Inc., 138-157.

Priesner, E., D. W. Reed, E. W. Underhill, and H. Bogenschutz. 1989. (Z,Z)-7,9-Dodecadienyl acetate, sex pheromone of *Epinotia tedella* Clerck (Lepidoptera: Tortricidae). Journal of Chemical Ecology 15(10): 2457-2464.

Reed, D. W. and M. D. Chisholm. 1985. Field trapping of three *Epinotia* species with (Z,Z)-(7,9-dodecadienyl acetate (Lepidoptera: Tortricidae) Journal of Chemical Ecology 11(10): 1389-1398.

Robinson, G. S., P. R. Ackery, I. J. Kitching, G. W. Beccaloni, and L. M. Hernández. 2011. HOSTS - a Database of the World's Lepidopteran Hostplants: *Eucosma aporema*. London: Natural History Museum. Retrieved March 6, 2012, from http://www.nhm.ac.uk/research-curation/research/projects/hostplants/search/index.dsml.

Sanchez, N. and P. Pereyra. 2004. Neotropical soybean borer, *Crocidosema aporema* (Walsingham, 1914) (Lepidoptera: Tortricidae). Encyclopedia of Entomoloty 1545-1546. Springer Netherlands.

Sauka, D. H., J. Sánchez, A. Bravo, and G. B. Benintende. 2007. Toxicity of *Bacillus thuringiensis* δ-endotoxins against bean shoot borer (*Epinotia aporema* Wals.) larvae, a major soybean pest in Argentina. Journal of Invertebrate Pathology 94: 125-129.

Walsingham, T. de G. 1914. Biologia Centrali Americana. Lepidoptera Heterocera, 4:225-392.

Mollusks

Primary Pests of Soybean (Full Pest Datasheet)

None at this time

Secondary Pests of Soybean (Truncated Pest Datasheet)

Cernuella virgata

Scientific name
Cernuella virgata Da Costa

Synonyms:
Cernuella virgatus, *Cernuella variabilis, Cernuella virgata* ssp. *variegata*, *Helicella maritime*, *Helicella variabilis*, *Helicella virgata*, *Helix virgata*

Common Name(s)
Maritime garden snail, Mediterranean snail, Mediterranean white snail, striped snail, vineyard snail, white snail

Type of Pest
Mollusk

Taxonomic Position
Class: Gastropoda, **Order:** Stylommatophora (Eupulmonata), **Family:** Hygromiidae (Helicidae)

Reason for Inclusion in Manual
CAPS Target: AHP Prioritized Pest List – 2009 through 2012 (as *Cernuella* spp.)

Pest Description
The shell of *C. virgata* is globose-depressed and white or yellowish-white in color with dark-brown bands or spots (Fig. 1, 2). Snail size is 6 to 19 mm (approx. $^{1}/_{4}$ to $^{3}/_{4}$ in) high x 8 to 25 mm (approx. $^{5}/_{16}$ to 1 in) wide. Shell size and banding patterns are reported to vary widely geographically throughout Southeastern Australia (Baker, 1988).

Figure 1. Banding of *C. virgata*. Photo courtesy of Tenby Museum

Size has been demonstrated as inversely proportional to population density (Baker, 1988). *Cernuella virgata* is considered polymorphic; banded and unbanded (more common) morphs have been found throughout Australia. Relative frequencies of each morph are likely correlated with site-specific factors such as predator pressure (Baker, 1988). The maritime gardensnail is relatively small and is characterized by prominent spiral banding on the shell (Fig. 1).

Symptoms/Signs

C. virgata is found atop plants during summertime (Fig. 3) and may also be found feeding on new growth earlier in the season. These snails aestivate on plant heads and stalks, which contaminates crops and clogs machinery. Areas previously infested with snails can prevent the re-establishment of a site as pastureland as livestock often reject slime-contaminated hay and forage (Baker, 2002).

Figure 2. *C. virgata*. Photo courtesy of L. Poggiani. http://www.lavalledelmetauro.it/.

Survey

CAPS-Approved Method*: Visual (Floyd, 2008).

*For the most up-to-date methods for survey and identification, see Approved Methods on the CAPS Resource and Collaboration Site, at http://caps.ceris.purdue.edu/.

Literature-Based Methods:

Visual survey: *Cernuella virgata* is a conspicuous crop pest that hides during the day. Surveys are best carried out at night using a flashlight or in the morning or evening following a rain event. It is easily seen, and attacked plants exhibit extensive rasping and defoliation. Like other mollusks, it can also be detected by signs of ribbon-like excrement and slime trails on plants and buildings.

Figure 3. Multiple *C. virgata* on tree trunk. Photo courtesy of L. Poggiani, http://www.lavalledelmetauro.it/.

At this time, a host risk map is available (Fig. 4). Surveys should take place at areas of greatest risk. The host risk map describes the relative density (on a scale of low to high) of susceptible hosts. This map shows that portions of North Dakota are at the greatest risk from this mollusk based on host availability. Portions of Arkansas, California, Colorado, Delaware, Idaho, Illinois, Indiana, Iowa, Kansas, Kentucky, Louisiana, Maryland, Michigan, Minnesota, Missouri, Mississippi, Montana, Nebraska, New York, North Carolina, North Dakota, Ohio,

Oklahoma, Oregon, Pennsylvania, South Dakota, Tennessee, Texas, Washington, and Wisconsin are also at moderate risk based on host availability.

Key Diagnostics/Identification

CAPS-Approved Method*:

Confirmation requires a morphological identification. All specimens should be submitted to Patrick Marquez (Western Region) or John Slapcinsky (Eastern Region). Both Domestic Identifiers are able to identify (even immature specimens) to the species level for this genus.

*For the most up-to-date methods for survey and identification, see Approved Methods on the CAPS Resource and Collaboration Site, at http://caps.ceris.purdue.edu/.

Literature-Based Methods: A morphological identification is required. *Cernuella virgata* is a relatively small snail (up to 15 mm in (approx. $^{9}/_{16}$ in) diameter) characterized by prominent spiral banding on the shell.

Cernuella virgata closely resembles the white Italian snail (*Theba pisana*) in appearance and pest status. *Cernuella virgata* can be differentiated from *T. pisana* by more pronounced spiral banding. Also, the umbilicus (hole about which the shell spirals) appears as a circular hole rather than being partially obscured as in the white Italian snail (CABI, 2007).

References

Baker, G.H. 1988. The life history, population dynamics and polymorphism of *Cernuella virgata* (Mollusca: Helicidae). Australian Journal of Zoology 36: 497-512.

CABI. 2007. Crop protection compendium: global module. Commonwealth Agricultural Bureau International, Wallingford, UK. http://www.cabi.org/compendia/cpc/.

Floyd, J. 2008. New Pest Response Guidelines: Temperate Terrestrial Gastropods. USDA-APHIS-PPQ-Emergency and Domestic Programs, Riverdale, Maryland.

Lissachatina fulica

Scientific Name
Lissachatina fulica Bowdich

Synonyms:
Achatina fulica

Common Name(s)
Giant African snail, African giant snail, Kalutara snail

Type of Pest
Mollusk

Taxonomic Position
Class: Gastropoda, **Order:** Pulmonata, **Family:** Achatinidae

Figure 1. The large size of the giant African snail. Photo courtesy of USDA-APHIS.

Reason for Inclusion in Manual
CAPS Target: AHP Prioritized Pest List – 2009 through 2013

Pest Description
Lissachatina fulica is distinctive in appearance and is readily identified by its large size and relatively long, narrow, conical shell (Fig. 1, 2).

Eggs: Elyptical, about 4 mm by 5 mm ($^{3}/_{16}$ in) in diameter, usually pale yellow, laid in clutches of 100 to 400 (Fig. 3) (USDA, 1982).

Juveniles: Similar to adults, but have a thinner, more brittle, translucent shell. Upon emergence, the juvenile shell is approximately 4 mm (approx. $^{3}/_{16}$ in) long (Denmark and Poucher, 1969; USDA-APHIS, 2005). Increase at a rate of 10 mm (approx. $^{3}/_{8}$ in) per month for first four months. The coloration is similar to adults. The columella is truncated.

Figure 2. Adult giant African snail. Photo courtesy of Matt Ciomperlik, USDA APHIS PPQ

Adults: Columella abruptly truncate (Burch, 1960). Columella and the parietal callus are white or bluish-white with no trace of pink (Bequaert, 1950). Shell size may be up to 8 inches (203 mm) in length and almost 5 inches (127 mm) in maximum diameter (Bequaert, 1950). Shell has seven to nine whorls and rarely as many as ten whorls (Bequaert, 1950). Shell color is reddish-brown with light yellowish, vertical (axial) streaks; or, light coffee colored. Protoconch is not bulbous. Body coloration can be either mottled brown or more rarely a pale cream color. The truncated columella is evident throughout the lifespan of the snail. The columella is generally concave. Snails with a lesser concaved columella tend to be somewhat twisted. Snails with a broader shell tend to have a more concave columella (Bequaert, 1950). In calcium-rich areas, the shells of the adults tend to be thicker and opaque (USDA-APHIS, 2005).

The giant African snail, *Lissachatina fulica*, is a polyphagous pest. This species is one of the most serious land snail pests known, reported to consume all growth stages of vegetables, cover crops, garden flowers, herbaceous ornamentals, and damaging many fruit and ornamental trees (USDA, 1982). Its preferred food is decayed vegetation and animal matter, lichens, algae and fungi. The bark of relatively large trees such as citrus, papaya, rubber and cocoa is also subject to attack. Poaceous crops (sugarcane, maize, rice) suffer little or no damage from this species. There are reports of *L. fulica* feeding more than 500 species of plants (CABI, 2007).

Figure 3. Giant African snail eggs. Courtesy of USDA APHIS.

A large infestation presents a nuisance problem with slime trails, excretions, and odors of decay when they die in large numbers. *Lissachatina fulica* has been shown to be a health hazard by transmitting the rat lungworm, *Angiostrongylus cantonensis*, which causes eosinophicillic meningoencepahlitis in humans. *Lissachatina fulica* has also been implicated in transmitting the following plant pathogens: *Phytophthora palmivora* on commercial pepper, coconut, betel pepper, papaya, and vanda orchid; *Phytophthora colocasiae* on taro; and *Phytophthora parasitica* on eggplant and tangerine (USDA, 1982).

Lissachatina fulica believed to be originally from East Africa, has become established throughout the Indo-Pacific Basin, including the Hawaiian Islands. This mollusk has also been introduced to the Caribbean islands of Martinique and Guadeloupe. Recently, the snails were detected on Saint Lucia and Barbados. Although many introductions are accidental via cargo or ships, some introductions were purposeful. The market for this snail species as food is expanding. In Africa and Asia, the medicinal properties of these snails are also being investigated. The U.S. Department of Agriculture has recently discovered and confiscated illegal giant African land snails from

commercial pet stores, schools and one private breeder. These snails were being used for science lessons in schools by teachers who were unaware of the risks associated with the snails and the illegality of possessing them. In 1966, a Miami boy smuggled three giant African land snails into the country. His grandmother eventually released them into a garden, and in seven years, there were more than 18,000 of them. The Florida state eradication effort took 10 years at a cost of $1 million.

Symptoms/Signs

Information specific to grape is not available. *Lissachatina fulica* is easily seen due to its large size, and attacked plants exhibit extensive rasping and defoliation. The weight of the number of snails on a plant can break the stems of some host species. *Lissachatina fulica* can also be detected by signs of ribbon-like excrement, and slime trails on plants and buildings.

In garden plants and ornamentals of a number of varieties, and vegetables, all stages of development are eaten. Cuttings and seedlings are the preferred food items. Young snails up to about 4 months feed almost exclusively on young shoots and succulent leaves. The bark of relatively large trees such as citrus, papaya, rubber and cocoa is also subject to attack. In these plants, damage is caused by complete consumption or removal of bark. The papaya appears to be the only fruit that is seriously damaged by *L. fulica*, largely as a result of its preference for fallen and decaying fruit (CABI, 2007).

Survey

CAPS-Approved Method*: Visual survey is the method to survey for *L. fulica.*

*For the most up-to-date methods for survey and identification, see Approved Methods on the CAPS Resource and Collaboration Site, at http://caps.ceris.purdue.edu/.

Literature-Based Methods:

Visual survey: The most effective method of survey for mollusks is through visual searching methods. Baited traps have been used in the past but are not effective for tropical species, such as the achatinids. *Lissachatina fulica* is a large and conspicuous crop pest that hides during the day. Surveys are best carried out at night using a flashlight, or in the morning or evenings following a rain event.

Surveys should occur in areas that are at greatest risk for establishment of *L. fulica.* A recent risk analysis by USDA-APHIS-PPQ-CPHST shows that portions of Alabama, Arizona, Arkansas, California, Florida, Georgia, Louisiana, Mississippi, New Mexico, North Carolina, Oklahoma, South Carolina, Texas, and Virginia are at low to moderate risk from *L. fulica*. Risk of *L. fulica* establishment is either low or unlikely in other parts of the continental United States based on climate and host availability.

Detailed survey information is available in the New Pest Response Guidelines available at http://www.aphis.usda.gov/import_export/plants/manuals/emergency/downloads/nprg_gas.pdf.

When using visual inspection methods for detection surveys an amount of bias and variation in sampling intensity is possible. In an effort to standardize sampling efforts, and thus approach a higher level of confidence in results, the following factors should be considered:

Seasonality: Conduct detection surveys on an ongoing basis, with repeated visits at the beginning, during, and/or just after the rainy season. Keep in mind that *Lissachatina fulica* remains active at a range of 9 to 29°C (48 to 84 °F). *Lissachatina fulica* begins hibernating at 2°C (35°F), and begins aestivation at 30°C (86 °F).

Time of Sampling: Plan surveys for early morning and overcast days. Achatinids are active on warm nights, early mornings, and overcast and rainy days. To maintain a consistent sampling time, conduct surveys in the early morning. On overcast days, conduct additional surveys throughout the day.
Micro Habitats: During the day, find snails in the following moist micro habitats: near heavily vegetated areas; under or near rocks and boulders; under discarded wooden boards and planks, fallen trees, logs, and branches; in damp leaf litter, compost piles, and rubbish heaps; under flower pots and planters; on rock walls, cement pilings, broken concrete, or grave markers; in gardens and fields where plants have been damaged by feeding snails and slugs; and at the base of the plants, under leaves, or in the “heart” of compact plants, such as lettuce or cabbage.

Evidence: While conducting a survey, look for the following clues that suggest the presence of snails: chewing damage to plants, eggs, juveniles and adults, empty snail shells, mucus and slime trails, large, ribbon-like feces, and an increase in rat population densities in an area.

Trapping: Use traps to supplement a visual inspection, if time and resources allow. Use commercial brands of slug bait to attract snails; however, due to the slow-acting effects of the molluscicide, these baits alone are not effective in trapping snails.

Note: Serious diseases are associated with the consumption and improper handling of certain mollusks (snails and slugs). Of particular concern, many mollusk species serve as intermediate hosts of nematodes and trematodes. While most cases of human infections result from consumption of raw or partially cooked snail meat, government inspectors, officers and field surveyors are at-risk due to the handling of live snail, samples, and potential exposure to mucus secretions. ***Wear neoprene gloves when handling mollusks and wash hands thoroughly after any mollusk survey or inspection activities.***

Key Diagnostics/Identification

CAPS-Approved Method*: Confirmation of *L. fulica* is by morphological identification. Identification should be verified by a malacologist at National Identification Services.

*For the most up-to-date methods for survey and identification, see Approved Methods on the CAPS Resource and Collaboration Site, at http://caps.ceris.purdue.edu/.

Literature-Based Methods:

Lissachatina fulica is distinctive in appearance and is readily identified by its large size and relatively long, narrow, conical shell. Reaching a length of up to 20 cm (7.9 in) the shell is more commonly in the range of 5 to 10 cm (2 to 4 in). See identification section in USDA-APHIS (2005).

References

Bequaert, J.C. 1950. Studies in the Achatinidae, a group of African Snails. Bulletin of the Museum of Comparative Zoology at Harvard College 105: 1-216.

Burch, J.B. 1960. Some snails and slugs of quarantine significance to the United States. ARS 82-1. Washington, DC: USDA–PPQ–Agricultural Research Service.

CABI. 2007. Crop Protection Compendium Wallingford, UK: CAB International. www.cabicompendium.org/cpc.

Denmark, H.A., and Poucher C., 1960. Some snails and slugs of quarantine significance to the United States. Plant Quarantine ARS 82-1. Agricultural Research Service, USDA, Washington, D.C.

USDA. 1982. Pests not known to occur in the United States or of limited distribution, No. 22: Giant African Snail. USDA-APHIS-PPQ.

USDA-APHIS. 2005. New Pest Response Guidelines. Giant African Snails: Snail Pests in the Family Achatinidae. USDA–APHIS–PPQ–Emergency and Domestic Programs–Emergency Planning, Riverdale, Maryland.

Nematodes

Primary Pests of Soybean (Full Pest Datasheet)

Rotylenchulus macrodoratus

Scientific Name
Rotylenchulus macrodoratus Dasgupta, Raski, and Sher

Common Name(s)
Reniform nematode

Type of Pest
Nematode

Taxonomic Position
Phylum: Nematoda, **Class:** Secernentea, **Order:** Tylenchida, **Family:** Hoplolaimidae

Reason for Inclusion in Manual
National Threat

Pest Description
Among the species of *Rotylenchulus* of major economic importance, *Rotylenchulus reniformis* and *R. parvus* are worldwide in distribution. *R. macrodoratus* occurs only in the Mediterranean region, particularly in France, Greece, Israel, Italy, and Malta.

Figure 1. Vermiform *R. macrodoratus* female. Photo courtesy of Nikos Volvlas.

This reniform nematode has semi-endoparasitic sedentary habits. Single-cell *R. macrodoratus* eggs measured 111 μm (98 to 119) x 44 μm (40 to 49), about twice as long as eggs of *R. parvus* (56 to 59 μm x 30 to 38 μm). The first stage juvenile appeared after 11 to 14 days, the second stage juvenile after 14 to 17 days, and hatching occurred 16 to 19 days after egg deposition. Second stage juveniles (J2) and following juvenile stages (J3 and J4) develop and attain the adult stage in the soil without feeding (Inserra and Vovlas, 1979).

The infective stages of *R. macrodratus* were the immature females, as reported for *R. parvus* and *R. reniformis.* The vermiform females (Fig. 1) penetrate host roots and become sedentary. Immature females were found in roots 14 to 16 days after inoculation. The anterior portion of their body remains embedded in the roots and the posterior portion protrudes from the root surface and swells. They establish a specialized feeding site (a mononucleate giant cell) in the stele. Swollen semi-endoparastic females (Fig. 2) were observed 25 to 31 days after inoculation, and 4 to 5 days thereafter fully developed females with the first eggs were found. After gonad maturation they deposit eggs in a gelatinous matrix (Fig. 2), which surrounds the female posterior body (Robinson et al., 1997). The complete lifecycle from egg to egg took about 45 to 55 days, somewhat longer than that of *R. parvus* (27 to 36 days) and more than twice that for *R. reniformis* (17 to 23 days) (Inserra and Vovlas, 1979).

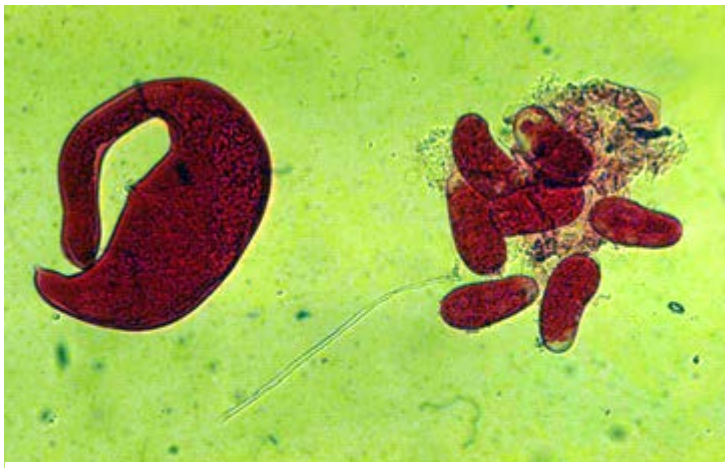


Figure 2. Adult female and eggs of *R. macrodoratus*. Photo courtesy of Nikos Volvlas.

Pest Importance

This nematode is common in the Mediterranean regions, where it parasitizes the root systems of fruit trees, ornamentals, and some herbaceous hosts.

Symptoms

Small swellings in the area of nematode penetration were noted in infested roots of *Dianthus* species. The symptom was not found on other hosts tested (Inserra and Vovlas, 1979). The detrimental effects of this nematode on the growth and yield of its economic hosts are unknown. Further studies are needed on the pathogenicity threshold limits and influence of population densities of this nematode on host-plant growth.

Known Hosts

This reniform nematode parasitizes many fruit crops and ornamental trees such as *Ceratonia siliqua* (carob), *Eurybotria japonica* (loquat), *Ficus carica* (fig), *Laurus nobilis* (laurel), *Nerium oleander* (oleander), *Olea europaea* (olive),*Prunus amygdalus* (along), *Pistacia vera* (pistachio), *Prunus armeniaca* (apricot), *Prunus domestica* (plum),*Vitis vinifera* (grape), *Quercus calliprinos* and *Q. farnetto* (oak).

Herbaceous hosts include: *Dianthus barbatus (*large-flowered sweet William*), Dianthus caryophyllus* (carnation), *Glycine max* (soybean), *Hedera ile* (ivy),

*Parietaria officinalis (*pellitory*),* and *Phlomis fruticosa* (phlomis).

Known Vectors (or associated insects)
Rotylenchulus macrodoratus is not a known vector and does not have any associated organisms.

Known Distribution
Rotylenchulus macrodoratus is a Mediterranean species, which occurs in France, Greece, Israel, Italy, and Malta (Robinson et al., 1997). It has recently been reported in South Africa (Van den Berg, 1998).

Potential Distribution Within the United States
No information available at this time

Survey
CAPS- Approved Method*: Has not been evaluated at this time.

*For the most up-to-date methods for survey and identification, see Approved Methods on the CAPS Resource and Collaboration Site, at http://caps.ceris.purdue.edu/.

Literatuter-Based Methods:
Soil/Host Plant Sampling: Soil and root samples were collected for *R. macrodoratus* and other nematode pests of olive by Tedeschini et al. (2002). To collect samples, the groves were divided into sampling blocks representing differences in soil texture, drainage patterns, or cropping history. A sample of 1 to 2 kg of soil and 10 g of roots was taken for nematode analysis from 5 to 20 subsamples collected. A sample of 100 ml of soil and 10 g of root was mixed and analyzed. Nematodes from soil samples were extracted by Oostenbrink's elutriator and the root samples by centrifugation. Nematodes were killed by heat and fixed in TAF (formalin and triethanolamine). For identification, temporary and fixed mount slides were prepared.

Key Diagnostics/Identification
CAPS- Approved Method*: Has not been evaluated at this time.

*For the most up-to-date methods for survey and identification, see Approved Methods on the CAPS Resource and Collaboration Site, at http://caps.ceris.purdue.edu/.

Literatuter-Based Methods:
The morphological characteristics of the vermiform stages of this reniform nematode are similar to those of *R. reniformis*. *R. macrodoratus* vermiform females, however, have a longer stylet than those of *R. reniformis* (22 to 26 vs. 16 to 21 μm). *R. macrodoratus* swollen females lack the characteristic spike-like mucro, which is present at the body posterior end of *R. reniformis* females.

References

Inserra, R.N. and Vovlas, N. 1980. The biology of *Rotylenchulus macrodoratus*. Journal of Nematology 12: 97-102.

Robinson, A.E., Inserra, R.N., Caswell-Chen, E.P., Vovlas, N., and Troccoli, A. 1997. Rotylenchulus species: identification, distribution, host ranges, and crop plant resistance. Nematropica 27: 127-180.

Tedeschini, J., Isufi, E., Pace, H., Stamo, B., Jovani, V., Shahini, Sh., Uka, R., Hasani, M., Baci, M., Pitts, C., Pfeiffer, D., Ferguson, L., Teviotdale, B., and McGiffen, M. 2002. Monitoring of crop pests and their natural enemies in olive production systems. www.ag.vt.edu/ipmcrsp/annrepts/annrep02/Albania/albania_topic1.pdf.

Van den Berg, E. 1998. New records and notes on known species of Hoplolaimidae (Nemata) in South Africa. Journal of Nematode Morphology and Systematics 1:29-46.

Secondary Pests of Soybean (Truncated Pest Datasheet)

None at this time

Plant Pathogens

Primary Pests of Soybean (Full Pest Datasheet)

Cowpea mild mottle virus (CPMMV)

Scientific Name
Cowpea mild mottle carlavirus

Common Name(s)
Bean angular mosaic virus, eggplant mild mottle virus, fuzzy vein virus, groundnut crinkle virus, groundnut Ngomeni mottle virus, psophocarpus necrotic mosaic virus, tomato pale chlorosis virus, voandzeia mosaic virus

Type of Pest
Plant pathogenic virus

Reason for Inclusion in Manual
National Threat

Pest Description
Cowpea mild mottle virus (CPMMV) has straight or slightly flexuous filamentous particles (Fig. 1), mostly measuring 650 x 12 nm, which sometimes have a loosely coiled external helix of unknown composition. It has physico-chemical properties typical of carlaviruses (Brunt and Kenten, 1973), and the structure of the 3' terminus of its genomic RNA is also similar to that of carlaviruses (Naidu et al., 1998). However, unlike definite aphid-borne carlaviruses, it can be

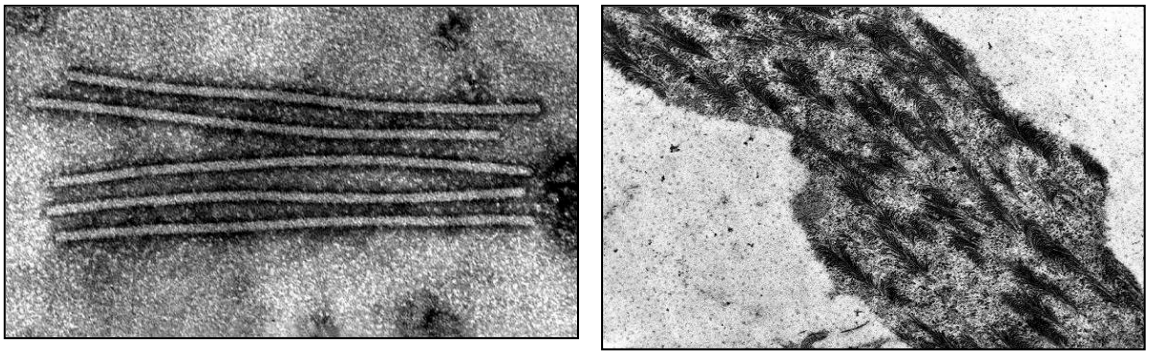


Figure 1. Transmission electron micrograph of CPMMV particles (left) and brush-like inclusions (right). Photo courtesy of Rothamsted Experimental Station (CABI, 2007).

transmitted by whiteflies (e.g. *Bemisia tabaci*) and induces brush-like or falcate inclusions (Fig. 1) within infected plants that are probably composed mostly of aggregated virus particles. It is, therefore, considered to be a tentative species of the carlavirus genus (Brunt, 1995).

Biology and Ecology

Bemisia tabaci, a whitefly, was first reported to be the natural vector of CPMMV by Iwaki et al. (1982). Laboratory transmissibility of the virus by this whitefly species has since been confirmed in Israel, Brazil, India, Nigeria, and Indonesia (CABI, 2007). The virus is not transmitted by a range of aphid vectors including *Aphis craccivora*, *A. fabae*, *A. glycines*, *A. gossypii, A. pisum*, *A. spiraecola*, and *Myzus persicae*, and (Brunt and Kenton, 1973; Iwaki et al., 1982; Thouvenel et al., 1982; Iizuka et al., 1984).

The virus was originally considered to be transmitted in the semi-persistent manner (Iwaki et al., 1982; Anno-Nyako, 1986). However, there is now cogent evidence that it is transmitted in the non-persistent manner (Costa et al., 1983; Muniyappa and Reddy, 1983). The ability to transmit CPMMV is retained for a maximum of 20-60 minutes.

The detailed epidemiology of CPMMV has yet to be investigated. Nevertheless, viruliferous whiteflies undoubtedly effect the transmission of virus from infected to healthy plants. Natural infection of perennial weed species has been reported in Kenya, Nigeria and India, and these are probably primary sources of infection for both tomatoes and leguminous crops. Similarly, when seed transmission occurs in leguminous crops, it may provide primary foci of infection for spread within a crop and transmission to adjacent tomato crops.

Reports of the seed transmissibility of CPMMV are contradictory. The original Ghanaian isolate, obtained from a seed-infected cowpea seedling, was subsequently shown to be seed-transmitted in cowpeas, soybeans, and French beans (*Phaseolus vulgaris*) (Brunt and Kenten, 1973). The virus was later reported to be seedborne to a level of 1 to 3% in cowpeas in India (Nain et al., 1994), to 0.9% in soybeans in Thailand (Iwaki et al., 1982), to 0.05 to 1.66% in 25 soybean cultivars in India, to unreported levels in soybeans in the Ivory Coast (Fauquet and Thouvenel, 1987), to unstated levels in cowpeas in India (Mali et al., 1989), and to 6 to 21% in bambara groundnuts (*Vigna subterranea*) in the Ivory Coast (Fauquet and Thouvenel, 1987).

However, seed transmission of the virus has not been detected by stringent tests in the following cases: French beans and soybeans in Brazil (Costa et al., 1983), peanuts and soybeans in India (Iizuka et al., 1984), cowpeas and soybeans in Nigeria and soybeans and peanuts in Indonesia (Horn et al., 1991). Seed transmissibility of the virus is thus probably dependent on the interaction between virus strain, plant genotype, duration of infection and, possibly, environmental conditions.

Pest Importance

Although the virus has a wide geographical distribution in Africa, Asia, Oceania and South America, its effect on the growth and yield of infected plants has been rarely studied.

CPMMV was reported to be of minor importance in cowpea crops in Papua New Guinea (Philemon, 1987), in mung beans and French beans (*Phaseolus vulgaris*) in Tanzania (Mink and Keswani, 1987), and in French beans and soybeans in Brazil (Costa et al., 1983). By contrast, the virus can cause yield losses of 64 to 80% in peanuts in Kenya (Bock et al., 1976, 1977). It also causes conspicuous leaf chlorosis and stunting, but unstated yield losses, of infected peanuts, soybeans, bambara groundnuts (*Vigna subterranea*) and winged beans (*Psophocarpus tetragonolobus*) elsewhere, including the Ivory Coast, India and Indonesia (Fauquet et al., 1979; Fortuner et al., 1979; Dubern and Dollet, 1981; Thouvenel et al., 1982; Fauquet and Thouvenel, 1987; Saleh et al., 1989; Reddy, 1991).

Symptoms/Signs

The virus causes conspicuous leaf chlorosis (Fig. 2) and stunting. Viral infection induces brush-like or falcate inclusions within infected plants (Fig. 1) that are probably composed mostly of aggregated virus particles. On cowpea, CPMMV cause diffuse chlorotic blotches on the primary leaves, systemic mottling, and leaf distortion.

Figure 2. Mosaic symptoms of CPMMV on soybean. Photo courtesy of Mitsuro Kameya-Iwaki (CABI, 2007).

In soybean, symptoms vary depending on the cultivar. Symptoms include slight vein clearing, leaf malformation, mosaic, mottling, crinkling of leaves, and downward curling or upward cupping of leaves (Thouvenel et al., 1982; Iwaki, 1986).

In peanut, the disease is characterized by stunting of plants, downward rolling, mottling, general chlorosis, necrotic lesions, and reduced size of leaflets (El Hassan et al., 1997). In tomato, the virus induced inconspicuous and transient narrow chlorotic banding of secondary and other minor leaf veins (so-called ‘fuzzy veins’) (Brunt and Phillips,

1981).

Known Hosts

Although most of its natural hosts are leguminous species, CPMMV also occurs naturally in tomatoes in Israel and Nigeria. Isolates of the virus are readily sap-transmissible experimentally to many species of the Fabaceae, and also to some species of the Amaranthaceae, Aizoaceae, Asteraceae, Chenopodiaceae, Cucurbitaceae, Pedaliaceae, Scrophulariaceae, Solanaceae and Sterculiaceae (CABI, 2007).

Major hosts

Arachis hypogaea (peanut), *Glycine max* (soybean), *Phaseolus vulgaris* (common bean), and *Vigna unguiculata* (cowpea)

Minor hosts

Calopogonium mucunoides (calopo), *Lycopersicon esculentum* (tomato), *Mucuna pruriens* (buffalo bean), *Phaseolus lunatus* (lima bean), *Phaseolus radiata* (black gram), *Psophocarpus tetragonolobus* (winged bean), *Solanum melongena* (egg plant), *Vicia faba* (broad bean), *Vigna subterranea* (bambara groundnut).

Wild hosts

Centrosema pubescens (centro), *Desmodium tortuosum* (Florida beggarweed), *Stylosanthes gracile*, and *Tephrosia villosa*

Known Vectors (or associated insects)

Cowpea mild mosaic virus is vectored by *Bemisia tabaci* (a whitefly).

Known Distribution

When it was first described in 1973, this virus was thought to be of only local, and possibly minor, importance in Ghana (Brunt and Kenten, 1973). However, a disease of peanuts, described as 'Ngomeni mottle' (Storey and Ryland, 1957) and since shown to be induced by CPMMV (Bock et al., 1976), occurred in Kenya at least 16 years previously. The virus has possibly also been long present but unrecognized in other countries.

Although no detailed surveys have been made to determine the extent of its geographical distribution, the virus is known to have a wide distribution in the following countries: **Africa:** Burkina Faso, Ivory Coast, Egypt, Ghana, Kenya, Malawi, Mozambique, Nigeria, Sudan, Swaziland, Tanzania, Togo, Uganda, Zambia; **Asia:** India, Indonesia, Israel, Japan, Jordan, Malaysia, Thailand, and Yemen; **Oceania:** Fiji, Papua New Guinea, the Solomon Islands; and **South America** Brazil (CABI, 2007).

Potential Distribution within the United States

Information is not available at this time, however, a range of legume and solanaceous crops are susceptible to this virus.

Survey

CAPS-Approved Method*: Has not been examined at this time.

*For the most up-to-date methods for survey and identification, see Approved Methods on the CAPS Resource and Collaboration Site, at http://caps.ceris.purdue.edu/.

Literature-Based Methods:

Visual Survey: There are no specific survey methodologies established for CPMMV. The disease has been traditionally detected based on the visual examination of plant hosts typical symptoms. Because symptoms are not specific for CPMMV, inoculation of indicator plants, ELISA, electron microscopy and/or PCR are necessary to confirm the presence of CPMMV.

Key Diagnostics/Identification

CAPS-Approved Method*: Has not been examined at this time.

*For the most up-to-date methods for survey and identification, see Approved Methods on the CAPS Resource and Collaboration Site, at http://caps.ceris.purdue.edu/.

Literature-Based Methods:

Indicator Plants: The virus is readily transmitted by mechanical inoculation of sap from infected crop plants to diagnostic herbaceous host species, the reactions of which are as follows:

Arachis hypogaea (peanut) - a few local necrotic lesions, rings or line patterns, and chlorosis, rolling and veinal necrosis of systemically infected leaves. Plants are severely stunted.

Beta vulgaris - a few fawn necrotic local lesions, but no systemic infection.

Cajanus cajan (pigeon pea) - severe chlorosis and distortion of systemically infected leaves and stunting of plants.

Chenopodium quinoa (quinoa) and/or *C. amaranticolor* (lambsquarters)- numerous local chlorotic or necrotic lesions, but no systemic infection.

Glycine max (soybean) - conspicuous chlorosis of systemically infected leaves and, sometimes, apical chlorosis.

Phaseolus vulgaris (bean) - chlorotic spotting of systemically infected leaves.

ELISA: The virus is best identified by serological methods of which enzyme-linked immunosorbent assay (ELISA) is the most useful (Antignus and Cohen, 1987; Mink and Keswani, 1987; Mali et al., 1989;); this method is also effective when using mixed antisera when screening for several viruses (Hampton et al., 1992).

Using ELISA detection methods, CPMMV, however, could not be detected in seeds from 60 cowpea pre-introductions from Botswana, India and Kenya (Gillaspie et al., 1995), in 4144 seeds harvested from seven CPMMV-infected soybean genotypes or in 214 seeds collected from CPMMV-infected peanut plants (cv. Gajah) (Horn et al., 1991).

Immunosorbent electron microscopy is also a very useful diagnostic procedure (Brunt et al., 1983; Gaspar et al., 1985). More recently, a polymerase chain reaction (PCR) procedure has been developed for the rapid and sensitive detection of CPMMV and other carlaviruses (Badge et al., 1996).

References

Anno-Nyako, F.O. 1987. Host range and symptomatology of an 'extra mild' isolate of cowpea mild mottle virus in Nigeria. Tropical Agriculture, UK, 64:206-208.

Antignus, Y. and Cohen, S. 1987. Purification and some properties of a new strain of cowpea mild mottle virus in Israel. Annals of Applied Biology, 110:563-569.

Badge, J., Brunt, A., Carson, R., Dagless, E., Karamagioli, M., Phillips, S., Seal, S, Turner, R., and Foster, G.D. 1996. A carlavirus-specific PCR primer and partial nucleotide sequence provides further evidence for the recognition of cowpea mild mottle virus as a whitefly-transmitted carlavirus. European Journal of Plant Pathology, 102:305-310.

Bock, K.R., Guthrie, E.J., Meredith, G., and Njuguna, J.G.M. 1977. Plant pathology. Report of the East African Agriculture and Forestry Research Organisation for 1975. Nairobi, Kenya: East African Agriculture and Forestry Research Organisation, 117-124.

Bock, K.R., Guthrie, E.J., Meredith, G., and Njuguna, J.G.M. 1979. Viruses of food legumes. Annual Report East African Agriculture and Forestry Research Organization, Record of Research, for 1976. Nairobi. Kenya, 138-139.

Brunt, A.A. 1995. Genus Carlavirus. In: Murphy F, Fauquet CM, Bishop DHL, Ghabrial SA, Jarvis AW, Martelli GP, Mayo MA, Summers MD, eds. Virus Taxonomy: Classification and Nomenclature of Viruses. Sixth Report of the International Committee on Taxonomy of Viruses. Archives of Virology, Supplement 10. Vienna: Springer-Verlag, 475-478.

Brunt, A.A. and Kenten, R.H. 1973. Cowpea mild mottle, a newly recognized virus infecting cowpea (*Vigna unguiculata*) in Ghana. Annals of Applied Biology, 74:67-74.

Brunt, A.A., and Phillips, S. 1981. 'Fuzzy vein', a disease of tomato (*Lycopersicon esculentum*) in Western Nigeria induced by cowpea mild mottle virus. Trop. Agric. (Guildford) 58(2): 177-180.

CABI. 2007. Crop Protection Compendium Wallingford, UK: CAB International. www.cabicompendium.org/cpc.

Costa, A.S., Gaspar, J.O., and Vega, J. 1983. Angular mosaic of the *Phaseolus vulgaris* variety Jalo, caused by a carlavirus transmitted by the white-fly *Bemisia tabaci.* Fitopatologia Brasileira, 8:325-337.

Dubern, J. and Dollet, M. 1981. Groundnut crinkle virus, a new member of the carlavirus group. Phytopathologische Zeitschrift, 101:337-347.

El-Hassan, S.M., Naidu, R.A., Ahmed, A.H., and Murant, A.F. 1997. A serious disease of groundnut caused by cowpea mild mottle virus in Sudan. Phytopath. Z. 145(7): 301-304.

Fauquet, C., Lamy ,D., and Thouvenel, J.C. 1979. Viral diseases of winged bean in the Ivory Coast. FAO Plant Protection Bulletin, 27:81-87.

Fauquet, C. and Thouvenel, J.C. 1987. Plant viruses in the Ivory Coast. Initiations, Documentations, Techniques, No. 46. Paris, France:ORSTOM, 243.

Fortuner, R., Fauquet, C., and Lourd, M. 1979. Diseases of the winged bean in Ivory Coast. Plant Disease Reporter, 63:194-199.

Gaspar, J.O., Beriam, L.O.S., Alves, M.N., Oliveira, A.R., and Costa, A.S. 1985. Serological identity of bean angular mosaic and cowpea mild mottle viruses. Fitopatologia Brasileira, 10:195-199.

Gillaspie, A.G. Jr., Hopkins, M.S., Pinnow, D.L., Hampton, R.O. 1995. Seedborne viruses in preintroduction cowpea seed lots and establishment of virus-free accessions. Plant Disease, 79:388-391.

Hampton, R.O., Albrechtsen, S.E., and Mathur, S.B. 1992. Seed health (viruses) of *Vigna unguiculata* selections from developing countries. Seed Science and Technology, 20:23-38.

Horn, N.M., Saleh, N., and Baliadi, Y. 1991. Cowpea mild mottle virus could not be detected by ELISA in soybean and groundnut seeds in Indonesia. Netherlands Journal of Plant Pathology, 97:125-127.

Iizuka, N., Rajeshwari, R., Reddy, D.V.R., Goto, T., Muniyappa, V., Bharathan, N., and Ghanekar, A.M. 1984. Natural occurrence of a strain of cowpea mild mottle virus on groundnut (*Arachis hypogaea*) in India. Phytopathologische Zeitschrift, 109:245-253.

Iwaki, M. 1986. Soybean crinkle leaf and cowpea mild mottle viruses. International Symposium on Virus Diseases of Rice and Leguminous Crops. Tropical Agriculture Research Series, 19:92-100.

Mali, V.R. and Nirmal, D.D. 1987. Properties of a cowpea mild mottle virus isolate from groundnut. Indian Phytopathology, 40:22-26.

Mali, V.R., Mundhe, G.E., and Shaikh, W.R. 1989. Sero-diagnosis of six cowpea seedborne viruses in India. Indian Journal of Virology, 5:45-55.

Mink, G.I. and Keswani, C.L. 1987. First report of cowpea mild mottle virus on bean and mung bean in Tanzania. Plant Disease, 71:557.

Muniyappa, V. and Reddy, D.V.R. 1983. Transmission of cowpea mild mottle virus by *Bemisia tabaci* in a non-persistent manner. Plant Disease, 67:391-393.

Naidu, R.A., Gowda, S., Satyanarayana, T., Boyko, V., Reddy, A.S., Dawson, W.O., and Reddy, D.V.R. 1998. Archives of Virology 143: 769-780.

Nain, P.S., Rishi, N., and Bishnoi, S.S. 1994. Profile of viral diseases of cowpea (*Vigna unguiculata*) in northern India. Indian Journal of Virology, 10:128-136.

Philemon, E.C. 1987, publ. 1990. Plant pathology note no 32. A virus disease of peanut. Harvest, 12:15-16. **Reddy, D.V.R.** 1991. Crop profile. Groundnut viruses and virus diseases: distribution, identification and control. Review of Plant Pathology, 70:665-678.

Saleh, N., Baliadi, Y., and Horn, N.M. 1989. Cowpea mild mottle virus naturally infecting groundnut in Indonesia. Penelitian Palawija, 4:32-35.

Storey, H.H. and Ryland, A.K. 1957. Viruses causing rosette and other diseases in groundnut. Annals of Applied Biology, 318-326.

Thouvenel, J.C., Monsarrat, A., and Fauquet, C. 1982. Isolation of cowpea mild mottle virus from diseased soybeans in the Ivory Coast. Plant Disease, 66:336-337.

Indonesian Soybean dwarf virus (ISDV)

Scientific Name
Indonesian Soybean Dwarf Luteovirus

Type of Pest
Plant pathogenic virus

Reason for Inclusion in Manual
National Threat

Pest Description
Little information is currently available on Indonesian soybean dwarf virus (ISDV). It was first reported in *Glycine max* from Bogor, Indonesia by Iwaki *et al.* (1980).

ISDV virions are isometric single stranded, positive sense RNA without an envelope. Virions are 26 nm in diameter, rounded in profile, without a conspicuous capsomere arrangement. Virions are found in the phloem of susceptible plants. In ultrathin sections, crystalline aggregates of spherical particles were observed in vacuoles of phloem cells of infected plants (Iwaki et al., 1980).

ISDV is transmitted by an aphid vector, *Aphis glycines* (soybean aphid) in a persistent manner. *A. glycines* is the only known vector for ISDV. The virus is retained when the vector molts, but the virus does not multiply in the vector. The virus is not transmitted by mechanical inoculation or contact between plants. It is not transmitted by seed or by pollen (Brunt et al., 1996).

Pest Importance
Although the virus is widely distributed in Indonesia, studies detailing crop loss and yield have not been completed.

Symptoms
On soybean, this virus causes stunting (dwarfing) with shortened petioles and internodes, mottling, and leaf malformation. Leaves are often rolled.

Known Hosts
Major hosts
Glycine max (soybean)

The only known host is soybean.

Known Vectors (or associated insects)
Indonesian soybean dwarf virus is vectored by *Aphis glycines* (an aphid).

Known Distribution
The virus is known to be present in Indonesia and Thailand (CABI, 2007).

Potential Distribution within the United States
Information is not available at this time. Surveys should be focused in areas that grow soybean and have the vector *Aphis glycines* present.

Survey
CAPS-Approved Method*: Has not been examined at this time.

*For the most up-to-date methods for survey and identification, see Approved Methods on the CAPS Resource and Collaboration Site, at http://caps.ceris.purdue.edu/.

Literature-Based Methods:
Visual Survey: Specific information is not available at this time, but like other viral diseases will involve visual survey of soybean fields for symptoms of ISDV.

Key Diagnostics/Identification
CAPS-Approved Method*: Has not been examined at this time.

*For the most up-to-date methods for survey and identification, see Approved Methods on the CAPS Resource and Collaboration Site, at http://caps.ceris.purdue.edu/.

Literature-Based Methods: Specific information is not available at this time.

References
Brunt, A.A., Crabtree, K., Dallwitz, M.J., Gibbs, A.J., Watson, L. and Zurcher, E.J. (eds.) (1996 onwards). `Plant Viruses Online: Descriptions and Lists from the VIDE Database. Version: 20th August 1996.' URL http://biology.anu.edu.au/Groups/MES/vide/.

CABI. 2007. Crop Protection Compendium Wallingford, UK: CAB International. www.cabicompendium.org/cpc.

Iwaki, M., Roechan, M., Hibino, H., Tochihara, H. and Tantera, D.M. 1980. A persistent aphidborne virus of **s**oybean, Indonesian soybean dwarf virus. Plant Disease 64: 1027.

Requested by CAPS Community (Full Pest Datasheet)

Phakopsora pachyrhizi

Scientific Name
Phakopsora pachyrhizi Syd. & P. Syd.

Synonyms:
Phakopsora calothea, P. erythrinae, P. sojae, P. vignae, Physopella pacyrizi, Uromyces sojae, Malupa sojae, Uredo erythrinae, Uredo sojae

Common Name(s)
Asian soybean rust, Asiatic soybean rust, soybean rust

Type of Pest
Fungus

Taxonomic Position
Phylum: Basidiomycota, **Class:** Urediniomycetes, **Order:** Uredinales, **Family:** Melampsoraceae

Reason for Inclusion in Manual
Requested by the CAPS community

Pest Description
Currently, there are two rust fungi that cause soybean rust, *Phakopsora pachyrhizi and P. meibomiae*. *P. pachyrhizi*, Asiatic soybean rust, has emerged as a major constraint of soybean production in both the eastern hemisphere (Australia, China, India, Taiwan, and Thailand) and in the western hemisphere (Brazil, Columbia, Costa Rica, Puerto Rico, and Hawaii). Another species of rust, *P. meibomiae* has been endemic in portions of South America for many years, but is considered less of a threat because it is not as aggressive as the Asiatic soybean rust. Both rust species have the same type of lesions and urediniospore morphology and thus cannot be distinguished except by using molecular tools. Asiatic soybean rust currently has a localized distribution within the United States It was detected for the first time in North American in Louisiana in November 2004 and, soon after, in other southeastern states of the United States. This pest description will focus on *P. pachyrhizi*.

A rust fungus may produce as many as five different spore stages in its life cycle (Table 1). Production of all five stages by *Phakopsora pachyrhizi*, the soybean rust pathogen, is uncertain (Green, 1984). *P. pachyrhizi* is described from the uredinial and telial stages. Like all rust fungi, *P. pachyrhizi* is an obligate parasite that requires living host cells.

Table 1. The five possible spore stages of a rust fungus.

STAGE	DESCRIPTION
0	Spermagonia bearing spermatia (n) and receptive hyphae (n)
I	Aecia bearing aeciospores (n+n)
II	Uredinia (uredia) bearing urediniospores (uredospores) (n+n)
III	Telia bearing teliospores (n+n → 2n)
IV	Basidia bearing basidiospores (n)

Spermatia (stage 0) and aecia (stage I) are not known to exist (Green, 1984).
Uredinia (stage II) are amphigenous (growing all around), most hypophyllous (on the under surfaces of leaves), minute, scattered or in groups on discolored lesions. Subepidermal in origin, the uredinia are surrounded by paraphyses arising from peridioid pseudoparenchyma; in addition, the uredinia have hymenial paraphyses. Openings are through the central apertures (ostioles). In appearance, the uredinia are pulverulent (appearing as if powdered); in color, uredinia are yellowish-brown to pale cinnamon-brown (Ono et al., 1992). Paraphyses (Fig. 2) are cylindric to clavate, 25 to 50 μm x 6 to 14 μm, slightly to conspicuously thickened apically (~18 μm). The color of the paraphyses ranges from pale yellowish-brown to colorless (Ono et al., 1992).

Figure 1. Soybean rust lesion with circular ostiole and urediniospores. Photo courtesy of USDA-ARS

Figure 2. *P. pachyrhizi* urediniospores and paraphyses, which appear identical to those of *P. meibomiae.* Photo courtesy of Mary Palm, USDA-APHIS-PPQ.

Urediniospores are sessile, obovoid to broadly ellipsoid, 18 to 34 μm to 15 to 24 μm, and minutely and densely echinulate (spiny) (Figs. 2, 3). The walls are uniformly about 1 μm thick. The color of the urediniospores ranges from pale yellowish-brown to colorless. In number, germ pores are mostly 4 to 8 (mostly 6, rarely 2 or 10). In position, germ pores are equatorial or scattered on the equatorial zone; on occasion, germ pores are scattered on or above the equatorial zone (One et al., 1992).

Telia (stage III) are hypophyllus, often intermixed with uredinia, pulvinate and crustose, chestnut-brown to chocolate-brown, subepidermal in origin, and 2- to 7-spore layered. The teliospores are one-celled, irregularly arranged, angularly subglobose, oblong to ellipsoid, and (10-)15 to 26 um x 6 to 12 um. The wall is uniformly about 1 um thick, sometimes slightly thickened (up to 3 um) apically in the uppermost spores, colorless to pale yellowish-brown (Ono et al., 1992).

In 1984, Green noted that 'no germination of teliospores had been reported'. However, in 1991, Saksirirat and Hoppe reported germination of teliospores.

Biology and Ecology

Unlike many pathogens that must find stomata, wounds or some opening before they are able to penetrate the host, soybean rust urediniospores are able to penetrate directly through the leaf cuticle and epidermis, making infection easier and quicker. The incubation period for the fungus is about 7 days; while the latent period is about 9 to 10 days (Melching et al., 1979). In a histological study, Marchetti et al. (1975) found hyphae in soybean mesophyll 20 hours after inoculation with urediniospores of *P. pachyrhizi* and frequently observed direct penetration from appresoria formed at the end of short germ tubes, usually less than 20 um long.

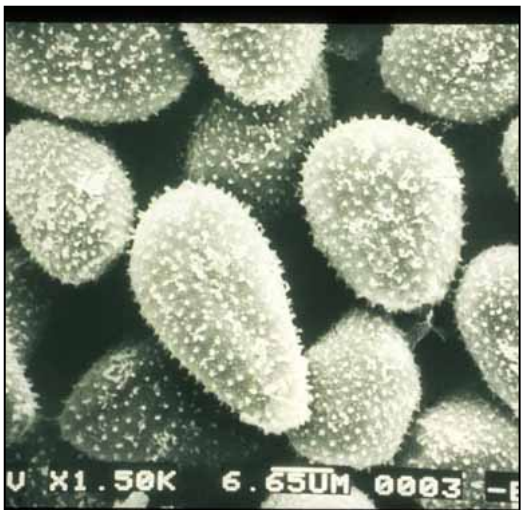


Figure 2. Scanning electron micrograph of soybean rust urediniospores showing spiny appearance. . Photo from the collection of Glen Hartman, USDA-ARS.

Phakopsora pachyrhizi is believed to have a heteroecious life cycle. However, pycnial and aecial stages have not been found. In warmer regions, volunteer crops, supplementary legume crops, and wild species may harbor the fungus throughout the year or during seasons in which soybeans are not cultivated, and may serve as a primary infection source. In colder regions where above-ground parts of annual hosts senesce during winter, no source of new infections in the soybean-growing season has been identified.

Urediniospores of *P. pachyrhizi* germinated between 10 and 28.5 °C, with a broad optimum in the range of 15 to 25 °C. At optimum temperatures, urediniospores germinate in 1 to 1.5 hours. Maximal infection of 'Wayne' soybean occurred at 20 to 25 °C with 10 to 12 hours of dew and at 15 to 17.5 °C with 16 to 18 hours of dew. The minimal dew period for infection was 6 hours at 20 to 25 °C and 8 to 10 hours at 15 to 17.5 °C. Infection did not occur above 27.5 °C (Marchetti et al., 1976). The temperature-moisture requirements for the

infection of soybeans by urediniospores of *P. pachyrhizi* would not preclude the establishment of the soybean rust fungus in all the major soybean growing areas of the United States (Marchetti et al., 1976).

Germinability and infectivity of urediniospores are reduced by exposure of the spores to dry and high temperature conditions prior to germination. Singh and Thapliyal (1977) reported that prior exposure of urediniospores to 35 °C for 6 hours prevented germination of an Indian isolate. Similarly, Kochman (1979) reported that germination of urediniospores on water agar at 21°C was significantly reduced by prior exposure of the spores to 28.5 to 42.5°C for 8 hours. According to Melching et al. (1989), urediniospores on unwetted soybean leaves progressively lost infectivity during sunny conditions, but exhibited enhanced infectivity after 1 or 2 days on dry foliage under cloudy conditions. After 8 days on dry foliage, no urediniospores were found to cause lesions following a 12 hour dew period at 18°C. Spores on leaves exposed to 4 or 6 hours of dew followed by drying for up to 4 days were able to infect when a 12 hour dew period was provided; however, they were less infectious than spores that had not been exposed to a brief initial wetting.

The formation of teliospores seems to be induced when infected plants are subjected to a temperature range below 20°C for at least 15 days. Yeh et al. (1981) reported that, on 20 soybean cultivars and nine other legume plants, teliospores were successfully induced when the inoculated plants were subjected to 12 hour photoperiods, 60 to 100% relative humidity (RH) and temperatures of 15 to 24°C. In the field, teliospores were produced only when the average daily temperature was below 20°C and the maximum temperature above 29°C. The authors further reported that telia and teliospores were formed on eight legume species when the infected hosts were inoculated and grown under a 12 hour photoperiod, at 60 to 100% RH, at a maximum day temperature of 24 ± 1°C and a minimum night temperature of 15 ± 1°C.

Dufresne et al. (1987) reported telial production in Taiwanese and Puerto Rican isolates. The two isolates were cultured on ‘Williams’ soybeans at two temperatures and three light intensities. The Taiwanese isolate produced telia after 21 and 30 days and the Puerto Rican isolate produced telia after 34 and 35 days at 10 and 15°C, respectively. At low light intensity (3.9 µE/m²/sec), the Taiwanese and Puerto Rican isolates produced telia after 29 and 33 days, respectively; at intermediate light intensity (5.3 µE/m²/sec) after 26 and 36 days, respectively; and at high light intensity (6.1 µE/m²/sec) after 22 and 34 days, respectively. The Taiwanese isolate produced larger lesions with a higher percentage of telia than the Puerto Rican isolate.

Saksirirat and Hoppe (1991) reported germination of teliospores. After treatment with 10 to 12 cycles of 24 hour wetting and 24 hour drying periods at room temperature, 65 to 70% of teliospores germinated at 20°C under artificial illumination of 5000 lux at 12 hour light/dark intervals. Only 25% of teliospores

germinated when the telia were treated with seven wetting and drying cycles. Higher germination rates were observed when telia were stored at 5°C for 5 to 6 months.

Figure 4. Soybean infected with soybean rust in Parana State BS near Londrina, Brazil; From left to right unsprayed, sprayed once with a fungicide and sprayed twice with a fungicide. Photo courtesy of Steve Koenning, North Carolina State University.

Pest Importance

Asian soybean rust is a serious disease of soybeans. Until recently this disease did not occur on soybean in the Western hemisphere, but it spread to South America in 2001 and was found for the first time in North America in November 2004. Soybean rust can be a devastating disease with yield losses up to 70 to 80% reported in some fields in Taiwan (Bonde et al., 1976). Plants that are heavily infested have fewer pods and smaller seeds that are of poor quality. In countries in which soybean rust is an established problem, losses range from 10 to 80 percent. The severity of losses varies depending on susceptibility of the soybean variety, time of the growing season in which the rust becomes established in the field, and weather conditions during the growing season.

Soybean rust spores can be carried long distances by wind currents. In 1998, spores were blown 1,350 miles down Africa from Uganda to Zimbabwe. Between 2001 and 2003, the disease spread more than 1,500 miles, from Paraguay to near the equator, infecting as much as 90% of Brazil's soybean acres on the way (APHIS, 2004). Although the exact source of the infection in the continental United States is unknown, a probable explanation is the spread of the disease from South American to the United States during the active hurricane season.

Unlike other rusts, *P. pachyrhizi* and *P. meibomiae* infect an unusually broad range of plant species, which increases the importance of the pest. *P. pachyrhizi* naturally infects 31 species in 17 genera of legumes, and 60 species in 26 other genera have been infected under controlled conditions. Twenty-four plant species in 19 genera are hosts for both species.

It has been estimated that yield losses from *P. pachyrhizi* could exceed 10% in most of the United States and up to 50% in the Mississippi Delta and southeastern United States. Currently, there is no resistance to soybean rust in any of the U.S. commercial soybean cultivars. Some fungicides are effective against *P. pachyrhizi* by slowing the spread of the pathogen enough so that normal seed set and pod fill can occur (Fig. 4). Widespread fungicide applications on soybeans in the United States, however, are not deemed cost effective. As a result this control option would be useful only for eradication on small acreages (Koenning et al., 2004).

Symptoms/Signs

The first symptom of soybean rust is chlorosis (Fig. 5A). Early symptoms of rust infection are found on leaves deep in the canopy, and look like tiny black specks scattered with mottled yellow areas (Fig. 5B). These yellow areas appear translucent if the affected leaves are held up to the sun. Asiatic soybean rust forms two types of lesions on leaves, tan and reddish brown. Lesions will contain one to three rust pustules, which are raised on the leaf surface. The lesions may have an angular appearance and be limited by leaf veins. Rust pustules may appear on cotyledons, leaves, petioles, stems, or pods, but are most likely to be observed as raised pustules on the under side of the leaf (abaxial) (Fig. 6). Soybean rust pustules are small (about the size of a pin head) and contain hundreds of spores. Spores are elliptical to obovoid in shape, colorless to yellowish or yellowish brown and minutely and densely spiny. Infected plants will senesce early and have smaller seeds with reduced yield (Koenning et al., 2004). For the rust to cause economic damage, first infections will probably have to occur before the R3 stage of soybean development.

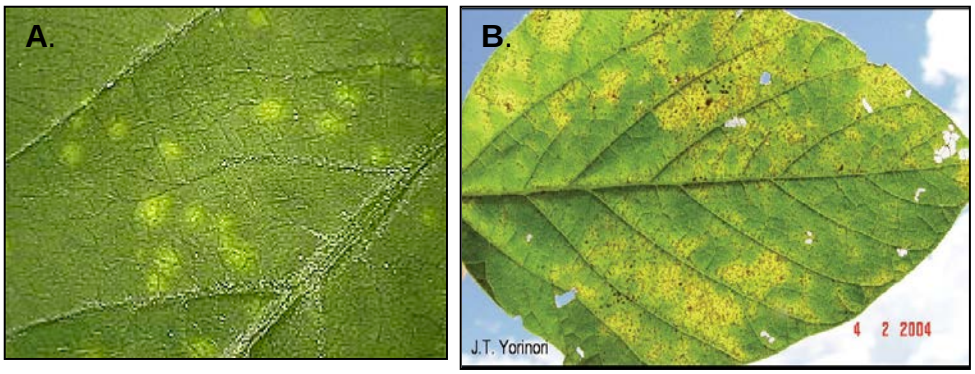


Figure 5. Early soybean rust infection symptoms on susceptible soybean. A) Chlorosis and B) black specks surrounded by mottled yellow areas. Photo courtesy of Glen Hartman and J.T. Yorinon.

On susceptible species/cultivars, infections result in small yellowish-brown or grayish-brown spots or lesions (TAN-type) (Fig. 7), which are delimited by vascular bundles. Several pustules ('pimple-like' structures) of urediniospores are formed on both adaxial and abaxial surfaces of the lesion, but more frequently on the abaxial surface. The lesions coalesce, become dark brown and are covered by buff or pale-brown spore masses as sporulation progresses. The tan lesions when mature, consist of small pustules with masses of tan colored urediniospores on the surface. Later in the season, the lesions become dark reddish-brown and crust-like; these are subepidemal telial clusters. When resistant species/cultivars are infected, minute, reddish-brown spots (RB-type) appear, on which only a few uredinial pustules are formed. Sporulation on RB-type lesions is much less than on TAN-type lesions.

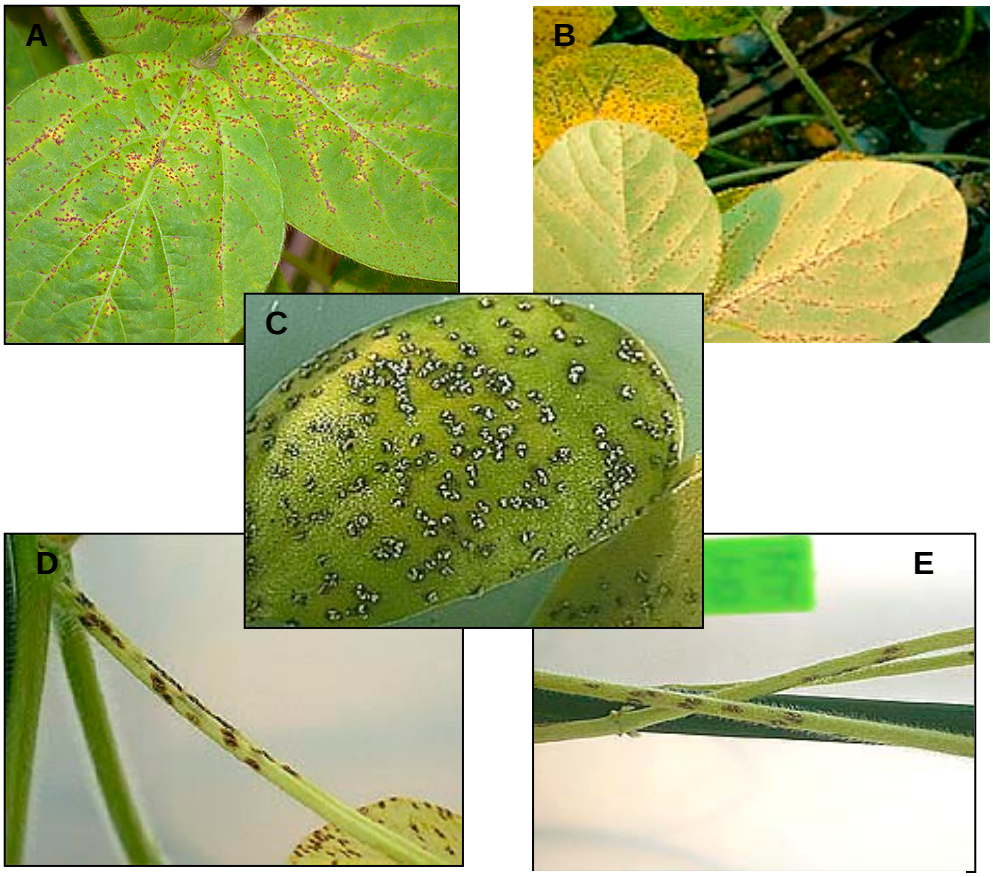


Figure 6. Soybean rust pustules caused by *Phakopsora pachyrhizi* (A) on the upper side of a soybean leaf, (B) on the under side of a soybean leaf, (C) on a soybean cotyledon, (D) on petioles, and (E) stems. Photo courtesy of Glen Hartman.

In soybean, *P. pachyrhizi* causes extensive necrosis of tissues in and around the penetration site. It may take weeks for productive uredia to appear within this necrotic zone. This is not typical of a majority of rust fungi. In soybean rust, the living hyphae must connect these uredia to food and water sources in living cells at distances up to perhaps 1 mm (Melching et al., 1979).

Known Hosts

Because of the confusion over the taxonomy of the pathogens causing soybean rust, *P. meibromiae* and *P. pachyrhizi*, the list of hosts of *P.pachyrhizi* may be incomplete; however, according to various recent references, a large number of legume species are host plants for *P. pachyrhizi. P. pachyrhizi* naturally infects 31 legume species in 17 different genera. *P. pachyrhizi* has been known to infect and sporulate in the field on 35 species in 18 genera of the Subfamily Papilionoideae in the Fabaceae. Among the naturally infected hosts, only *Crotalaria anagyroide*s, *Glycine max*, *Pachyrhizus erosus*, *Phaseolus lunatus*, and *Vigna unguiculata* serve as hosts of another soybean rust fungus, *Phakopsora meibomiae*, which occurs exclusively within the Americas.

Figure 7. Tan-type of soybean rust lesion. Photo courtesy of Glen Hartman.

Major hosts
Cajanus cajan (pigeon pea), *Glycine max* (soybean), *Lupinus* (lupine), *Pachyrhizus erosus* (yam bean), *Phaseolus* spp. (beans), *Pueraria montana* var. *lobata* (kudzu), and *Vigna unguiculata* (cowpea).

Minor hosts
Calpogonium mucunoides, *Erthrina subumbrans* (December tree), *Erythrina variegata* (Indian coral tree), *Kennedia prostrate*, *Kennedia rubicund*a, *Mucuna* (velvetbeans), *Pueraria phaseoloides* (tropical kudzu), *Vicia villosa* (winter vetch), and *Voandzeia subterranea* (bambara groundnut).

Because kudzu is a common weed in the southeastern United States it might serve as a continental source of inoculum.

Wild hosts
Glycine soja (wild soybean).

Known Distribution

Asia: Bangladesh, Cambodia, China, India, Indonesia, Japan, Korea, Laos, Malaysia, Myanmar, Nepal, Philippines, Russian Federation, Singapore, Sri Lanka, Thailand, Vietnam. **Africa:** Congo Democratic Republic, Ethiopia, Ghana, Mozambique, Nigeria, Sao Tome and Principe, Sierra Leone, South Africa, Sudan, Tanzania, Uganda, Zambia, Zimbabwe. **North America:** United States. **Caribbean:** United States Virgin Islands, Puerto Rico. **South America:** Argentina, Bolivia, Brazil, Paraguay, Uruguay, Venezuela. **Oceania:** Australia, Cook Islands, Federated states of Micronesia, Guam, New Caledonia, Niue, Papua New Guinea, Tonga, Vanuata.

Asian soybean rust was first observed in Japan in 1902. Until recently the pathogen was distributed throughout Asia and Australia. It was reported from Hawaii in 1994. In the late 1990's Asian soybean rust was found in Africa and in 2001 was reported in South America. As of 2004, Asian soybean rust in the Americas was known from Argentina, Bolivia, Brazil, Paraguay, and Uruguay.

In November 2004, *P. pachyrhizi* was found for the first time in Louisiana and, soon thereafter, in other southeastern U.S. states. Many earlier reports of *Phakopsora pachyrhizi* in the Americas are erroneous. The reports of *P. pachyrhizi* prior to 1992 actually refer to *P. meibomiae*, a similar-looking rust that also occurs on soybeans and numerous other legumes. In a monograph of the genus Phakopsora Ono et al. (1992) discussed the morphological differences between *P. pachyrhizi* and *P. meibomiae*, although it is difficult to separate them based on morphology with certainty. A molecular test for differentiating these species was published by Frederick et al. (2002) and its use is essential for the accurate identification of these two species.

Potential Distribution within the United States

Soybean rust has a localized distribution within the United States. It was detected for the first time in North America in Louisiana in November 2004 (Scheider et al., 2005) and, soon after, in other southeastern states of the United States (Hernandez, 2005). It was found on the alternate host kudzu (*Pueraria montana* var. *lobata*) in Florida in March 2005. It was also observed on Florida beggarweed (*Desmodium tortuosum*) in Georgia in November 2005. It has also been reported at times in Alabama, Arkansas, Georgia, Hawaii, Illinois, Indiana, Iowa, Kansas, Kentucky, Minnesota, Mississippi, Missouri, Nebraska, North Carolina, Oklahoma, South Carolina, South Dakota, Tennessee, Texas, and Virginia.

Predictive models suggest that conditions in Georgia, South Carolina, Virginia, and North Carolina are favorable for development of an epidemic of soybean rust. The soybean rust pathogen is primarily tropical in distribution and would be able to survive over winter in only the most southern portions of United States (southern Florida and Texas).

Survey

CAPS-Approved Method*: Has not been evaluated at this time.

*For the most up-to-date methods for survey and identification, see Approved Methods on the CAPS Resource and Collaboration Site, at http://caps.ceris.purdue.edu/.

Literature-Based Methods:

Visual Survey: The disease is detected by inspecting the abaxial surface of leaves for uredinial pustules that are powdery and buff and pale brown. The disease is diagnosed both macroscopically by the characteristic symptoms and microscopically by abundantly paraphysate uredinia with pale-yellowish brown or almost colorless, echinulate uredinospores (CABI, 2007).

One of the challenges of identifying Asian soybean rust is that the early stage of the disease can look like other leaf diseases of soybean brown spot, bacterial blight, bacterial pustule, frogeye leaf spot, *Cercospora* leaf blight and downy mildew). In general, to check a field for rust: walk through the entire field in a standard scouting pattern (e.g. a 'W'-shaped pattern), periodically stop and examine the soybean plants, look low and deep into the canopy of the plants, and closely examine the plants for mottled yellow leaves with 'tell-tale' pustules (pimple-like structures) on the underside. Areas in the field with distinct yellowing or browning of the leaves, or areas of dense canopy development, should be targeted in addition to the areas covered by the standard scouting pattern.

Key Diagnostics/Identification

CAPS-Approved Method*: Has not been evaluated at this time.

*For the most up-to-date methods for survey and identification, see Approved Methods on the CAPS Resource and Collaboration Site, at http://caps.ceris.purdue.edu/.

Literature-Based Methods:

P. pachyrhizi is considered an Australasian species of soybean rust and *P. meiborniae* is a new world species. *P. pachyrhizi* is the species currently causing damage in the Southern Hemisphere. Both have the same type of lesions and urediniospore morphology, and thus cannot be distinguished except by using molecular tools. Classical and real-time PCR techniques were developed by Frederick et al. (2002) to detect soybean rust and distinguish the soybean pathogens *Phakospora pachyrhizi* and *P. meiobomiae*.

Easily Confused Species

The early stage of the disease can look like other leaf diseases of soybean, including brown spot, bacterial blight, bacterial pustule, frogeye leaf spot, *Cercospora* leaf blight and downy mildew.

Pustules of Asian soybean rust are clustered alongside the veins and have pores from which masses of urediniospores are released.

Bacterial pustule and bacterial bight: Bacterial pustule caused by *Xanthomonas axonopodis* pv. *glycines* and bacterial blight caused by *Pseudomonas savastanoi* pv. *glcinea* produce spots similar to those formed by the soybean rust fungus on the discolored leaf lesions. However, the bacterial spots are at first water-soaked in appearance and later ooze out slimy bacterial masses instead of powdery spore masses in the rust. Bacterial pustule is also rare in commercial soybean varieties, since most if not all are resistant to this disease. A hand lens may aid in seeing the raised nature of the pustule. Also, placing leaves in a plastic bag with a moist paper towel for twenty four hours may cause the pustules to erupt, thus making identification easier.

Brown spot: *Septoria glycines* can be confused with *Phakopsora pachyrhizi*. Two distinct types of brown spot lesions have been described on soybeans. The most common type is an angular reddish brown lesion surrounded by a chlorotic halo and is associated with plants grown from yellow seeds. The other type is an angular dark brown lesion without the surrounding chlorosis and is associated with plants grown from green seeds. In contrast, lesions of Asian soybean rust are initially yellow flecks or tan to brown or reddish-brown pinpoint spots on the upper leaf surface. As they get older, these lesions develop pale brown pustules on the undersides of leaves from which masses of uredionospores are released.

Cercospora leaf blight: Only the upper leaf surface is discolored and no pustules are found on the underside of the leaf.

Downy mildew: Downy mildew is easily distinguished from Asian soybean rust by the growth of grayish to pale-purplish tufts of sporangiophores and sporangia on the lower leaf surface during humid conditions, whereas, Asian soybean rust forms pustules with pores releasing a powdery brownish-red mass of uredionospores.

Frogeye leaf spot: Lesions of frogeye leaf spot do not have pustules. Lesions of frogeye leaf spot are larger and have distinct purple to reddish-brown margins.

References

Bonde, M.R., Melching. J.S. , and Bromfield, K.R. 1976. Histology of the susceptible pathogen relationship between Glycine max and *Phakopsora pachyrhizi,* the cause of soybean rust. Phytopathology 66: 1290-1294.

CABI. 2007. Crop Protection Compendium Wallingford, UK: CAB International. www.cabicompendium.org/cpc.

Dufresne, L.A., Bean, G.A., Bonde, M.R., and Goth, R.W. 1987. Effects of temperature and light intensity on telia development by Puerto Rico and Taiwan isolates of *Phakopsora pachyrhizi,* the soybean rust fungus. Plant Disease 71: 629-631.

Green, A. 1984. Soybean Rust. Pests Not Known to Occur in the United States of Limited Distribution. No. 56. USDA-APHIS-PPQ.

Frederick, R.D., Snyder, C.L., Peterson, G.L. and Bonde, M.R. 2002. Polymerase chain reaction assays for the detection and discrimination of the soybean rust pathogens *Phakopsora pachyrhizi* and *P. meibomiae*. Phytopathology 92: 217-227.

Kochman, K. 1979. The effect of temperature on development of soybean rust (*Phakopsora pachyrhizi*). Australian Journal of Agricultural Research 30: 273-277.

Koenning, S., Mueller, J., Kemerait, R., Phipps, P., and Stromberg, E. Soybean Rust. Soybean Disease Information Note 8. North Carolina, Georgia, South Carolina, and Virginia Cooperative Extension Service. http://www.ces.ncsu.edu/depts/pp/notes/Soybean/soy008/soy008.htm.

Marichetti, M.A., Uecker, F.A., and Bromfield, K.R. 1975. Uredial development of *Phakopsora pachyrhizi* in soybeans. Phytopathology 65: 822-823.

Marichetti, M.A., Melching, J.S., and Bromfield, K.R. 1976. The effects of temperature and dew period on germination and infection by uredospores of *Phakopsora pachyrhizi*. Phytopathology 66: 461-463.

Melching, J.S., Bromfield, K.R., and Kingsolver, C.H. 1979. Infection, colonization, and uredospore production on Wayne soybean by four cultures of *Phakopsora pachyrhizi*, the cause of soybean rust. Phytopathology 69: 1262-1265.

Melching, J.S., Dowler, W.M., Koogle, D.L., and Royer, M.H. 1989. Effects of duration, frequency, and temperature of leaf wetness periods on soybean rust. Plant Disease 73: 117-122.

Ono, Y., Buritica, P., and Hennen, J.F. 1992. Delimitation of *Phakopsora*, *Physopella*, and *Cerotelium* and their species on Leguminosae. Mycological Research 96: 825-850.

Saksirirat, W. and Hoppe, H.H. 1991. Teliospore germination of soybean rust fungus (*Phakopsora pachyrhizi*). Journal of Phytopathology 132: 339-342.

Schneider, R.W., Hollier, C.A., and Whitam, H.K. 2005. First report of soybean rust caused by *Phakopsora pachyrhizi* in the continental United States. Plant Disease 89: 774.

Singh, K.P. and Thapliyal, P.N. 1977. Some studies on the soybean rust caused by *Phakopsora pachyrhizi*. Indian Journal of Mycology and Plant Pathology 7:27-31.

USDA APHIS. 2004. Soybean rust factsheet. http://www.aphis.usda.gov/lpa/pubs/fsheet_faq_notice/faq_phsoybeanrust.html.

Yeh, C.C., Tschanz, A.T., and Sinclair, J.B. 1981. Induced teliospore formation of *Phakopsora pachyrhizi* on soybeans and other hosts. Phytopathology 71: 1111-1112.

Weeds/Parasitic Plants

Primary Pests of Soybean (Full Pest Datasheet)
None at this time

Secondary Pests of Soybean (Truncated Pest Datasheet)

Alectra vogelii

Scientific Name
Alectra vogelii Benth.

Synonyms:
Alextra angustifolia, Alextra merkeri, Alextra scharensis

Common Name(s)
Yellow witchweed, cowpea witchweed

Type of Pest
Hemiparasitic plant

Taxonomic Position
Class: Magnoliopsida, **Order:** Scrophulariales. **Family:** Scrophulariaceae

Reason for Inclusion in Manual
CAPS Target: AHP Prioritized Pest List - 2009

Pest Description
Flowers: Flowers are five-lobed, sulfur yellow to pale orange (Fig. 1), bell shaped with large horseshoe shaped stigma. Plant height ranges from 30 to 45 cm tall, often as a single stem, but sometimes branch near ground level. Flowers are borne individually on short stems in the axils of the upper leaves. The corolla, formed of five petals which are fused into a tube for the bottom half, is bell-shaped when open, 1.6 to 1 cm in diameter, and somewhat longer than the calyx. Petals are generally pale yellow and may or may not have three deep red veins. Both flower forms can be found in the same stand of *A. vogelii*. Anthers and filaments are glabrous. After flowering, the corolla withers and remains covering the developing globose seed capsule, which eventually swells to approximately 5 mm in diameter.

Leaves: Leaves are 1.5 to 3.5 cm long by 0.3 to 1.5 cm wide and are hairy. Leaf margins vary from five or six sharp teeth to two to five widely spaced teeth, with some plants having entire margins.

The chromosome number (2n) is 38.

Pest Importance

Alectra vogelii is a parasitic weed found in major leguminous crops, including chickpea, cowpea, soybean, and runner bean. In 1929, one report estimated a 20% loss in yield for cowpea crops in Kenya. In 1966, the Agricultural Department for Botswana reported a loss of 24,000 acres in cowpea due to 'yellow witchweed'. In 1977, on-farm trials in Botswana produced no cowpea yields in 6 out of 25 blackyeye crops. In 1979, a blackeye cowpea trial had an average yield of 602 kg/ha and 100 kg/ha for the non-infested and infested fields, respectively. Yield losses of 15% are reported for peanut production in Nigeria, and a 30 to 50% reduction in bambara nut yields in South Africa. A ten year crop rotation study found that long-term rotation with non-crop hosts did not reduce the density of *Alectra* infestations.

Figure 1. Mature *A. vogelii* plant (left) and close up of flowers (right). Photos courtesy of C.R. Riches (CABI, 2007).

Symptoms/Signs

Symptoms associated with *A. vogelii* include: stunted crop plants with smaller leaf area, shorter leaf petioles, and increased shoot/root ratios. Roots are bright orange below soil surface. Stems and leaves are conspicuously hairy. The dust-like seeds have a complex structure. An outer cell layer of the testa is modified into a cone or a 'trumpet-like' structure about 1 mm long, within which the 'kernel' of the seed, measuring about 0.15 mm by 0.25 mm, is suspended. The surface of the seed coat is covered in indentations.

Known Hosts

Major hosts
Vigna unguiculata (cowpea)

Minor hosts
Arachis hypogaea (peanut), *Glycine max* (soybean), *Lablab purpureus* (hyacinth bean), *Mucuna pruriens* (Buffalobean), *Phaseolus acutifolius* (tepary bean), *Phaseolus coccineus* (runner bean), *Phaseolus radiata*, *Phaseolus vulgaris* (common bean), and *Voandzeia subterranea* (bambara groundnut)

Wild hosts
Acanthospermum hispidum (bristly starbur)

Known Distribution

A. vogelii is distributed throughout semi-arid areas of tropical Africa and subtropical southern Africa, from Swaziland and South Africa in the south, to Burkina Faso and Mali in the west, to Kenya in the east. This species is closely associated with cropping and is rarely found in natural areas. *A. vogelii* is distributed throughout semi-arid areas of tropical and sub-tropical Africa. In the Nigerian savannahs, it can be found in cowpea crops, which are also attacked by *Striga gesnerioides*, and it has been reported as the major parasite of the crop in the northern Guinea savannah (Lagoke, 1989). Elsewhere in West Africa, infestations tend to be more localized, as in southern Mali. *A. vogelii* has replaced *S. gesnerioides* as an important constraint to cowpea production in eastern, central, and particularly southern Africa.

Potential Distribution within the United States

A recent host analysis by USDA-APHIS-PPQ-CPHST indicates portions of Arkansas, Illinois, Indiana, Iowa, Kansas, Kentucky, Maryland, Michigan, Minnesota, Mississippi, Missouri, Nebraska, North Dakota, Ohio, South Dakota, and Tennessee are at the greatest risk of *A. vogelii* establishment based on susceptible host presence.

Survey

CAPS-Approved Method*: Conduct a visual survey and collect suspected plants.

*For the most up-to-date methods for survey and identification, see Approved Methods on the CAPS Resource and Collaboration Site, at http://caps.ceris.purdue.edu/.

Literature-Based Methods:
Visual Survey: Conduct a visual survey for *A. vogelii.* The plant is an annual; vine/climber; shrub; herbaceous; seed propagated. Flowers are five-lobed, sulfur yellow to pale yellow, and bell-shaped. Hairy stems and leaves on parasitic weed, combined with stunted crop plants.

As *A. vogelii* is largely dependent on annual cropping, environmental requirements mirror those of its major hosts cowpea, bambara, peanut and soybean in sub-Saharan Africa. By and large, infestations are found in semi-arid areas with a short growing

season of 4 to 6 months, below 1500 m altitude. The parasite is most commonly found in areas of mono-modal rainfall with a long dry season as in Botswana or the Guinea savannah of West Africa, but it is also a pest in bimodal rainfall areas as in north-west and coastal Tanzania. Although crops are not produced during the cold dry season in the range of the parasite, frost at the end of the growing season will kill host plants surviving in crop residue on residual moisture and will prevent further seed production by *A. vogelii.* Host crops are largely associated with free-draining sands and sandy-loams.

Climatic amplitude (estimates):
- Mean annual rainfall: 520 to 1000 mm
- Rainfall regime: summer; bimodal
- Dry season duration: 6 to 7 months
- Mean annual temperature: 19 to 26ºC
- Mean maximum temperature of hottest month: 29 to 38ºC
- Mean minimum temperature of coldest month: 6 to16ºC
- Absolute minimum temperature: -3 to 0ºC

Key Diagnostics/Identification

CAPS-Approved Method*: Confirmation of *A. vogelii* requires morphological identification by a qualified botanist. Characteristics of the flowers, leaves, seeds, and roots can be used to distinguish *A. vogelii*.

*For the most up-to-date methods for survey and identification, see Approved Methods on the CAPS Resource and Collaboration Site, at http://caps.ceris.purdue.edu/.

Literature-Based Methods: The Federally Noxious Weeds Disseminules of the U.S. provides keys and fact sheets that can help identify seeds of Federally Noxious Weeds, including *Alectra* spp. (Scher and Walters, 2010). *A. vogelli* can be confused with *Striga* species and nutrient deficiencies.

References

Anota, T. 1991. Le Striga au Zaire. In: Lagoke STO, Hoevers R, M'Boob SS, Traboulsi R, eds. Improving *Striga* Management in Africa, Proceedings of the second General Workshop of the Pan-African Striga Control Network, Accra, Ghana: Food and Agriculture Organisation of the United Nations, 135-137.

Bagnall-Oakeley, H., Gibberd, V., and Nyongesa, T.E. 1991. The incidence and control of *Alectra vogeli*i in Embu district, Kenya. Proceedings of the 5th international symposium of parasitic weeds, Nairobi, Kenya, 24-30 June 1991., 340-345.

Beck, B.D.A. 1987. The effect of attack by *Alectra vogelii* (Benth.) on the yield of njugo beans (*Vigna subterranea* (L.)). Proceedings of the 4th international symposium on parasitic flowering plants 79-82.

Berner, D.K., Awad, A.E., and Aigbokhan, E.I. 1994. Potential of imazaquin seed treatment for control of *Striga gesnerioides* and *Alectra vogelii* in cowpea (*Vigna unguiculata*). Plant Disease, 78:18-23.

Botha, P.J. 1948. The parasitism of *Alectra vogelii* Benth. With special reference to the germination of its seeds. Journal of South African Botany, 14:63-80.

CABI. 2007. Crop Protection Compendium Wallingford, UK: CAB International. www.cabicompendium.org/cpc.

de la Harpe, A.C., Visser, J.H., and Grobbelaar, N. 1980. Photosynthetic characteristics of some South African parasitic flowering plants. Zeitschrift fur Pflanzenphysiologie, 103:265-275.

Engler, A. 1922. Alectra Thunb. Botanisches Jahrbücher für Systematik, Pflanzengeschichte und Pflanzengeographie, 57:613.

Greathead, D.J. and Milner, J.E.D. 1971. A survey of *Striga spp.* (Scrophulariaceae) and their insect natural enemies in East Africa with a discussion on the possibilities of biological control. Tropical Agriculture, 48:111-124.

Hattingh, I.D. 1954. Geelblom control. Farming in South Africa, May edition: 525-526.

Herb, R., Visser, J.H., and Schildknecht, H. 1987. Recovery, isolation and preliminary structural investigation of germination stimulants produced by *Vigna unguiculata* Walp. cv. Saunders Upright. Proceedings of the 4th international symposium on parasitic flowering plants. 351-366.

Hoffman, G. and Diara, S. 1991. Une nouvelle approche de l'Etude des plantes parasites des cultures: diagnostic de terrain. In: Lagoke STO, Hoevers R, M'Boob SS, Traboulsi R, eds. Improving Striga Management in Africa, Proceedings of the second General Workshop of the Pan-African Striga Control Network, Accra, Ghana: Food and Agriculture Organisation of the United Nations, 43-50.

Hutchinson, J. and Dalziel, J.M. 1963. Flora of West Tropical Africa. Volume 2, 2nd edition. London, UK: Crown Agents.

Kureh, I. and Alabi, S.O. 2003. The parasitic angiosperm *Alectra vogelii* (Benth.) can influence the growth and nodulation of host soybean (*Glycine max* (L.) Merrill). Crop Protection, 22(2):361-367.

Lagoke, S.T.O. 1989. *Striga* in Nigeria. FAO Plant Production and Protection Paper, No. 96:68-75.

Melchior, H. 1941. Die Gattung Alectra Thunb. Notizblatt der botanischen garten und museum Berlin-Dahlem, 15:423-447.

Müller, S., Hauck, C., and Schildknecht, H. 1992. Germination stimulants produced by *Vigna unguiculata* Walp cv. Saunders Upright. Journal of Plant Growth Regulation, 11:77-84.

Olmstead, R.G., de Pamphilis, C.W., Wolfe, A.D., Young, N.D., Elisons, W.J., and Reeves, P.J. 2001. Disintegration of the Scrophulariaceae. American Journal of Botany, 88:348-361.

Parker, C. and Riches, C.R. 1993. Parasitic weeds of the world: biology and control. Wallingford, UK; CAB International, xx + 332 pp.

Philcox, D. 1990. Alectra Thunb. In: Launert E, (Ed.), Flora Zambesiaca. Volume 8. Part 2. Scrophulariaceae. London, UK: Flora Zambesiaca Managing Committee.

Ramaiah, K.V., Safa, S.B., and Musselman, L.J. 1988. *Alectra* and *Striga aspera* in Burkina Faso. Haustorium, 19.

Riches, C.R. 1989. The biology and control of *Alectra vogelii* Benth. (Scrophuliariacea) in Botswana. PhD thesis, Reading, UK: University of Reading.

Riches, C.R. 2001. *Alectra vogelii* Benth. - a constraint to cowpea production in southern Africa. In: Mgonja MA, Chivinge OA, Monyo ES, eds. Striga research in Southern Africa and strategies for regionalized control options. Bulawayo, Zimbabwe: International Crops Research Institute for the Semi-arid Tropics, 87-91.

Riches, C.R., Hamilton, K.A., and Parker, C. 1992. Parasitism of grain legumes by Alectra species (Scrophulariaceae). Annals of Applied Biology, 121:361-370.

Salako, E.A. 1984. Observations on the effect of *Alectra vogelii* infestation on the yield of groundnut. Tropical Pest Management, 30:209-211.

Scher, J., and Walters, D. 2010. Federally Noxius Weed Disseminules of the U.S. Edition 2. Main page. http://keys.lucidcentral.org/keys/v3/FNWE2/, http://keys.lucidcentral.org/keys/v3/FNWE2/key/FNW_Seeds/Media/Html/fact_sheets/Alectra.htm.

Singh, B.B. 2000. Breeding cowpea varieties with combined resistance to different strains of Striga gesnerioides. In: Haussmann BIG, Hess DE, Koyama MI, Grivet L, Rattunde HFW, Geiger HH, eds. Breeding for Striga resistance in cereals, Weikersheim, Germany: Margraf Verlag, 261-270.

Stroud, A. and Parker, C. 1989. A Weed Identification Guide for Ethiopia. Rome, Italy: Food and Agriculture Organization.

USDA-APHIS. 2003. Federal Noxious Weed List. USDA, APHIS PPQ, Riverdale, USA. http//www.aphis.usda.gov/ppq/permits/fnwsbycat-e.PDF.

Visser, J.H. 1978. The biology of *Alectra vogelii* Benth., an angiospermous root parasite. Beitrage zur Chemischen Kommunikation in Bio- und Okosystemen, 279-294.

Visser, J.H. and Johnson, A.W. 1982. The effect of certain strigol analogues on the seed germination of *Alectra*.. South African Journal of Botany, 1(3):75-76.

Appendix A: Diagnostic Resource Contacts

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Appendix B: Glossary

Abaxial: Concerning the surface of a structure that is turned away from the structure's primary axis, pertaining to the lower surface of a leaf.

Abdomen: The posterior of the 3 main body divisions of an insect. Bears no functional legs in the adult stage.

Adaxial: Located on the side or directed toward the axis, pertaining to the upper surface of a leaf.

Aedeagus: In male insects, the penis or intermittent organ, situated below the scaphium and enclosed in a sheath.

Aestivate: To pass the summer in a dormant or torpid state.

Agglutinate: To cause to adhere, as with glue.

Amphigenous: Growing all around.

Anal: In the direction or position of the anus, near the anus or on the last abdominal segment.

Anal plate: 1) Lepidoptera larvae: The shield–like covering of the dorsum of the last segment; 2) Embryonic larvae: tergum XI, 3)Cocciids: a pair of triangular or semicircular sclerites at the cephalic end of the caudal cleft.

Antenna (pl. Antennae): One of the paired segmented sensory organs borne one on each side of the head, maybe referred to as horns or feelers.

Antennal club: A variable number of segments of the antennal flagellum usually identified by a change in shape or form from preceding segments. The antennal club is always apical, is sometimes arbitrarily delimited by segment number and always includes the terminal segment.

Anterior: In front, before.

Anther: Pollen-bearing portion of a flower.

Aperture (pl. Aperatures): To uncover, to open. Any opening in a wall, surface or tube.

Apical: At, near, or pertaining to the apex of any structure.

Arthropod: Any of numerous invertebrate animals of the phylum Arthropoda, including the insects, crustaceans, arachnids, and myriapods, that are

characterized by a chitinous exoskeleton and a segmented body to which jointed appendages are articulated in pairs.

Autoecious: In reference to rust fungi, producing all spore forms on one species of host plant (see heteroecious).

Axil: The angle formed by the leaf petiole and the stem.

Axillary: Pertaining to or placed within an axil.

Basal: Pertaining to the base or point of attachment to or nearest the body.

Blight: Sudden, severe, and extensive spotting, discoloration, wilting, or destruction of leaves, flowers, stems, or entire plants.

Callus (pl. calli): 1) A hard lump or mound-like, rounded swelling of the integument, such as a swelling at the base of the wing articulating with the thorax; 2) Heteroptera: the thickened or raised spots on the thorax, especially of Pentatomidae.

Calyx: The outer-most group of leaves surrounding the flower; the external-most part of the flower.

Capsomere: Protein subunits that serve as components of the viral capsid.

Carlavirus: (Siglum of carnation latent virus.) Member of a group of plant viruses with slightly flexuous, rod-shaped particles containing a single molecule of linear RNA, most of which are transmitted by aphids in a noncirculative manner.

Caudal: At or towards the anal (tail) end.

Cell wall: Protective, resistant, but permeable structure secreted externally to the cell membrane in plants, bacteria, fungi, and certain other organisms.

Cephalic: Pertaining to the head.

Chlorosis (adj. chlorotic): Failure of chlorophyll development, caused by disease or a nutritional disturbance; fading of green plant color to light green, yellow, or white.

Chorotic: Abnormal condition of plants in which the green parts lose their color or turn yellow as a result of chlorophyll production due to disease or lack of light.

Cinereous: Possessing the qualities of ash-colored, grey tinged with black.

Clavate: Club-shaped.

Coecum: A blind sac or tube. Term applied to a series of appendages opening into the alimentary canal at the junction of a crop and chylific ventricle.

Chorion: The outer shell or covering of the insect egg.

Comovirus: (Siglum of cowpea mosaic virus). Member of a group of multicomponent plant viruses with small, isometric particles containing two linear RNA species, readily transmitted mechanically and by beetles.

Corolla: Petals, collectively.

Corpus bursae: The sac-like portion of the bursa copulatrix which bears the ostium bursae.

Costa: 1) An elevated ridge that is rounded at its crest; 2) the thickened anterior margin of a wing, typically referring to the forewing; 3) the vein extending along the anterior margin of the wing from base to the point of junction with the subcosta.

Cotyledon: A leaf of the embryo of a seed plant, which upon germination either remains in the seed or emerges, enlarges, and becomes green; also called seed leaf.

Cremaster: 1) The apex of the last segment of the abdomen; 2) the terminal spine or hooked process of the abdomen of subterranean pupa. Used to facilitate emergence from the earth; 3) an anal hook by which some pupae are suspended.

Crustose: A crust-like growth form that is closely attached to the substrate.

Cucullus: 1) A hood, a hood-shaped covering or structure; 2) genitalia of male Lepidoptera: terminal part of the harpe.

Cultivar: A plant type within a species, resulting from deliberate genetic manipulation, which has recognizable characteristics (color, shape of flowers, fruits, seeds and height or form).

Cuticle: The noncellular outer layer of the body wall of an arthropod.

Cuticula: The outer body wall of an insect; “thin skin”.

Detritus: Material which remains after disintegration; rubbing away or the destruction of structure; fragmented material; any disintegrated or broken matter.

Deutonymph: The third instar of a mite.

Diapause: A condition of restrained development and reduced metabolic activity, which cannot be directly attributed to unfavorable environmental conditions. Regarded by

entomologists to involve a resting period of an insect, especially of larvae in winter. (hibernation, quiescence).

Dicotyledonous (dicots): A flowering plant with two seed leaves characterized by embryos with two cotyledons, net veined leaves, flower parts in fours or fives.

Dimorphism: A genetically controlled, non-pathological condition in which individuals of a species are characterized by distinctive or discrete patterns of coloration, size or shape. Dimorphism can be a seasonal, sexual or geographic manifestation.

Direct penetration: Penetration of plant tissues by a pathogen through barriers such as leaf cuticle by chemical and physical means (e.g. penetration peg).

Diverticulum: A tube, sac or invagination originating on the wall of a vessel or the alimentary canal and closed at the distal end.

Dorsal: On the upper surface.

Ductus bursae: The duct in female Lepidoptera extending from the ostium to the bursa copulatrix.

Ductus seminalis: Female Lepidoptera: the tube or canal connecting the bursa copulatrix with the common oviduct.

Echinulate: Having small spines projecting from cell walls.

Eclosion: Hatching from the egg.

Elytra: The anterior leathery or chitinous wings of beetles.

Envelope: Virology: a protein covering that packages the virus's genetic information.

Epidermis: The cellular layer of the skin; secreting the cuticula of insects.

Exoskeleton: The entire body wall, to the inner side of which muscles are attached; the outside skeleton in insects.

Facultative diapause: May or may not need to diapause; not required for development.

Feces: Excrement; the eliminated wastes of the digestive process.

Falcate: (Of spores) sickle-shaped.

Fascia: 1) Anatomy: a thin layer of connective tissue that covers, supports, binds, or connects muscles or body organs; 2) Taxonomy: a transverse band or broad line.

Femur (pl. femora): The third and usually the stoutest segment of the insect leg. Articulated with the body via the trochanter and bearing the tibia at its distal margin.

Forewing: The anterior wing of an insect which is attached to the mesothorax.

Frass: Plant fragments made by a wood-boring insect usually mixed with excrement; solid larval insect excrement.

Frontal suture: 1) The suture between the front and the clypeaus; 2) Diptera: separates the frontal lunule from the part of the head above it; 3) Coleoptera: clypeal suture or the suture formed by the arms of the epicranial suture.

Fuscous: Of, or pertaining to dark brown, approaching black; a plain mixture of black and red.

Gastropod: Any of a large class (Gastropoda) of mollusks, usually with a univalve shell or no shell and a distinct head bearing sensory organs, such as snails and slugs.

Germ Pore: An unthickened spot in a spore or conidial wall through which a germ tube may form.

Germ Tube: Hypha resulting from an outgrowth of the spore wall and cytoplasm after germination.

Glabrous: Without hairs.

Globose: Descriptive of structure which is spherical or globular in shape.

Grub: An elongate, whitish insect larva. The term is loosely applied to all insects, but more specifically applied to the larvae of Coleoptera, and some Hymenoptera.

Hamate: Hooked; bent at the end into a hook.

Haustorium (pl. haustoria): Specialized branch of a parasite formed inside host cells to absorb nutrients.

Hemiparasitic: Obtaining water and nutrients from the roots of other plants then manufacturing food through photosynthesis.

Heteroecious: Pertaining to a rust fungus requiring two unrelated host plants for completion of its life cycle (see autoecious).

Hindwing: The posterior wing of an insect, attached to the metathorax.

Host cell: A cell that is infected by a virus or another type of microorganism.

Humeral: Pertaining to the shoulder; located in the anterior basal portion of the wing.

Humeral callus: Diptera: each of the anterior angles of the prescutum of the mesothorax, usually a more-or-less rounded tubercle.

Hyaline: Like glass, transparent colorless.

Hypophyllous: On the underside of a leaf surface.

Imago: The adult stage or sexually mature insect.

Incubation Period: The time between penetration of a host by a pathogen and the first appearance of disease symptoms; the time during which microorganisms inoculated onto a medium are allowed to grow.

Inoculate: 1) To communicate a disease to (a living organism) by transferring its causative agent into the organism; 2) to implant microorganisms or infectious material into (a culture medium).

Inoculum: Pathogen or its parts, capable of causing infection when transferred to a favorable location.

Intercalary: Formed or situated somewhere between apex and base of a given structure.

Interspaces: 1) Coleoptera: the plane surface between elytral striae; 2) Lepidoptera: spaces between wing veins not included in closed cells; 3) Orthoptera: a deep incision or sulcus on the posterior margin of the metasternum.

Isometric: Usually used for virus particles to describe those that are icosahedral in structure and appear approximately round.

Juxta: Male Lepidoptera: a sclerite beneath the aedeagus and to which it may be hinged or fused; part of the fultura inferior.

Labrum: The 'upper lip', forming the roof of the preoral cavity and mouth; derived from the first head segment.

Lamella: A thin plate or leaf-like process; a parademe.

Larvae (pl. for larva): An early, free-living immature form of any animal that changes structurally when it becomes an adult usually by complex metamorphosis.

Latent Period: The time between infection and the production of new inoculum; the time after a vector has acquired a pathogen and before it can be transmitted.

Leaf spot: A plant disease lesion typically restricted in development in the leaf after reaching a characteristic size.

Lesions: Localized diseased area or wound.

Luteovirus: Literally "yellowish". Member of a group of plant viruses with isometric particles containing one molecule of linear RNA, mainly confined to the phloem, and usually not mechanically transmitted but transmitted in nature by aphids in a circulative manner.

Mandible: The first pair of jaws in insects, stout and tooth-like in chewing insects, needle or sword-shaped in piercing-mouthed sucking insects; the lateral upper jaws of biting insects; in muscoid larvae, the mouth hooks.

Medial: Referring to, or at the middle of a structure.

Membranous: Tissue which is thin, pliable and semi-transparent; like a membrane.

Mesophyll: The photosynthetic tissue of a leaf, located between the upper and lower epidermis. Mesophyll is commonly differentiated into palisade parenchyma and spongy parenchyma.

Mesothorax: The second or middle thoracic segment which bears the middle legs and anterior wings.

Metathorax: The third (and last) segment of the thorax.

Micropyle: A very small opening in the outer coat of an ovule, through which the pollen tube penetrates; the corresponding opening in the developed seed; one of the minute openings in the insect egg, through which spermatozoa enter in fertilization.

Microtrichium (pl. microtrichia): Small, sclerotized non-innervated cuticular projects on the body and wings of insects; also found on the tracheae.

Mildew: Thin coating of mycelial growth and spores on the surfaces of infected plant parts.

Molt: A process of shedding the exoskeleton, ecdysis.

Monocotyledonousc (monocot) An embryo having a single cotyledon.

Mosaic: Disease symptom characterized by non-uniform coloration, with intermingled normal, light green and yellowish patches, usually caused by a virus; often used interchangeably with mottle.

Mucro: Nematodes: A stiff or sharp point abruptly terminating an organ.

Multivoltine: Pertaining to organisms with many generations in a year or season.

Necrotic: Death of cells or tissue, usually accompanied by black or brown darkening.

Neonate: Newly born individual.

Nematode: Non-segmented roundworm (animal), parasitic on plants or animals, or free living in soil or water.

Obovoid: Egg-shaped, with the narrow end outward.

Obligate: Restricted to a particular set of environmental conditions, without which an organism cannot survive. (e.g., an obligate parasite can survive only by parasitizing another organism.)

Ocellus (pl. ocelli): A simple eye of an insect or other arthropod.

Ooze: Mass of bacterial cells mixed with host fluids.

Ostium bursae: Ostium is the external genitalic opening of female Lepidoptera. Bursae the opening of the bursa copulatrix in Lepidoptera, equivalent to the vulva of female insects having the genital opening on the eighth segment.

Oviparous: Lay eggs.

Ovoid: Egg-like in shape or appearance.

Oviposit (oviposition): To deposit or lay eggs or ova. The act of depositing eggs.

Ovipositor: The external, tubular part of the female reproductive system through which eggs are passed. The ovipositor may be rigid and fixed in length or flexible and telescopic.

Ovisac: A receptacle for eggs.

Palp (pl. palpi): Finger-like, usually segmented appendage of the maxilla (maxillary palp) and labium (labial palp).

Papilla (pl. papillae): A hump or swelling.

Papillae anales: Lepidoptera: A pair of lobes at the apex of the female abdomen which are used in oviposition.

Parasite (adj. parasitic): Organism that lives in intimate association with another organism on which it depends for its nutrition; not necessarily a pathogen.

Parasitoid: A parasite that kills its host.

Parenchyma: The primary tissue of higher plants, composed of thin-walled cells and forming the greater part of leaves, roots, the pulp of fruit, and the pith of stems.

PCR (acronym for polymerase chain reaction): A technique used to amplify the number of copies of a specific region of DNA in order to produce enough of the DNA for use in various applications such as identification and cloning.

Pestiferous: Producing or breeding infectious disease. Infected with or contaminated by an epidemic disease.

Petiole: 1) Botany: stalk portion of a leaf; 2) Insect: Apocrital Hymenoptera; the narrow second (and sometimes third abdominal segments that precede the gaster) forming the 'waist'.

Pheromone: A substance given off by one individual that causes a specific reaction by other individuals of the same species, such as sex attractants, alarm substances etc.

Phloem: The vascular tissue in vascular plants, that conducts and distributes sugars and other dissolved foods from the places the food is produced to the places the food is needed or stored.

Phytophagous: Plant eating.

Polyphagous (Polyphagy): Eating many kinds of food.

Polyvoltine: See multivoltine.

Posterior: A term of position pertaining to a structure situated behind the axis. Toward the rear, caudal or anal end of the insect; opposed to anterior.

Process (pl. processus): A projection from the surface, margin or appendage.

Pronotum: The upper (dorsal) plate of the prothorax.

Proleg: 1) Any process or appendage that serves the purpose of a leg; 2) specifically, the pliant, non-segmental abdominal legs of caterpillars and some sawfly larvae. Not true segmented appendages.

Prothorax: The first segment of the thorax.

Protonymph: The second instar of a mite.

Pseudoparenchyma: A mass of hyphae arranged together to form a tissue like structure. A dense tissue formed by hyphae becoming twisted and fixed together where the hyphal components of the tissue are no longer distinguishable.

Pulverulent: Appearing as though covered with a fine powder.

Pulvinate: Cushion, cushion-shape, flattened pads or pad-like.

Pupa (pl. pupae): The stage between the larva and adult in insects with complete metamorphosis, a nonfeeding and usually inactive stage.

Pupation: Becoming a pupa.

Pustule: A blister-like spore mass breaking through a plant epidermis.

Raster: Scarabaeoid larvae: a complex of specifically arranged bare areas, setae and spines on ventral surface of last abdominal segment, anterior of anus.

Raceme: A type of inflorescence in which flowers are formed on individual stalks along a main axis or peduncle.

Reniform: Kidney-shaped.

Restriction fragment length polymorphism (RFLP): A variation in DNA sequence that is easily recognized because it occurs at a site where a restriction enzyme cuts a specific sequence, producing DNA fragments of varying lengths. RFLP's often serve as genetic markers.

Reticulate: Descriptive of surface sculpture, usually the insect's integument, that is covered with net-like lines.

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Rachis: Elongated main axis of an inflorescence.

Rust: A disease caused by a specialized group of basidiomycetes that often produces spores of a rusty color.

Scab: Roughened, crustlike diseased area on the surface of a plant organ.

Scarification: The physical or chemical treatment given to some seeds in order to weaken the seed coat sufficiently for germination to occur.

Sclerenchyma (adj. sclerenchymatous): Tissue made up of thick-walled plant cells.

Sclerite: A hardened body wall plate bounded by sutures or membranous areas.

Sclerotized: Hardened.

Scutellum: A sclerite of the thoracic notum; the mesoscutellum appearing as a more or less triangular sclerite behind the pronotum, especially in Hemiptera.

Semi-looper: A caterpillar in which 1-2 pairs of the abdominal legs are absent and movement is restricted to progression only in small loops (of the Noctuoidea superfamily).

Semilunar: In the form of a half crescent.

Senesce: To decline in stature, vigor and capacity following maturity.

Septate: With cross walls; having septa.

Serotype: A subdivision of virus strains distinguished by protein or a protein component that determines its antigenic specificity.

Sessile: Used in reference to a leaf, leaflet, flower, floret, fruit, ascocarp, basidiocarp, etc., without a stalk, petiole, pedicel, stipe or stem; (of nematodes) permanently attached; not capable of moving about.

Seta (pl. setae): A bristle; commonly known as hairs.

Single stranded, positive sense RNA: Also known as a sense-strand RNA virus, a virus whose genetic information consists of a single strand of RNA that is the positive (or sense) strand which encodes mRNA (messenger RNA) and protein. Replication in positive-strand RNA viruses is via a negative-strand intermediate. Examples of positive-strand RNA viruses include polio virus, Coxsackie virus, and echovirus.

Spiracles: A breathing pore; in the plural the lateral openings on the segments of the insect body through which air enters the trachaea.

Spore: A specialized reproductive body in fungi (and some other organisms), containing one or more cells, capable of developing into an adult.

Sporulate (sporulation): To produce spores.

Stele: Central cylinder of vascular tissue (especially in roots).

Stigma: Portion of a flower that receives pollen and on which the pollen germinates.

Stipule: One pair of leaf-like structures, spines, glands, or scales at the leaf base or along a petiole.

Stria (pl. striae): Descriptive of the surface sculpture, usually the insect's integument, that is marked with numerous parallel, fine, impressed lines.

Strigula: A fine, short, transverse mark or line.

Stunting: Reduction in height of a vertical axis resulting from a progressive reduction in the length of successive internodes or a decrease in their number.

Subepidermal: Beneath the epidermis.

Suture: Gastropods, the spiral line that marks the junction of the whorls; in chitons, the junction between girdle and valves.

Tarsus (pl. tarsi): The leg segment immediately beyond the tibia, consisting of one or more segments or subdivisions.

Tegumen: Lepidoptera: the tergum in male genitalia. A structure shaped as a hood or inverted trough, positioned dorsal of the anus; the uncus articulates with its caudal margin, derived from the ninth abdominal tergum.
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Teneral: Describing the imago or adult shortly after emergence from the nymphal or pupal stage when the integument is not hardened or its color has not matured.

Termen: The outer margin of a wing, between the apex and the posterior or anal angle.

Testa: Seed coat.

Thorax: The body region behind the head, which bears the legs and wings.

Tibia (pl. tibiae): The fourth segment of the leg, between the femur and tarsus.

Tibial spur: The spur or spurs frequently borne near to or at the distal end of the tibia.

Transverse: Pertaining to structures which are wider than long; running across or cutting the longitudinal axis at right angles.

Truncate: Pertaining to structures which end abruptly as if cut at a right angle to the longitudinal axis.

Tubercle: A little solid pimple or small button, in Sphecoidea rounded lobes of the dorsal lateral margin of the pronotum; in caterpillars, body structures of the character, sometimes bear setae.

Urediniospore (also urediospore, uredospore): The asexual, dikaryotic, often rusty-colored spore of a rust fungus, produced in a structure called a uredinium; the "repeating stage" of a heteroecious rust fungus, i.e. capable of infecting the host plant on which it is produced.

Uredinium (also uredium; pl. uredinia): Fruiting body (sorus) of rust fungi that produces urediniospores.

Urogomphi: Fixed or mobile processes found on the terminal segments of certain larvae; variously termined styli, cerci, pseudocerci, corniculi.

Valva: Harpagones or two lateral sclerites which cover the ovipositor when not in use.

Vascular bundle: Strand of conductive tissue, usually composed of xylem and phloem (in leaves, small bundles are called veins).

Vector: Literally a bearer; specifically a host of a disease transmissible to another species of organism.

Vein clearing: Disappearance of green color in or around leaf veins (a common symptom associated with virus infection).

Ventral: Pertaining to the under surface of abdomen.

Vermiform: Worm-shaped.

Vesica: Lepidoptera: the penis, or terminal part of the aedeagus. Vesica is membranous and eversible; typically held within the tubular part of the aedeagus but everted and inflated during copulation.

Virion: Complete virus particle.

Virus: A submicroscopic,intracellular, obligate parasite consisting of a core of infectious nucleic acid (either RNA or DNA) usually surrounded by a protein coat.

References Used:

Agrios, G.N. 1997. Plant Pathology, 4th Edition. San Diego: Academic Press.

Arneson, P.A. Accessed 2006. On-Line Glossary of Technical Terms in Plant Pathology. http://ppathw3.cals.cornell.edu/glossary/Glossary.htm.

D'Arcy, C. J., D. M. Eastburn, and G. L. Schumann. 2001. Illustrated Glossary of Plant Pathology. *The Plant Health Instructor.* http://www.apsnet.org/education/IllustratedGlossary/default.htm.

Dictionary.com. Accessed 2006. Lexico Publishing Group. www.dictionary.com.

Gordh, G. and D.H. Headrick. 2001. A Dictionary of Entomology. New York: CABI Publishing.

Insect Basics Glossary. Accessed 2006. University of California, Berkeley, CA. http://www.cnr.berkeley.edu/citybugs/allaboutbugs/Glossary.htm.

IPM Glossary. Accessed 2006. North Carolina State Integrated Pest Management. http://ipm.ncsu.edu/AG271/glossary.html.

Purdue Entomology Glossary. Accessed 2006. http://www.entm.purdue.edu/entomology/courses/306/306glossary.html.

Appendix C: FY10 & 11 CAPS Prioritized Pest List and Commodity Matrix

AHP Prioritized Pest List for FY10

Rank	Scientific Name	Common Name	Taxonomic Group	Pest Source
1	*Agrilus biguttatus*	Oak splendor beetle	Arthropod	NAFC-ExFor Pest
1	*Platypus quercivorus*	Oak ambrosia beetle	Arthropod	NAFC-ExFor Pest
2	*Cronartium flaccidum*	Scots pine blister rust	Fungus	NPDRS
3	*Helicoverpa armigera*	Old World bollworm	Arthropod	OPIS A List
4	*Thaumetopoea processionea*	Oak processionary moth	Arthropod	EPICA
5	*Tomicus destruens*	Pine shoot beetle	Arthropod	NAFC-ExFor Pest
6	*Dendrolimus superans*	Siberian silk moth	Arthropod	Western Region 2004 Exotic Pest List/NAFC-ExFor Pest
7	*Spodoptera litura*	Cotton cutworm	Arthropod	EPPO A1 List
8	*Otiorhynchus dieckmanii*	Wingless weevil	Arthropod	NPAG
9	*Ceroplastes japonicus*	Japanese wax scale	Arthropod	OPIS A List
10	*Unaspis yanonensis*	Arrowhead scale	Arthropod	OPIS A List
11	*Phytophthora alni*	Alder root rot	Fungus	Western Region 2004 Exotic Pest List/NAFC-ExFor Pest
12	*Ralstonia solanacearum* race 3 biovar 2	Bacterial wilt	Bacterium	OPIS A List
13	*Achatina fulica*	Giant African snail	Mollusk	OPIS A List
14	*Lymantria mathura*	Pink gypsy moth	Arthropod	Western Region 2004 Exotic Pest List/NAFC-ExFor Pest
15	*Leucoptera malifoliella*	Pear leaf blister moth	Arthropod	Western Region 2004 Exotic Pest List
16	*Ditylenchus angustus*	Rice stem nematode	Nematode	Western Region 2004 Exotic Pest List
17	*Ceroplastes destructor*	Soft wax scale	Arthropod	OPIS A List
18	*Chilo suppresalis*	Asiatic rice borer	Arthropod	Western Region 2004 Exotic Pest List
19	Veronicellidae spp.		Mollusk	Mollusk Team
20	*Dendrolimus pini*	Pine-tree lappet	Arthropod	NAFC-ExFor Pest
21	*Spodoptera littoralis*	Egyptian cottonworm	Arthropod	OPIS A List
22	*Chalara fraxinea*	Ash dieback	Fungus	NPAG
23	*Monochamus sutor*	Small white-marmorated longhorned beetle	Arthropod	NAFC-ExFor Pest
24	*Planococcus minor*	Passionvine mealybug	Arthropod	OPIS A List
25	*Tuta absoluta*	Tomato leaf miner	Arthropod	NPAG
26	*Nysius huttoni*	New Zealand wheat bug	Arthropod	NPAG
27	*Candidatus* Phytoplasma australiense	Phytoplasma yellows	Phytoplasma	NPAG
28	*Meloidogyne indica*	Citrus root-knot nematode	Nematode	OPIS A List
29	*Raffaelea quercivora*	Japanese oak wilt	Fungus	CAPS Oak Commodity Survey
30	*Monacha spp.*		Mollusk	Mollusk Team
31	*Oxycarenus hyalinipennis*	Cotton seed bug	Arthropod	National CAPS Committee Pest Prioritization Subgroup
32	*Eudocima fullonia*	Fruit piercing moth	Arthropod	Western Region 2004 Exotic Pest List
33	*Thaumatotibia leucotreta*	False codling moth	Arthropod	OPIS A List
34	*Phytoplasma* AP-MLO	Apple proliferation	Phytoplasma	EPPO A2 list
35	*Monochamus saltuarius*	Japanese pine sawyer	Arthropod	NAFC-ExFor Pest
36	*Mycosphaerella gibsonii*	Needle blight of pine	Fungus	EPPO A1 List
37	*Onopordum acaulon*	Horse thistle	Plant	APHIS Weed Team
38	*Diabrotica speciosa*	Cucurbit beetle	Arthropod	EPPO A1 list
38	*Harpophora maydis*	Late wilt of corn	Fungus	NPDRS
38	*Xanthomonas oryzae*	Bacterial leaf streak, bacterial blight	Bacterium	OPIS A List
39	*Adoxophyes orana*	Summer fruit tortrix moth	Arthropod	Western Region 2004 Exotic Pest List
40	*Archips xylosteanus*	Variegated golden tortrix	Arthropod	National CAPS Committee Pest Prioritization Subgroup

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AHP Prioritized Pest List for FY10

Rank	Scientific Name	Common Name	Taxonomic Group	Pest Source
41	*Meloidogyne fujianensis*	Asian citrus root-knot nematode	Nematode	OPIS A List
41	*Meloidogyne jianyangensis*	Citrus root-knot nematode	Nematode	OPIS A List
41	*Meloidogyne mingnanica*	Citrus root-knot nematode	Nematode	OPIS A List
42	*Meloidogyne paranaensis*	Parana coffee root-knot nematode	Nematode	OPIS A List
43	*Meloidogyne citri*	Asian citrus root-knot nematode	Nematode	OPIS A List
44	*Candidatus* Phytoplasma prunorum	European stone fruit yellows	Phytoplasma	QUADS
45	*Cernuella* spp.	Exotic species	Mollusk	OPIS A List
46	*Cochlicella* spp.	Exotic species	Mollusk	OPIS A List
47	*Meloidogyne artiellia*	British root-knot nematode	Nematode	OPIS A List
48	*Heterodera latipons*	Mediterranean cereal cyst nematode	Nematode	OPIS A List
49	*Meloidogyne donghaiensis*	Donghai root-knot nematode	Nematode	OPIS A List
50	*Heterodera cajani*	Pigeonpea cyst nematode	Nematode	OPIS A List
50	*Heterodera sacchari*	Sugar cane cyst nematode	Nematode	OPIS A List
51	*Meloidogyne fallax*	False Columbia root-knot nematode	Nematode	OPIS A List
52	*Rhynchophorus ferrugineus*	Red palm weevil	Arthropod	NPAG

Shading denotes a rank value shared by two or more pests; pests with the same rank received identical AHP Scores.

FY10 Commodity Matrix

Scientific Name	Common Name	Almonds (*Prunus dulcis*)	Apples (*Malus* spp.)	Asparagus (*Asparagus* spp.)	Barley (*Hordeum* spp.)	Beans (*Phaseolus* spp.)	Broccoli (*Brassica oleracea*)	Cantaloupes (*Cucumis* spp.)	Carrots (*Daucus carota*)	Celery (*Apium graveolens*)	Citrus (*Citrus* spp.)	Corn (*Zea* spp.)	Cotton (*Gossypium* spp.)	Cucumbers (*Cucumis* spp.)	Grapes (*Vitis* spp.)	Lettuce (*Lactuca* spp.)	Oats (*Avena* spp.)	Onions (*Allium* spp.)	Peaches (*Prunus persica*)	Peanuts (*Arachis* spp.)	Pears (*Pyrus* spp.)	Potatoes (*Solanum tuberosum*)	Rice (*Oryza* spp.)	Sorghum (*Sorghum* spp.)	Soybeans (*Glycine* spp.)	Strawberries (*Fragaria* spp.)	Sunflower (*Helianthus* spp.)	Tomatoes (*Solanum lycopersicum*)	Wheat (*Triticum* spp.)	Pine (*Pinus* spp.)	Other Softwood Trees*	Soft Hardwood Trees*	Hardwood Trees*	Pest Commodity Total
Achatina fulica	Giant African snail		■			■	▲	▲	■		■	■	■	▲	■	■		■			▲		■	■	■	■		■	■				■	■	21
Adoxophyes orana	Summer fruit tortrix moth		▲								■		■		■				▲	■	▲					■	■						■	■	11
Agrilus biguttatus	Oak splendor beetle																																■	▲	2
Archips xylosteanus	Variegated golden tortrix		■								■								■		■										■	■	■	7
Candidatus Phytoplasma australiense	Phytoplasma yellows					■		■						■	▲				■								▲						■	■	8
Candidatus Phytoplasma prunorum	European stone fruit yellows	■	■																▲														■	■	5
Cernuella spp.					■	■					■															■		■		■	■	■	■	■	10
Ceroplastes destructor	Soft wax scale		■								▲										■												■	■	5
Ceroplastes japonicus	Japanese wax scale		■								▲				■				■		■												■	■	7
Chalara fraxinea	Ash dieback																																■		1
Chilo suppressalis	Asiatic rice borer						■					▲												▲	▲				■	■					6
Cochlicella spp.					■				■																					■		■	■	■	6
Cronartium flaccidum	Scots pine blister rust																														▲				1
Dendrolimus pini	Pine-tree lappet																														▲	■			2
Dendrolimus superans	Siberian silk moth																														▲	▲	■	■	4
Diabrotica speciosa	Cucurbit beetle		■	■		■	■	■	■	■	■	▲	■	■	■	■	■	■	■	▲		▲	■	■	▲	▲	■	■	■	▲				■	26
Ditylenchus angustus	Rice stem nematode																							▲											1
Eudocima fullonia	Fruit piercing moth		■					■			▲			■	■				■		■						■		▲				■	■	11
Harpophora maydis	Late wilt of corn											■																							1
Helicoverpa armigera	Old World bollworm		■		▲	▲	■	■	■		▲	▲	▲	■		■	■	■	■	▲		▲	■	▲	▲	■	▲	▲	▲	▲	■	■	■	27
Heterodera cajani	Pigeonpea cyst nematode					▲																													1
Heterodera latipons	Mediterranean cereal cyst nematode				▲				■									▲			■									▲					5
Heterodera sacchari	Sugar cane cyst nematode											■												▲	■										3
Leucoptera malifoliella	Pear leaf blister moth		▲																■		▲												■	■	5
Lymantria mathura	Pink gypsy moth		■																		■										■	■	▲	▲	6
Meloidogyne artiellia	British root-knot nematode				■	■	■			■								▲							■				■	■					8
Meloidogyne citri	Asian citrus root-knot nematode										■																		■						2
Meloidogyne donghaiensis	Donghai root-knot nematode										■																								1
Meloidogyne fallax	False Columbia root-knot nematode			■		■			▲			■				■					■		▲				■		■						9
Meloidogyne fujianensis	Asian citrus root-knot nematode										■																								1
Meloidogyne indica	Citrus root-knot nematode										■																								1
Meloidogyne jianyangensis	Citrus root-knot nematode										■																								1
Meloidogyne mingnanica	Citrus root-knot nematode										■																								1
Meloidogyne paranaensis	Parana coffee root-knot nematode																								■	■			■						3
Monacha spp.																																			0
Monochamus saltuarius	Japanese pine sawyer																														▲	▲			2
Monochamus sutor	Small white-marmorated longhorned beetle																														▲	▲		■	3
Mycosphaerella gibsonii	Needle blight of pine																														▲				1
Nysius huttoni	New Zealand wheat bug				■		■									■	■				■						■	■		■					8
Onopordum acaulon	Horse thistle																																		0
Otiorhynchus dieckmanni	Wingless weevil															■																			1
Oxycarenus hyalinipennis	Cotton seed bug		■						■	■					■					■		■							■				■	■	9
Phytophthora alni	Alder root rot																																▲		1
Phytoplasma AP-MLO	Apple proliferation		▲												■							■												■	4
Planococcus minor	Passionvine mealybug			■		■	■	■		■	■	■	■	■	■						■	■	▲	■		■	■	■	■		■	■	■	■	21
Platypus quercivorus	Oak ambrosia beetle																																■	▲	2
Raffaelea quercivora	Japanese oak wilt																																	▲	1
Ralstonia solanacearum race 3 biovar 2	Bacterial wilt					■							■								■		▲						▲						5
Rhynchophorus ferrugineus	Red palm weevil		■																														■	■	3

FY10 Commodity Matrix

Scientific Name	Common Name	Almonds (*Prunus dulcis*)	Apples (*Malus* spp.)	Asparagus (*Asparagus* spp.)	Barley (*Hordeum* spp.)	Beans (*Phaseolus* spp.)	Broccoli (*Brassica oleracea*)	Cantaloupes (*Cucumis* spp.)	Carrots (*Daucus carota*)	Celery (*Apium graveolens*)	Citrus (*Citrus* spp.)	Corn (*Zea* spp.)	Cotton (*Gossypium* spp.)	Cucumbers (*Cucumis* spp.)	Grapes (*Vitis* spp.)	Lettuce (*Lactuca* spp.)	Oats (*Avena* spp.)	Onions (*Allium* spp.)	Peaches (*Prunus persica*)	Peanuts (*Arachis* spp.)	Pears (*Pyrus* spp.)	Potatoes (*Solanum tuberosum*)	Rice (*Oryza* spp.)	Sorghum (*Sorghum* spp.)	Soybeans (*Glycine* spp.)	Strawberries (*Fragaria* spp.)	Sunflower (*Helianthus* spp.)	Tomatoes (*Solanum lycopersicum*)	Wheat (*Triticum* spp.)	Pine (*Pinus* spp.)	Other Softwood Trees*	Soft Hardwood Trees*	Hardwood Trees*	**Pest Commodity Total**
Spodoptera littoralis	Egyptian cottonworm		■	■		▲	■	▲	▲	■	▲	▲	▲	▲	▲	▲		▲		▲	■	▲	▲	▲	▲	■	▲	▲	■	■	■	■	**28**
Spodoptera litura	Cotton cutworm		▲	■		▲	▲	■	■	■	▲	▲	▲	■	▲	■		▲	■	▲	■	▲	▲	▲	▲	▲	▲	▲	■		■	▲	■	**28**
Thaumatotibia leucotreta	False codling moth					■					▲	▲	▲		■				▲					■				■				■	▲	**10**
Thaumetopoea processionea	Oak processionary moth																													▲		■	■	**3**
Tomicus destruens	Pine shoot beetle																													■				**1**
Tuta absoluta	Tomato leaf miner																					■						▲						**2**
Unaspis yanonensis	Arrowhead scale										▲																							**1**
Veronicellidae spp.						■	■	■	■		■			■		■				■									■					**9**
Xanthomonas oryzae	Bacterial leaf streak, bacterial blight																						▲											**1**
	Total Pests Per Commodity:	**1**	**16**	**5**	**6**	**14**	**10**	**9**	**9**	**5**	**21**	**12**	**10**	**9**	**12**	**9**	**5**	**5**	**12**	**11**	**13**	**9**	**10**	**10**	**9**	**10**	**8**	**17**	**10**	**12**	**12**	**25**	**27**	

Legend:

▲ = Primary host

■ = Other host

***Key to Forest Product Categories:**

Other Softwood Trees (Genera: *Abies, Casuarina, Cupressus, Juniperus, Larix, Picea, Pseudotsuga, Tsuga*)

Soft Hardwood Trees (Genera: *Acacia, Albizia, Alnus, Asimina, Castanea, Catalpa, Celtis, Elaeagnus, Fraxinus, Liquidambar, Magnolia, Melaleuca, Persea, Platanus, Populus, Paulownia, Sabal, Salix, Tamarix, Tilia, Ulmus*)

Hardwood Trees (Genera: *Acer, Aleurites, Amelanchier, Betula, Carpinus, Carya, Castanopsis, Cornus, Crataegus, Diospyros, Eucalyptus, Fagus, Ilex, Juglans, Lithocarpus, Malus, Melia, Morus, Prunus, Quercus, Sapium, Sorbus, Vaccinium*)

Commodities in Decreasing Order of Value:**

Corn (*Zea* spp.), Soybeans (*Glycine* spp.), Wheat (*Triticum* spp.), Cotton (*Gossypium* spp.), Tomatoes (*Lycopersicon* spp.), Grapes (*Vitis* spp.), Potatoes (*Solanum* spp.), Apples (*Malus* spp.), Citrus (*Citrus* spp.), Peanuts (*Arachis* spp.), Lettuce (*Lactuca* spp.), Rice (*Oryza* spp.), Sorghum (*Sorghum* spp.), Barley (*Hordeum* spp.), Strawberries (*Fragaria* spp.), Almonds (*Prunus dulcis*), Onions (*Allium* spp.), Peaches (*Prunus persica*), Carrots (*Daucus carota*), Cucumbers (*Cucumis* spp.), Beans (*Phaseolus* spp.), Sunflower (*Helianthus* spp.), Pears (*Pyrus* spp.), Celery (*Apium graveolens*), Broccoli (*Brassica oleracea*), Cantaloups (*Cucumis* spp.), Oats (*Avena* spp.), Asparagus (*Asparagus* spp.). **Not included in the ranking by value:** Pine (*Pinus* spp.), Other Softwood trees, Soft Hardwood trees, Hardwood Trees.

****** NASS. 2008. Agricultural Prices 2007 Summary. Agricultural Statistics Board, National Agricultural Statistics Service, United States Department of Agriculture.

Appendix D: FY12 CAPS Prioritized Pest List and Commodity Matrix

AHP Prioritized Pest List for 2012 by Rank **Appendix D**

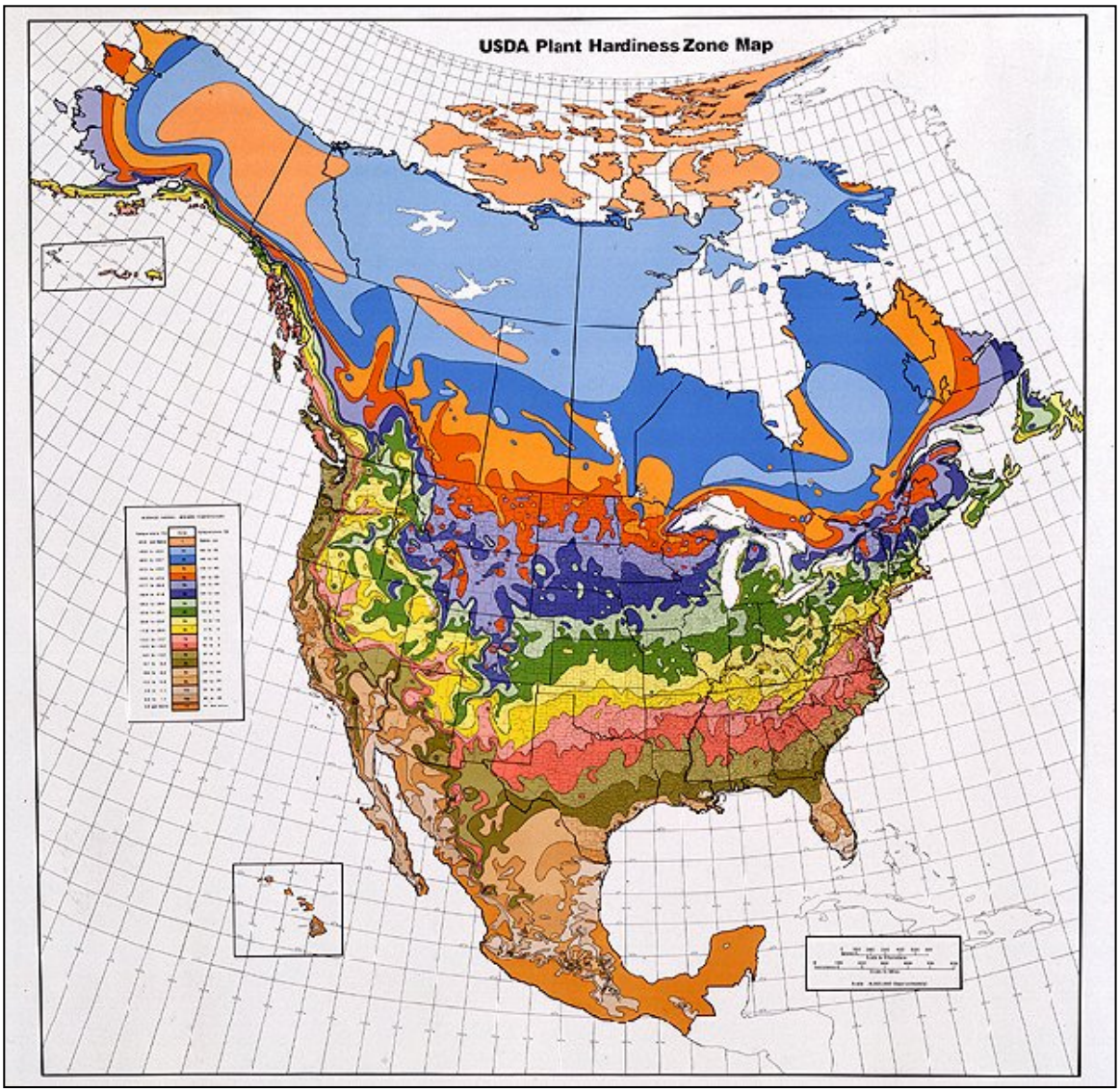
Rank	Scientific Name	Common Name	Kingdom	Phylum	Class	Order	Family
1	*Agrilus biguttatus* (F.)	Oak Splendor Beetle	Animalia	Arthropoda	Insecta	Coleoptera	Buprestidae
1	*Platypus quercivorus* (Murayama)	Oak Ambrosia Beetle	Animalia	Arthropoda	Insecta	Coleoptera	Platypodidae
2	*Cronartium flaccidum* (Alb. & Schwein.) G. Winter	Scots Pine Blister Rust	Fungi	Basidiomycota	Urediniomycetes	Uredinales	Cronartiaceae
3	*Helicoverpa armigera* (Hübner)	Old World Bollworm	Animalia	Arthropoda	Insecta	Lepidoptera	Noctuidae
4	*Tremex fuscicornis* (Fabricius)	Tremex Wood Wasp	Animalia	Arthropoda	Insecta	Hymenoptera	Siricidae
5	*Thaumetopoea processionea* (L.)	Oak Processionary Moth	Animalia	Arthropoda	Insecta	Lepidoptera	Notodontidae
6	*Tomicus destruens* (Wollaston)	Pine Shoot Beetle	Animalia	Arthropoda	Insecta	Coleoptera	Scolytidae
7	*Dendrolimus sibiricus*	Siberian Silk Moth	Animalia	Arthropoda	Insecta	Lepidoptera	Lasiocampidae
7	*Dendrolimus superans*	Sakhalin silk moth	Animalia	Arthropoda	Insecta	Lepidoptera	Lasiocampidae
8	*Spodoptera litura* (F.)	Cotton Cutworm	Animalia	Arthropoda	Insecta	Lepidoptera	Noctuidae
9	*Otiorhynchus dieckmanii* (Mangano)	Wingless Weevil	Animalia	Arthropoda	Insecta	Coleoptera	Curculionidae
10	*Ceroplastes japonicus* (Green)	Japanese Wax Scale	Animalia	Arthropoda	Insecta	Hemiptera	Coccidae
11	*Unaspis yanonensis* (Kuwana)	Arrowhead Scale	Animalia	Arthropoda	Insecta	Hemiptera	Diaspididae
12	*Phytophthora alni* (Brasier & Kirk)	Alder Root and Collar Rot	Chromista	Oomycota	Oomycetes	Pythiales	Pythiaceae
13	*Ralstonia solanacearum* race 3 biovar 2 (Smith)	Bacterial Wilt	Bacteria	Proteobacteria	Betaproteobacteria	Burkholderiales	Ralstoniaceae
14	*Achatina fulica* (Bowditch)	Giant African Snail	Animalia	Mollusca	Gastropoda	Stylommatophora	Achatinidae
15	*Lymantria mathura* (Moore)	Rosy Moth	Animalia	Arthropoda	Insecta	Lepidoptera	Lymantriidae
16	*Massicus raddei* (Blessig)	Mountain oak longhorned beetle	Animalia	Arthropoda	Insecta	Coleoptera	Cerambycidae
17	*Leucoptera malifoliella* (Costa)	Pear Leaf Blister Moth	Animalia	Arthropoda	Insecta	Lepidoptera	Lyonetidae
18	*Ditylenchus angustus* (Butler)	Rice Stem Nematode	Animalia	Nematoda	Secernentea	Tylenchida	Anguinidae
19	*Ceroplastes destructor* (Newstead)	Soft Wax Scale	Animalia	Arthropoda	Insecta	Hemiptera	Coccidae
20	*Chilo suppresalis* (Walker)	Asiatic Rice Borer	Animalia	Arthropoda	Insecta	Lepidoptera	Pyralidae
21	Veronicellidae spp. (*V. cubensis*, *V. sloanei*).	Veronicellid Slugs	Animalia	Mollusca	Gastropoda	Stylommatophora	Veronicellidae
22	*Dendrolimus pini* (L.)	Pine-Tree Lappet	Animalia	Arthropoda	Insecta	Lepidoptera	Lasiocampidae
23	*Spodoptera littoralis* (Boisduval)	Egyptian Cottonworm	Animalia	Arthropoda	Insecta	Lepidoptera	Noctuidae
24	*Chalara fraxinea* (T. Kowalski)	Ash Dieback	Fungi	Ascomycota	Ascomycetes	Incertae sedis	Incertae sedis
25	*Monochamus sutor* (L.)	Small White-Marmorated Longhorned Beetle	Animalia	Arthropoda	Insecta	Coleoptera	Cerambycidae
26	*Planococcus minor* (Maskell)	Passionvine Mealybug	Animalia	Arthropoda	Insecta	Hemiptera	Pseudococcidae
27	*Tuta absoluta* (Meyrick)	Tomato Leaf Miner	Animalia	Arthropoda	Insecta	Lepidoptera	Gelechiidae
28	*Dendrolimus punctatus* (Walker)	Masson pine moth	Animalia	Arthropoda	Insecta	Lepidoptera	Lasiocampidae
29	*Nysius huttoni* (White)	Wheat Bug	Animalia	Arthropoda	Insecta	Hemiptera	Lygaeidae
30	*Pieris brassicae* (L.)	Large White Butterfly	Animalia	Arthropoda	Insecta	Lepidoptera	Pieridae
31	*Candidatus* Phytoplasma australiense (R.E. Davis et al.)	Australian Grapevine Yellows	Bacteria	Firmicutes	Mollicutes	Acholeplasmatales	Acholeplasmataceae
32	*Raffaelea quercivora* (Kubono & Shin. Ito)	Japanese Oak Wilt	Fungi	Ascomycota	Ascomycetes	Ophiostomatales	Ophiostomataceae
33	*Monacha* spp. (*M. cantiana*, *M. syriaca*)	Helicid Snail	Animalia	Mollusca	Gastropoda	Stylommatophora	Hygromiidae
34	*Oxycarenus hyalinipennis* (Costa)	Cotton Seed Bug	Animalia	Arthropoda	Insecta	Hemiptera	Lygaeidae
35	*Eudocima fullonia* (Clerck)	Fruit Piercing Moth	Animalia	Arthropoda	Insecta	Lepidoptera	Noctuidae
36	*Thaumatotibia leucotreta* (Meyrick)	False Codling Moth	Animalia	Arthropoda	Insecta	Lepidoptera	Tortricidae
37	*Candidatus* Phytoplasma mali (Seemuller & Schneider)	Apple Proliferation	Bacteria	Firmicutes	Mollicutes	Acholeplasmatales	Acholeplasmataceae
38	*Monochamus saltuarius* (Gebler)	Japanese Pine Sawyer	Animalia	Arthropoda	Insecta	Coleoptera	Cerambycidae
39	*Tetranychus roseus* (Gutierrez)	No common name, a spider mite	Animalia	Arthropoda	Arachnida	Acari	Tetranychidae
40	*Diprion pini* (L.)	Conifer Sawfly	Animalia	Arthropoda	Insecta	Hymenoptera	Diprionidae
41	*Mycosphaerella gibsonii* (H. Evans)	Needle Blight Of Pine	Fungi	Ascomycota	Ascomycetes	Mycosphaerellales	Mycosphaerellaceae
42	*Onopordum acaulon* (L.)	Horse Thistle	Plantae	Magnoliophyta	Magnoliopsida	Asterales	Asteraceae
43	*Paysandisia archon* (Burmeister)	No common name, a palm borer	Animalia	Arthropoda	Insecta	Lepidoptera	Castniidae
44	*Monilia polystroma* (van Leeuwen)	Asiatic Brown Rot	Fungi	Ascomycota	Incertae sedis	Incertae sedis	Incertae sedis
45	*Diabrotica speciosa* (Germar)	Cucurbit Beetle	Animalia	Arthropoda	Insecta	Coleoptera	Chrysomelidae
45	*Xanthomonas oryzae* pv. *oryzae* [(Ishiyama) Swings et al. & *X. oryzae* pv. *oryzicola* (Fang et al.) Swings et al.]	Bacterial Leaf Streak, Bacterial Blight	Bacteria	Proteobacteria	Gammaproteobacteria	Xanthomonadales	Xanthomonadaceae

45	*Harpophora maydis* (Samra, Sabet & Hing.) W. Gams	Late Wilt Of Corn	Fungi	Ascomycota	Ascomycetes	Incertae sedis	Incertae sedis
46	*Cameraria ohridella* (Deschka & Dimić)	Horse Cheetnut Leaf Miner	Animalia	Arthropoda	Insecta	Lepidoptera	Gracillariidae
47	*Panolis flammea* (Denis & Schiffermüller)	Pine Beauty Moth	Animalia	Arthropoda	Insecta	Lepidoptera	Noctuidae
48	*Adoxophyes orana* (Fischer von Röeslerstamm)	Summer Fruit Tortrix Moth	Animalia	Arthropoda	Insecta	Lepidoptera	Tortricidae
49	*Archips xylosteanus* (L.)	Variegated Golden Tortrix	Animalia	Arthropoda	Insecta	Lepidoptera	Tortricidae
50	*Meloidogyne* spp. (*M. citri, M. donghaiensis, M. fujianensis, M. indica, M. jianyangensis, M. minganimica*)	Citrus Root-Knot Nematodes	Animalia	Nematoda	Secernentea	Tylenchida	Heteroderidae

Rank	Scientific Name	Common Name	*Allium* spp. (Onion)	*Apium graveolens* (Celery)	*Arachis* spp. (Peanut)	*Asparagus* spp. (Asparagus)	*Avena* spp. (Oat)	*Brassica* spp.	*Citrus* spp. (Citrus)	*Cucumis* spp. (Cantaloupe)	*Cucumis* spp. (Cucumber)	*Daucus carota* (Carrot)	*Fragaria* spp. (Strawberry)	*Glycine* spp. (Soybean)	*Gossypium* spp. (Cotton)	*Helianthus* spp. (Sunflower)	*Hordeum* spp. (Barley)	*Lactuca* spp. (Lettuce)	*Malus* spp. (Apple)	*Oryza* spp. (Rice)	*Phaseolus* spp. (Bean)	*Prunus dulcis* (Almond)	*Prunus persica* (Peach)	*Pyrus* spp. (Pear)	*Solanum lycopersicum* (Tomato)	*Solanum tuberosum* (Potato)	*Sorghum* spp. (Sorghum)	*Triticum* spp. (Wheat)	*Vitis* spp. (Grape)	*Zea* spp. (Corn)	*Pinus* spp. (Pine)	Other Softwood Trees*	Soft Hardwood Trees*	Hardwood Trees*	Pest Commodity Total
14	*Achatina fulica*	Giant African Snail	■		▲			▲	■	▲	▲	■		■	■	■		■	■	■	■				■	■	■		■	■			■	■	21
48	*Adoxophyes orana*	Summer Fruit Tortrix Moth			■				■				■	■	■				▲				▲	▲					■				■	■	11
1	*Agrilus biguttatus*	Oak Splendor Beetle																															■	▲	2
49	*Archips xylosteanus*	Variegated Golden Tortrix							■										■				■	■								■	■	■	7
46	*Cameraria ohridella*	Horse Chestnut Leaf Miner																																▲	1
31	*Candidatus* Phytoplasma australiense	Australian Grapevine Yellows								■	■		▲								■			■			■			▲			■	■	9
37	*Candidatus* Phytoplasma mali	Apple Proliferation																	▲				■	■						■				■	5
19	*Ceroplastes destructor*	Soft Wax Scale							▲										■					■									■	■	5
10	*Ceroplastes japonicus*	Japanese Wax Scale							▲										■				■	■						■			■	■	7
24	*Chalara fraxinea*	Ash Dieback																															■		1
20	*Chilo suppressalis*	Asiatic Rice Borer						■												▲						■		▲	■		▲				6
2	*Cronartium flaccidum*	Scots Pine Blister Rust																														▲			1
22	*Dendrolimus pini*	Pine-Tree Lappet																														▲	■		2
28	*Dendrolimus punctatus*	Masson pine moth																														▲			1
7	*Dendrolimus sibiricus*	Siberian Silk Moth																														▲	▲	■	■	4
7	*Dendrolimus superans*	Sakhalin silk moth																														▲	▲	■	■	4
45	*Diabrotica speciosa*	Cucurbit Beetle	■	■	▲	■	■	■	■	■	■	■	■	▲	■	■		■	■	■	■			■		■	▲	■	▲	■	▲				■	26
18	*Ditylenchus angustus*	Rice Stem Nematode																		▲																1
40	*Diprion pini*	Conifer Sawfly																														▲				1
35	*Eudocima fullonia*	Fruit Piercing Moth							▲	■	■		■						■					■	■	▲				■			■	■	11
45	*Harpophora maydis*	Late Wilt Of Corn																													▲				1
3	*Helicoverpa armigera*	Old World Bollworm	■		▲		■	■	▲	■	■	■	■	▲	▲	▲	▲	■	■	■	▲			■		▲	▲	▲	▲		▲	▲	■	■	27
17	*Leucoptera malifoliella*	Pear Leaf Blister Moth																	▲					■	▲								■	■	5
15	*Lymantria mathura*	Rosy Moth																	■						■							■	■	▲	▲	6
16	*Massicus raddei*	Mountain oak longhorned beetle																																▲	1
50	*Meloidogyne spp. (M. citri, M. donghaiensis, M. fujianensis, M. indica, M. jianyangensis, M. mingnanica)*	Citrus Root-Knot Nematodes							▲																	■										2
33	*Monacha spp.*	Helicid Snail						■							■			■				■														4
44	*Monilia polystroma*	Asiatic Brown Rot																	▲				▲	▲	▲											4
38	*Monochamus saltuarius*	Japanese Pine Sawyer																														▲	▲			2
25	*Monochamus sutor*	Small White-Marmorated Longhorned Beetle																														▲	▲		■	3
41	*Mycosphaerella gibsonii*	Needle Blight Of Pine																														▲				1
29	*Nysius huttoni*	Wheat Bug					■	■					■			■	■	■							■				■							8
42	*Onopordum acaulon*	Horse Thistle																																		0
9	*Otiorhynchus sulcatus*	Wingless Weevil																■																		1
34	*Oxycarenus hyalinipennis*	Cotton Seed Bug													■				■					■	■	■				■	■			■	■	9
47	*Pancalis flammea*	Pine Beauty Moth																														▲	▲	▲		3
43	*Paysandisia archon*	No common name, a palm borer																																■		1
12	*Phytophthora alni*	Alder Root and Collar Rot																																▲		1
30	*Pieris brassicae*	Large White Butterfly	■					▲						▲																						3
26	*Planococcus minor*	Passionvine Mealybug		■	■	■		■	■	■	■		■	■	■	■				■	■				■	■	■			■	■		■	■	21
1	*Platypus quercivorus*	Oak Ambrosia Beetle																																■	▲	2
32	*Raffaelea quercivora*	Japanese Oak Wilt																																	▲	1
13	*Ralstonia solanacearum* race 3 biovar 2	Bacterial Wilt			■										■							■				▲	▲									5
23	*Spodoptera littoralis*	Egyptian Cottonworm	▲	■	▲	■		■	▲	▲	▲	▲	■	▲	▲	▲		▲	■	▲	▲				■	▲	▲	▲	▲	▲	▲	■	■	■	■	28
S	*Spodoptera litura*	Cotton Cutworm	▲	■	▲	■		▲	▲	■	■	■	▲	▲	▲	▲		■	▲	▲	▲				■	▲	▲	▲	■	▲	▲		■	▲	■	28
39	*Tetranychus roseus*	No common name, a spider mite																																		0
36	*Thaumatotibia leucotreta*	False Codling Moth							▲						▲							■		▲		■		▲		■	▲			■	▲	10

Appendix E: USDA Plant Hardiness Zone Map
(see http://www.usna.usda.gov/Hardzone/ushzmap.html for interactive map)

The preceding map was produced in 1990 by the US Department of Agriculture (USDA). This version shows in detail the lowest temperatures that can be expected each year in the United States, Canada, and Mexico.



These temperatures are referred to as "average annual minimum temperatures" and are based on the lowest temperatures recorded for each of the years 1974 to 1986 in the United States and Canada and 1971 to 1984 in Mexico. The map shows 10 different zones, each of which represents an area of winter hardiness for the plants of agriculture and our natural landscape. It also introduces zone 11 to represent areas that have average annual minimum temperatures above 40 F (4.4 C) and that are, therefore, essentially frost free. Actual temperature ranges for each zone are given below. Zones

2-10 in the map have been subdivided into light- and dark-colored sections (a and b) that represent 5 F (2.8 C) differences within the 10 F (5.6 C) zone.

Zone	Fahrenheit	Celsius
1	Below -50 F	Below -45.6 C
2a	-50 to -45 F	-42.8 to -45.5 C
2b	-45 to -40 F	-40.0 to -42.7 C
3a	-40 to -35 F	-37.3 to -39.9 C
3b	-35 to -30 F	-34.5 to -37.2 C
4a	-30 to -25 F	-31.7 to -34.4 C
4b	-25 to -20 F	-28.9 to -31.6 C
5a	-20 to -15 F	-26.2 to -28.8 C
5b	-15 to -10 F	-23.4 to -26.1 C
6a	-10 to -5 F	-20.6 to -23.3 C
6b	-5 to 0 F	-17.8 to -20.5 C
7a	0 to 5 F	-15.0 to -17.7 C
7b	5 to 10 F	-12.3 to -14.9 C
8a	10 to 15 F	-9.5 to -12.2 C
8b	15 to 20 F	-6.7 to -9.4 C
9a	20 to 25 F	-3.9 to -6.6 C
9b	25 to 30 F	-1.2 to -3.8 C
10a	30 to 35 F	1.6 to -1.1 C
10b	35 to 40 F	4.4 to 1.7 C
11	above 40 F	above 4.5 C

Appendix F: 2008 Exotic Pest Detection Survey Order Form

2008 EXOTIC PEST DETECTION SURVEY ORDER FORM

Please fill out an order form for each state in which traps will be placed. States with more than one "ship-to" address should fill out a separate order form for each address.

If there is any trapping to be done within a state, an order form must be submitted to Otis **even if that state is not ordering any new lures**. Simply fill out the "number traps to be placed" column and put a "0" in the "number dispensers requested" column. This will provide the information to compile the annual report.

Lures for Khapra Beetle (*Trogoderma granarium*), European Cherry Fruit Fly (*Rhagoletis cerasi*) and Swede midge (Contarinia nasturtii) are to be purchased from commercial sources, but again, please indicate on the order form any trapping to be done for these species. All other lures are provided by the Otis Pest Survey, Detection and Exclusion Laboratory. Traps and liners are to be purchased locally or regionally.

If you have any questions about exotic pest survey trapping, please consult your **Exotic Pest Detection Manual**, and if there are remaining questions, please call or email Natalie Leva.

All orders for pheromone dispensers should be sent to:

Natalie Leva
USDA, APHIS, PPQ
Otis Pest Survey, Detection and Exclusion Lab
Building 1398
Otis ANGB, MA 02542

Tel: 508-563-9303 x 255
Fax: 508-564-4398
E-mail: natalie.m.leva@aphis.usda.gov

All orders should be received in the Otis lab by January 25, 2008.

Please use a Street Address, not a P.O. Box, for the Ship to Address and make sure you fill out all * fields, thank you.

State in which trap will be placed:	Date order placed:

Name: _____

Organization: _____

Address 1 _____

Address 2 _____

Phone _____

Email

Ship to Address

*Name: _____

*Organization _____

*Address 1 _____

Address 2 _____

*Phone _____

Email

Requested Ship Date: _____

Other Requests: _____

THE OTIS LAB DOES NOT SUPPLY ANY TRAPS OR TRAP PARTS

FOR OTIS USE ONLY:	
Shipment Date:	
Shipment Date:	☐
Shipment Complete:	☐

*Name: _____ *State: _____

Common Name / Species	Code	# Traps to be Placed	# Traps Ordered	# Trap Liners Ordered	Pheromone Dispenser Type	Frequency of Dispenser Replacement	# Dispensers Requested (Lures)
Leek Moth *Acrolepiopsis assectella*	LEM				Rubber septa	2 weeks	
Summer fruit tortrix moth *Adoxophyes orana*	ADOX				Rubber septa	12 weeks	
Apple Tortrix *Archips fuscocupreanus*	AF				Rubber septa	4 weeks	
Silver Y moth *Autographa gamma*	AG				Rubber septa	4 weeks	
Peach Fruit Moth *Carposina niponensis*	CN				Polycap	4 weeks	
Asiatic rice borer *Chilo suppressalis*	CS				Rubber septa	4 weeks	
Maize borer *Chilo partellus*	CP				Rubber septa	4 weeks	
Tomato Looper *Chrysodeixis chalcites*	TL				Rubber septa	4 weeks	
False codling moth *Cryptophlebia leucotreta*	FCM				Rubber septa	8 weeks	
Plum fruit moth *Cydia funebrana*	PFM				Rubber septa	4 weeks	
**Siberian Moth *Dendrolimus superans sibiricus*	SM				Rubber septa	4 weeks	
Cherry bark tortrix *Enarmonia formosana*	CBT				Rubber septa	4 weeks	
Light brown apple moth *Epiphyas postvittana*	LBAM				Rubber septa	4 weeks	
European grape berry moth *Eupoecilia ambiguella*	EA				Rubber septa	6 weeks	
Old World Bollworm *Helicoverpa armigera*	HA				Rubber septa	4 weeks	
Pear leaf blister moth *Leucoptera malifoliella*	PLBM				Rubber septa	10 weeks	
Grape vine moth *Lobesia botrana*	LB				Rubber septa	3 weeks	
*Rosvy moth *Lymantria mathura*	RM				String	12 weeks	
Nun moth *Lymantria monacha*	NM				Laminate	12 weeks	
Cabbage moth *Mamestra brassicae*	MB				Polycap	12 weeks	
(No Common Name) *Pandemis heperana*	PH				Rubber septa	4 weeks	
European cherry fruit fly *Rhagoletis cerasi*	RC				Polycap	2 weeks	OTIS DOES NOT SUPPLY
Egyptian cottonworm *Spodoptera littoralis*	ECL				Laminate	12 weeks	
Rice cutworm (cotton leafworm) *Spodoptera litura*	CL				Laminate	12 weeks	
Khapra beetle *Trogoderma granarium*	KB				Rubber septa	4 weeks	OTIS DOES NOT SUPPLY
Cherry ermine moth *Yponomeuta padellus*	CEM				Rubber septa	12 weeks	
Apple ermine moth *Yponomeuta malinellus*	AEM				Red Rubber septa	12 weeks	
***Swede midge *Contarinia nasturtii*	SWM				Polycap	12 weeks	OTIS DOES NOT SUPPLY

*Pending Lure Availability
***Jackson Trap

**Requires modified milk carton GM traps, modification available upon request

2008 Exotic Pest Order Form

Plastic Bucket Trap Protocol

The plastic bucket trap is a long-lasting insect trap used in conjunction with a lure to monitor or detect various species of moths. The plastic bucket trap is the preferred trap for some moth species as it is able to catch large numbers of moths without damaging some of their identifying characters. The trap has four parts: 1) lid, 2) lure basket with cap, 3) funnel, and 4) bucket. The trap is available in various color combinations. For PPQ programs, the trap consists of a green lid, yellow funnel, and white bucket. Fig. 1 is a photograph of a trap cut in half.

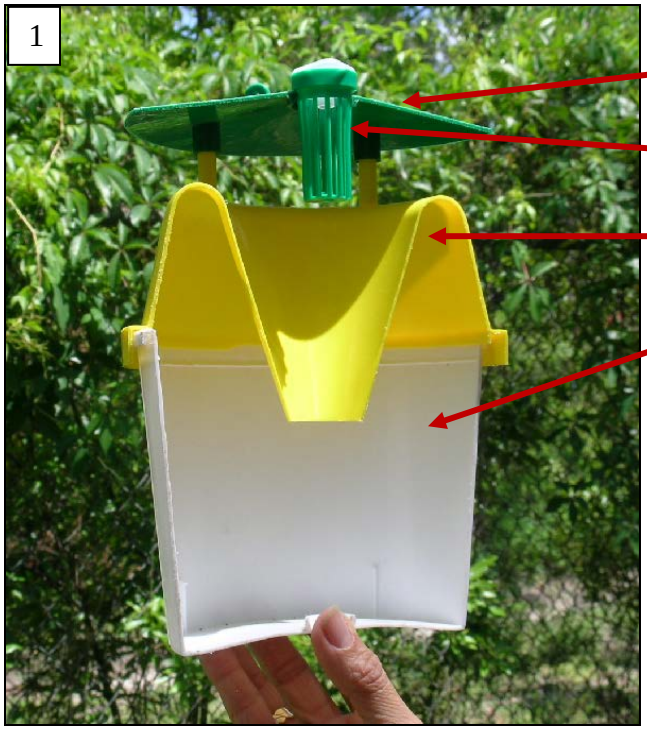


Fig. 1. Plastic bucket trap cut in half to show its interior.

Follow the steps below to prepare the bucket traps for use in the field.

1. Pheromone

Unwrap a pheromone lure and place it inside the lure basket. Handle lures with gloves (see Fig. 4). Close the basket with a cap and insert the basket through the circular opening on the center of the lid (Fig. 2). If the cap no longer snaps snuggly into the trap lid opening, secure it with a piece of tape.

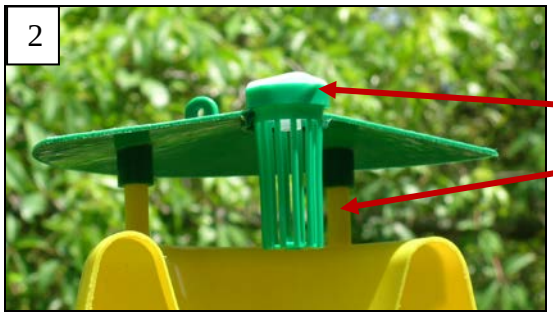
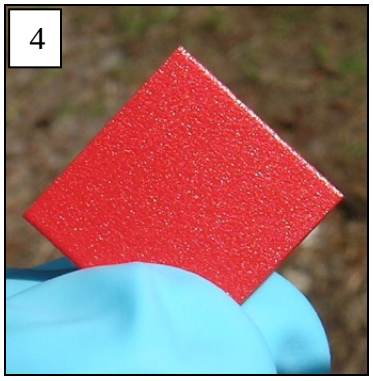


Fig. 2. Lure basket with cap inserted through center of lid.

The synthetic pheromone is embedded in a small rubberized square (as seen in the photos below) or septum (similar to a pencil eraser). If the lure is flat and small (Figs. 3 and 4) you may attach the lure to a small paper clip and fold the clip so that the lure does not fall out of the basket. If a lure basket is not available, attach the lure to a cork with a pin and place the cork in the lid's opening. Always carry extra corks.

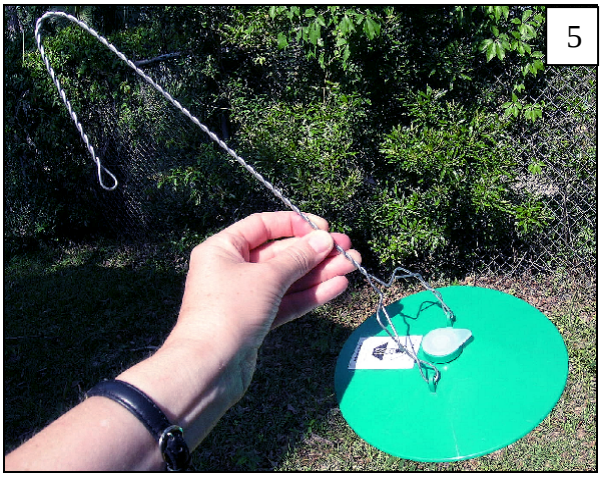


Figs. 3 and 4. Lure made of a small rubberized square with embedded synthetic pheromone chemicals.

When not in use, the lures should be stored, unwrapped, in a freezer not used for food or drinks. MSDS documents for the pheromones to be used should be available and should be read.

2. Handle

Attach a wire handle to the lid through its two loops, as shown in the photos below (Figs. 5 and 6). A wire handle is usually included with each purchased trap. If a handle is not included, is lost, or is damaged and needs to be replaced, make one with a 12-inch long wire or with string, but the latter does not last as long as the wire.



Figs. 5 and 6. Wire handle attached to trap's lid.

3a. Sponge

Place a dry cellulose sponge in the bottom of the trap, as shown in Fig. 7. The sponge will absorb rainwater (except for extremely heavy amounts) that may enter the trap, keeping the moths somewhat dry.

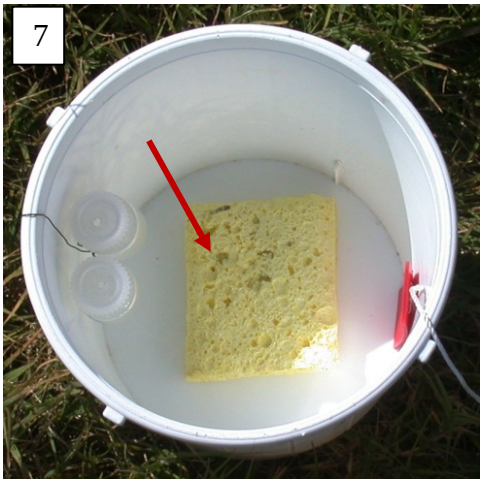


Fig. 7. Cellulose sponge inside the trap.

3b. Wire screen

Alternatively, the bottom part of the trap, the bucket, requires two modifications. Drill two to four drain holes in the bottom (see Fig. 8). If water remains in the trap, the killing agent (the pesticide) can spoil; in addition, the trapped insects may decay, making identification impossible.

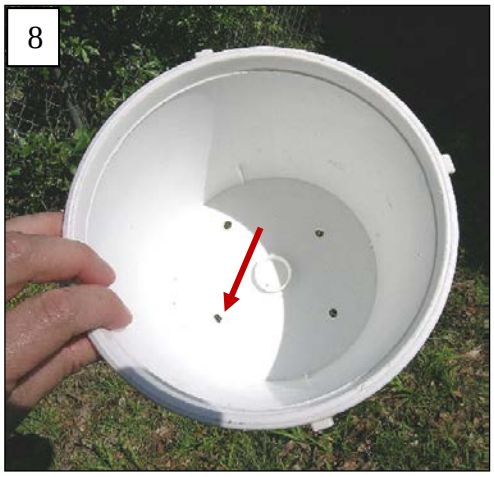
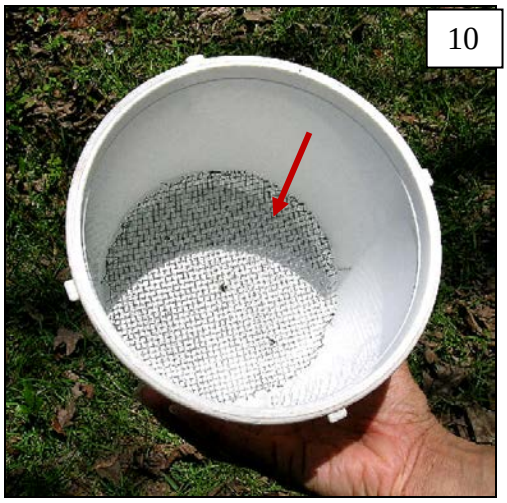


Fig. 8. Bucket with four drilled holes.

Then, add a wire screen slightly larger than the bucket bottom's inside diameter (Figs. 9 and 10). The screen keeps the pesticide strip(s) and the moths from getting too wet from rainwater accumulated in the trap. Prepare a cardboard template for long term use. Cut the wire mesh with metal-cutting scissors.



Figs. 9 and 10. Metal wire screen inside the bucket.

4. Insecticidal strips

Place two insecticidal strips (Figs. 11 and 12), which kill the moths that enter the bucket. The active ingredient in the strips is Dichlorvos, also known as DDVP and Vapona. The strips should be handled with gloves. Read and have available the MSDS documents for this product. Store unopened strips in a freezer not used for food or drink. Rain, wind, high heat or an abundance of captured moths may reduce its potency from 3 to 4 weeks to a week or less. If using only one kill strip, change it every 2 weeks.

Figs. 11 and 12. Pesticide strips.

5. Label the trap

Attach a rain-proof printed label (see Fig. 13) or handwrite a note with a water-proof black marker on the bucket trap. It should indicate that the trap belongs to a state or a PPQ program. Include a phone number in case someone has concerns or questions about the trap.

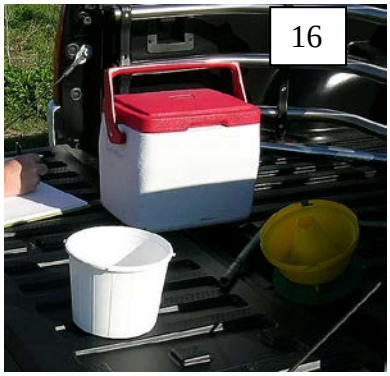
Fig. 13. Label on the trap's lid.

6. Placement of traps

The traps function best when placed in the open, away from foliage, as illustrated on Fig. 14. When hung under foliage, the 3-dimensional shape of the pheromone plume (chemical in the air) is disrupted and the effectiveness of the trap is much reduced. Hang the traps from such places as greenhouse roofs or in the open using metal rods (see Fig. 14) or other materials.

Figs. 14. Trap set away from foliage, in open field.

In the field, transfer the caught moths to labeled zip-loc bags and store them in a cooler (Figs. 15 and 16). Place them overnight in a freezer to kill any surviving specimens.



Figs. 15 and 16. Moths placed in a ziploc bag and stored in a cooler.

Prior to shipping, screen the samples. Remove any moth vastly different from the target and all other arthropods (beetles, flies, spiders). Write on PPQ Form 391 the approximate number of moths being submitted. Place an absorbent paper, such as a piece of a paper towel, inside each plastic bag to reduce moisture and to pad the specimens for their protection. The specimens should be well padded inside a box to prevent the specimens from being crushed or otherwise damaged. If longer-term storing is necessary, freezing works best, but refrigeration is acceptable as well.

The general recommendation for maintenance of the plastic bucket traps is to wash them occasionally with soap and water to keep them clean, and to store them indoors, or at least protected from sun, rain and dust. Keep the wire handle and the wire screen in good repair. The traps can be used multiple times and for multiple species since the chemicals degrade quickly in outdoor conditions. These traps usually last more than 5 years.

This protocol is designed to aid in the detection of exotic moths of concern by giving instructions on how to use generic plastic bucket traps. All photos were taken by J. Brambila and R. Meagher. These instructions are primarily based on work by R. Meagher.

This aid was prepared by Julieta Brambila (USDA/APHIS/PPQ Eastern Region), Lisa Jackson (USDA/APHIS/PPQ/CPHST), and Dr. Robert L. Meagher (USDA/ARS/CMAVE), on April 2010.