Cotton Commodity-based Pest Survey

Cooperative Agricultural Pest Survey (CAPS)
November 2011

Cover Images: Michelle Walters – USDA –APHIS-PPQ-CPHST Phoenix, AZ, http://www.cottonman.com/cotton_bolls.htm, and http://www.cotton-bales.com/.

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Draft Log

November 2011 – Draft sent for CPHST review

December 2011 – Draft sent for CAPS review

March 2012 – Final draft posted on CAPS Resource and Collaboration site

August 2016 – Removed outdated maps, updated mapping information

Introduction to Reference

History of Commodity-Based Survey

The CAPS community is made up of a large and varied group of individuals from federal, state, and university organizations who use federal (and other) funding sources to survey for and (in some cases) diagnose exotic and invasive plant pests. By finding pests early, eradication efforts will likely be less expensive and more efficient. For more information on 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. From this list each year, states 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 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 may 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 more efficiently used 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 with developing a commodity-based survey strategy in support of the CAPS program. Two types of end products are 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 (Appendix C and D) and the Select Agent list (http://www.aphis.usda.gov/programs/ag_selectagent/ or http://www.aphis.usda.gov/programs/ag_selectagent/ag_bioterr_toxinlist.shtml). The AHP prioritized pest lists for FY 11' and FY 12' are also given in Appendices C and D. Additional pests may be added if they are cited in scientific 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 or industry. 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 annual surveys.

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 that are relevant to the state in terms of risk. 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 soybean, corn, grape, potato, small grains, stone fruit, oak forests, and pine forests.

Cotton Commodity Survey Reference

The *Cotton Commodity-based Survey Reference* (CSR) is a companion document to the *Cotton Commodity-based Survey Guidelines* (CSG). Both documents are intended to be tools to help survey professionals develop surveys for exotic pests of cotton. The *Cotton CSR* is a collection of detailed data sheets on exotic pests of cotton. 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.

In contrast, the *Cotton 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 *Cotton 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 *Cotton 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 Cotton 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 generally (but not always) 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 B has a listing of NIS and Domestic Identifier contact information.

Organisms Included in the Cotton Survey Reference

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

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

2. Organisms are then divided by their pest status on cotton [*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 2012 CAPS National Survey Guidelines (http://caps.ceris.purdue.edu/webfm_send/1063; http://pest.ceris.purdue.edu/services/napisquery/query.php?code=cam2012) 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.

Introduction to Cotton

In 2010, 10,773,000 acres were planted to cotton in the United States. The United States produces over 20% of the world's cotton and exports more than $3 billion annually. Cotton is an important fiber, oil, and seed crop. Cotton fiber is used in apparel (64%), home furnishings (28%), and industrial products (8%). Cottonseed is used mainly in livestock feed, and cottonseed oil is used in food products. In 2010, the United States produced 18.2 million 480 lb. bales of cotton and exported 14.5 million 480 lb. bales. Cotton is grown in 17 states across the "Cotton Belt" in the southern United States: Alabama, Arizona, Arkansas, California, Florida, Georgia, Kansas, Louisiana, Mississippi, Missouri, New Mexico, North Carolina, Oklahoma, South Carolina, Tennessee, Texas, and Virginia (Fig. 1).

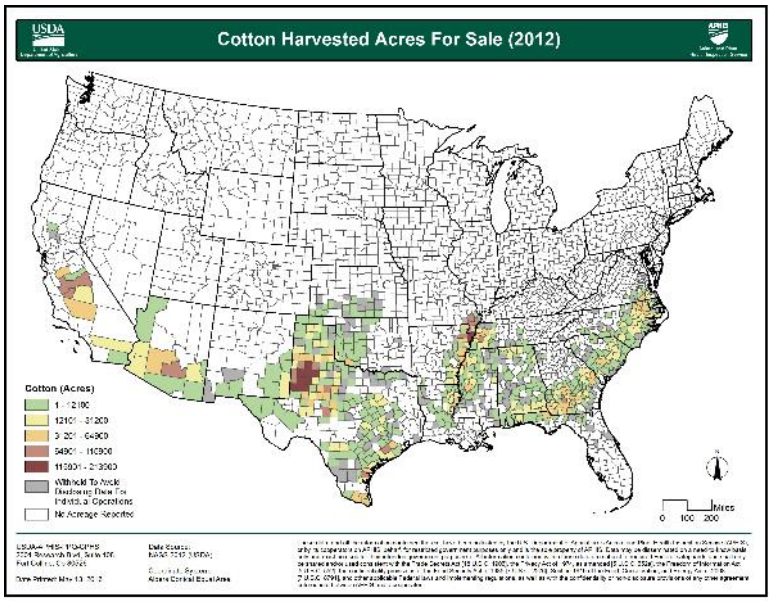

Figure 1. Cotton (*Gossypium* spp.) acreage by county in the continental United tates. Photo courtesy of USDA-APHIS-PPQ-CPHST

Growth and Development of Cotton

Cotton growth and development of roots, leaves, branches, squares (flower buds), flowers, and bolls follows a definite sequence. Two species are grown in the United States, upland (*Gossypium hirsutum*) and American Pima (*G. barbadense*); the pattern of development applies to both species, with a few differences. Cotton is a perennial, but is grown as an annual crop.

Germination
Growth begins when the cotton seed absorbs water from the soil. At germination, the radicle grows downward and becomes the taproot. The hypocotyl elongates and arches until it pushes through the soil surface. By the time the hypocotyl emerges, the cotyledons are producing energy for early growth.

Roots
The taproot grows to a depth of about 6 feet (1.83 m). By the time the seedling emerges from the soil, the root is about 10 inches (25.4 cm) deep and forming lateral branches. Most lateral roots occur in the upper 2 feet (0.61 m) of soil. The root system is well established by flowering, and little root growth occurs after fruit set.

Main stem and leaves
As the main stem develops from the apical meristem, it forms nodes from which main stem leaves and branches grow. Each node above the cotyledons bears a leaf and one branch. Leaves and branches are situated in a spiral three fifths of a turn around the main stem.

Branches
Branches develop from buds at each node. Lower branches are vegetative; the first fruiting branch occurs at the fifth node or higher. Fruiting branches grow in a series of segments; each segment grows from an axillary bud of the previous segment. This growth pattern gives fruiting branches a ‘zigzag’ appearance.

Squares (Flower buds)
Squares consist of concentric whorls. Until the flower opens, only the outer whorl is visible (Fig. 2A). The outer whorl is made up of three bracts, which are tightly closed around the bud. If the square is damaged, these bracts will flare open and turn yellow before the square drops from the plant.

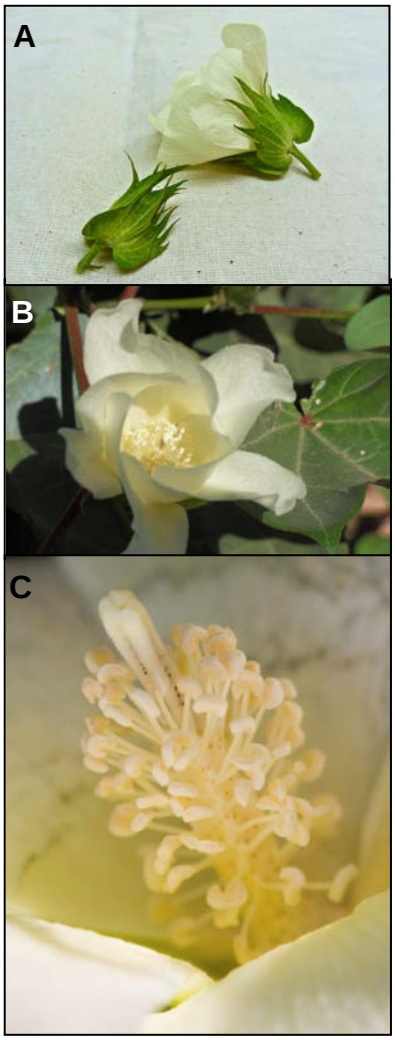

Figure 2. (A) Upland cotton square and flower, (B) Upland cotton flower, (C) Pistil of an upland cotton flower. Photos courtesy of Lydia Brown – University of Arizona (A&B) and Stephen Ausmus-USDA ARS (C).

Flowers
Cotton flowers usually bloom for just one day. In upland cotton, flowers are creamy white the first day (Fig 2), but turn red as they close and wilt the second day. Pima cotton has yellow flowers with a purple spot near the base. Like upland cotton flowers, Pima flowers fade in a day but do not change color.

At the base of the flower is the pistil (Fig. 2C). Numerous stamens surround the pistil. Each stamen bears a pollen-producing anther. The ovary at the base of the pistil is made up of two or more seed chambers called locules or “locks”. Upland cotton flowers usually have four or five locks; Pima flowers usually have three. Locks house ovules, which, when fertilized, produce seed and lint.

Bolls, seed, and lint

Cotton flowers are self-pollinating and self-compatible and do not require pollinators. Outcrossing is not necessary, but it is possible and results in viable seeds. Wind movement, honey bees, native bees, and other insects can contribute to pollination. While the seed develops, cotton fibers develop from the outermost cells of the seed coat. Each fiber takes 18 to 21 days to elongate fully. At maturity, the lint dries. Shortly after, the boll dries and splits open, exposing the lint and seeds.

References:

EPA. 2009. Major crops grown in the United States- Ag 101-Agriculture. http://www.epa.gov/oecaagct/ag101/cropmajor.html.

University of California. 1984. Integrated pest management for cotton in the western region of the United States. University of California, Division of Agriculture and Natural Resources Publication 3305: 9–12.

USDA-NASS (United States Department of Agriculture, National Agricultural Statistics Survey). 2010. Cotton area harvested, yield, and production by type—States and United States: 2009 and forecasted December 1, 2010.

Arthropods

Primary Pests of Cotton (Full Pest Datasheet)

Anthonomus grandis

Scientific Name
Anthonomus grandis Boheman

Synonyms:
Anthonomas grandis grandis

Common Name(s)
Boll weevil and southeastern boll weevil

Type of Pest
Weevil

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

Reason for Inclusion In Manual
PPQ Program Pest

Pest Description
There are three forms of *A. grandis* found in the United States, the Mexican boll weevil (intermediate), thurberia weevil (*A. grandis thurberiae*), and the southeastern boll weevil (*A. grandis grandis*) (EPPO, n.d.). This datasheet deals specifically with *A. grandis grandis* unless otherwise stated.

Figure 1. Larva of *Anthonomus grandis grandis* in cotton square. Image courtesy of Alton N. Sparks, Jr., University of Georgia, www.bugwood.org.

Figure 2. Adult boll weevil, *Anthonomus grandis grandis*. Image courtesy of Anyi Mazo-Vargas. University of Puerto Rico, www.bugwood.org.

Eggs: Eggs are slightly elliptical, opaque, and 1 mm (0.04 in.) long (Leigh et al., 1996).

Larvae: Larvae (Fig. 1) have a distinct head, are C-shaped, and cream colored with a tan head (Leigh et al., 1996) and legless (Matthews, 1989). Larvae can grow to 9.5 mm (0.375 in.) (Leigh et al., 1996).

Pupae: Pupae are similar to the adult form as their snout, legs, and wings are visible through the cuticula (Parrott et al., 1970; Roach, 1973; Leigh et al., 1996).

Adults: Adults (Fig. 2) are 3.2 to 8.5 mm (0.125 to 0.33 in.) in length. Colors of adults can range from tan to dark gray and sometimes dark brown (Parencia, 1978). Black colored weevils may occur, but these are not common (Bartlett, 1967; McGovern et al., 1974). The upper side of the elytra is pale with fine parallel lines (EPPO, n.d.). Adults are covered densely with gray pubescence (Matthews, 1989). Adults have a curved snout and the femur of the front leg has a large double tooth (Leigh et al., 1996).

Biology and Ecology:
Development is completed in 2.5 to 3 weeks when conditions are favorable (Smith and Harris, 1994) with the average development time taking from 20 to 80 days (Loftin, 1946). Higher temperatures and humidity increase the rate of development (Smith and Harris, 1994). Up to seven generations a year may develop in the extreme southern parts of the United States cotton belt (Smith and Harris, 1994).

Adults that have recently emerged prefer feeding on the pollen of open flowers. After a few days, the elongated rostrum allows adults to penetrate the flower bud (also called the square) (Wagner et al., 1996).

Figure 3. Damage caused by *Anthonomus grandis grandis*. An egg-laying puncture may be seen on the upper right edge of the square on the left; a typical feeding puncture appears on the square on the right. Image courtesy of Clemson University - USDA Cooperative Extension Slide Series, www.bugwood.org.

In spring, females usually lay eggs on flower-buds singly unless populations are high and there are few buds (Smith and Harris, 1994) in which case, eggs may then be laid on young bolls (Parencia, 1978). Oviposition occurs during the daytime soon after mating (EPPO, n.d.). Females can lay from 100 to 300 eggs (Matthews, 1989). Eggs hatch in three to five days. Larvae bore into buds or bolls where they feed for seven to twelve days (Smith and Harris, 1994). Weevils take longer to develop in bolls versus squares (Loftin, 1946). Larvae go through three instars (Matthews, 1989). The pupal stage then lasts 3 to 5 days before adults emerge (Smith and Harris, 1994). Females can begin laying eggs three to four days after emergence (Cross, 1983). Adults make deep punctures in which the female lays a single egg (Fig. 3). The wound is then sealed with frass and a mucous secretion (Smith and Harris, 1994).

New generations will continue to occur until the host plant is killed by cold weather (Parencia, 1978). Adults then hibernate, staying in the surface litter and surrounding areas around the crop (Smith and Harris, 1994). Diapausing adults prefer leaf litter over

well-drained soil under deciduous trees (Matthews, 1989). A high mortality (95%) of hibernating adults is common (EPPO, n.d.).

Figure 4. Adult *Anthonomus grandis grandis* damaging a bud (Image courtesy of Clemson University - USDA Cooperative Extension Slide Series, www.bugwood.org).

Symptoms/Signs

Both feeding and oviposition cause damage to the plant (Smith and Harris, 1994). Both males and females make feeding punctures while feeding (Parencia, 1978). Yellowish frass may be present around the feeding area (Leigh et al., 1996).

When initially attacked, a small puncture (either for oviposition or feeding) can be seen on the side of the flower bud (Fig. 3 & 4).

With oviposition wounds, the bracts around plant buds will flare and turn yellow. Damaged buds and young bolls will drop to the ground after a few days. Large bolls may not drop but can be invaded by microorganisms. Infested locs, the cotton segments of the boll, produce little if any cotton and the quality is inferior (Smith and Harris, 1994). Bolls are hollowed out as the larvae feed (Leigh et al., 1996).

Several larvae may develop in a single boll when food is scarce; this can lead to the entire boll being ruined (Loftin, 1946).

Larvae can be found in holes in the lint when conducting visual surveys (Leigh et al., 1996).

Pest Importance

This species initially spread from Mexico into the southern tip of Texas around 1892 (Burke et al., 1986). *A. grandis grandis* moved across the cotton production area quickly, reaching the Atlantic seaboard of Georgia in 1916 and close to the northern limits of cotton production by 1922 (Loftin, 1946). During the first few years, losses in newly invaded areas ranged from one-third to one-half of their cotton yields; this threatened to ruin the cotton industry, which at the time made up the majority of southern agriculture and industry (Loftin, 1946).

The quick movement coupled with the South's dependence on cotton led to serious consequences: bankrupted farmers, merchants, and bankers, deserted farms and homes, and demoralized laborers and tenants (Loftin, 1946). Loftin (1946) sums up its

impact by stating that *A. grandis grandis* “affected both the economic and social welfare of more Americans than any other insect”.

According to Smith and Harris (1994) this is the most costly insect pest of cotton in the United States, with yield losses over 8% annually. Control of this pest adds cost to cotton production and is estimated to average $75 million annually in the United States (Smith and Harris, 1994). *A. grandis grandis* has cost American cotton producers more than $15 billion in yield losses and control since its introduction (National Cotton Council, n.d.).

Since its discovery in the United States, research has been conducted, first to determine how to slow the spread and later to determine how to eradicate and control the pest (Loftin, 1946). The impact of this pest has also led to the development of new varieties of cotton that were more resistant to *A. grandis* attacks (Loftin, 1946).

The serious impacts of this pest led to the National Cotton Council requesting increased research and technology development from the United States to aid in the eradication of *A. grandis* (Smith and Harris, 1994). This eventually led to an eradication trial located in northeast North Carolina and southern Virginia towards the eastern extremity of the cotton belt (Smith and Harris, 1994). This trial was successfully completed in 1980 and included:

- Late season insecticide treatments,
- Pheromone traps to monitor populations,
- Diflubenzuron applications when needed,
- Sterile weevil releases,
- Defoliant applications to destroy food and breeding sites before stalk destruction,
- Stalk destruction soon after harvest, and
- Monitoring for other pests (primarily bollworms) and treatments when needed (Smith and Harris, 1994).

The successfulness of the program led to its spread to other parts of the United States, including North and South Carolina (1983 to 1985); Arizona, southern California, and northwestern Mexico (beginning in 1985); and Georgia, Florida, and portions of Alabama (beginning in 1986) (Smith and Harris, 1994). As of 2009, it has been successfully eradicated from Alabama, parts of Arkansas, Arizona, California, Florida, Georgia, Kansas, Kentucky, parts of Louisiana, Mississippi, Missouri, New Mexico, North Carolina, Oklahoma, South Carolina, parts of Tennessee, parts of Texas, and Virginia (Grefenstette, 2009).

Detailed information on the eradication of *A. grandis grandis* can be found in (King et al., 1996).

Known Hosts

Principle plant hosts for *A. grandis* are found in four closely related genera within the Malvaceae family: *Cienfuegosia*, *Gossypium*, *Hampea*, and *Thespesia* (Cross et al., 1975).

Major Hosts: *Cienfuegosia affinis* (fly mallow), *C. drummondii* (yellow flymallow), *C. rosei* (fly mallow), *Gossypium barbadense* (Creole cotton; wild host), *G. davidsonii* (Davidson's cotton), *G. harknessii* (San Marcos hibiscus), *G. hirsutum* (upland cotton), *G. laxum*, *G. lobatum*, *G. thurberi* (Thurber's Cotton), *Hampea nutricia*, *H. rovirosae*, and *Thespesia populnea* (Portia tree) (Cross et al., 1975).

Minor Hosts: *Cienfuegosia digitata* (fly mallow), *C. heterophylla* (variable leaf flymallow), *C. hildebrandtii* (fly mallow), *C. yucatanensis* (Yucatan flymallow), *Gossypium areysianum*, *G. armourianum*, *G. gossypioides*, *G. klotzschianum*, *G. longicalyx*, *G. raimondii*, *G. robinsonii*, *G. somalenses*, *Hibiscus syriacus* (rose of Sharon; not a good host), *Pseudabutilon lozani* (Lozano's false Indianmallow), *Sphaeralcea angustifolia* (copper globemallow), and *Thespesia lampas* (common mallow) (Cross et al., 1975; Matthews, 1989; Smith and Harris, 1994; EPPO, n.d.).

Marginal hosts: *Cienfuegosia heterophylla*, *Hibiscus syriacus*, *Pseudabutilon lozani*, and *Sphaeralcea angustifolia* (Cross et al., 1975).

Adults may feed on the following: *Abutilon giganteum* (Indian mallow), *A. hirtum* (Florida Keys Indian mallow), *Hibiscus* (*Abelmoschus*) *esculentus* (okra), *H. mutabilis* (Dixie rosemallow), *H. rosa-sinensis* (shoeblackplant), *Malvaviscus drummondii* (wax mallow), *Sphaeralcea ambigua* (desert globemallow), *Sphaeralcea coulteri* (Coulter's globemallow), *Sphaeralcea emoryi* (Emory's globemallow), *Wissadula contracta*, and *Sida* spp. (fanpetals) (Cross et al., 1975).

Both Matthews (1989) and Smith and Harris (1994) state that *C. drummondii* (yellow flymallow), is the most important host of *A. grandis grandis* in the United States as it serves as a reservoir for the species. Other wild hosts may also help maintain populations of *A. grandis grandis* when cotton is unavailable.

Pathogens or Associated Organisms Vectored

This species is not known to vector any pathogens; however, wounds caused by *A. grandis grandis* may be invaded by microorganisms.

Known Distribution

A. grandis grandis is indigenous to Central America (Matthews, 1989).

This species is currently found in: **Caribbean:** Cuba, Dominican Republic, Haiti, Martinique, and St. Kitts-Nevis. **Central America:** Belize, Costa Rica, El Salvador, Guatemala, Honduras, and Nicaragua. **North America:** Mexico and United States. **South America:** Argentina, Brazil, Colombia, Ecuador, Paraguay, and Venezuela (EPPO, 2007).

Distribution within the United States

Adults are strong fliers and can migrate large distances. The weevil's initial spread from Texas to other cotton growing states in the United States is believed to have happened almost entirely by natural dispersal (Smith and Harris, 1994). During the spread of this pest in the United States, the average annual spread was 80 km (49.7 miles). They can also be moved large distances by wind dispersal (Smith and Harris, 1994) or by human-mediated means (Kim and Sappington, 2004).

This species spread from Mexico into the southern tip of Texas around 1892 (Burke et al., 1986).

As of 2009, eradication for this pest is actively occurring in Arkansas, Louisiana, Missouri, Tennessee, Texas, and parts of northern Mexico. Post eradication activities are occurring in states where *A. grandis grandis* was previously eradicated: Alabama, parts of Arkansas, Arizona, California, Florida, Georgia, Kansas, Kentucky, parts of Louisiana, Mississippi, Missouri, New Mexico, North Carolina, Oklahoma, South Carolina, parts of Tennessee, parts of Texas, and Virginia (Grefenstette, 2009).

Detailed descriptions on the history, management and eradication efforts can be found in King et al. (1996). Information on the current status of this pest in the United States can be found on the PPQ Program website found here: http://www.aphis.usda.gov/plant_health/plant_pest_info/cotton_pests/index-bw.shtml.

Pathway

EPPO (2007) states that *A. grandis grandis* may be able to travel through international trade on cotton through both seeds and stored products.

This species initially entered the United States through natural dispersal. Its movement throughout the United States was largely through flight and local movement of cotton products to and from public gins, including un-ginned cotton and cotton seed (Loftin, 1946). This species can disperse long distances (Kim and Sappington, 2004).

This species can also spread through human-mediated assistance through road vehicles, movement of infested machinery and equipment, and movement of infested host or habitat material.

Survey

CAPS-Approved Method*:

Trap and lure. The trap and lure combination is the boll weevil trap, boll weevil lure, and an insecticidal strip. The lure is available in either a two-or four-week duration formulation; however, the four-week lure is the most appropriate for CAPS surveys (Schoenholz, 2011). The lure dispenser type is a plastic square.

The boll weevil trap is a plastic trap made up of three parts: 1) a green plastic body, 2) a molded screen cone that fits on top of the body, and 3) a collection chamber at the top

of the trap. Traps must include an insecticidal strip to contain the weevils and prevent predation in traps. The lure and insecticidal strip are placed in the collection chamber.

IPHIS Survey Supply Ordering System Product Names:

1) Boll Weevil Trap
2) Boll Weevil Lure - 4 Week
3) Boll Weevil Insecticide Strip

IMPORTANT
Before planning a boll weevil survey it is IMPERATIVE that you contact your PPQ Regional Cotton Program Manager to determine if your state should survey for this pest. CAPS surveys should NOT be conducted in areas that have PPQ Boll Weevil Program trapping programs.

Time of year to survey:
Weevils begin emerging once average night temperatures reach 20-21°C (68-69.8°F) (Smith and Harris, 1994). Emergence from overwintering sites takes place from April to the end of June/ early July for about 90% of the population (Schoenholz, 2011). Emergence can extend to early August in the most northerly part of the insect's distribution (Matthews, 1989). To survey for migrating populations, surveys may be initiated later in the season (August-October) (Schoenholz, 2011).

More information can be found on the USDA-APHIS-PPQ program pest page for *A. grandis grandis* which can be found here: http://www.aphis.usda.gov/plant_health/plant_pest_info/cotton_pests/index-bw.shtml.

Trap Placement:
Traps should be placed atop a 1.2 to 1.5 meter (4 to 5 feet) reed or stake driven into the ground to a depth where it will stand up straight (Schoenholz, 2011).

*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/.

Key Diagnostics/Identification

CAPS-Approved Method*: Morphological.

Samples are screened for the presence of the boll weevil. Level 1 screening for this pest requires experience sorting wood-borers and bark beetles to morpho-species. Initial screening should be performed using the following characters:

1. Face terminates with a long snout (rostrum).

2. Antennae elbowed, originate just over half way down the rostrum.

3. Body 3.2 to 8.4 mm (0.125 to 0.33 inches) long, reddish brown, covered in whitish hair-like scales.

4. Pronotum with a longitudinal white midline.

5. Femur swollen with double-pointed teeth on the inner margin.

Suspect boll weevil samples will then be forwarded to experienced eradication program personnel or entomology taxonomists at the state department of agriculture or a land grant university within the state for Level 2 Examination.

Boll weevil identification requires specialized training and family-level identification expertise. If such expertise is unavailable, samples should be forwarded directly to the designated identifier.

If program personnel believe the specimen is a boll weevil, they will initiate appropriate response activity (intensified trapping, ground and aerial treatments).

Suspect *Anthonomous grandis grandis* specimens will be forwarded via overnight mail, with a telephone alert, to:

Areas EAST of the Mississippi River (AL, FL, GA, KY, MS, NC, SC, TN, and VA)

Dr. Richard L. Brown
Mississippi Entomological Museum
100 Twelve Lane
Mississippi State, MS 39762
Phone: (662) 325-2990
Fax: (662) 325-8837
E-mail: RBrown@entomology.msstate.edu

Designated States WEST of the Mississippi River (AR, CA, KS, LA, MO, NM, OK, and TX)

Kira Metz
USDA APHIS PPQ
412 Minnie Belle Heep Center,
TAMU 2475
College Station, TX 77843-2475
Phone: (979) 862-3052 ; Cell: (979) 450-5492
E-mail: kira.metz@aphis.usda.gov

Please make sure to include form 391 “Specimens for Determination” with each sample.

Arizona
The Arizona Cotton Council screens trap samples and differentiates *Anthonomous grandis grandis* from *Anthonomous grandis thurberiae*, which occurs in Arizona and

feeds on wild cotton. Questionable specimens are sent to CPHST Mission lab for molecular analysis.

For instructions on how to submit specimens, refer to the **Guidelines for Submitting Wood Borer and Bark Beetle (WBBB) Specimens for Identification** at http://caps.ceris.purdue.edu/taxonomic_services/wbbb_sample_submission.

*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:
Recently, adult weevils collected in Chihuahua, Mexico were successfully identified using a new molecular diagnostic tool. The tool was developed to help distinguish the *Anthonomus grandis* southeastern variant, which attacks commercial cotton, from the thurberia weevil variant, that prefers the use of wild cotton hosts.

A PCR method has been developed to differentiate *A. grandis grandis* from other superficially similar weevils, but it does not distinguish *A. grandis grandis* from the sub-specific weevil *A. grandis thurberiae* (Kim et al., 2009). Characteristics to compare the three forms of *A. grandis* can be found in Clark and Burke (1986).

More information can be found on the USDA-APHIS-PPQ program pest page for *A. grandis grandis* which can be found here: http://www.aphis.usda.gov/plant_health/plant_pest_info/cotton_pests/index-bw.shtml.

Easily Confused Pests

A key to distinguish *A. grandis* from four other species in the "*A. grandis* group" is found in Jones and Burke (1997). It includes *A. townsendi*, *A. hunteri*, *A. mallyi*, and *A. palmeri* (Jones and Burke, 1997). *A. hunteri* is known to only develop on *Hampera trilobata*, an endemic plant to the Yucatan Peninsula, Belize, and northern Guatemala (PaDIL, 2010). None of these species are currently known to be present in the United States.

The similar-looking sub-species, *A. grandis thurberiae,* should only be of concern in surveys conducted in the Southwestern United States.

Commonly Encountered Non-targets

There are some superficially similar weevils that can be attracted to components of the *A. grandis grandis* lure or trap color including *A. eugenii* (pepper weevil), *A. eugenii musculus* (cranberry weevil), and *Curculio caryae* (pecan weevil) (Kim et al., 2009). They may be hard to distinguish from *A. grandis grandis* if the weevil samples are partially destroyed by predatory insects.

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Helicoverpa armigera

Scientific Name
Helicoverpa armigera (Hübner)

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

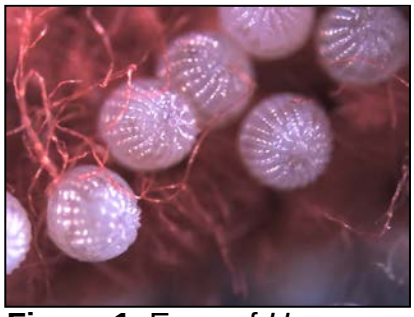

Figure 1. Eggs of *H. armigera.* Photo courtesy of Aneel Mohite, Rashtrasant Tukdoji Maharaj Nagpur University, India, www.wikipedia.com.

Common Name(s)
Old world bollworm, scarce bordered straw worm, corn earworm, cotton bollworm, African cotton bollworm, tobacco budworm, tomato grub, tomato worm, gram pod borer, and green bollworm.

Type of Pest
Moth

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

Reason for Inclusion in Manual
CAPS Target: AHP Prioritized Pest List – 2003 through 2012

Pest Description
Eggs: Eggs are tiny (0.5 mm, 0.02 in.) and nearly spherical with flattened bases (Fig. 1) (CAPS, 2010; EPPO, 2003; Pearson, 1958). The apical area around the micropyle is smooth, but the rest of the egg is ribbed. Each egg has about 24 ribs; alternate ribs are slightly shorter (Pearson, 1958). Young eggs are yellowish-white, but as they age turn brown (Fitt, 1994). Eggs are laid singly (Carter and Hargreaves, 1986).

Figure 2. *H. armigera* larva. Photo courtesy of Gyorgy Csoka, Hungary Forest Research Institute, www.bugwood.org.

Larvae: Larval color darkens with successive molts. Color is influenced by diet and ranges from bluish green to brownish red (Fowler and Lakin, 2001). Color is extremely variable, ranging from shades of green, straw yellow, black, pink, or reddish brown (EPPO, 2003).

Freshly emerged first instars are translucent whitish yellow. The head, prothoracic shield, supra-anal shield, and prothoracic shield are dark brown-black. Larvae appear spotted, because the setae above the spiracular line are on raised black pinacula and because of the dark rims of the spiracles (Pearson, 1958; King, 1994; Bhatt and Patel, 2001; EPPO, 2003). Second instar larvae are yellow-green with dark thoracic legs (CAPS, 2010). The mid-dorsal line is broad, dark brown or black. The dorsal line is edged with lighter bands. There is a broad, lighter-colored band on the lateral line on which the dark spiracles show clearly (Pearson, 1958). The thoracic and anal plates are often the same color as the integument, but sometimes the thoracic plate is darker with distinct maculation. Thoracic legs are brown. The prolegs are the same color as the body. Dorsal hairs just behind the head are transparent (EPPO, 2003). Fully-grown larvae are 30 to 40 mm (1.18 to 1.57 in.) long (Pearson, 1958; King, 1994; EPPO, 2003) (Fig. 2).

Pupae: Pupae are mahogany-brown, 14 to 20 mm (0.55 to 0.79 in.) long, and smooth (EPPO, 2003). Pupae are rounded anteriorly and posteriorly, with "two tapering parallel spines at the posterior tip" (Pearson, 1958). Pupation occurs in the top 1 to 7 inches (2.54 to 17.8 cm) of soil (Carter and Hargreaves, 1986).

Figure 3. *H. armigera* adult. Photo by Dr. Michael Barry 06-14-2006, copyright © Somerset Moth Group 2011, www.somersetmothgroup.org.uk.

Adult: *H. armigera* is a typical stout bodied noctuid moth (Fig. 3) about 18 mm long (0.71 in.) with a 40 mm (1.57 in.) wingspan (Pearson, 1958). The forewings range from dull yellow to olive-gray to brown with darker brown markings (Pearson, 1958). Sometimes, wings have a pinkish suffusion, especially on the underside of wings (Pearson, 1958). The brown markings form a distinct spot halfway between the apex and base, a smaller spot nearer the base, and a broad band across the "broadest part of the wing parallel to its distal margin" (Pearson, 1958). The hindwing is pale with "strongly marked veins and a broad, dark apical border with two lighter spots in it" (Pearson, 1958).

Biology and Ecology

H. armigera overwinters as a pupa in the soil. Moths emerge in May to June, depending on the latitude. The moths copulate within days of emergence from the pupa, but only if they have fed on nectar (Pearson, 1958). Each female can lay about 1000 eggs (Hill, 1983). Plants in flower are more attractive for oviposition than those without flowers (Firempong and Zalucki, 1990b). Oviposition on cotton is rare until flower buds form; in general, oviposition by *H. armigera* is closely associated with the period of budding and flowering in its host plants (Pearson, 1958). Females prefer pubescent leaves to smooth leaves and relatively taller plants when ovipositing (Firempong and Zalucki, 1990b; King, 1994). Eggs are deposited mainly on upper sides of leaves, but sometimes also

on squares in cotton (Pearson, 1958). The number of larval instars ranges from five to eight, but six is most common (Hardwick, 1965). Larvae drop off the plant and pupate in the soil.

H. armigera has overlapping generations, so it is difficult to determine the number of completed generations per year. Typically *H. armigera* completes two to five generations per year in temperate and subtropical climates. Under favorable conditions and tropical climates, there may be up to eleven generations per year (Tripathi and Singh, 1991; King, 1994; Fowler and Lakin, 2001). Temperature and host availability have the most influence on *H. armigera* population size, number of generations, and seasonality (King, 1994).

The eggs have a lower developmental threshold of 10.5°C (51°F) and require a thermal constant of 51 degree days to develop. Larval development requires 215.1 degree days based on an 11.3°C (52°F) threshold. Pupation requires 151.8 degree days based on a 13.8°C (57°F) threshold (Jallow and Matsumura, 2001).

H. armigera undergoes facultative pupal diapause induced by low temperatures (15 to 23°C; 59 to 73°F) and shortening day lengths (11 to 14 hours per day) (CABI, 2010). In some areas, a summer pupal dormancy is induced by prolonged hot, dry conditions (≥37°C (≥99°F) during 3rd instar) (Nibouche, 1998).

Moths may migrate long distances under adverse conditions (King, 1994; Zhou et al., 2000; Casimero et al., 2001; Shimizu and Fujisaki, 2002; CABI, 2010). Moths disperse up to 10 km (6.21 miles) during non-migratory flights and up to 250 km (155.3 miles) during migratory flights, which occur when host quality or availability decline (Saito, 1999; Zhou et al., 2000; Casimero et al., 2001; Fowler and Lakin, 2001).

For further information, see Dominguez Garcia-Tejero (1957), Pearson (1958), Hardwick (1965), Cayrol (1972), Delattre (1973), Hackett and Gatehouse (1982), King (1994), and CABI (2010).

Figure 4. Damage/hole on a young cotton square. Photo courtesy of O.P. Sharma, NCIPM, New Delhi. India, www.bugwood.org.

Symptoms/Signs

H. armigera shows preference for flower buds, flowers, and developing fruits of host plants (Pearson, 1958), but may also feed on foliage. Feeding often causes holes in reproductive structures with frass accumulation alongside (Pearson, 1958). Larvae may enter plant structures, and detection may require cutting into the plant.

On cotton: Eggs are laid on the upper surfaces of cotton leaves and on squares (Pearson, 1958). Larvae feed

preferentially on growing tips and reproductive structures and damage terminals, flower buds, and bolls (Fitt, 1994). Larvae bore clean, circular holes into flower buds, some fruits, and cotton bolls of all sizes (Hill, 1983). Young larvae feed preferentially in the upper canopy of cotton on new shoot tissue and small squares (Dillon and Fitt, 1995). Larger later instar larvae often feed with only the front end of their bodies inside the holes they make (Fig. 4). Frass accumulates between the bract and attacked buds or bolls (Pearson, 1958).

Pest Importance

Helicoverpa derives its pest status from factors that enable it to colonize agricultural systems: high fecundity, high mobility, polyphagy, and facultative diapause (Fitt, 1989). It has one of the widest distributions of any agricultural pest (Fitt, 1989).

H. armigera has been reported causing serious losses throughout its range, in particular to cotton, tomatoes, and corn. 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, 2010). For example, on cotton, two to three larvae on a plant can destroy all the bolls within two weeks (CABI, 2010); second and third instar larvae can cause up to 65% loss to cotton yields (Ting, 1986). In corn, they consume kernels and on tomatoes they invade fruits, preventing development and causing shed (CABI, 2010). In South African citrus, a severe bollworm infestation may destroy more than 80% of flowers and developing fruit and damage more than 50% of late-hanging fruit if left uncontrolled (Bedford, 1968; Begemann and Schoeman, 1999). In pigeon pea, an important grain legume, *H. armigera* causes yield losses of up to 100% in some years and locations, and worldwide losses to pigeon pea of more than $300 million per year (Thomas et al., 1997).

Known Hosts

H. armigera is polyphagous, and larvae attack at least 60 cultivated and 67 wild host plants (Pogue, 2004). Worldwide, *H. armigera* has been reported on over 180 cultivated hosts and wild species in at least 45 plant families (Venette et al., 2003). Predominant host families are Asteraceae, Fabaceae, Leguminaceae, Malvaceae, Poaceae, and Solanaceae (Fitt, 1989). The most important crop plants are cotton, corn, and grain sorghum (Reed, 1965; Roome, 1975; Hackett and Gatehouse, 1982; Pogue, 2004). Larvae attack leaves, shoots, flower buds, and fruit (EPPO, 2003).

H. armigera preferred hosts are tobacco, maize, and sunflower. Cotton, soybean, and lucerne are intermediately preferred hosts, and cabbage, pigweed, and linseed are least preferred hosts (Firempong and Zalucki, 1990a).

Major hosts: *Abelmoschus esculentus* (okra), *Allium* spp. (onions, garlic, leek, etc.), *Arachis hypogaea* (peanut), *Avena sativa* (oats), *Brassicaeae* (cruciferous, mustard crops), *Cajanus cajan* (pigeon pea), *Capsicum annuum* (bell pepper), *Carthamus tinctorius* (safflower), *Cicer arietinum* (chickpea, gram), *Citrus* spp., *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), *Pelargonium* spp. (geranium), *Pennisetum glaucum* (pearl millet), *Phaseolus* spp. (beans), *Phaseolus vulgaris* (common bean), *Pinus* spp. (pines), *Pisum sativum* (pea), *Prunus* spp. (stone fruit), *Solanum esculentum* (tomato), *Solanum melongena* (eggplant), *Solanum tuberosum* (potato), *Sorghum bicolor* (sorghum), *Triticum* spp. (wheat), *Triticum aestivum* (wheat), *Vigna unguiculata* (cowpea), and *Zea mays* (corn) (CABI, 2010).

Poor hosts:
Vitis vinifera (grape) (Voros, 1996)

Wild hosts:
Acalypha spp. (copperleaf), *Amaranthus* spp. (pigweed, amaranth), *Datura metel* (datura), *Gomphrena, Hyoscyamus niger* (black henbane), and *Sonchus oleraceus* (annual sowthistle) (Gu and Walter, 1999; CABI, 2010). For a complete list of hosts, see Venette et al. (2003).

Pathogens or Associated Organisms Vectored

Helicoverpa armigera is not a known vector and does not have any associated organisms.

Known Distribution

H. armigera has one of the widest distributions of any agricultural pest (Fitt, 1989). It is widely distributed throughout tropical and subtropical areas of Africa, Asia, Australia, Pacific Islands, New Zealand, the Mediterranean region, and the Middle East. (EPPO/CABI, 1997).

Africa: Algeria, Angola, Benin, Botswana, Burkina Faso, Burundi, Cameroon, Cape Verde, Central African Republic, Chad, Congo, Cote d'Ivoire, Egypt, Eritrea, Ethiopia, Gabon, Gambia, Ghana, Guinea, Kenya, Lesotho, Libya, Madagascar, Malawi, 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. **Asia:** Afghanistan, Armenia, Azerbaijan, Bangladesh, Bhutan, Brunei Darussalam, Cambodia, China, Cocos Islands, Republic of Georgia, India, Indonesia, Iran, Iraq, Israel, Japan, Jordan, Kazakhstan, Korea, Kuwait, Kyrgyzstan, Laos, Lebanon, Malaysia, Myanmar (Burma), Nepal, Philippines, Saudi Arabia, Singapore, Sri Lanka, Syria, Tajikistan, Thailand, Turkey, Turkmenistan, United Arab Emirates, Uzbekistan, Vietnam, and Yemen. **Europe:** Albania, Bulgaria, Cyprus, Finland, France, Germany, Greece, Hungary, Italy, Macedonia, Malta, Portugal, Romania, Russian Federation, Serbia and Montenegro, Slovenia, Spain, Switzerland, and Ukraine. **Oceania:** American Samoa, Australia, Belau, 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 (CABI, 2010).

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, Arkansas, Arizona, Florida, Georgia, Louisiana, Mississippi, Oklahoma, South Carolina, and Texas have the greatest risk for *H. armigera* establishment based on host availability and climate within the continental United States. Areas of most states, however, have a low to moderate risk for *H. armigera* establishment.

Because of transgenic cotton varieties, damage from *H. armigera* on U.S. cotton may be mitigated. *Bt* cotton with a single *cry*1A gene or stacked with *cry2*A has shown satisfactory protection against *H. armigera* (Gujar et al., 2007). As of 2007, there was no evidence of field-level resistance to *Bt* cotton (Gujar et al., 2007). Resistance management programs are required for *Bt* cotton in the United States, Australia, India, and some other countries (Gujar et al., 2007).

Survey

CAPS-Approved Method*: The CAPS-approved method is a trap and lure combination.

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

1) Plastic bucket trap
2) *Heliothis* trap
3) Texas (Hartstack) trap

The Lure Product Name is "*Helicoverpa armigera* Lure." The lure is effective for 28 days (4 weeks).

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 instructions on using the plastic bucket trap. The Texas (Hartstack) trap is not available commercially. See Hartstack et al. (1979) or Johnson and McNeil (1994) for images and trap design.

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:
Lures: A mixture of 97% (Z)-11-Hexadecenal and 3% (Z)-9-Hexadecenal was found to lure male moths (Pawar et al., 1988; Loganathan and Uthamasamy, 1998; Loganathan et al., 1999; Su et al., 2006). Septa impregnated with 1 mg of pheromone were the most effective, and should be replaced every 13 days (Loganathan et al., 1999). Pawar et al. (1988) described and used the ICRISAT standard trap. The ICRISAT trap's "inability to capture any *H. zea* or *H. virescens* compared with other traps (wire cone, plastic cone, and sticky traps) (Lopez et al., 1990) contrasts markedly with its relative efficiency for *H. armigera* in India (Pawar et al., 1988)" (King, 1994). The differences in trap efficiency are most likely due to fundamental behavioral differences between insect species (King, 1994). Other effective traps include funnel traps and Texas (Hartstack) traps (Kant et al., 1999). Place trap just above crop height (Pawar et al., 1988), or at 1.8 m (5.91 ft.) (Kant et al., 1999).

Sampling: In cotton, only the upper terminal portion of the plant is sampled because the majority of eggs and young larvae occur in the top portion of plants (Dillon and Fitt, 1995). Older larvae feed on older fruit lower in the canopy, but the majority of eggs and young larvae occur in the top 20 cm (7.9 in.) of the cotton plant, so terminal sampling is suitable (Dillon and Fitt, 1995). However, Bt cotton as a host may alter these distributions based on the distribution and potency of the toxin. "Bloom tags" or senesced floral structures stuck to the top of the developing boll may be an important site for inspection of young larvae.

Key Diagnostics/Identification

CAPS-Approved Method*: Confirmation of *Helicoverpa armigera* is by morphological identification. *H. armigera* can be visually screened to some degree, but definitive screening and identification requires dissection. *Helicoverpa armigera* and the native, abundant species, *Helicoverpa zea* are very similar looking. Final identification is by dissection of (adult) male genitalic structures (CAPS, 2010). A trained taxonomist must confirm any specimens.

*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: *H. armigera* and *H. zea* cannot be reliably identified without dissection of the male genitalia (Pogue, 2004). Adult *H. armigera* may be identified by differences in genitalia (Common, 1953; Kirkpatrick, 1961; Hardwick, 1965).

A screening aid is available for *H. armigera* at: http://caps.ceris.purdue.edu/webfm_send/552.

Instructions for dissecting *H. armigera* are available at:

http://caps.ceris.purdue.edu/webfm_send/551 and
http://caps.ceris.purdue.edu/webfm_send/550.

Figure 6. (left) *Helicoverpa zea* larval size by days, Clemson University - USDA Cooperative Extension Slide Series. (center) Larva feeding on cotton, Fudd Graham, Auburn University, and (right) adult moth, Steve L. Brown, University of Georgia, www.bugwood.org.

Easily Confused Pests

H. armigera can be easily confused with very similar pests from the noctuid subfamily Heliothinae (Figs. 6 & 7). *Helicoverpa zea* and *Heliothis virescens* are present in the United States. Several other noctuid pests can be confused easily with *H. armigera*, including *Helicoverpa assulta* (not known in the United States), *Helicoverpa punctigera (*not known in the United States) (Kirkpatrick, 1961; CABI, 2010) and *Heliothis subflexa.*

Figure 7. (left) Larvae of *H. virescens,* R.J. Reynolds Tobacco Company Slide Set, R.J. Reynolds Tobacco Company, (top, right) *H. virescens* larva. Photo courtesy of John L. Capinera, University of Florida (bottom, right) Adult moths of *Helicoverpa armigera* (left) and *Heliothis virescens* (right), W. Billen, Pflanzenbeschaustelle, Weil am Rhein, www.bugwood.org.

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Ostrinia furnacalis

Scientific Name

Ostrinia furnacalis Guenée

Synonyms: *Ostrinia damoalis, Ostrinia salentialis, Botys damoalis, Botys furnacalis, Botys salentialis, Hapalia damoalis, Micractis nubilalis, Micractis varialis, Pyrausta damoalis, Pyrausta furnacalis, Pyrausta nubilalis, Pyrausta nubilalis salentialis, Pyrausta polygoni, Pyrausta salentialis, Pyrausta vastatrix,* and *Spilodes kodzukalis.*

Figure 1. *O. furnacalis* egg mass. Photo courtesy of James Litsinger (CABI 2010).

In 1854, Guenée originally described the Asian corn borer as *Botys furnacalis*. It was subsequently re-described by many authors or repeatedly misidentified as the European corn borer, *Ostrinia nubilalis*. Much of the literature prior to 1966 treats *O. furnacalis* as *O. nubilalis*. Muturra and Munroe (1970) revised the genus *Ostrinia*, confirming the status of *O. furnacalis* as a valid species separate from the European corn borer *O. nubilalis* and synomizing it with *Botys (Pyrausta) damoalis*, *Botys (Pyrausta) salentialis*, *Pyrausta polygoni*, *P. vastatrix*, and *Spilodes kodzukalis*. Many other variants and combinations of these names are present in the literature.

Common Name(s)

Asian corn borer, Asian maize borer, Asiatic corn borer, China corn borer, and Oriental corn borer

Type of Pest

Moth

Taxonomic Position

Class: Insecta, **Order:** Lepidoptera, **Family:** Crambidae

Reason for Inclusion In Manual

CAPS Target: AHP Prioritized Pest List – 2006 through 2009

Pest Description

Eggs: The size of individual eggs ranges from 0.6 to 1 mm (0.02 to 0.04 in.) in diameter. Eggs are laid in white masses, overlapping in a fish-scale pattern. Usually these masses contain 20 to 40 eggs (Fig. 1) (Nafus and Schreiner, 1991). Eggs are light yellow or cream in color, turning black as larvae develop within.

Larvae: Freshly emerged larvae are about 1 to 2 mm (0.04 to 0.08 in.) long with a dark brown head and white body (Fig. 2a). Young larvae are pink or a yellowish-gray in color. The head capsule gets progressively lighter in color as larvae develop. Mature larvae are about 19 to 25 mm (0.75 to 0.98 in.) long with a medium to dark brown head and

creamy white to gray body. Raised and darkened lateral wart-like spots are evident on each body segment of the late instar larvae (Fig. 2b) (CAPS, 2010).

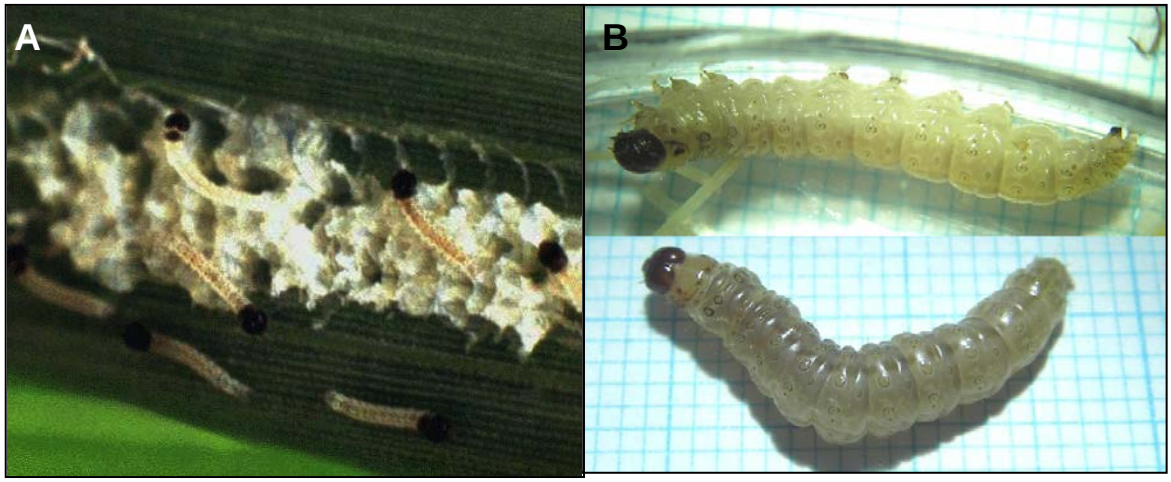

Figure 2. 1st instar larvae (A) and late instar larva of *O. furnacalis* (B). Notice the darkened lateral spots. Photos courtesy of James Litsinger, CABI and M. Nanjyo (www. jpmoth.org), respectively.

Pupae: Pupae are about 12 to 17 mm (0.47 to 0.67 in.) long with a tiny hook or cremaster at the tail end of the abdomen (Lee et al., 1980; CAPS, 2010). The head end is rounded and medium to dark brown in color.

Adults: Male moth wingspan is about 20 to 26 mm (0.79 to 1.02 in.); female moth wingspan is 26 to 30 mm (1.02 to 1.18 in.). The forewings are straw-colored to brown (Fig. 3), and the hindwings are light yellow to white in color. Females are slightly lighter colored than males. There is some geographical variation in wing marking. Male specimens from the tropics have more reddish-brown scales intermixed with the ground color, which makes the line markings more obvious. In female specimens, the lines and markings are more dark brownish gray color (CAPS, 2010).

Figure 3. Adult *O. furnacalis*. Photo courtesy of K. Yamamoto, www.jpmoth.org.

Biology and Ecology

Cotton is an alternative host of the Asian corn borer *Ostrinia furnacalis*. In some areas, the pest moves between cotton and cornfields. Populations in tropical areas without distinct wet and dry seasons are continuously active and can have as many as 12 continuous and overlapping generations (Camarao, 1976). In subtropical and tropical areas with distinct wet and dry seasons, populations and damage levels in corn are highest during or immediately following the wet season (Tseng, 1981; Morallo-Rejesus, 1985). In temperate zones, *O. furnacalis* has one to a few distinct generations per year

based on observations in both corn and cotton fields (Nafus and Schreiner, 1991; Li et al., 1999; Liu and Hou, 2004).

These populations undergo diapause as pupae. Moisture is the likely trigger for the start and end of diapause in corn (Hussein et al., 1983; Hussein and Kameldeer, 1988; Sapin et al., 2006). At the end of the season, the final instar larvae diapause in plant residues. Pupae are located wherever the last feeding occurred. They can be found in tunnels in the stem residue of cotton or corn depending on the planting time of corn.

The first generation moths emerge in May and copulate within days of emergence from the pupae. Most females oviposit during late May and early June (CGRIS-ICGR-CAAS, 2011). Oviposition usually takes place over a 20-day period for each generation (He et al., 2006). Eggs are generally deposited in white, fish-scale like masses on the underside of fully expanded middle to upper leaves on corn plants (Hussein et al., 1983; Legacion and Gabriel, 1988). They hatch in three to five days. The eggs within a mass hatch synchronously and the newly hatched larvae remain near the mass for 20 minutes to five hours (Nafus and Schreiner, 1991). After June, the newly emerged larvae chew holes in the cotton leaves and burrow into the main stem, fruiting branches, flower buds, white flowers, and green bolls of cotton plant (He et al., 2004; He et al., 2006; CGRIS-ICGR-CAAS, 2011). Rainfall during cotton growth is an important climatic factor that influences the regional population dynamics of the pest during a season. In corn populations, rainfall increases moth activity and oviposition, and humidity is favorable for egg development (Hussein et al., 1983; Hussein and Kameldeer, 1988; Sapin et al., 2006).

Full larval development requires five to seven instars and lasts 16 to 46 days based on host availability, diet, season, and weather (Nafus and Schreiner, 1991; Liu and Hou, 2004). The pupal period lasts four to nine days. Adults emerge in the late afternoon to early evening (4 to 8 pm) and start mating 19 to 24 hours later. Oviposition begins one to two days after mating (Camarao, 1976). Adults are active throughout the night with the activity peak in the early evening and in the morning before sunrise, and can live up to 14 days (females tend to live longer than males) (Nafus and Schreiner, 1991).

Larvae are capable of moving from plant to plant via silk, on self-created threads via wind, and on strands already attached to multiple plants. It is also common for adult moths to have long distance dispersal and immigration into cornfields especially when they first emerge.

Symptoms/Signs

On cotton: Asian corn borer larvae cause economic damage in cotton by attacking the terminals, new leaves, young squares, and white flowers (He et al., 2004; He et al., 2006). The larvae also burrow into the main stem, flower buds, fruiting branches, and green bolls, which may cause excessive vegetative branching and bunchy growth (He et al., 2004; He et al. 2006). Small, dark colored entry holes can be found on the stem or bolls. An accumulation of frass around these feeding sites is common. The stem, terminal or branches above the entry holes turn yellow, wilt and eventually die. This

damage leads to a loss of squares and green bolls and decreases the crop yield. The Asian corn borer also consumes fiber and seeds in the infested bolls (Fig. 4).

Figure 4. Symptoms of *O. furnacalis* infestation on cotton boll. Photo courtesy of CGRIS-ICGR-CAAS 2011 http://icgr.caas.net.cn/disease/default.html.

Pest Importance

O. furnacalis is an important component of the lepidopteran pest complex of cotton in China (He et al., 2004; He et al., 2006). The damage to cotton by the Asian corn borer is evident in areas where cotton, spring corn, and summer corn are planted concurrently or large scale wheat-cotton intercropping is planted (CGRIS-ICGR-CAAS, 2011). In these areas, the larvae of the first generation may cause severe damage on cotton, and the larvae from the second and third generation mainly cause damage on corn (CGRIS-ICGR-CAAS, 2011).

Transgenic cotton varieties have been successful in decreasing damage ratings of *O. furnacalis* in both laboratory and field studies (He et al., 2004; He et al., 2006). The presence of transgenic cotton in the United States may decrease the likelihood of *O. furnacalis* establishment.

Known Hosts

Because of confusion about the taxonomy of *O. furnacalis*, there are issues with the reported host range. Some host records may actually be from other species of *Ostrinia*. Further studies are needed to verify the host range. Schreiner et al. (1990) report that the growth and survival rates of the *O. furnacalis* larvae were higher on sweet corn ears than on any other potential host tested. The second best host was Johnson grass (*Sorghum halepense*). Only two larvae completed development on pepper and *Brachiaria mutica* (para grass), while only one developed on wildcane (*Saccharum spontaneum*). In this study, although reported as minor hosts, *Eleusine indica* (Indian goosegrass), *Pennisetum purpureum* (elephant grass), and *Phragmites karka* (tall reed) did not support any Asian corn borers through their complete development.
The following are the reported hosts according to the literature at this time.

Major hosts:
Gossypium hirsutum (cotton), *Panicum miliaceum* (millet), *Sorghum bicolor* (sorghum), *Sorghum halepense* (Johnson grass), *Zea mays* (maize/corn), and *Zingiber officinale* (ginger) (Lewvanich, 1973; Young, 1979; Schreiner et al., 1990; Nafus and Schreiner, 1991; Talekar et al. 1991).

Minor hosts:
Amaranthus spp., *Artemisia spp.* (wormwoods), *Apocynum cannabium* (Indian hemp), *Artemesia* spp., *Blumea lacera*, *Brachiaria mutica* (buffalo grass), *Cannabis sativa*

(hemp), *Capsicum annuum* (bell pepper), *Coix lacryma-jobi* (Job's-tears), *Eleusine indica* (finger millet), *Emex spp.* (Emex), *Humulus lupulus* (hops), *Oryza sativa* (rice), *Panicum virid*, *Pennisetum glaucum* (pearl millet), *Pennisetum spp.* (feather grass), *Phaseolus spp.* (bean), *Phragmites karka* (tall reed), *Phytolacca spp.* (pokeweed), *Polygonum* spp. (knotweed), *Polytoca macrophylla, Populus spp.* (poplars), *Rheum rhabarbarum* (rhubarb), *Rumex dentatus, Saccharum officinarum* (sugarcane), *Saccharum robustum*, *Saccharum spontaneum* (wild sugarcane), *Setaria italica* (foxtail millet), *Setaria viridis* (barbed bristlegrass), *Solanum melongena* (eggplant), *Themeda intermedia, Urochloa mutica* (tall panicum), and *Vigna sinensis* (cowpea) (CAPS, 2010).

Pathogens or Associated Organisms Vectored

Ostrinia furnacalis is not a known vector and does not have any associated organisms. However, the injuries produced by this borer may increase fungal and bacterial infections (Dalmacio et al., 2007).

Known Distribution

Because of confusion about the taxonomy of *O. furnacalis*, there may be issues with the reported distribution. The countries listed here and in the literature may not represent the true distribution of *O. furnacalis.*

Asia: Afghanistan, Brunei Darussalam, Cambodia, China, India, Indonesia, Japan, Korea, Laos, Malaysia, Myanmar (Burma), Pakistan, Philippines, Singapore, Sri Lanka, Thailand, and Vietnam. **Europe:** Russian Federation. **Oceania:** Australia, Micronesia, Guam, Northern Mariana Islands, Papa New Guinea, and Solomon Islands.

Potential Distribution within the United States

A recent risk analysis by USDA-APHIS-PPQ-CPHST indicates that areas of Illinois, Indiana, Iowa, Kentucky, Louisiana, Minnesota, Nebraska, Ohio, South Dakota, Texas, and Wisconsin have the greatest risk for *O. furnacalis* establishment based on host availability and climate within the United States.

Survey

CAPS- Approved Method*: The CAPS-approved method is a trap and lure combination.

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

1) Wing Trap Kit, Paper
2) Wing Trap Kit, Plastic

The Lure Product Name is "*Ostrinia furnacalis* Lure." The lure is effective for 28 days (4 weeks).

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

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: *Ostrinia furnacalis* can be identified by visual inspection (egg masses, larvae, damage) and with pheromone traps that attract adult males. Because of similarities in life stages to the European corn borer (*Ostrinia nubilalis*) and contamination of traps with *O. nubilalis* males, proper identification (via adult genitalia) is still necessary in areas that have no known establishment of *O. furnacalis*.

Trapping: The components of the sex pheromone of *O. furnacalis* have been analyzed (Cheng et al., 1981; Luo et al., 1983; Du et al., 1986; Yeh et al., 1989; Zhao et al., 1990). While all populations of *O. furnacalis* produce a pheromone with a mixture of (*E*)- and (*Z*)-12-tetradecanyl acetates, in many instances different populations produce and are attracted to significantly different proportions of these two stereoisomers. Reports from Japan (Huang et al., 1998) and China (Cheng et al., 1981) state that populations prefer pheromones containing 36 to 39%, 44%, and 54% of the (*E*) stereoisomer. Boo and Park (1998) summarize the ratios of the two stereoisomers used in six studies for *O. furnacalis*.

In addition, a third compound, tetradecanyl acetate was discovered and used in the pheromone mixture to attract males in Taiwan (Kou et al., 1992). The addition of tetradecanyl acetate did not enhance or suppress male response in the study. The addition of the compound in its natural ratio, however, resulted in a decrease in trap catches in China (Cheng et al., 1981; Chen et al., 1982).

Kou et al. (1992) used plastic tubes containing (*E*) - and (*Z*)-12-tetradecanyl acetates and tetradecanyl acetate in a 48: 37: 15 ratio inside wing-shaped sticky traps. Traps were placed 32.8 feet (10 m apart) and 2.6 to 3.3 feet (0.8 to 1 m) above the ground. Catches were compared with blank traps and traps baited with 2- to 3-day old virgin females every two days. Jackman et al. (1983) tested four types of traps: metal basin trap, plastic basin trap, Pherocon® 1C sticky trap, and Biotrap® sticky traps. No significant differences were observed between traps, although differences did exist between bait used (virgin female versus synthetic pheromone). Cheng et al. (1981) used simple water traps 20-cm (7.9-in.) diameter vessel filled with water and detergent to reduce surface tension). A paper roll was impregnated with the pheromone solution and supported 1 to 1.5 cm (0.4 to 0.6 in.) above the water surface. Trap height ranged from 2.6 to 3.3 feet (0.8 to 1 m) depending on the height of the plants, and lures were changed each night. Jackman et al. (1984) also used water traps. A circular plastic pan 30 cm (11.8 in.) in diameter and 9 cm (3.5 in.) deep was placed in a wood frame at 1.2 meters (3.94 ft.) high and supplied with a 1 cm (0.4 in.) thick wooden cover 8 cm (3.1 in.) above the pan top. Basins were filled with 5 cm (2 in.) of water and about 1 teaspoon of commercial laundry detergent.

Russell IPM (United Kingdom) manufactures a pheromone lure for *O. furnacalis*. They suggest using a Delta trap to monitor this insect. A bucket trap, however, may be used in dusty conditions or where there is a high moth population density. Two traps per hectare are recommended for small fields or fields with uneven topography, while one trap per two hectares is recommended for large scale field and homogenous land. The trap should be placed near the highest point of the plant using support posts approximately 1 meter high or higher if the crop is higher. Additional information can be found at:
http://www.russellipm-agriculture.com/insect.php?insect_id=211&lang=en.

For the European corn borer (ECB), *Ostrinia nubilalis*, traps used also include various 'cone' style traps, including the Scentry (cloth) design and the Harstack (metal screen) trap. These are currently used mostly for scouting (determining population levels in a field as a prelude to deciding whether or not to apply a treatment).
Black light (UV) traps have also been used for ECB monitoring, but these traps catch many insect pests including moths (CAPS, 2010).

Visual survey: *O. furnacalis* is detected in the field by surveying standing crops for egg batches or damage by larvae (cavities) (CAPS, 2010).

Key Diagnostics/Identification

CAPS-Approved Method: Confirmation of *O. furnacalis* is by morphological identification. A trained taxonomist should confirm identification of samples.

*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 similarities as larvae and adults, adult genitalia must be examined to differentiate between *Ostrinia* species (Mutuura and Munroe, 1970).

Easily Confused Pests

O. furnacalis is closely related to *O. nubilalis*, the European corn borer (Fig. 6). It is thought to have similar biology and ecology and a number of parasites in common. When pheromone traps for *O. furnacalis* are placed in the field, adult males of the European corn borer are often caught.

Figure 6. Larva (left) and adult (right) of *Ostrinia nubilalis*, the European corn borer. Photos courtesy of Ronald Smith, Auburn University and Frank Peairs, Colorado State University, www.bugwood.org.

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Oxycarenus hyalinipennis

Scientific Name
Oxycarenus hyalinipennis (Costa)

Synonyms:
Aphanus hyalinipennis and *Aphanus tardus* var. *hyalinipennis*

Common Name(s)
Cotton seed bug, dusty cotton stainer

Type of Pest
Hemipteran -True bug

Taxonomic Position
Class: Insecta, **Order:** Hemiptera, **Family:** Lygaeidae

Reason for Inclusion In Manual
CAPS Target: AHP Prioritized Pest List 2008 through 2012

This datasheet was prepared for CAPS surveys; however, it is also appropriate for use by cotton industry scouts and extension agents for early detection surveys of the cotton seed bug.

Figure 1. *Oxycarenus hyalinipennis* adult, dorsal and side view. Photos courtesy of Natasha Wright, Florida Department of Agriculture and Consumer Services, www.bugwood.org.

Pest Description
Eggs: "Oval 0.28 x 0.95 mm, longitudinally striated, pale yellow becoming pink" (Henry, 1983).

Nymphs: "Head and thorax brownish-olivaceous, abdomen pinkish. Fifth instar darker brown on head and thorax, wingpads distinct, extending to at least third abdominal segment" (Henry, 1983).

Adults: "Newly emerged individuals pale pink but rapidly turn black. Length of male about 3.8 mm (0.15 in.); female 4.3 mm (0.17 in.). Male abdomen terminates in round lobe, while female is truncate. The insects have three tarsal joints and a pair of ocelli. Second antennal segment usually in part pale yellow. Hemelytra hyaline and usually whitish; clavus, base of corium, and costal vein more opaque than rest. Setae of 3 different types: [1)] More or less erect stiff setae, blunt at tip terminating in 4 to 7 small teeth; [2)] normal, straight, tapering setae; and [3)] very thin, curved, flat-lying setae" (Henry, 1983) (Figure 1, 2).

Biology and Ecology

Once host plant seeds open, *O. hyalinipennis* begins to feed, mate, and lay eggs (Henry, 1983). *O. hyalinipennis* cannot pierce unopened bolls to feed (Henry, 1983). Copulation occurs within one day of adult emergence during hot months, while mating occurs within two to three days in cooler months. Oviposition occurs during the evening or night (Hammad et al., 1972). In Florida, however, oviposition has been observed during the day within the floss and on the surface of the split bolls (Derksen, 2012). After mating, females lay approximately 20 eggs on the lint of the opened bolls (Henry, 1983). Ananthakrishnan et al. (1982) found that females prefer immature capsules and bolls when ovipositing, usually laying eggs between the calyx and fruit wall. The total amount of eggs laid per female is dependent on both temperature and host material (Holtz, 2006). Eggs are laid singly or in groups of two to four eggs; rarely are more laid at the same time (Hammad et al., 1972).

Figure 2. Adult *O. hyalinipennis*. Photos courtesy of Laura Loru, ISE-CNR www.bugwood.org.

Eggs hatch around four days later when temperature is optimum (around 35°C (95°F)) (Henry, 1983). After nymphs hatch, they will cluster together for approximately half an hour before they begin searching for food (Hammad et al., 1972). The nymphs go through five nymphal instars in as little as two weeks, depending on temperature (Henry, 1983).

Both adults and nymphs of *O. hyalinipennis* feed on seeds (Henry, 1983). *O. hyalinipennis* may also feed on the leaves and young stems of host plants to obtain moisture (Fig. 3, 4) (Ananthakrishan et al., 1982).

An entire generation can be completed in 20 days with three to four generations occurring annually (Henry, 1983). Some sources have reported that *O. hyalinipennis* can complete as many as seven generations per year (Hammad et al., 1972). Hammad et al. (1972) found that temperature affects the duration of the life stages, especially when relative humidity is constant. *O. hyalinipennis* displays local migratory behavior in which it moves between host plants throughout the year (Holtz, 2006).

The last generation hibernates on “branches or leaves of grass and weeds or other such shelters” and does not feed or mate until host food is available again (Henry, 1983). The last generation flies to resting places (not necessarily host plants) and clusters together until host material is available again (Schaefer and Panizzi, 2000).

Schaefer and Panizzi, (2000) suggest that *O. hyalinipennis* intentionally tries to avoid feeding on the vegetative parts of the host plant [cotton] in order to avoid harming hosts with their toxic saliva. This would allow the plant to produce more valuable food seeds at a later time (Schaefer and Panizzi, 2000). This may be why *O. hyalinipennis* undergoes facultative diapause when seeds are shed versus feeding on plant material (Schaefer and Panizzi, 2000).

A detailed description of biology can be found in Kirkpatrick (1923), including life history information for different host plants.

Symptoms/Signs

Lint of cotton will be stained pinkish from the crushed insects (Henry, 1983). Although cotton seeds appear normal from the outside, the embryos are shriveled and discolored (Kirkpatrick, 1923) and weight loss can occur up to 15% (Schaefer and Panizzi, 2000). Also, seed germination can be severely reduced (Hill, 1983; Schaefer and Panizzi, 2000).

O. hyalinipennis has been described feeding on several types of fruit trees; the damage due to feeding appears like greasy spots that exude light-colored gum (Henry, 1983). When crushed, nymphs and adults of *O. hyalinipennis* emit a characteristic foul smell (Hill, 1983).

A detailed description of damage on cotton can be found in Kirkpatrick (1923).

Figure 3. Adult and nymphal cotton seed bugs on cotton bolls. Photos courtesy of Karolynne Griffiths, USDA-APHIS.

Pest Importance

O. hyalinipennis is considered a serious cotton pest in Egypt that causes weight loss in cottonseed, decreased germination, and decreased oil quality of the seed (Henry, 1983). It is also considered a major pest in Southeast Asia, India, and Africa on both cotton and okra (Hill, 1983). Both adults and nymphs can cause damage by sucking oil from mature seeds (Ananthakrishan et al., 1982; Rajashekhargouda et al., 1983). Stored, un-ginned cotton may also be attacked by *O. hyalinipennis* (Henry, 1983).

O. hyalinipennis has also been recorded causing tree fruit damage in Israel; damage included greasy spots caused by adults sucking on fruits and disfigurement of the fruits

caused by feces of the bug (Avidov and Harpaz, 1969). Fruit damage may be due to the toxic saliva of *O. hyalinipennis* (Schaefer and Panizzi, 2000).

Figure 4. Adult cotton seed bug in a crevice of plant branches (left) and on okra (right). Photos courtesy of Julio A. Navarro, USDA-APHIS.

Known Hosts

O. hyalinipennis primarily feeds on seeds of plants in the Malvaceae family, particularly *Gossypium* spp. (cotton). In addition to cotton, this pest has also been intercepted on certain fruits and vegetables including apple, avocado, corn, dates, figs, grapes, peach, okra, pineapple, and pomegranate, as well as hibiscus (USDA, 2009).

Holtz (2006) classified the following as 'true hosts' of *O. hyalinipennis* within the Malvales order:

Abelmoschus spp. (*A. esculentus* (okra), *A. moschatus* (musk okra)), *Abutilon* spp. (*A. crisper* (bladdermallow), *A. guineese*, *A. indicum* (monkeybush), *A. mauritianum*), *Althaea* spp. (*A. rosea* (hollyhock)), *Cola* spp. (cola), *Corchorus* spp. (*C. olitorius* (nalta jute)), *Dombeya* spp., *Gossypium* spp. (*G. barbadense* (Gallini cotton), *G. hirsutum* (Bourbon cotton)), *Hibiscus* spp. (*H. cannabinus* (kenaf), *H. mutabilis* (Dixie rosemallow), *H. sabdariffa* (roselle), *H. titiaceus* (sea hibiscus), *H. trionum* (Venice mallow)), *Malva* spp. (*M. rotundifolia* (low mallow)), *Malvastrum* spp. (false mallow), *Pavonia* spp. (swampmallow), *Phymosia umbellata*, *Sida* spp. (*S. acuta*, *S. cordifolia* (llima), *S. mollis*, *S. rhombifolia* (Cuban jute), *S. rhomboidea*), *Sphaeralcea* spp. (globemallow), *Sterculia* spp., *Triumfetta* spp., *Urena lobata* (Caesarweed), and *Wissedula amplissima*.

Holtz (2006) also listed reported hosts outside of the Malvales order:
Asclepias spp., *Cydonia* spp., *Diospyros* spp. (Malabar ebony) *Eriodendron* spp., *Ficus carica* (fig), *Malus* spp. (apple), *Persea americana* (avocado), *Phoenix dactylifera* (date-palm), *Prosopis juliflora* (mesquite), *Prunus* spp. (stonefruit), *Pyrus* spp. (pear), *Ricinus communis* (castor beans), *Spondias mangifera*, *Vigna sinensis* (blackeyed pear), *V. unguiculata* (cowpea), *Vitis* spp. (grape), and *Zea mays* (corn).

Pathogens or Associated Organisms Vectored

This pest is not currently known to vector any pathogens or other associated organisms.

Known Distribution

Africa: Algeria, Angola, Benin, Botswana, Burkina Faso, Burundi, Canary Islands, Congo, Cote d'Ivoire, Egypt, Ethiopia, Ghana, Guinea, Kenya, Madagascar, Malawi, Mali, Mauritania, Morocco, Mozambique, Namibia, Niger, Nigeria, Rwanda, Sao Tome and Principe, Senegal, Somalia, South Africa, Sudan, Swaziland, Tanzania, Togo, Tunisia, Uganda, and Zimbabwe. **Asia:** Bangladesh, Burma (Myanmar), Cambodia, China, India, Iran, Iraq, Israel, Laos, Libya, Pakistan, Philippines, Saudi Arabia, Sri Lanka, Syria, Thailand, Turkey, Vietnam, and Yemen. **Caribbean:** Bahamas, Cayman Islands, Cuba, Dominican Republic, Puerto Rico, Turks and Caicos Islands, and U.S. Virgin Islands. **Europe:** Austria, Bosnia and Herzegovina, Bulgaria, Croatia, France, Germany, Greece, Hungary, Italy, Portugal, Russia, Serbia, Slovakia and Spain. **North Armerica:** United States (Florida). **South America:** Argentina, Bolivia, Brazil, and Paraguay (Grillo Ravelo, 1993; Baranowski and Slater, 2005; Fauna Europaea, 2005).

Potential Distribution within the United States

Henry (1983) suggests that *O. hyalinipennis* could survive in southern California and other southern states with climates similar to the pest's range in Africa, Asia, and Europe. Holtz and Borchert (2006) predict that *O. hyalinipennis* could complete 4 to 7 generations a year in all cotton growing areas of the United States. Using lifecycle data, it was predicted that *O. hyalinipennis* could complete a maximum of seven generations in California, Arizona, Texas and Florida (Holtz and Borchert, 2006). According to Holtz (2006), a potential Malvales host of *O. hyalinipennis* is found in every U.S. state. A recent risk analysis by USDA-APHIS-PPQ-CPHST, indicates that most states in the United States have areas that are at low to moderate risk for *O. hyalinipennis* establishment based on climate and host availability. This pest was recently found in 2010 in Monroe County, Florida (FDACS, 2010) and also in Puerto Rico and the U.S. Virgin Islands (USDA, 2010). The pest was only ever detected at two locations in the lower Keys and has been identified nowhere else throughout Monroe County, Florida in spite of repeated surveys.

Pathway

The cotton seed bug may be introduced into the continental United States through natural movement. In recent years, it has been observed steadily extending its distribution northward through the Caribbean (Smith and Brambila, 2008), though there have been no definitive studies conducted on the specific mode of spread. The cotton seed bug has been documented flying short distances, but may also be aided by wind. The occurrence of hurricanes or tropical storms may aid the spread of the cotton seed bug from Caribbean islands to the continental United States (USDA-APHIS, 2010).

"The cotton seed bug moves easily in trade, even with commodities that are not known as hosts (CAPS, 2007; Henry, 1983). The cotton seed bug has been intercepted 570 times at U.S. ports-of-entry since 1984, primarily on cut

flowers or fruit for consumption. A few interceptions have also been recorded on plants for propagation. Seventy percent of those interceptions have occurred since 2000. The cotton seed bug has been found to move in both baggage and commercial cargo shipments. Sixty-six percent of the interceptions were recorded in permit cargo, and 26 percent were recorded in baggage. Eighty-four percent of the interceptions occurred on plants not known as hosts (PestID, 2010)" (USDA-APHIS, 2010).

Survey

CAPS-Approved Method*: Visual inspection is the approved method to survey for *O. hyalinipennis*.

Survey site selection

Surveys should be conducted in high risk areas where the introduction of cotton seed bug is most likely. This includes cultivated and wild cotton stands in states near the Caribbean islands, where the pest is known to be established. In addition, cotton fields near transit sites or distribution centers that receive items from countries with known infestations should also be targeted for regular surveys, as these items may carry hitchhiker bugs (USDA-APHIS, 2010).

Time of year to survey

Surveys should be carried out when the host plants are in seed. Surveyors for cotton should examine crops when host plants have newly matured bolls and dry seeds (Derksen et al., 2009). For early detection surveys, surveying during "the quiescent period [of the host] is not recommended, due to the cryptic nature of the cotton seed bug" (USDA-APHIS, 2010).

Sampling protocol

All samples should be processed at the field location.

1. Sample size

Choose the appropriate sample size based on the type of cotton to be surveyed.

a. Wild cotton stands
In isolated, wild cotton stands, sample from as close to twenty plants as possible. Inspect five bolls per plant or as many as possible.

Or

b. Large commercial cotton fields
Determine the acreage of the field and ask for permission to survey the field. Ask the grower if it is possible to cut bolls (destructive sampling). Ideally, plants from 10% of the field should be visually inspected; however, if resources are not available to survey at this level, survey as close to it as possible. Insect samples can be taken where necessary (not necessarily from every plant or boll) and bolls do not have to be removed to collect insect specimens.

2. Survey design
This species occurs in a clumped distribution so it is important to walk into the field and visually inspect portions of the entire field. The highest risk zone in the field is the entrance edge (where the vehicles enter the field from the highways or roads).

3. Visual inspection
Walk through the field and visually inspect bolls on cotton plants. The entire cotton plant should be inspected. The insects may be in other places than just the bolls; the insects may rest in hiding places outside the boll, on the leaves, etc. Be especially attentive and inspect the bolls and plants thoroughly.

If there are no signs of the insect in either the field edges or the areas visually examined, it is not necessary to take samples. Infestations are very obvious; the bolls look like they have fleas. If small, black insects are observed (preferably Lygaeidae if they can be distinguished, or just Heteroptera), prepare to collect the insect specimens by one of the following methods and follow the instructions in the **Collect insect specimens** section below:

a. Cut the boll from the plant. This is the preferred method as it allows the surveyor to closely examine the boll.

Or

b. Leave the boll on the plant, but open the boll with your fingers. This method is appropriate if permission cannot be obtained to remove bolls.

4. Insect specimen collection
At the collection location, collect insects that are hiding within the bolls by one of the following methods:

a. Open a gallon-sized plastic bag (does not need to be re-sealable) and tap the bolls into the bag, dislodging the insects into the bag. **Important: Remove the bolls.** Squirt 70% isopropyl alcohol into the bag to kill the insects and make it easier to transfer them to a vial (Brambila and Smith, personal communication, 2011).

Or

b. Beat the bolls over a tray or paper sheet and use an aspirator to collect the insects (Brambila and Smith, personal communication, 2011).

Use either method and then transfer the insects to a vial and cover with 70% isopropyl alcohol. **DO NOT transport live insects, cut bolls, or cotton seeds.** Leave all plant material in the field.

*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 Method:

Visual observation

"The cotton seed bug has been observed on tree trunks, on the undersides of both living and dead leaves, pods of leguminous plants, cracks in telephone poles or wooden posts and fences, under bark, in old nests of *Polistes,* in crevices between strands of barbed wire, on dried flower heads, among roots of grasses, underneath sheath-leaves of maize and sugarcane, in stored cotton, or in artificial traps such as old sacks on poles or in hedges near cotton fields (Kirkpatrick, 1923). It could also be found in leaf litter beneath cotton plants, or occasionally on the leaves (Smith and Brambila, 2008), in dry fruit pods, under tree bark, between planks of wooden structures, or in dry grass and leaf litter (Adu-Mensah and Kumar, 1977)" (USDA-APHIS, 2010).

"Trees appear to be a preferred resting location of the cotton seed bug. Rough barked trees are more attractive than smooth barked trees. The cotton seed bug was rarely observed on willows or poplars. Very few bugs were observed on date-palms and mulberries. The more common trees included various species of *Ficus*, *Acacia*, and some *Eucalyptus*. Colonies on the trees may be detected near the ground, up to a height of 6–7 meters (Kirkpatrick, 1923). Sweep-netting of weeds between cotton rows, or along field edges is not recommended except in cases where there is a high likelihood that the pest is present. It may be useful to sweep remaining vegetation after removing infested host material. Kirkpatrick (1923) found significant numbers of the cotton seed bug when old sacking was tied to bushes during the winter quiescent period" (USDA-APHIS, 2010).

UV-lights

"UV-light traps are not recommended for surveying for the cotton seed bug except in cases where there is a need to confirm eradication or enhance detection of a known population. UV-light traps are not pest specific, and consequently are cumbersome and time-consuming for sampling and identification purposes. In addition, it is unclear whether or not UV-light traps would be an effective monitoring tool for the cotton seed bug. Kirkpatrick (1923) demonstrated positive phototropism in laboratory experiments; however, when Kirkpatrick placed light traps at night in the direct path that the cotton seed bug was known to use between a tree and nearby field where they were coming from, no individuals were captured. It was concluded that the cotton seed bug did not migrate at night, and was not attracted to light at night" (USDA-APHIS, 2010).

"Conversely, Nakache and Klein (1992) noted that the cotton seed bug was strongly attracted to light at night in Israel. Additional research regarding the efficacy of UV-light traps is needed" (USDA-APHIS, 2010).

UV-light traps were used as part of a sentinel site survey in Florida. "Sentinel sites were located in areas where the pests' preferred host plants (cotton, okra and kenaf) could be found and UV-light traps were placed in areas related to a potential pathway for *O. hyalinipennis* to enter Florida. Typically, sentinel sites were chosen in pathways that were easily accessible (parks with open admission, roadsides, etc.) and had several host plants for inspection. Traps were placed in or around a point of entry and were checked at least once a month" (Derksen et al., 2009).

Key Diagnostics/Identification

CAPS-Approved Method*: Morphological examination of adults is needed to confirm identification. A field screening aid is available for *O. hyalinipennis* on the CAPS website at http://caps.ceris.purdue.edu/webfm_send/529. Final identification should be confirmed by dissecting and examining adult male internal structures (Brambila, 2010).

*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 similarities, adult genitalia should be examined for final identification between *O. hyalinipennis* and other similar *Oxycarenus* species (Brambila, 2010). Slater and Baranowski (1994) reported that *O. hyalinipennis* may be differentiated from *O. bokalae* by examining the clavus and pygophore opening. The coloration of the first five abdominal segments could distinguish *O. hyalinipennis* from *O. albidipennis*, *O. pallidipennis* and *O. congoensis* (Slater and Baranowski, 1994).

Easily Confused Pests

This pest can be mistaken for *Oxycarenus bokalae* which is not currently present in the United States. *O. hyalinipennis* can be differentiated by examining the clavus which will be either completely or mostly pale, brick-red to white, whereas in *O. bokalae* the clavus will be almost uniformly dark brown or black (Slater and Baranowski, 1994). Also, the pygophore opening in *O. hyalinipennis* tapers evenly to a triangular point, whereas in *O. bokalae*, the opening "is broad with the side margins arcuate and triangularly tapering to a sharply or bluntly pointed distal end" (Slater and Baranowski, 1994). *O. hyalinipennis* may also be mistaken for *O. albidipennis*, *O. pallidipennis* and *O. congoensis*, but these species can be differentiated due to their "orange-red coloration of the first five abdominal segments" (Slater and Baranowski, 1994).

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Pectinophora gossypiella

Scientific Name
Pectinophora gossypiella Saunders

Synonyms:
Depressaria gossypiella Saunders
Ephestia gossypiella Saunders
Gelechia gossypiella Saunders
Gelechiella gossypiella Saunders
Platyedra gossypiella Saunders

Common Name(s)
Pink bollworm

Type of Pest
Moth

Figure 1. *P. gossypiella* larvae. Image courtesy of Peggy Greb, USDA Agricultural Research Service, www.bugwood.org.

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

Reason for Inclusion In Manual
PPQ Program Pest

Pest Description
Eggs: Elongate oval, flattened; about 1 mm long and 0.5 mm broad (0.04 by 0.02 in.); the shell is pearly white, with a finely wrinkled surface. When newly laid, the egg has a slightly greenish tint. At maturity it turns reddish (Busck, 1917).

Larvae: The larvae (Fig. 1) are initially white with a dark head. The full grown larvae are 10 to 12 mm (0.39 to 0.47 in.) long and are white with a double red band on the upper portion of each segment (Mukuka et al., 2002).

Figure 2. *P. gossypiella* adult. Image courtesy of Mississippi State University Archive, Mississippi State University, www.bugwood.org.

Pupae: The pupa is 8 to 10 mm (0.31 to 0.39 in.) long, rather plump, reddish brown; posterior end pointed and terminating in a short, stout, upwardly turned hooklike cremaster; entire surface finely pubescent; no long setae, spines or hooks, except on last joint. When mature, the pupa becomes much darker; the imago's eyes can be seen prominently under the gena of the pupal skin, and the segmentation of the adult antennae and legs becomes discernible (Busck, 1917).

Adults: Moths (Fig. 2) are brown with a wingspan of 15 to 20 mm (0.59 to 0.79 in) (Mukuka et al., 2002).

P. gossypiella adults are small, dark-brown moths measuring about 12 to 20 mm (0.47 to 0.79 in) across the wings (USDA, 1948). The head is reddish brown in color with pale, iridescent scales. Antennae are brown and the basal segment bears a pecten of five or six long, stiff, hair-like scales. The labial palpi are long and curved upwards: the second segment bears a slightly furrowed hairy brush on the underside that becomes smooth distally and the terminal segment is shorter than the second. The proboscis is scaled.

Forewings are elongated-oval, pointed at the tips and bearing a wide fringe. The ground color of the forewings is brown and they have fine dark scales that form vague patches in the region of the medial cells and at the wing base. The apical portion of the wing is dark brown with a transverse, light-colored band. Sometimes the wing bears a round medial spot.

The hind wings are broader than the fore wings, trapezoidal in form and silvery gray with a darker, iridescent hind margin. The wing fringe is ochreous and darker at the base and apex.

Legs are brownish black with transverse, ochreous bands in the form of rings. The abdomen is ochreous toward the upper side, dark brown laterally and covered with ochreous-brown scales on the underside.

In the genitalia, the male uncus is broad at the base, tapering to a point and the aedeagus has a hooked tip. The female ovipositor is weakly sclerotized (CABI, 2010).

Biology and Ecology
This species is adapted for areas with low rainfall and a long growing season (Noble, 1969).

Eggs are laid singly or in small batches on the green cotton boll, the calyx, or the flower; they are more commonly found on the apex of the green boll on the longitudinal depressions (Busck, 1917). Before bolls are present, eggs are laid singly on squares, stems, and terminal buds; bolls are preferred when present (Noble, 1969). One to four eggs per boll are commonly seen although as many as 20 can be found on one boll (Busck, 1917). Hatching occurs in four to six days (Mukuka et al., 2002). Females can lay approximately 300 eggs (Mukuka et al., 2002).

After hatching, the larva tunnels into the boll and begins feeding on the soft inner wall or the partitions of the boll (Busck, 1917). The larva will usually bore close to the apex and tunnel downward towards the bottom seeds (Busck, 1917). From here it begins partially feeding on seeds while moving back up towards the tip of the boll where it ends as a full grown larva (Busck, 1917). Feeding lasts 10 to 15 days (Noble, 1969). Development is completed in a single square or boll; larvae do not move between structures (Noble, 1969). If buds are less than 10 days old, larvae will die (Ingram, 1994).

There are four instars total; only the last instar has the pink tint referred to in its common name (Busck, 1917). The larval period lasts 20 to 30 days during the summer and much longer during colder months (Busck, 1917). Overwintering occurs in the larval stage within the seeds (Busck, 1917), old bolls, leaf litter, or at gins or seed-storage facilities (Noble, 1969).

Pupation occurs in a spun cocoon (Noble, 1969) within the seeds although larvae may pupate in the soil if disturbed (Busck, 1917). The pupal period lasts 10 to 20 days (Busck, 1917).

The adult is small and sluggish, hiding during the day mainly in ground cover (Busck, 1917). Flight occurs at dusk, with adults flying to the nearest cotton boll to mate and lay eggs (Busck, 1917). Mating can occur on the first night after emergence and can occur more than once (Noble, 1969). Although adults have the capability for strong flight, they usually only infest nearby or adjoining fields (Busck, 1917). Adults are too sluggish for sustained flight (Busck, 1917). Pre-oviposition is about two days (Matthews, 1989). After oviposition, the moths live from 1.5 to 2 weeks (Noble, 1969). Under favorable conditions, an entire life cycle can be completed in as little as 35 days. There may be four to six overlapping generations per year (Busck, 1917).

Symptoms/Signs

Entry holes made by *P. gossypiella* larvae are difficult to see. If opened, the caterpillars are easy to find (Fig. 3). Bolls damaged by the larvae fail to open completely (Mukuka et al., 2002). Other signs include fruit shedding, lint damage, seed loss (CABI, 2010), rotted bolls, and discolored lint or seed (Leigh et al., 1996). Flowers may also be damaged (Ingram, 1994).

Figure 3. *P. gossypiella* larva on a section of a cotton boll with attendant damage. Image courtesy of David Pierce, USDA-APHIS-PPQ-WR.

Larvae may be hard to see at the early larval stages, but close examination may reveal the small entrance hole. The entrance hole may have a small amount of reddish frass, empty egg shells, or small larvae mining within the boll wall when dissected (Busck, 1917). Larval mines on the inner carpel wall can be seen from the outside (Noble, 1969). Also, a round exit hole about 2 mm (0.08 in.) in diameter can be found in the carpel (Noble, 1969; Matthews, 1989).

The shell of the infested bolls will eventually become discolored taking on a reddish or black color (Busck, 1917). It should be noted that this discoloration may also occur from other injuries besides *P. gossypiella* and should not be used for definitive diagnosis (Busck, 1917).

In square infestations, the bloom is usually prevented from opening due to the larval webspinning (Noble, 1969). In boll infestations, older bolls (20+ days) are usually attacked; younger bolls will be attacked if older bolls are scarce or the *P. gossypiella* population is high (Noble, 1969). Younger bolls will have the greatest damage (Noble, 1969).

Larvae do not attack cotton leaves or shoots (Busck, 1917). Bud and flower damage only occurs early in the season (Ingram, 1994).

Pest Importance

P. *gossypiella* is currently found in almost all cotton growing countries of the world (Naranjo et al., 2001) and is considered one of the most important economic pests in the world (Raulston et al., 1996). In the United States, it attacks two cultivated crops, cotton and okra (Wagner et al., 1996). Damage can reduce the lint yield by 50% and can lead to a decrease in seed oil (Busck, 1917).

As the larvae eat, they tunnel and soil the lint, which causes slowed growth of the cotton plant (Busck, 1917). The boll may then either rot or open prematurely and imperfectly (Busck, 1917). Un-infested parts of the boll will have slowed growth and the cotton lint will depreciate in value (Busck, 1917).

Boll feeding leads to reduction of quality of lint (discoloration, reduced fiber length, and strength) and seed (Noble, 1969). If damaged lint fibers are mixed with undamaged lint, it will reduce the average grade of the crop (Parencia, 1978). It can also lead to reduced yield in weight in high infestations (Noble, 1969). Quantity of seed cotton can be reduced as well (Noble, 1969). Severe infestations can lead to crop loss (Matthews, 1989).

Known Hosts

The major host of *P. gossypiella* is *Gossypium hirsutum* (cotton). Other hosts include *Abelmoschus esculentus* (okra), other *Gossypium* spp. (cotton), *Gossypium tomentosum*, and *Hibiscus* spp. (Busck, 1917; EPPO, 2007). Incidental hosts include species in the family Malvaceae (EPPO, 2007).

CABI (2010) also list the following species as host plants of *P. gossypiella*: *Abutilon* spp. (Indian mallow), *A. indica* (country mallow), *Althaea* spp. (hollyhocks), *Gossypium arboretum*, *Hibiscus* spp. (rosemallows), *H. cannabinus* (kenaf), *H. sabdariffa* (Jamaica sorrel), and *Medicago sativa* (alfalfa, lucerne).

Noble (1969) lists the following as alternative hosts of *P. gossypiella*:

Malvaceae (mallow family):
Abutilon amplum, *A. hirtum*, *A. hypoleucum*, *A. incanum**, *A. indicum*, *A. lignosum**, *A. otocarpum*, *A. trisulcatum*, *Althaea rosea* (hollyhock), *Callirhoe involucrata* var. *lineariloba* (poppy-mallow), *Fugosia australis*, *Hibiscadelphus hualalaiensis*, *Hibiscus abelmoschus**, *H. aculeatus**, *H. bifurcatus*, *H. brasiliensis**, *H. cannabinus**, *H. cardiophyllus**, *H. coccineus** (scarlet-rosemallow), *H. coulteri* (desert-rosemallow), *H. dasycalyx**, *H. denudatus* (paleface-rosemallow), *H. divaricatus*, *H. drummondii*, *H. esculentus** (okra), *H. furcellatus* var. *youngianus*, *H. heterophyllus*, *H. incanus**, *H. lambertianus**, *H. lasiocarpus* (woolly-rosemallow)*, *H. leucophyllus**, *H. ludwigii*, *H. militaris** (rosemallow), *H. mutabilis** (cotton-rosemallow), *H. panduraeformis*, *H. rosa-sinensis* (rose of China), *H. sabdariffa* (roselle), *H. syriacus* (shrubby althea), *H. tiliaceus*, *H. trilobus*, *H. tubiflorus*, *H. vitifolius*, *Kosteletzkya althaeifolia*, *K. virginica*, *Malachra capitata**, *Malva parviflora*, *M. sylvestris* (high-mallow), *Malvastrum coromandelianum*, *Malvaviscus arboreus* (false-mallow), *M. drummondii** (waxmallow), *Pseudabutilon lozani** (false-abutilon), *Sida cordifolia**, *S. corrugata*, *S. spinosa** (prickly-sida), *S. virgata*, *Thespesia danis*, *T. grandiflora*, *T. lampas*, and *T. populnea** (tulip tree).

Euphorbiaceae (spurge family):
*Croton capitatus** (croton), *C. texensis** (croton), and *Ricinus communis** (castorbean).

Leguminosae (pea family):
Acacia wrightii (tree cat's-claw), *Daubentonia punicea** (coffeebean), *Gleditsia triacanthos** (honeylocust), and *Prosopis juliflora* var. *glandulosa* (mesquite).

Convolvulaceae (morning-glory family):
Ipomoea crassicaulis (Texas brush morning-glory).

Tiliaceae (linden family):
Corchorus olitorius (jute).

Bombacaceae (bombax family):
Bombax munguba.

Cochlospermaceae (cochlospermum family):
Cochlospermum regium.

*Diapausing larvae were found to survive the winter in seed pods (Noble, 1969).

Pathogens or Associated Organisms Vectored

This pest is not currently known to vector any pathogens or other associated organisms. However, feeding by *P. gossypiella* larvae can lead to secondary rot on the host plant (Mukuka et al., 2002). Exit holes can predispose the bolls to *Aspergillus flavus* infections (Henneberry et al., 1978; Ingram, 1994).

Known Distribution

This pest has a large distribution throughout the world. It is thought to have originated in India.

Areas of distribution include: **Africa:** Algeria, Angola, Benin, Burkina Faso, Burundi, Cameroon, Central African Republic, Chad, Cote d'Ivoire, Egypt, Ethiopia, Ghana, Kenya, Libya, Madagascar, Malawi, Mali, Mauritius, Morocco, Mozambique, Niger, Nigeria, Rwanda, Senegal, Seychelles, Sierra Leone, Somalia, Sudan, Tanzania, Togo, Tunisia, Uganda, Zaire, Zambia, and Zimbabwe. **Asia:** Afghanistan, Bangladesh, Cambodia, China, India, Indonesia, Iran, Iraq, Israel, Japan, Jordan, Korea, Laos, Malaysia, Myanmar (Burma), Pakistan, Philippines, Saudi Arabia, Syria, Taiwan, Sri Lanka, Thailand, Turkey, Vietnam, and Yemen. **Caribbean:** Antigua and Barbuda, Bahamas, Barbados, Cuba, Dominica, Dominican Republic, Grenada, Guadeloupe, Haiti, Jamaica, Martinique, Montserrat, Puerto Rico, Saint Lucia, Saint Kitts-Nevis, Saint Vincent and the Grenadines, Trinidad and Tobago, and Virgin Islands (British). **Europe:** Cyprus, Greece, Macedonia, Montenegro, Romania, and Spain. **North America:** Mexico and United States. **Oceania:** Australia, Fiji, French Polynesia, New Caledonia, Northern Mariana Islands, Papua New Guinea, Samoa, and Vanuatu. **South America:** Argentina, Bolivia, Brazil, Colombia, Guyana, Paraguay, Peru, Uruguay, and Venezuela (Mukuka et al., 2002; EPPO, 2007).

Distribution within the United States

Infestations first occurred in the United States in 1917 on cotton in Texas (Naranjo et al., 2001). This population was apparently eradicated only to have the same area reinfested in the 1930s through suspected moth migration from Mexico (Raulston et al., 1996). Since its original introduction, it has spread to other states including Arizona, Arkansas, California, Florida, Georgia, Louisiana, New Mexico, and Oklahoma (Naranjo et al., 2001). Several eradications and re-infestations have occurred over the years. *P. gossypiella* is actively being eradicated from Arizona, New Mexico, and Texas and managed/suppressed in California (Schoenholz, 2011). This species has been present in Hawaii since the early 1900s (Ingram, 1994).

Pathway

This species has been introduced into many new areas through infested cottonseed, including Brazil, Mexico, the West Indies, the Philippines, the United States, and possibly Australia (Naranjo et al., 2001).

This pest can also move through infested okra as well as any material that has been contaminated with infested cottonseed including baled lint, mechanical cotton pickers, transport vehicles, and oil mill products (Noble, 1969).

Adults can also fly long distances (Noble, 1969), although they are usually considered too sluggish for sustained flight (Busck, 1917). Adult dispersal may be aided by wind (Raulston et al., 1996).

Several items are regulated to prevent the spread and reintroduction of pink bollworm, including all parts of cotton, seed cotton, cotton waste, used cotton-harvesting equipment, and okra. A list of these can be found here: http://www.aphis.usda.gov/plant_health/plant_pest_info/cotton_pests/downloads/bollw.pdf or in the Code of Federal Regulations, 301.52.

Survey

CAPS-Approved Method*:

Trap with lure. The CAPS-Approved Method will follow the PPQ Pink Bollworm Program recommendations. The trap is the pink bollworm delta trap (Fig. 4). This trap is orange in color and has each of the three interior surfaces coated in adhesive.

Figure 4. Delta trap used for *P. gossypiella*. Image courtesy of John Crowe, USDA-APHIS.

Lures should be changed every 2 weeks. The PPQ Cotton Program changes the lures on a weekly basis; however, every 2 weeks is acceptable for CAPS (Schoenholz, 2011). In excessively dusty conditions, lures should be changed weekly as moth specimens may be too dusty to properly identify (Schoenholz, 2011).

IPHIS Survey Supply Ordering System Product Names:

1) Paper Delta Trap, 3 sticky sides, Orange
2) Pink Bollworm Lure

IMPORTANT:
Before planning a pink bollworm survey it is IMPERATIVE that you contact your PPQ Regional Cotton Program Manager to determine if your state should survey for this pest. CAPS surveys should NOT be conducted in states that are part of the PPQ Pink Bollworm eradication program (AZ, CA, NM, and TX). In addition, some cotton-producing states have wet climates that are not conducive to pink bollworm development.

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).

Time of year to survey:
Traps are set out shortly after crop planting and remain until either defoliation or harvest or a killing freeze occurs (Leggett et al., 1994; Grefenstette et al., 2009).

Trap placement:

Traps should be placed level with the crop canopy.

*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/.

Key Diagnostics/Identification

CAPS-Approved Method*: Morphological. Samples are screened for the presence of pink bollworm. Level 1 screening for this pest requires experience working with Lepidoptera in sticky traps. Initial screening should be performed using the following characters:

1. Labial palpi long and curved upward, their second joint with two black horizontal bands.

2. Hind wing dusky, with apex pointed, margin fringed.

3. Forewing narrow, mottled brown or grayish-brown with poorly defined black spots, with apex pointed and fringed.

4. Forewing approximately 6.3 to 8.7 mm (0.25 to 0.34 inches) in length in males, 6.9 to 10 mm (0.27 to 0.39 inches) in length in females.

Suspect pink bollworm specimens should then be forwarded to the designated identifier:

Areas EAST of the Mississippi River:

Dr. Julieta Brambila
USDA, APHIS, PPQ
1911 SW 34th Street
Gainesville, FL 32608
Tel.: (352) 372-3505, ext. 438
E-mail: julieta.brambila@aphis.usda.gov

Areas WEST of the Mississippi River:

Kira Metz
USDA APHIS PPQ
412 Minnie Belle Heep Center,
TAMU 2475
College Station, TX 77843-2475
Phone: (979) 862-3052 ; Cell: (979) 450-5492
E-mail: kira.metz@aphis.usda.gov

Please include form 391 “Specimens for Determination” with each sample. Please follow the CAPS-approved sticky trap sample submission guidelines at http://caps.ceris.purdue.edu/guidelines/2011/apdx_e2.

The ***Pectinophora gossypiella* Pink Boll Worm Field Screening Aid and Diagnostic Aid** is available at the following link: http://caps.ceris.purdue.edu/screening/pectinophora_gossypiiella.

*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 thorough description of the moth and larvae can be found in Busck (1917). A description of all stages can be found in Noble (1969) and Matthews (1989).

An online key for *P. gossypiella* was recently released and can be found here: http://itp.lucidcentral.org/id/pbw/ (Hughes and Moore, 2011). This key offers ID support for field surveys and includes an adult and larval key with similar pests that may be found when surveying for *P. gossypiella* and many detailed images.

Easily Confused Pests

P. gossypiella is similar to both *P. scutigera* and *P. endema* in markings, venation, and morphology (Hodges, 1984). Descriptions to tell the males and females of each species apart can be found in Hodges (1984). A key to differentiate *P. gossypiella* and *P. scutigera* larvae (second to fourth instar) can also be found in Hodges (1984).

Busck (1917) states that several caterpillar species found in the United States in cotton bolls have been previously mistaken for *P. gossypiella*. These include (*Platynota*) *Sparganothis idaeusalis* and *Sparganothis rostrana* (Busck, 1917). These species are usually leaf-rollers but may enter open cotton bolls (Busck, 1917). *Pyroderces rileyi* may also be found in open cotton bolls, their color resembling the color of older instar *P. gossypiella* (Busck, 1917). Hughes and Moore (2011) state that the larvae of *Crocidosema plebejana*, *Dicymolomia julianalis*, and *Pyroderces rileyi* are similar to larvae of *P. gossypiella,* all of which are found in the United States. Distinguishing characteristics are found in Hughes and Moore (2011).

Matthews (1989) states that *P. gossypiella* may also be confused with *Mometa zemiodes* and *Pyroderces simplex*. Both species look similar, but *M. zemiodes* tends to feed on mature seeds and *P. simplex* is found in damaged open bolls (Matthews, 1989).

Hughes and Moore (2011) list similar adult species that are found in the United States including *Epilechia catalinella*, *Gelechia* sp., *Ofatulena duodecemstriata*, *Platyedra subcinerea*, and *Pyroderces rileyi*.

Commonly Encountered Non-targets

Both males and females of *Gelechia* sp. are attracted to the pink bollworm delta traps. Other moths that have been found in pink bollworm delta traps include *Epilechia catalinella* and *Ofatulena duodecemstriata* (Hughes and Moore, 2011).

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Spodoptera littoralis

Scientific Name
Spodoptera littoralis Boisduval

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

The two Old World cotton leafworm species *S. littoralis* and *S. litura* are allopatric, and their ranges cover Africa and Asia, respectively. Many authors have treated them as the same species.

Common Name(s)
Egyptian cotton leafworm, Cotton leafworm, Mediterranean climbing cutworm, tobacco caterpillar, tomato caterpillar, Egyptian cotton worm, Mediterranean brocade moth, and 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 2003 through 2012

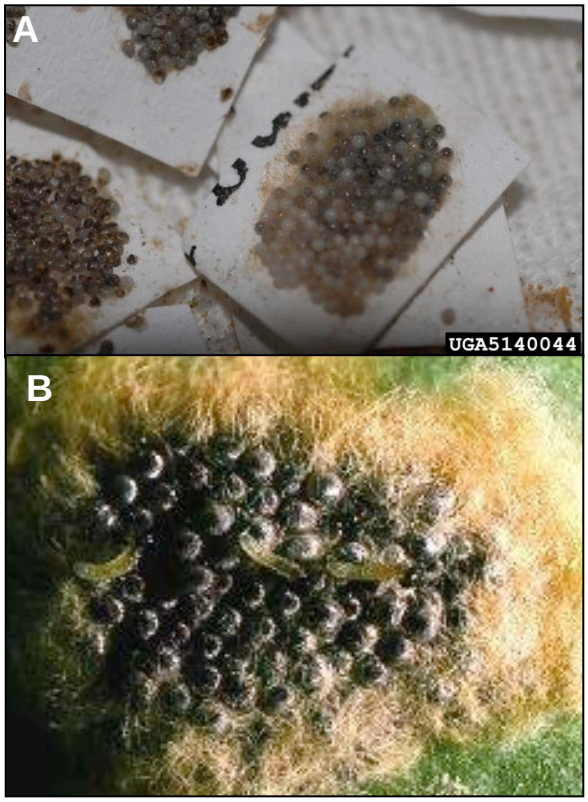

Figure 1. *Spodoptera* egg masses (A); eggs and neonates (B). Eggs are laid in batches covered with orange-brown hair scales. Photos courtesy of Esmat M. Hegazi, University of Alexandria, www.bugwood.org and http://www.defra.gov.uk/planth/pestnote/spod.htm, respectively.

Pest Description
Eggs: Eggs are about 0.6 mm (0.024 in.) in diameter, spherical, and somewhat flattened in shape. They are laid in batches covered with orange-brown hairy scales derived from the tip of the abdomen of the female moth (Fig. 1). Eggs are usually whitish-yellow in color, turning black as they age (Pinhey, 1975).

Larvae: Newly emerged larvae are about 2 to 3 mm (0.07 to 0.12 in.) long with a black head and white body. They are difficult to detect visually. Mature larvae are 40 to 45 mm (1.57 to 1.77 in.) long, hairless and cylindrical, tapering towards the posterior (Fig. 2). Fully developed larval body color varies from blackish-gray to dark green to reddish-

brown or whitish-yellow. Dark and light longitudinal bands are present on the sides of the body. From a dorsal view, two dark semilunar spots can be seen laterally on each body segment except the prothorax. The spots on the first and eighth abdominal segments are larger than the others, interrupting the lateral lines on the first segment. Larvae are nocturnal and can be found at the base of plants during the day (CAPS, 2010). Brown and Dewhurst (1975) described the larva of *S. littoralis*.

Figure 2. Larva of *S. littoralis*. Photo courtesy of Biologische Bundesanstalt für Land-und Forstwirtschaft Archive, Biologische Bundesanstalt für Land- und Forstwirtschaft, www.bugwood.org.

Pupae: Freshly formed pupae are green with a reddish color on the abdomen, becoming dark reddish-brown after a few hours. The general shape is cylindrical with the dimension of 14 to 20 x 5 mm (0.55 to 0.79 x 0.20 in.), tapering towards the posterior (CAPS, 2010). The last segment ends in two strong, straight hooks (Pinhey, 1975).

Adults: Moths are about 15 to 20 mm (0.59 to 0.79 in.) long, with a gray to brown body (Fig. 3). The wingspan ranges from 30 to 38 mm (1.18 to 1.49 in.). The forewings are gray to reddish-brown with paler lines along the veins. In male moths, bluish areas occur on the forewing base and tip (Fig. 3). The hindwings are grayish white, iridescent with gray margins, and usually lack darker veins (EPPO, 1997). The ocellus is marked by two or three oblique whitish stripes.

Figure 3. Adult moth of *S. littoralis*. Photos courtesy of Bernard Fransen, www.bugwood.org.

Biology and Ecology

S. littoralis is a multivoltine species that does not undergo a diapause stage. Female moths lay most of their egg masses (20 to 1,000 eggs per moth) on the lower leaf surface of younger leaves or upper parts of the plant (CAPS, 2010). Anderson and Alborn (1999) stated that *S. littoralis* preferred to oviposit on small plants (three-to-four leaf stage) that had been fed upon by 3rd or 4th instar larvae (72%) over non-damaged control plants. However, when using larger plants (eight to 10 true leaves), the preference was reversed with only 31% of eggs deposited on induced (previously fed upon) plants. Eggs begin to hatch after 28.6 degree days (DD) at a base temperature of 14.8°C (59°F). The optimal temperature for egg hatch is 28 to 30°C (82 to 86°F) (CAPS, 2010).

Full larval development requires six instars. The developmental time of the pest varies from two to seven weeks, depending on the temperature, relative humidity, and other similar factors (Clapham, 1980). Young larvae remain on the underside of the leaves, feed actively on cotton leaves, and move about vigorously during the day (Clapham, 1980). On cotton, the first three larval instars feed mainly on the lower surface of the leaves, whereas later instars feed on both surfaces. Third and fourth instars remain on the plant, but do not feed during the day. Older instars spend a large proportion of their time off the plant and in the soil throughout the day and return to the plant at night (CAPS, 2010).

A mature larva creates a clay cocoon about 3 to 5 cm (1.2 to 2 in.) below the soil surface, in which it usually pupates within five to six hours (Clapham, 1980). Emergence of adult moths occurs at night. Adults have a life span of five to ten days. Adults are nocturnal and are most active between the hours of 8 pm and midnight.

Symptoms/Signs

On most crops, damage can be caused by extensive feeding by larvae, leading to complete stripping of the plants. Corn stems are often mined by *S. littoralis* and young grains in the ear may also be damaged.

On cotton: On cotton, the pest may cause considerable damage by feeding on the leaves, fruiting points, flower buds, and occasionally bolls. Damage of *S. littoralis* consists of feeding scars and skeletonization caused by larvae feeding on the underside of leaves. Damage starts with numerous small feeding points, and finally spreads over the entire leaf. Later, holes and bare sections are found on leaves, young stalks, bolls, and buds resulting from feeding activities of the pest. In some cases, the shoot tips above a feeding hole turn yellow, wilt, and eventually die (CAPS, 2010).

Pest Importance

S. 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 cutworm on seedlings. It can attack numerous economically important crops all year round (EPPO, 1997; CAPS, 2010). On cotton, the pest may cause considerable damage by feeding on the leaves, fruiting points, flower buds, and occasionally on bolls. When groundnuts (peanuts) are infested, larvae first selectively feed on the young folded leaves, 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. Larvae also often badly damage the pods of cowpeas and the seeds they contain. In tomatoes, larvae burrow into the fruit, which is thus rendered unsuitable for consumption. Numerous other crops are attacked, mainly on their leaves (CAPS, 2010)

Damage caused by *S. littoralis* in Europe was minimal until about 1937. In 1949, there was a catastrophic population explosion in southern Spain, which attacked alfalfa, potatoes, and other vegetable crops. At present, this noctuid pest is economically important in Cyprus, Israel, Malta, Morocco, and Spain (except the north). In Italy, it is of great importance on protected crops of ornamentals and vegetables (Inserra and

Calabretta, 1985). In Greece, *S. littoralis* causes slight damage in Crete on alfalfa and clover only. In North Africa, tomato, *Capsicum* spp. (peppers), cotton, corn, and other vegetables are affected. In Egypt, it is one of the most serious cotton pests (CAPS, 2010).

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

Known Hosts

S. littoralis is highly polyphagous and has hosts in over 40 plant 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), *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 esculentum* (tomato), *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).

Pathogens or Associated Organisms Vectored

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

Known Distribution

The northerly distribution limit of *S. littoralis* in Europe is related to the climatic zone in which winter frosts are not frequent. It occurs throughout Africa and extends eastward into Turkey and north into eastern Spain, southern France and northern Italy. However, this boundary is probably the extent of migrant activity only (CAPS, 2010). 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. Therefore, low winter temperatures are an important limiting factor affecting the northerly distribution, especially in a species without known diapause (Miller, 1976; Sidibe and Lauge, 1977).

Africa: Algeria, Angola, Benin, Botswana, Burkina Faso, Burundi, Cameroon, Cape Verde, Central African Republic, Chad, Comoros, Congo, DR Congo, Cote d'Ivoire, Egypt, Equatorial Guinea, Eritrea, Ethiopia, Gabon, Gambia, Ghana, Guinea, Kenya, Liberia, Libya, Madagascar, Malawi, Mali, Mauritania, Mauritius, Morocco, Mozambique, Namibia, Nigeria, Reunion, Rwanda, Senegal, Siera Leone, Somalia, South Africa, Sudan, Swaziland, Tanzania, Togo, Tunisia, Uganda, Zambia, and Zimbabwe. **Asia:** Afghanistan, Bangladesh, Brunei, and India. **Europe:** France, Germany, Greece, Italy, Malta, Portugal, Spain, and United Kingdom. **Middle East:** Bahrain, Cyprus, Iran, Iraq, Israel, Jordan, Lebanon, Oman, Saudi Arabia, Syria, United Arab Emirates, and Yemen. **Oceania:** American Samoa.

Potential Distribution within the United States

This pest has been intercepted at U.S. ports on plant parts such as leaves and flowers. The potential range of *S. littoralis* in U.S. 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 able to periodic spread into northern states and even Canada by late summer or early fall. Venette et al. (2003) suggest that about 49% of the continental United States would be suitable for *S. littoralis.* A recent risk analysis by USDA-APHIS-PPQ-CPHST shows that portions of Alabama, Arkansas, Florida, Georgia, Louisiana, Mississippi, South Carolina, and Texas are at the greatest risk from *S. littoralis*. Portions of most states within the continental United States have low to moderate risk of *S. littoralis* establishment based on climate and host range.

Survey

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

The Lure Product Name 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, 2010)
Trapping: 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 mixture of the minor sex pheromone component, (9*Z*, 12*Z*)-9,12-tetradecadienyl acetate and the major component (9*Z*,11*Z*)-9,11-tetradecadienyl acetate was widely used in Israel to attract males.

Sex-pheromone baited delta traps remained attractive for approximately two weeks, but effectiveness declined after three to four weeks of use (Ahmad, 1988). To monitor male flight activity in vegetable production areas, delta traps were placed 1.7 m (5.57 ft.) 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 four weeks of use (Ahmad, 1988). Traps are deployed at a similar height (1.5 m; 4.92 ft.) to monitor male flight in cotton (Salem and Salama, 1985). Catches in pheromone traps do not correlate as well with densities of egg masses in cotton fields as do 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. 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. A trained taxonomist should confirm all specimens. 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 to 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).

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. *S. littoralis* is difficult to distinguish from *S. litura* without close examination of the genitalia. See the Field Diagnostics and Wing Diagnostics aids by Brambila (2008a, b) for additional information (http://caps.ceris.purdue.edu/webfm_send/553 and http://caps.ceris.purdue.edu/webfm_send/554).

*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: Observation of adult genitalia is often the only method to separate species.

Easily Confused Pests

S. littoralis is also confused with *S. dolichos*, *S. ornithogalli*, *S. latifascia* and other *Spodoptera* species (present in the United States). Consult a trained taxonomist for confirmation of samples.

S. littoralis is often confused with *S. litura.* The variability and similarity of the two species makes it difficult to correctly distinguish them. 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 Mochida (1973) and Brown and Dewhurst (1975).

Figure 5. *Spodoptera exigua* larva, Frank Peairs, Colorado State University, www.bugwood.org.

Figure 6. Adult moths of *S. ornithogalli,* female (left) and male (right). Photos courtesy of Jim Vargo, Mississippi Entomological Museum, http://www.mothphotographersgroup.msstate.edu/species.php?hodges=9669.

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. *S. 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. 5), but *S. littoralis* larvae are light or dark brown, while *S. exigua* are brown or green. *S. littoralis* is also larger than *S. exigua* (Venette et al., 2003).

Adults of *S. littoralis* are almost nearly identical in appearance to the yellow striped armyworm *S. ornithogalli* (Fig. 6), a common pest in the United States. The hindwings of female *S. littoralis* are darker than those of *S. ornithogalli* (USDA 1982 as cited in CAPS 2010).

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Spodoptera litura

Scientific Name
Spodoptera litura Fabricius

Synonyms:
Spodoptera littoralis, Mamestra albisparsa, Noctua elata, Noctua histrionica, Noctua litura, Prodenia ciligera, Prodenia declinata, Prodenia evanescens, Prodenia glaucistriga, Prodenia litura, Prodenia littoralis, Prodenia subterminalis, Prodenia tasmanica, and *Prodenia testaceoides*

Common Name(s)
Rice cutworm, taro caterpillar, tobacco budworm, cotton leafworm, cluster caterpillar, cotton worm, Egyptian cotton leafworm, tobacco caterpillar, tobacco cutworm, tobacco leaf caterpillar, and 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 2012

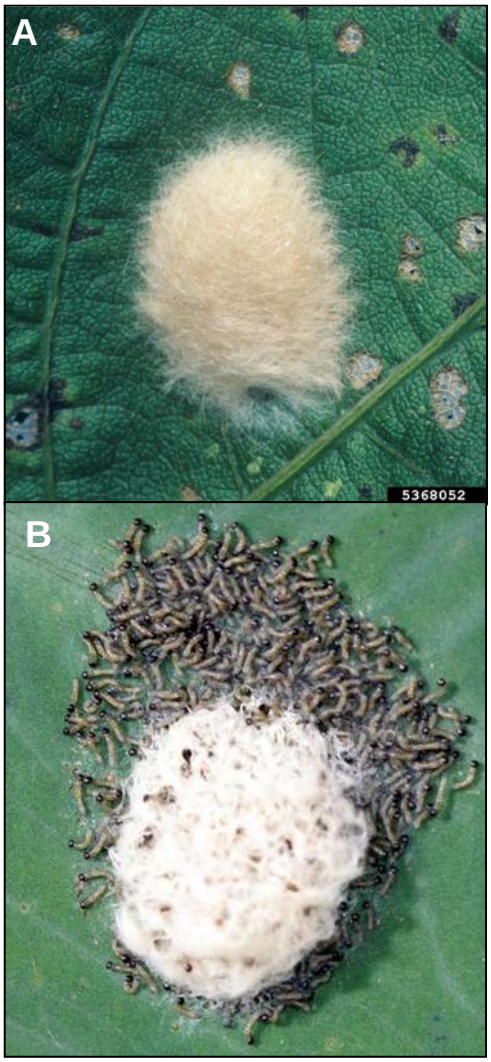

Figure 1. (A) Eggs covered with hairy scales. Photo courtesy of Merle Shepard, Gerald R. Carner, and P.A.C Ooi, Insects and their Natural Enemies Associated with Vegetables and Soybean in Southeast Asia, www.bugwood.org (B) Emerging larvae, Photo courtesy of Amy Carmichael, Queensland University of Technology, www.wikipedia.org.

Pest Description
Eggs: Eggs are about 0.4 to 0.7 mm (0.016 to 0.028 in.) in diameter, spherical, and somewhat flattened in shape. They are sculptured with about 40 longitudinal ribs. Eggs are laid in batches covered with pale orange-brown or pink hairy scales derived from the tip of the abdomen of the female moth (Fig. 1A) (Pearson, 1958; CABI, 2010). The female scales make egg batches look like an irregular furry mass on the underside of leaves (CAPS, 2010). Eggs are pearly green, and turn black as they age (CABI, 2010).

Larvae: Freshly hatched larvae are small and blackish green with a distinct black band on the first abdominal segment. Mature larvae are 40 to 50 mm (1.57 to 1.97) long with

two large black spots on the first and eighth abdominal segments (Hill, 1975; CABI, 2010). Fully-grown larvae are stout and smooth with scattered short setae. The head is shiny black and has conspicuous black tubercles, each with a long hair on each segment. The color of fully-grown larvae is not constant: it varies from dark gray to dark brown or black. Larvae sometimes have yellow dorsal and lateral stripes of unequal width (Fig. 2). The lateral yellow stripe is bordered dorsally with series of semilunar black marks. When disturbed, the larvae curl into a tight spiral with the head protected in the center (CAPS, 2010).

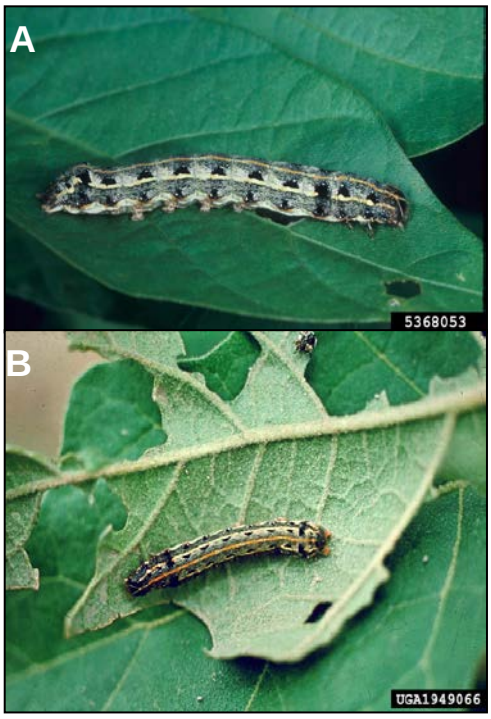

Figure 2. *S. litura* larvae. Photos courtesy of (A) Merle Shepard, Gerald R.Carner and P.A.C Ooi, Insects and their Natural Enemies Associated with Vegetables and Soybean in Southeast Asia, and (B) K. Kiritani, www.bugwood.org .

Pupae: Pupae are 18 to 22 mm (0.71 to 0.86 in.) long, reddish brown in color, and enclosed inside rough earthen cases in the soil. The last abdominal segment ends in two strong hooks (CABI, 2010).

Adults: Adults are 14 to 18 mm (0.55 to 0.71 in.) long with a 28 to 38 mm (1.1 to 1.5 in.) wingspan (Hill, 1975). The body is whitish to yellowish in color and is suffused with pale red. Forewings are dark brown with lighter shaded lines and stripes (Fig. 3A). Hindwings are whitish with a violet sheen, the margin is dark brown, and wing venation is brown (Fig. 3B). The thorax and abdomen are orange to light brown with hair-like tufts on the dorsal surface. The head is covered with tufts of light and dark brown scales.

See Schmutterer (1969) and Brown and Dewhurst (1975) for additional information.

Biology and Ecology

Two to five days after emergence, female moths lay 50 to 300 eggs in masses on the lower surface of leaves (preferred). The eggs hatch in three to four days (Chari and Patel, 1983). A single female lays a total of 1500 to 2500 eggs in about six to eight days. Castor bean is the most preferred host for ovipositing females (Chari and Patel, 1983). Freshly irrigated fields are also very attractive to ovipositing females (CAPS, 2010). Three peak periods of egg laying have been observed in the third weeks of June, July, and in mid-August. Groups of freshly hatched larvae feed 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 (CAPS, 2010). *S. litura* larvae generally pass through six instars. 1st to 3rd instars generally remain on the underside surface of leaves. The 4th to 6th instars drop onto the ground, root around 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°F). Mean female longevity was 8.3 days and mean fecundity was 2673 eggs. Mean male longevity was 10.4 days. Male and female moths do not mate on the night of emergence; 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°C).

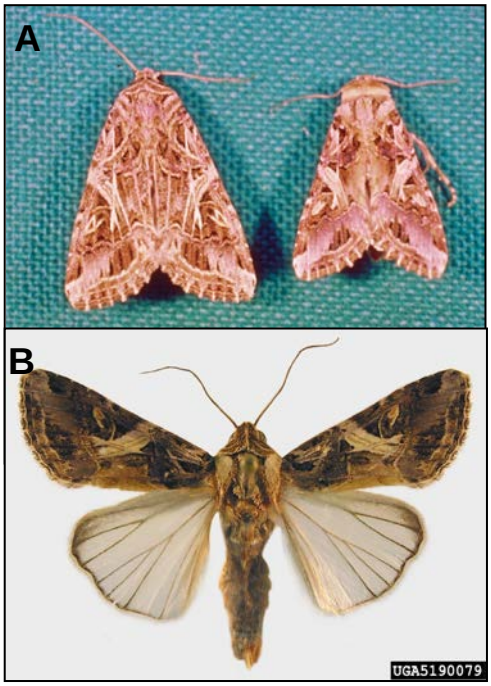

Figure 3. *S. litura* adults. Photos courtesy of (A) K. Kiritani and (B) Natasha Wright, Florida Department of Agriculture and Consumer Services, respectively, www.bugwood.org.

Fecundity is adversely affected by high temperature and low humidity (about 960 eggs laid at 30°C and 90% RH and 145 eggs at 35°C (95°F) and 30% RH) (EPPO/CABI, 1997). 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 and 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). A 24-hour 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* and resulted in a shorter larval period, 100% pupation, a shortened pupal period, and 100% adult emergence.

The eggs hatch is about four days in warm areas or up to 11 to 12 days in winter (EPPO/CABI, 1997). 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 required 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 11:30 PM and 12:30 AM under controlled conditions. The duration of mating behaviors 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 male was 10.3 and per female was 3.1 (Ahmed et al., 1979). Ohbayashi et al. (1973) showed two peaks in mating behavior at 11:00 PM (3 hours after initiation of a dark period) and a minor peak at 3:00 AM (1 hour before the end of the dark period).

The pre-pupal and pupal period of *S. litura* is spent in earthen cells or cocoons in the soil and lasts about 11 to 13 days at 25°C (77°F). 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 (4.7 in.) deep. Most larvae pupated at a 4 cm (1.6 in.) depth across soil types.

Symptoms/Signs

On most crops, damage can be caused by extensive feeding by larvae, leading to complete stripping of the plants. Larvae are leaf eaters but sometimes act as a cutworm with crop seedlings. *S. litura* feeds on the lower surface of leaves (Fig. 4) and causes feeding scars and skeletonization of leaves. Early larval stages remain together, radiating out from the egg mass. Initial small feeding points eventually spread over the entire leaf. Later stages are solitary (CAPS, 2010). 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).

Figure 4. *S. litura* damage on persimmon leaf. Photo courtesy of Yuan-Min Shen, Taichung District Agricultural Research and Extension Station, www.bugwood.org.

On cotton: The pest may cause considerable damage by feeding on leaves and bolls. Leaves are heavily damaged and bolls have large holes. Yellowish-green to dark green larval excrement may surround bore holes in bolls (EPPO /CABI, 1997).

On corn: Corn stems are often mined by *S. litura* and young grains in the ear may also be damaged (EPPO/CABI, 1997).

On tobacco: Leaves develop irregular, brownish-red patches and the stem base may be gnawed off (EPPO/CABI, 1997).

Pest Importance

S. litura is an extremely serious pest, the larvae of which can defoliate many economically important crops. It is 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 injure tubers and roots. Hosts include field crops grown for food and fiber, plantation and forestry crops, as well as certain weed species (CABI, 2010).

Most studies on the economic impact of *S. litura* have been conducted in India, where it is a serious pest of a variety 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). On tobacco in

India, it was estimated that two, four, and eight larvae per plant reduced yield by 23 to 24, 44.2 and 50.4%, respectively (Patel et al., 1971). On *Colocasia esculenta* (taro), an average of 4.8 4th instar larvae per plant reduced yield by 10%. Aroid tuber crops (including taro) suffered yield losses of up to 29% as a result of infestation by *S. litura*, *Aphis gossypii* (cotton or melon aphid), and spider mites (Pillai et al., 1993). Larvae (2.3 and 1.5) reduced yield of aubergines (eggplant) and *Capsicum*, respectively in glasshouses by 10% (Nakasuji and Matsuzaki, 1977). 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 (CAPS, 2010). Larvae also attack exposed tubers when young succulent leaves were unavailable (CABI, 2010). *S. litura* is also a pest of sugarbeet, with infestations presenting in March and peaking in late March and April (Chatterjee and Nayak, 1987). Severe infestations in beet lead to the skeletonization of leaves, as well as feeding holes in roots that render 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.

S. 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 (CAPS, 2010). In controlled experiments on soybeans in India, crops chemically protected from *S. litura* and other pests yielded over 42% more than crops that were not sprayed (Srivastava et al., 1972).

S. litura is responsible for brown flag syndrome in banana (Ranjith et al., 1997) and 5 to 10% fruit damage in grapes (Balikai et al., 1999).

Insecticide resistance has been reported in India (Armes et al., 1997; Kranthi et al., 2001) and Pakistan (Ahmad et al., 2007).

Known Hosts

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, aubergine (eggplant), *Brassica* spp. (mustards), *Capsicum* spp. (peppers), cucurbits, bean, potato, sweet potato, grape, and cowpea. Other hosts include ornamentals, wild plants, weeds, and shade trees (for example, *Leucaena leucocephala,* a shade tree of cocoa plantations in Indonesia). Balasubramanian et al. (1984) found 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* spp., *Brassica oleracea*

var. *botrytis* (cauliflower), *Brassica oleracea* var. *capitata* (cabbage), *Camellia sinensis* (tea), *Capsicum frutescens* (chili), *Castilla 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), *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* spp. (roses), *Sesbania grandiflora* (agati), *Solanum esculentum* (tomato), *Solanum melongena* (aubergine, eggplant), *Solanum tuberosum* (potato), *Sorghum bicolor* (sorghum), *Syzygium aromaticum* (clove), *Tectona grandis* (teak), *Theobroma cacao* (cocoa), *Trifolium* spp. (clover), *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).

Pathogens or Associated Organisms Vectored

S. litura is not a known vector and does not have any associated organisms.

Known Distribution

S. litura is widely distributed throughout tropical and temperate Asia, Australasia, and the Pacific Islands (Kranz et al., 1977).

Asia: Afghanistan, Bangladesh, Brunei Darussalam, Cambodia, China, Christmas Island, Cocos Islands, India, Indonesia, Iran, Japan, Korea, Laos, Lebanon, Malaysia, Maldives, Myanmar, Nepal, Oman, Pakistan, Philippines, Singapore, Sri Lanka, Syria, Thailand, and Vietnam. **Europe:** Russia. **Africa:** Reunion. **North America:** United States (Hawaii). **Oceania:** American Samoa, Australia, Belau, Cook Islands, Federated states of Micronesia, Fiji, French Polynesia, Guam, Kiribati, Marshall Islands, New Caledonia, New Zealand, Niue, Norfolk Island, Northern Mariana Islands, Papua New Guinea, Pitcairn Islands, Samoa, Solomon Islands, Tonga, Tuvalu, Midway Islands, Wake Island, Vanuatu, and the Wallis and Futuna Islands.

Potential Distribution within the United States

The pest has been present in Hawaii since 1964 (CABI, 2010). *S. litura* was identified in a sample from a Miami-Dade County, Florida nursery in April 2007. Pheromone traps were placed over a nine square mile area and have yielded no additional finds. A recent risk analysis by USDA-APHIS-PPQ-CPHST shows that portions of Alabama, Arkansas, California, 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 many areas of the United States.

Survey

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

The Lure Product Name 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: (From Venette et al. 2003; CABI, 2010)
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). Yang et al. (2009) successfully used (*Z,E*)-(9,11)- tetradecadienyl acetate and (*Z,E*)-(9,12)-tetradecadienyl acetate in a 10:1 ratio for trapping *S. litura* in China. The compounds are most effective in a ratio between 4:1 and 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) and in China in a 10:1 ratio from NewCon Incorporated (Yang et al., 2009). 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 (6.56 ft.) above the ground level caught significantly more male *S. litura* than those placed at higher or lower heights [ranging from 0.5 to 4.0 m (1.64 to 13.12 ft.)]. Ranga Rao et al. (1991) suggest trap placement at 1 m (3.3 ft.).

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; Singh and Sachan, 1993; Ranga Rao et al., 1991). 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; Nakamura, 1977; Oyama, 1977). Kamano et al. (1976) also mentioned a trap composed of two cylindrical parts and four cones made of wire screen that open 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; 17.7 x 3.9 in.) 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. *S. 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. Sweep net for adults and larvae at dawn or dusk. Third instar larvae rest in upper soil layers during the day. 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.

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).

Key Diagnostics/Identification

CAPS-Approved Method*: Confirmation of *S. litura* is by morphological identification. *S. litura* is difficult to distinguish 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).

*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). *S. 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 and by using wing diagnostics are available at these websites: http://caps.ceris.purdue.edu/webfm_send/555 and http://caps.ceris.purdue.edu/webfm_send/556.

For additional images, including photos of host damage see http://www.padil.gov.au/viewPestDiagnosticImages.aspx?id=418.

Easily Confused Pests

S. litura is often confused with *S. littoralis.* The variability and similarity of the two species makes it difficult to correctly distinguish them. 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), 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. *S. litura* is not established in the continental United States, but has been reported in Hawaii.
S. litura is also confused with *S. dolichos*, *S. ornithogalli*, *S. pulcella* and other *Spodoptera* species (present in the United States).

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Thaumatotibia leucotreta

Scientific Name
Thaumatotibia leucotreta Meyrick

Synonyms:
Cryptophlebia leucotreta, Argyroploce leucotreta

Common Name(s)
False codling moth, citrus codling moth, orange moth, and orange codling moth

Type of Pest
Moth

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

Reason for Inclusion In Manual
CAPS target: AHP Prioritized Pest List 2003 through 2012

Pest Description
Eggs: Eggs are flat, oval, and whitish (Hill, 1987). They are 0.9 to 1 mm (0.035 to 0.039 in.) long (Hill, 1987; USDA-APHIS-PPQ, 2010-2011). The egg surface has a shiny, reticulate sculpture (USDA-APHIS-PPQ, 1983; USDA-APHIS-PPQ, 2010-2011). Newly laid eggs are white to cream in color, and turn reddish as they age. The black head capsule of the larva becomes visible through the shell as hatching nears (Daiber, 1979).

Figure 1. Larvae of *T. leucotreta.* Photos courtesy of (A) M.J. van der Straten, © Plant Protection Service, the Netherlands, http://ukmoths.org.uk and (B) Tertia Grové, Institute for Tropical and Subtropical Crops, www.bugwood.org.

Larvae: First instar larvae are 1 to 1.3 mm (0.039 to 0.051 in.) long and creamy white with tiny black spots. Each spot has a short hair. The head is dark brownish black (USDA-APHIS-PPQ, 2010–2011). Subsequent instars are a characteristic pink-red that is less intense on the underside. The fifth instar is 12 to 20 mm (0.47 to 0.79 in.) long. The coloring is a diffuse pink with orange yellow on the sides, top, and legs (Fig. 1A & B). The head is light maroon, and the pronotum is yellowish brown (Fig. 1A). The pronotum juts out in front, to both sides, and to the rear. The anal comb has two to seven teeth (USDA-APHIS-PPQ, 1983; USDA-APHIS-PPQ, 2010–2011). There are no characteristics that allow larval identification with an unaided eye (Sofberg, 1948).

Pupae: The pupae are encased in a lightly woven cocoon with soil and leaf fragments. The pupae are 7 mm (0.28 in.) long and yellow to dark brown. Segments have transverse row spines. It is possible to determine the sex using morphological differences on pupal case (Fig. 2) (Daiber, 1979). Males are smaller than females and have two knobs side by side in the center on the ventral side of the ninth abdominal segment; females lack these knobs (USDA-APHIS-PPQ, 1983; USDA-APHIS-PPQ, 2010–2011).

Figure 2. Pupae of *T. leucotreta*. Photo courtesy of J.H. Hofmeyr, Citrus Research International, www.bugwood.org.

Adults: Adult *T. leucotreta* bodies are 6 to 9 mm (0.24 to 0.35 in.) long and 2.5 mm (0.098 in.) wide. The wingspan of adult female is 17 to 20 mm (0.67 to 0.79 in.), and the wingspan of adult male is 15 to 18 mm (0.59 to 0.71 in.). Color is grayish brown to dark brown or black (Fig. 3). The forewings are broad and elongated with a black triangular patch and are fringed with hairs. Hindwings are a lighter grayish brown and darker toward the outer margins (Fig. 3A & B). The male can be distinguished by a large, pale gray genital tuft (Fig. 3C), hind wings with a dense brush of grayish white hairs (Fig. 3A), hind tibia with tufts of modified scales on the inner side (Fig. 3D), and slightly reduced hindwings with a deep semicircular pocket in the anal angle (Fig. 3A) (Karvonen, 1983; USDA-APHIS-PPQ, 1983; USDA-APHIS-PPQ, 2010–2011). The male and female genitalia are illustrated by Bradley (1959) and Larsen (1978) (Karvonen, 1983).

Biology and Ecology

In South Africa, false codling moth has four to six non-discrete generations per year (Georgala, 1969; Sofberg, 1954); populations are low in the spring but increase in summer (Daiber, 1976). Females lay individual eggs (100 to 250 per female) on fruit or foliage (Catling and Aschenborn, 1974; Daiber 1978). Leaves are the preferred oviposition sites, and eggs are most often laid on the upper side of leaves (Daiber, 1976). The female may lay her eggs on leaves adjacent to maturing fruits (Daiber, 1976). In cotton, most eggs are laid on large green bolls at the bottom and center of the plant (Reed, 1974).

Females tend to oviposit on prematurely ripened fruit or wounded fruit when compared to healthy fruit at a normal state of development (Newton and Mastro, 1989). Neonate larvae penetrate the fruit and complete development within the fruit. Mature larvae leave the fruit and spin cocoons near the soil or in bark crevices. Diapause or a resting stage has not been recorded.

Daiber (1980) showed that *T. leucotreta* adults live longest at 15°C (59°F) and most eggs were laid at 25°C (77°F). Oviposition at 20 and 25°C (68 and 77°F) increased rapidly soon after the first egg was laid but only gradually at 15°C (59°F) to reach peak numbers some time after the initial egg lay. Very few eggs were laid at 10°C (50°F).

T. leucotreta most likely cannot survive outside of tropical and subtropical regions (Bradley, 1959; Bradley et al., 1979; Karvonen, 1983).

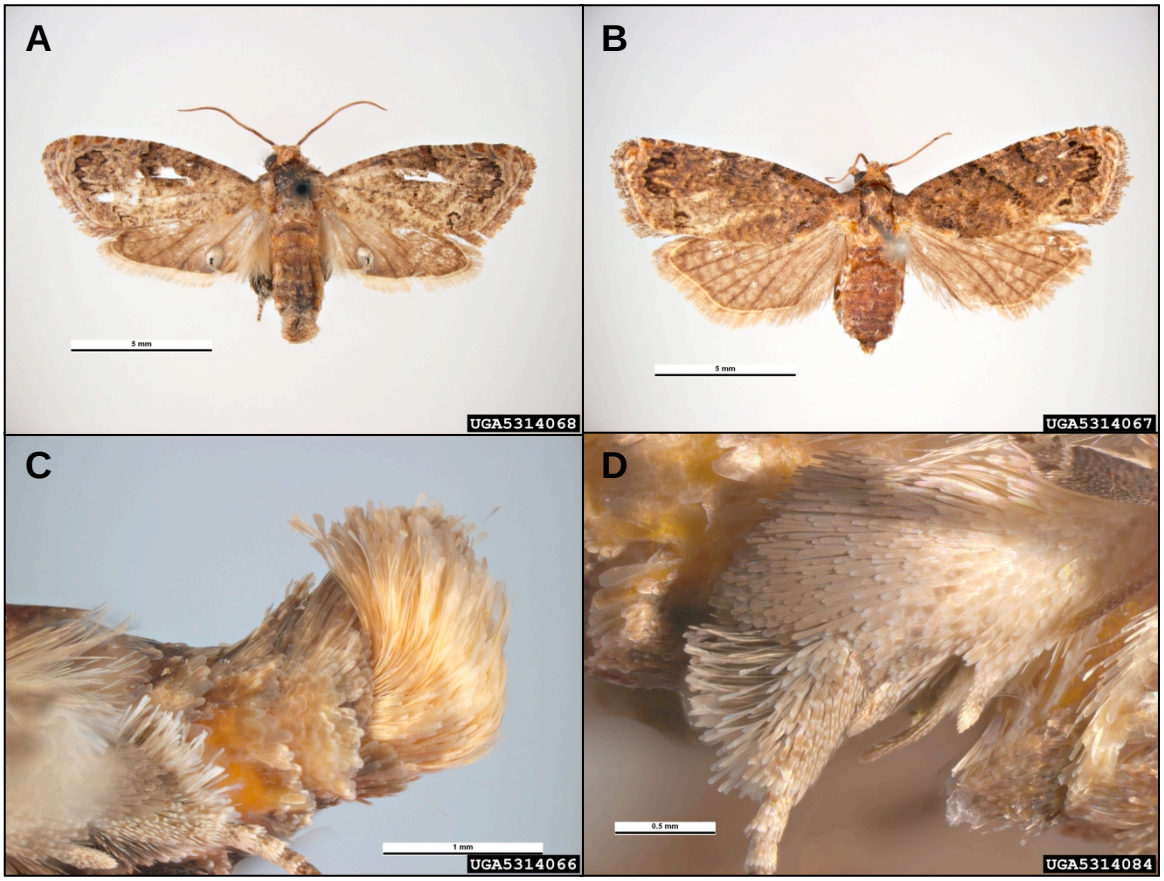

Figure 3. (A) Male, (B) female adults of *T. leucotreta*. (C) Abdomen (posterior-lateral view), (D) hind tibia (right) of male adult, Photos courtesy of Pest and Diseases Image Library, www.bugwood.org.

Symptoms/Signs

From (CAPS, 2010):

In general, the habit of internal feeding by false codling moth larvae displays few symptoms. Emerging larvae bore into a fruit and usually feed just below the fruit surface. Cannibalism among young larvae ensures that usually only one caterpillar matures in each fruit. When full-grown the larvae bore their way out of the fruit to seek a site for pupation, the rind around the point of infestation takes on a yellowish-brown color as the tissue decays and collapses. Larval feeding and development can affect fruit development at any stage and may cause premature fruit drop.

In Cotton: Eggs are laid on large green bolls, usually singly, but sometimes a few are laid together (Reed, 1974; Hill, 1987). An average of eight eggs may be found on a single fruit (Hill, 1987). False codling moth feeds mainly on large, but not mature, green bolls (Reed, 1974; van der Geest and Evenhuis, 1991). Larvae often wander for a short time on fruit before mining into the boll (Reed, 1974; van der Geest and Evenhuis,

1991). Larvae mine into the boll wall. Dark frass may be found at the point of entry (Hill, 1987; van der Geest and Evenhuis, 1991). The younger larvae feed almost entirely inside the boll wall itself, but the older larvae penetrate the inner septum and feed on the developing seeds and lint (Reed, 1974; Hill 1987). Larvae stay inside the boll until just before pupation (Reed, 1974). Larval penetration of cotton bolls facilitates entry of other microorganisms that can rot and destroy the boll, especially in humid conditions (Reed, 1974). The cultivars Edranol, Hass, and Pinkerton were the most susceptible to attack by false codling moth (Stibick, 2006).

See USDA-APHIS-PPQ New Pest Response Guidelines (2010-2011) for information on other crops.

Pest Importance

The false codling moth is a major pest of cotton and citrus in many parts of Africa. In unsprayed citrus orchards in South Africa, average crop loss in 1980 to 1981 was 4.3 to 8.1% (Schwartz and Anderson, 1983).

In Cotton: *T. leucotreta* is usually a minor pest, but sometimes it is a seriously damaging pest of cotton (Hill, 1987). In Uganda by 1969, *T. leucotreta* was the most damaging pest of sprayed cotton; over 90% of late green bolls on later sown cotton were damaged by false codling moth (Reed, 1974). In the early 1970's in Uganda, over 90% of rotten cotton bolls had insect damage, at least 60% of which was caused by *T. leucotreta* (Reed et al., 1970; 1974).

According to California Department of Food and Agriculture (CDFA) (2008), commonly grown agricultural hosts in California for false codling moth include citrus, grapes, peach, plum, cherry, beans, tomato, pepper, persimmon, apricot, olive, pomegranate, English walnut, and corn. Based on its status as a pest in Africa, establishment of false codling moth in California and/or in other parts of the United States could result in significant economic losses. False codling moth would likely be a significant production and quarantine issue for numerous agricultural commodities. In California alone, the annual combined gross value of the top ten agricultural commodities which would be directly impacted by this pest is over $7.1 billion, which amounts to 22% of the total agricultural value for the State (USDA NASS, 2007).

Known Hosts

False codling moth feeds on more than 70 host plants (CABI, 2010). Economically important hosts include avocado, banana, bean, cacao, carambola, castor bean, citrus (*Citrus sinensis*, *Citrus spp.*), coffee, corn (*Zea mays*), cotton, cowpea, English walnut, grape, guava, macadamia nut, mango, okra, olive, peach, pepper/pimento, persimmon, plum, pineapple, pomegranate, sorghum, and tea (Venette et al., 2003).

Major hosts:
Abelmoschus esculentus (okra), *Abutilon hybridum* (flowering maple), *Abutilon* x *hybridum* (Chinese lantern), *Ananas comosus* (pineapple), *Averrhoa carambola* (carambola), *Camellia sinensis* (tea), *Capsicum* spp. (peppers), *Citrus* spp., *Coffea*

arabica (coffee), *Gossypium* spp. (cotton)*, Litchi chinensis* (litchi), *Macadamia spp.* (macadamia)*, Mangifera indica* (mango), *Olea* spp. (olive), *Persea americana* (avocado), *Prunus armeniaca* (apricot), *Prunus domestica* (plum), *Prunus persica* (peach), *Prunus spp.* (cherry), *Psidium guajava* (guava), *Punica granatum* (pomegranate), *Quercus* spp. (oak, acorns), *Ricinus communis* (castor bean), *Sorghum bicolor* (sorghum), and *Zea mays* (corn).

Minor / Wildhosts:
Abutilon spp. (Indian mallow), *Acacia nilotica* (acacia), *Acacia tortilis* (umbrella thorn)*, Annona cherimola* (cherimoya), *Annona glabra* (pond apple), *Annona muricata* (soursop), *Annona reticulata* (Bullock's heart, custard apple), *Annona squamosa* (sugar apple), *Azanza garckeana* (snot apple), *Bauhinia galpinii* (red bauhinia), *Bequaertiodendron magalismontanum* (stamvrug), *Butyrospermum parkii* (shea butter tree), *Caesalpinia pulcherrima* (pride-of-Barbados), *Caesalpinia* spp. (nicker), *Calotropis procera* (sodom apple), *Capparis tomentosa* (African caper), *Carya illinoensis* (pecan), *Cassia petersiana* (monkey pod), *Catha edulis* (khat), *Ceiba pentandra* (kapok), *Chrysophyllum cainito* (star apple), *Chrysophyllum palismontatum* (stamvrugte), *Cola nitida* (bitter cola), *Combretum apiculatum* (apiculatum), *Combretum apiculatum* (rooibos), *Combretum zeyheri* (raasblaar), *Cyphomandra betacea* (tree tomato), *Diospyros mespiliformis* (Jjakkalsbessie), *Diospyros* spp. (persimmon), *Englerophytum magalismontanum, Eriobotrya japonica* (loquat), *Eugenia uniflora* (Surinam-cherry), *Ficus capensis* (wild fig), *Flacourtia indica* (governor's-plum), *Garcinia mangostana* (mangosteen), *Harpephyllum caffrum* (kaffir-plum), *Hibiscus cannabinus* (kenaf), *Hibiscus spp.* (hibiscus), *Juglans regia* (English walnut), *Juglans spp.* (walnut), *Solanum* (*Lycopersicon*) *esculentum* (tomato), *Mimusops zeyheri* (Transvaal red milkwood), *Musa paradisiaca* (banana), *Pennisetum purpureum* (elephant grass), *Phaseolus lunatus* (lima bean), *Phaseolus* spp. (bean), *Physalis ixocarpa* (tomatillo) *Physalis* spp. (groundcherry), *Piper spp.* (pepper), *Podocarpus falcatus* (yellowwood), *Podocarpus* spp. (plum pine), *Pseudolachnostylis maprouneifolia* (kudu-berry), *Royena pallens* (pale-branched Royena), *Saccharum officinarum* (sugarcane), *Schotia* spp. (boerboon), *Sclerocarya birrea* (marula) *Sechium edule* (chayote), *Sida* spp. (fanpetals), *Solanum melongena* (eggplant), *Synsepalum dulcificum* (miraculous berry), *Syzygium cordatum* (waterbessie), *Syzygium jambos* (rose-apple), *Theobroma cacao* (cacao),*Triumfetta* spp. (bur weed), *Vangueria infausta* (wild medlar) *Vigna* spp. (cowpea), *Vitis* spp. (grape), *Xeroderris stuhlmannii* (wing bean), *Ximenia caffra* (suurpruim), *Yucca* spp. (yucca), and *Ziziphus* spp. (jujube).

Pathogens or Associated Organisms Vectored
T. leucotreta is not a known vector and does not have any associated organisms. The wounds produced by *T. leucotreta*, however, can provide an entrance for pathogens, especially under humid conditions (Reed, 1974).

Known Distribution
Africa: Angola, Benin, Burkina Faso, Burundi, Cameroon, Cape Verde, Central African Republic, Chad, Congo, Eritrea, Ethiopia, Gambia, Ghana, India, Ivory Coast, Kenya, Madagascar, Malawi, Mauritius, Mozambique, Niger, Nigeria, Reunion, Rwanda, Saint

Helena, Senegal, Sierra Leone, Somalia, South Africa, South Sudan, Sudan, Swaziland, Tanzania, Togo, Uganda, Zambia, Zimbabwe

Potential Distribution within the United States

From CAPS (2010):

Increased international trade and tourism between the United States and many African countries in recent years has increased the risk of introduction of this pest. Since 1984, false codling moth has been intercepted over 1500 times on 99 plant taxa at 34 U.S. ports of entry. In June 2005, live *T. leucotreta* caterpillars were found at California's border stations inside previously cold treated Clementine citrus from South Africa. Its discovery in California is a new record for the Americas. *T. leucotreta* is not known to be established in California.

On June 16, 2005, California Department of Food and Agriculture (CDFA) inspectors found one live and one dead larva on a shipment of South African clementines at the California border station in Needles. Both a CDFA lab and the USDA Systematic Entomology Laboratory (SEL) Specialist identified the larvae as False Codling Moth (FCM), *Thaumatotibia leucotreta* Meyrick. The fruit had entered the United States in the port of Philadelphia (PA) off the vessel Nova Zembla. Initial review of the cold treatment records did not reveal failures in the treatment. On June 20, a second live larva was intercepted on a separate shipment of South African clementines in California. This shipment came on the vessel Fuji Star on June 14, 2005. This larva was identified by CDFA as FCM, *T. leucotreta*. An eradication program would be triggered if two moths were detected within one life cycle and within three miles of each other, or a mated female was found, or any immature stage (egg, larva, or pupa) was found. FCM has not triggered an eradication project in California at this time. Survey using traps and some fruit sampling continue around the Ventura County find (CAPS, 2010).

A recent risk analysis by USDA-APHIS-PPQ-CPHST shows that portions of Alabama, Arkansas, California, Florida, Georgia, Louisiana, Mississippi, North Carolina, South Carolina, and Texas are at the greatest risk from *T. leucotreta*. Establishment of *T. leucotreta* is precluded in the northern United Stated based on climate and host range.

Survey

CAPS-Approved Method*: The CAPS-approved method is a trap and lure combination.

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

1) Wing Trap, Paper
2) Wing Trap, Plastic

The Lure Product Name is "*Thaumatotibia leucotreta* Lure." The lure is effective for 56 days (8 weeks).

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 Note: The wing trap and the diamond trap are both effective traps for *Thaumatotibia leucotreta*. In order to standardize data and trap procurement, it is preferable that states use the wing trap. However, if states find reason to use the diamond trap, it is acceptable for negative data reporting. Diamond traps will not be available through the IPHIS Survey Supply Ordering System.

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:

For surveys in cotton, fields near high-risk areas such as citrus and stone fruit should be monitored using pheromone traps. The pheromone traps should be placed at a frequency of 1 trap per 4 hectares. Traps should be inspected weekly. Cotton should also be inspected visually for the presence of FCM during the growing season. The first four rows bordering citrus or stone fruit orchards should be examined carefully.

Trapping: Male *T. leucotreta* are attracted to a two-component blend of (*E*)-8-dodecenyl acetate and (*Z*)-8-dodecenyl acetate. These components are most effective when used in a ratio between 70:30 and 30:70 (*E*:*Z*) (Persoons et al., 1977; Venette et al., 2003). Schwartz and Anderson (1983) used PVC pipe type traps in citrus orchards. The traps were baited with 10µL synthetic female pheromone in rubber septa. The lures were replaced every 8 weeks (Schwartz and Anderson, 1983). Burger et al. (1990) found that the presence of 7-vinyldecyl acetate strongly reduced the attractiveness of the pheromone (*E*)- and (*Z*)-8-dodecenyl acetate. More recently, Newton et al. (1993) refined the sex pheromone and reported that a 90:10 ratio was optimal. Stibick (2006) recommends utilizing a 50:50 ratio.

A loading rate of 0.5 to 1.0 mg per septum was found to attract the greatest number of males. The pheromone blend (1 mg applied to a rubber septum) has been used effectively with Pherocon 1C traps to capture male *T. leucotreta* (Newton et al., 1993). Delta traps have also been used, but these have performed less well than either the Hoechst Biotrap or Pherocon 1C traps. Traps using closed polyethylene vials to dispense pheromones captured more moths than traps using rubber septa (using a 50:50 blend of (*E*)- and (*Z*)-8-dodecenyl acetate). Lures should be replaced every eight weeks. Traps should be placed approximately 5 ft. (1.5 m) high. Hofmeyr and Burger (1995) developed a prototype controlled release dispenser that was capable of releasing sex pheromone without replacement for more than seven months. Pheromone traps (homemade sticky trap with unspecified pheromone blend) have been used to monitor the number of *T. leucotreta* adult males in citrus orchards (Daiber, 1978) and detect the presence of the pest in peach orchards (Daiber, 1981).

Pheromone lures with (*E*)- and (*Z*)-8-dodecenyl acetate may also attract *Cydia cupressana* (native), *Hyperstrotia spp*., *Cydia atlantic*a (exotic), *Cydia phaulomorpha* (exotic) and *Cryptophlebia peltastica* (exotic).

Visual survey: Visual inspections of plant materials may be used to detect eggs, larvae, and adults of *T. leucotreta* (USDA, 1984). Look for plants showing signs of poor growth or rot, holes in bolls, adults hidden in foliage, and crawling larvae. Surveys are best conducted during warm, wet weather when the population of the pest increases (USDA, 1984). Eggs will commonly be found on fruits, foliage, and occasionally on branches (USDA, 1984). However, eggs are small and laid singly, which makes them difficult to detect. On corn, *T. leucotreta* has been reported laying eggs on the husk of the ear. Fruit should be inspected for spots, mold, or shrunken areas with 1 mm (0.039 in.) exit holes in the center. On citrus fruits and other fleshy hosts, dissections are needed to detect larvae; larvae are likely to be found in the pulp (USDA, 1984). Infested fruits may be on or off the tree. In cotton, older larvae may be found in open bolls and cotton seed (USDA, 1984). Occasionally adults may be observed on the trunk and leaves of trees in infested orchards (USDA, 1984). For field crops, such as corn, the whole plant is the recommended sample unit. Because larvae of *T. leucotreta* have a strongly aggregated spatial distribution among corn plants, a large sample size (>60 plants) is recommended; however at low densities of the pest (<1 larva/plant), sample sizes needed to detect the pest may be prohibitively large.

Soil Sampling: Collect soil samples within 200 yards of any larval or egg detection and at any spot where dropped, especially prematurely dropped, fruit occur. Soil samples should consist of loose surface soil and any debris. Examine soil for larvae, cocoons, and pupae.

Not recommended: Light trapping is ineffective (Catling and Aschenborn, 1974).

Key Diagnostics/Identification

CAPS-Approved Method*: Confirmation of *T. leucotreta* is by morphological identification. Larval specimens must be examined under a dissecting microscope preferably by a screener experienced with the arrangement of setae on Lepidoptera larvae.

*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: *Thaumatotibia leucotreta* can be distinguished from other species by host range and morphological characteristics. A tool for identifying larvae of leafrollers and a job aid is provided in Appendix D and Appendix E, respectively, of the New Pest Response Guideline to False Codling Moth that can help you determine if you have a possible larva of false codling moth (available at http://www.aphis.usda.gov/import_export/plants/manuals/emergency/downloads/nprg-fcm.pdf). The job aid from Appendix E is also available at

http://caps.ceris.purdue.edu/webfm_send/544. Sofberg (1948) provides a detailed description of larval structures that distinguish FCM from other larvae.

See Padil website for additional FCM images, including diagnostic characters (http://www.padil.gov.au/pests-and-diseases/Pest/Main/136276).

Timm et al. (2007) provides diagnostic characters and keys to identify *T. leucotreta* using both morphological and molecular methods. Morphology of the final instar larvae and pupae of *T. leucotreta* is described and used to develop keys to distinguish closely related species. Mitochondrial cytochrome oxidase I (*COI*) gene was sequenced from *T. leucotreta* and can be used as an alternate means of identification of this species. Molecular methods may be the only method for providing accurate and rapid identification since many pest species are intercepted as eggs or very young larvae.

Easily Confused Pests

T. leucotreta is easily confused with many *Cydia* spp. including codling moth (*Cydia pomonella* Linnaeus) (Fig. 5) (Bradley et al., 1979; USDA-APHIS-PPQ, 2010–2011). Male *T. leucotreta* can be distinguished from other tortricid species by its specialized hind wing, which is slightly reduced and has a circular pocket of fine hairlike black scales overlaid with broad weakly shining whitish scales in anal angle, and its heavily tufted hind tibia (Fig. 3) (Bradley et al., 1979).

Early instar larvae are creamy white in color and can be confused with fruit fly larvae in some cases. The distinct brown black head of *T. leucotreta* larvae, however, make it readily distinguishable from fruit fly larvae (Economides, 1979).

Figure 5. *Cydia pomonella* (codling moth). Photo courtesy of John Davis, www.bugguide.net.

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Arthropods

Secondary Pest of Cotton (Truncated Datasheets)

Figure 1. (A) *A. orana* larva, (B) adult. Photos courtesy of Jae-Cheon Sohn, www.bugwood.org

Adoxophyes orana

Scientific Name
Adoxophyes orana Fischer von Röslerstamm

Synonyms:
Adoxophyes congruana Walker, *Adoxophyes fasciata* Walsh, *Adoxophyes reticulana* Hübner, *Adoxophyes tripsiana, Acleris reticulana, Capua congruana, Capua orana, Capua reticulana* Hübner, *Cacoecia reticulana, Tortrix orana* Fischer von Röslerstamm, and *Tortrix reticulana* Hübner

Common Name(s)
Summer fruit tortix moth, apple peel tortricid, smaller tea tortrix, and tea tortrix (smaller).

Type of Pest
Moth

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

Reason for Inclusion In Manual
CAPS target: AHP Prioritized Pest List 2006 through 2012

Pest Description
Eggs: Egg clusters are shiny yellow; black dots appear as larvae develop within the eggs. Eggs are flattened and deposited in groups of 20 to 100 usually on the upper surface of apple leaves and the underside of pear leaves (van der Geest and Evenhuis, 1991; INRA, 2005).

Larvae: Mature larvae are 16 to 22 mm (0.63 to 0.87 in.) long. The head and prothoracic shield are shiny brown in young caterpillars and honey-colored in older larvae. The body and anal plate range from dark or olive green to yellow green (Fig. 1A). Pinacula are small and yellow. The thoracic legs are light brown (van der Geest and Evenhuis, 1991). The larvae are very agile (INRA, 2005).

Pupae: Pupae are dark brown and 10 to 11 mm (0.39 to 0.43 in.) long (van der Geest and Evenhuis, 1991). Larvae pupate in folded leaves or between two leaves stuck together (INRA, 2005).

Adults: The head and thorax are beige. Wingspan ranges from 17 to 19 mm (0.67 to 0.75 in.) in males and from 19 to 22 mm (0.75 to 0.87 in.) in females. The male moth's forewing ranges from ochre to rust-brown and bears darker brown markings; the female moth's forewing is gray-brown with subtle brown markings (Fig. 1B). Hindwings are light gray in males, light brown in females (van der Geest and Evenhuis, 1991; INRA, 2005).

Symptoms/Signs

Larvae feed on flower buds, leaves, new shoots, and occasionally on fruit (Fig. 2). Feeding rarely causes enough foliar damage to affect plant growth. Feeding damage on fruit heals poorly and creates entry for pathogens (INRA, 2005). Damaged leaves are deformed and necrotic and may appear wilted, yellow, shredded, or dead. Larvae fold and roll leaves and use webbing to hold them together. Injured shoots have lesions. Early instars may damage flower buds and flowers; evidence of flower feeding includes webbing and external damage. Summer generation larvae feed extensively and severely damage fruit. Feeding on fruits or pods causes scabs, lesions, or pitting, and frass may be present. On fruit crops, larvae prefer to feed sheltered under a leaf bound to fruit by silk. Feeding on fruit sometimes covers several square centimeters of the fruit surface (Charmillot and Brunner, 1989).

Figure 2. Larval damage on leaves and fruit. Photos courtesy of Magnus Gammelgaard, www.plant-diseases.com.

On cotton, damage is most likely to be foliar, but no record of damage to cotton was found in the literature. Cotton is listed as a host plant, but no specific damage is described (DeJong et al., 1971; Savopoulou-Soultani et al., 1985; Shu et al., 2002; Vasilev, 1924). Potentially, the pest could create lesions on bolls that would allow entry of pathogens.

Survey

CAPS-Approved Method*: The CAPS-approved method is a trap and lure combination.

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

1) Paper Delta Trap, 2 sticky sides, Brown
2) Paper Delta Trap, 2 sticky sides, Green
3) Paper Delta Trap, 2 sticky sides, Orange

The Lure Product Name is "*Adoxophyes orana* Lure." The lure is effective for 84 days (12 weeks).

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: Trap should be used with ends open. Trap color is up to the State and does not affect trap efficacy.

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:

Trapping: The most effective trapping method for *A. orana* is sex-pheromone baited traps. Two synergistic isomers are required to attract *A. orana*: both (*Z)*-9-tetradecenyl acetate and (*Z*)-11-tetradecenyl acetate (Meijer et al., 1972). If either isomer is present in the *E* conformation, the chemicals will not be effective and will have an inhibitory effect (Meijer et al., 1972; Davis et al., 2005). Alone, these chemicals will not attract *A. orana* (Meijer et al., 1972). The most effective ratio is 9:1 (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: Visual sampling may also be used to inspect plants for eggs, larvae, and pupae. Survey for *A. orana* may include visual inspection for larvae on flowers in the spring and shoots and fruit in the summer (Charmillot and Brunner, 1989). Rolled leaves are easily seen in the early to mid summer, but rolled leaves must be separated and examined to confirm the presence of *A. orana* (Charmillot and Brunner, 1989),. Eggs may be observed on the stems and leaves, and pupal cocoons may be found in leaves, on stems, or in mummified pods/seeds. Visual methods are time consuming, so they 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. Light traps are not recommended because they 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 and climate within the continental United States. Areas of the southeastern United States, California, Illinois, Indiana, Maryland, Missouri, Oklahoma, and Texas have the highest risk of *A. orana* establishment.

Key Diagnostics/Identification

CAPS-Approved Method*: Confirmation requires morphological identification. *Adoxophyes orana* may occur in mixed populations with other morphologically similar species, including other *Adoxophyes* species. Final identification requires dissection of male genitalic structures, so a trained taxonomist should be consulted.

*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 they are hidden in leaves, leafrollers are difficult to detect. Distinguishing between males and females of adult *Adoxophyes* is difficult. According to Yasuda (1998), the color and pattern variation of the forewing and morphological resemblance among *Adoxophyes* species make it difficult to distinguish between species. *A. orana* closely resembles two U.S. species, *Adoxophyes negundana* and *A. furcatana.* There are slight differences in male genitalia; an appropriately trained entomologist should confirm any identification.

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Autographa gamma

Figure 1. *A. gamma* larvae. Photos courtesy of (A) copyright Ben Emmett, ADAS UK, hdc.org.uk, (B) Charles Olsen, USDA –APHIS-PPQ, www.bugwood.org.

Scientific Name
Autographa gamma L.

Synonyms
Phytometra gamma and *Plusia gamma*

Common Name(s)
Silver Y moth and beet worm

Type of Pest
Moth

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

Reason for Inclusion In Manual
CAPS Target: AHP Prioritized List 2006 through 2009

Pest Description
Eggs: Eggs are semi-spherical and 0.57 mm (0.022 in.) in diameter. They are strongly and irregularly ribbed with 28 or 29 ribs (Paulian et al., 1975; Carter, 1984). Eggs are initially yellowish-white, but as they age, they turn yellowish-orange and later brown. They are laid singly or in small groups on the underside of leaves (Hill, 1987).

Larvae: The larvae are “semiloopers” with only three pairs of prolegs: two pairs of abdominal prolegs and one pair of anal prolegs (abdominal segments 5, 6, and 10) (Fig. 1B) (Carter, 1984; Hill, 1987; INRA/HYPP Zoology, 2011). The caterpillar ranges from bright green to dark olive green. There is a dark green dorsal line edged with white (Fig. 1) (Jones and Jones, 1984). The spiracular line is yellowish, edged above with green (CAPS, 2010). There are several white transverse lines between the spiracular line and the dorsal dark green line (Jones and Jones, 1984). Some larval forms have white spots. The head has a dark patch under the ocelli (Fig. 1B) or is entirely black and glossy (Fig. 1A) (Emmett, 1980). Maximum length ranges from 20 to 40 mm (0.79 to 1.57 in.). The larvae complete five instars (Dochkova, 1972).

Pupae: *A. gamma* pupates on the lower leaf surface or within the first centimeter of soil (Dochkova, 1972). The pupa is 20 mm (0.79 in.) long, black, shiny, and encased in a loose, web-like cocoon (Jones and Jones, 1984). The cremaster is “strongly developed,

ridged, with two outcurved, spatulate spines and six terminally coiled setae" (Carter, 1984).

Adult: Forewings are marbled silvery gray to brown to velvety black. There is a distinct white/silver "Y" or Greek letter gamma (γ) in the center of the forewing (Fig. 2A & B). Wingspan is 36 to 40 mm (1.42 to 1.57 in.) (Jones and Jones, 1984). The hindwings are light brown with a dark brown marginal border (Nazmi et al., 1980).

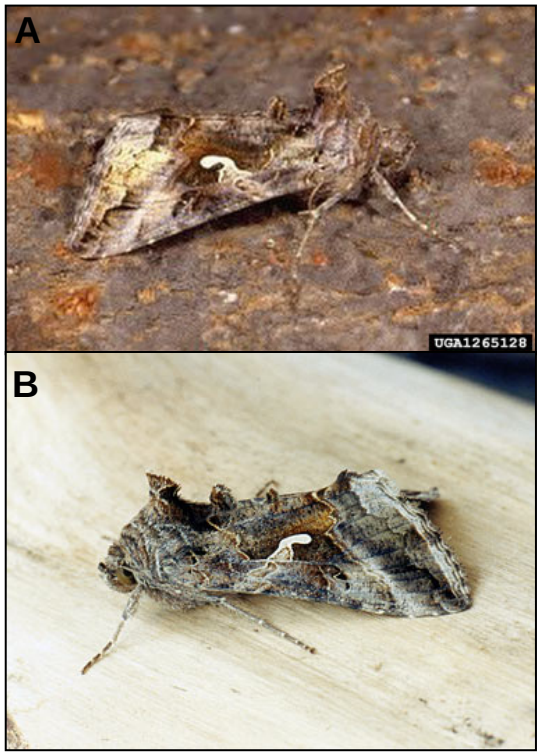

Figure 2. *A. gamma* moth. (A) Photo courtesy of Paolo Mazzei, www.bugwood.org, (B) Photo courtesy of Ian Kimber, UK, moths.org.uk.

Symptoms/Signs

Young larvae often skeletonize leaves or cut petioles; older larvae will eat the entire leaf, beginning from the edges of the leaf and working inward. Larvae prefer older leaves. During the day, caterpillars can be found flattened against the undersides of leaves. If disturbed, caterpillars drop from the plant. Eggs are also found on the undersides of leaves, laid singly or in small groups (CAPS, 2010).

Survey

CAPS-Approved Method*: The CAPS-approved method is a trap and lure combination. The trap is Plastic Bucket Trap. The lure is effective for 28 days (4 weeks).

The Lure Product Name 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:
The sex pheromone of *A. gamma* is composed of (*Z*)-7-dodecenyl acetate (*Z*7-12:Ac) and(*Z*)-7-dodecenol (*Z*7-12:OH); *Z*7-12:Ac is the major component (Tóth et al., 1983; Mazor and Dunkelblum, 2005). (*Z*)-7-dodecen-1-yl acetate and (*Z*)-7-dodecen-1-ol in 100:1 and 95:5 mixtures attract male *A. gamma* moths (Tóth et al., 1983). Traps can be baited with rubber septum dispensers with 1 mg dispensing rates. Lures should be replaced every 4 weeks. Optimum trap height is 1.5 m (4.92 ft.) (Terytze et al., 1987).

The pheromone of *A. gamma* also attracts the following Lepidoptera in the United States: *Anagrapha ampla, Anagrapha falcifera, Autographa ampla, Autographa biloba, Autographa californica, Caenurgia* spp., *Epismus argutanus, Geina periscelidatyla, Helvibotys helvialis, Lacinipolia lutura, Lacinipolia renigera, Ostrinia nubilalis, Pieris rapae, Polia* spp., *Pseudoplusia includes, Rachiplusia ou, Spodoptera ornithogalli, Syngrapha falcifera,* and *Trichoplusia ni* (CAPS, 2010).

Surveys should be focused where the greatest risk for establishment occurs. A recent risk analysis by USDA-APHIS-PPQ-CPHST indicates that many states in the United States have a low to moderate risk rating for *A. gamma* establishment based on host availability and climate within the continental United States. Establishment is precluded in most of the northern states. Areas of Arkansas, Kentucky, Louisiana, Mississippi, North Carolina, Oklahoma, and Virginia have the highest risk of *A. gamma* establishment.

Key Diagnostics/Identification

CAPS-Approved Method*: Confirmation of *A. gamma* is requires morphological identification by a trained entomologist/taxonomist.

*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. gamma* is most reliably identified by inspection of wing venation and genitalia (Nazmi et al., 1980; USDA, 1986).

Easily Confused Pests

Several Noctuids at several life stages may be confused for *A. gamma.* In the United States, the most important species include: *Trichoplusia ni* (cabbage looper), *A. californica* (alfalfa looper), *Syngrapha celsa* (plain silver-Y or western conifer looper), and *A. pseudogamma* (delicate silver-Y). Other species that occur in Africa, Asia, and Europe include: *Cornutiplusia circumflexa* (Essex Y). *Syngrapha interrogationis* (scarce silver Y) occurs in the UK (Venette et al., 2003). See Nazmi et al. (1980) for comparison and similarities between closely related species.

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Diabrotica speciosa

Scientific Name

Diabrotica speciosa Germar

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

Common Name(s)
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 through 2012

Pest Description
From CAPS (2010):
Germar first described *Diabrotica speciosa* 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 (0.029 x 0.014 in.), and clear white to pale yellow (EPPO, 2005). 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 (0.047 in.) long, and mature third instars are about 8.5 mm (0.33 in.) long. Larvae are subcylindrical and chalky white; head capsule is dirty yellow to light brown; epicraneal and frontal sutures are lighter with long, light brown setae; mandibles are reddish dark brown; antennae and palpi are pale yellow. The body is covered by sparse, short, dark setae; has a light brown irregular prothoracic plate; and has a 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: Mature third instars build an 8 x 4 mm (0.31 x 0.16 in.) oval cell in the soil in which they pupate. Pupae are 5.8 to 7.1 mm (0.16 x 0.28 in.) long and white. Females have a pair of tubercles near the apex. Tenerals (freshly emerged, soft bodied adults) remain in the soil for about three days.

Adults: Baly (1886), Araujo Marques (1941), and Christensen (1943) provide full descriptions of *D. speciosa.* Adults are 5.5 to 7.3 mm (0.22 to 0.29 mm) long. Antennae are filiform, dark reddish-brown to black, and 4 to 5 mm (0.16 to 0.20 in.) long (Fig. 1); the first three basal antennal segments are lighter colored. The general color is grass green (USDA, 1957). Head color ranges from reddish brown to black. The labrum, scutellum, metathorax, tibiae, and tarsi are black. The elytra each have three large, yellow, oval transverse spots; basal spots (those nearest the head) are larger and usually reddish toward the humeral callus. Ventrally, the head and metathorax are dark brown, the prothorax is green, and the mesothorax and abdomen are light brown or yellow-green. The pronotum is bi-foveate, convex, smooth, shiny, and 1⁄4 wider than long. Male antennae proportionally are longer than female antennae; antennae are shorter than the body in both sexes (Araujo Marques, 1941). Males have an extra sclerite on the apex of the abdomen that makes it look blunt, compared with the relatively pointed female apex (EPPO, 2005).

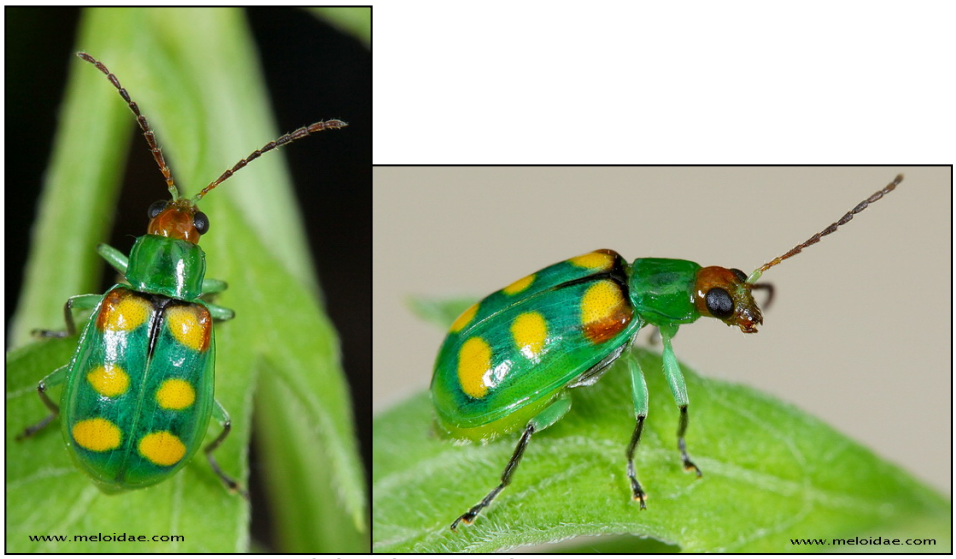

Figure 1. Adults of *Diabrotica speciosa*. Photos courtesy of Stanislav Krejcik, www.meloidae.com.

Symptoms/Signs

D. speciosa larvae attack the roots of host plants. Adults feed on leaves, flowers, and fruits. 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 because of a reduction in nutrient uptake.

On Cotton: Most likely, larvae would attack roots, causing death of young plants and decreased vigor in larger plants. Adults might feed on leaves and squares, but would likely do the most damage to floral structures. Damage to flowers could negatively impact fruiting and, therefore, boll development.

On corn: Larval attack on young plants produces a typical condition known as 'goose neck', in which the plant exhibits stunted growth, reduced vigor, and bent first few internodes, sometimes to such an extent that the plant lies on the ground. The larvae feed on the tassels, preventing pollination and reducing the number of ripening grains from the tip of the ear to the base (EPPO, 2005).

On peanuts, groundnut, and potatoes: The larvae cause external damage or short bores on the pods or tubers similar to those of several other pests such as wireworms and other chrysomelids (EPPO, 2005).

On grape: Adult beetles attack 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. Insect feeding on the stigma causes flower abortion and, as a consequence, clusters show low numbers of flowers and fruits that are below market standards (Roberto et al., 2001).

Survey

CAPS-Approved Method*: Visual inspection is the approved method to survey for *D. speciosa*.

*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 inspection of adults is effective and easy because *D. speciosa* feeds on plants during the day. Detecting larvae is more difficult, because they feed underground. First instars are very difficult to sample; larger larvae may be observed feeding on roots immediately after the host plant is removed from soil. Extensive damage to the host may occur before larvae are detected. Sampling and counting methods have not been developed for *D. speciosa* (Fisher and Bergman, 1986).

Trapping: Adults *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). Arruda-Gatti et al. (2006) found that transparent green traps baited with 1,4dimethoxybenzene caught significantly more beetles than baited yellow, white, and transparent traps. Analogs 1,4-dimethoxybenzene are not effective in luring *D. speciosa* (Marques et al., 2009).

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 (CAPS, 2010).

Surveys should be focused where the greatest risk for establishment occurs. A recent risk analysis by USDA-APHIS-PPQ-CPHST indicates that many states in the United States have a low to moderate risk rating for *D. speciosa* establishment based on host availability and climate within the continental United States. Areas of Arkansas, Illinois, Indiana, Kansas, Kentucky, Louisiana, Mississippi, North Carolina, Oklahoma, and Tennessee have the highest risk of *D. speciosa* establishment.

Key Diagnostics/Identification

CAPS-Approved Method*: Confirmation of *D. speciosa* is by morphological identification. *Diabrotica speciosa* is almost identical to *D. balteata* (Fig. 2), which is widely present in the southern United States. Confirmation by a chrysomelid specialist is required.

*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: Same as above.

Figure 2. Adult banded cucumber beetle, *Diabrotica balteata*. Photo courtesy of John L. Capinera, University of Florida.

Easily Confused Pests

D. speciosa may be easily confused with the banded cucumber beetle, *Diabrotica balteata* (Fig. 3). Other rootworms (western corn rootworm, southern corn rootworm) are easily distinguished from *D. speciosa* as adults by markings on elytra.

Those with little experience with *Diabrotica* may confuse *D. speciosa* with *Diabrotica viridula* (not present in the United States) and other pestiferous *Diabrotica* species in South America, so confirmation by a chrysomelid specialist is required.

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Eutetranychus orientalis

Scientific Name
Eutetranychus orientalis Klein

Synonyms:
Eutetranychus anneckei, *Eutetranychus latus, Eutetranychus monodi*, *Eutetranychus sudanicus*, *Anychus latus, Anychus orientalis*, and *Anychus ricini*

Common Name(s)
Citrus brown mite, oriental mite, oriental red mite, oriental red spider mite (Avidov and Harpaz, 1969), oriental spider mite, Lowveld citrus mite (in South Africa), and citrus mite (EPPO/CABI, 1997).

Type of Pest
Mite

Taxonomic Position
Class: Arachnida, **Order:** Acarina, **Family:** Tetranychidae

Reason for Inclusion In Manual
CAPS Target: AHP prioritized Pest List 2006 through 2009

Pest Description
The genus *Eutetranychus* is characterized by its empodium, which is reduced to a small protuberance (Avidov and Harpaz, 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; CAPS 2010).

Eggs: The eggs of *E. orientalis* are oval or circular with a disc-shaped appearance (Fig. 1), 140 µm in diameter (Avidov and Harpaz, 1969), and flattened. They come to a point dorsally but lack the long dorsal stalk of other spider mites. Newly laid eggs are bright and hyaline, but later they become a yellow, parchment-like color (Smith-Meyer, 1981; CAPS 2010).

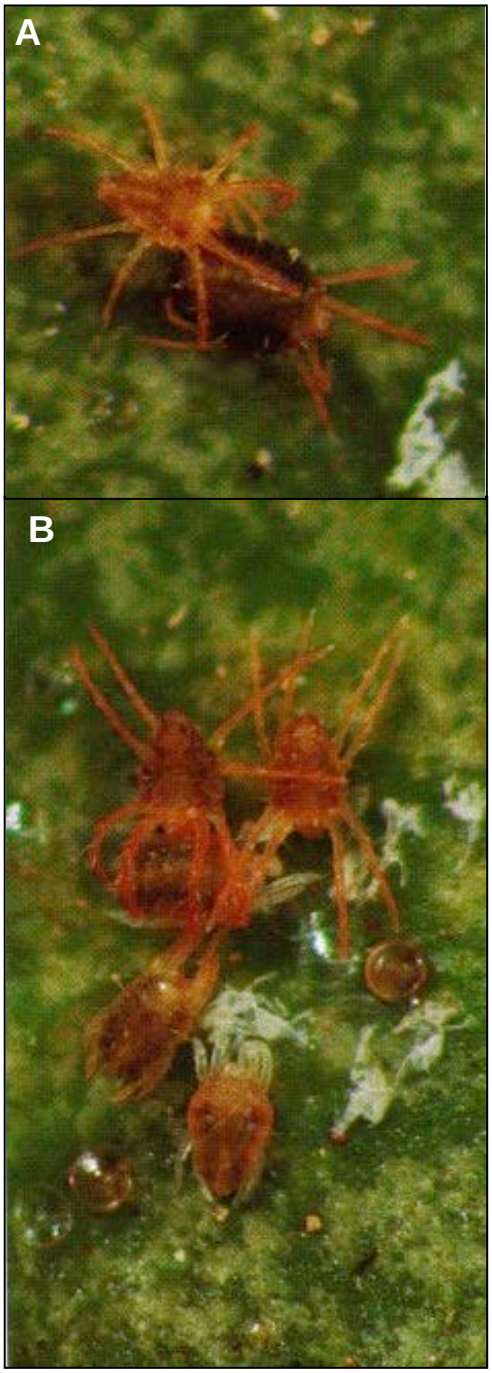

Figure 1. *E. orientalis* (A) Male on top of darker female, (B) males clustering around pre-adult female, photos courtesy of Smith et al. 1997, EPPO/CABI

Larvae: The average size of the nymph of *E. orientalis* is 190 x 120 µm. The protonymph is pale-brown to light green, with legs shorter than the body and an average size of 240 x 140 µm. The deutonymph is pale-brown to light green with an average size of 300 x 220 µm (EPPO/CABI, 1997; CAPS, 2010,).

Adults: Adult females 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 yellow-brown (Fig. 1 & 2). The average size is 410 x 280 µm (CAPS 2010).

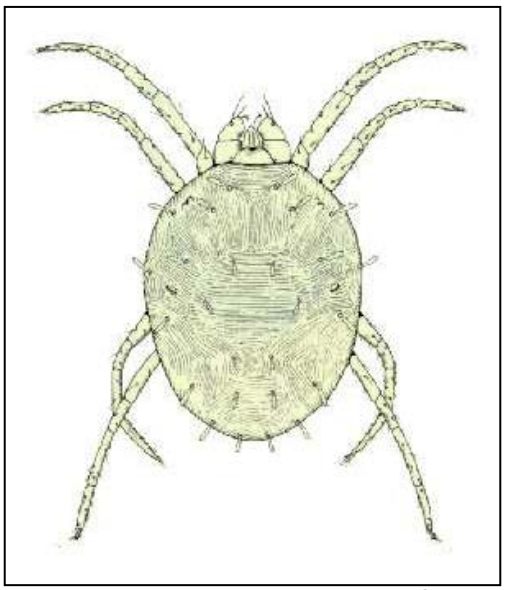

Figure 2. Adult of *Eutetranychus orientalis.* Drawing courtesy of CSIRO Entomology, Australia.

Male adults are much smaller than the females. They are elongate and triangular in shape with long legs (leg about 1.5 x body length) (CAPS, 2010). The body setae are short and cannot be observed with a 10x lens (Smith-Meyer, 1981; Dhooria and Butani, 1984; EPPO/CABI, 1997).

Technical Description: Jeppson et al. (1975) and Smith-Meyer (1987) provide diagnostic descriptions of *Eutetranychus orientalis*. *E. orientalis* has the following combination of characters: striae on the prodorsum longitudinal and tuberculated; striae between the second (d/sub/1) and third (e/sub/1) dorsocentral setae longitudinal or V-shaped; the 13 pairs of dorsal body setae all arise from basal tubercles and vary in length and shape; dorsolateral setae on the body (c2), (d2), (e2), (f2) are long, lanceolate and subspatulate or broadly spatulate; dorsocentral setae (c1), (d1), (e1), (f1), (h1) short and spatulate, lanceolate, or subspatulate; first pair of dorsocentral setae (c1), first pair of dorsal lateral setae (c2), and humeral setae (c3) all more or less in line; third (e1) and fourth (f1) dorsocentral setae form a square; terminal sensillum (spinneret) of palptarsus three times as long as broad; coxa II with one seta; tactile setal formulae (I-IV): femora 8-6-(3-4)-(1-2), genua 5-5-2-2, tibiae 9-6-6-7; chromosome number (n)=3 (CAPS, 2010).

For additional information see: EPPO/CABI (1997) http://www.eppo.org/QUARANTINE/insects/Eutetranychus_orientalis/EUTEOR_ds.pdf.

Symptoms/Signs

All active stages of *E. orientalis* feed and molt on the upper side of fully expanded leaves (Hill, 1987). On citrus, the mite starts feeding on the upper side of the leaf along the midrib and then spreads to the lateral veins. The area around the feeding site turns gray, and during infestation the entire leaf surface appears chlorotic due to a large number of gray spots (Avidov and Harpaz, 1969; EPPO/CABI, 1997; CAPS, 2010). Pale

yellow streaks develop along the midrib and veins. Little webbing is produced. 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 dieback of branches, which may result in defoliated trees. Lower populations in dry areas can produce the same effect (EPPO/CABI, 1997; CAPS, 2010).

Survey

CAPS-Approved Method*: Visual inspection is the approved 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: *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 mite spreads via wind, and new infestations commonly occur at 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 in most states in the continental United States pest establishment is unlikely. Risk for *E. orientalis* establishment based on climate and host availability is low in Arizona, California, Nevada, Texas, and Utah. Risk is low to moderate in Florida.

Hall (1992) discusses sampling strategies for spider mites in orange groves. The author's sampling method consisted of examining 16 leaves per tree, five trees within a small area of trees, and three areas per block. Leaves are collected by removing four leaves from each of the north, east, south, and west sides of a tree. Leaves are placed into separate plastic bags. The bags are 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 mites. Leaves are placed in a jar filled with 0.5% liquid dishwashing soap and 0.5% standard bleach (5% NaCl) (each % by volume) in distilled water. The liquid soap breaks up surface tension; the bleach dissolves any webbing. The author showed that this liquid sampling procedure collects 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 and Butani (1984) 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 certain 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.

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Planococcus minor

Scientific Name

Planococcus minor Maskell

Synonyms:
Planococcus pacificus, Planococcus psidii, Dactylopius calceolariae minor, Pseudococcus calceolariae minor, and *Pseudococcus minor.*

Common Name(s)
Passionvine mealybug and Pacific mealybug

Type of Pest
Mealybug

Taxonomic Position
Class: Insecta, **Order:** Homoptera, **Family:** Pseudococcidae

Reason for Inclusion In Manual
CAPS Target: AHP Prioritized Pest List 2003 through 2012

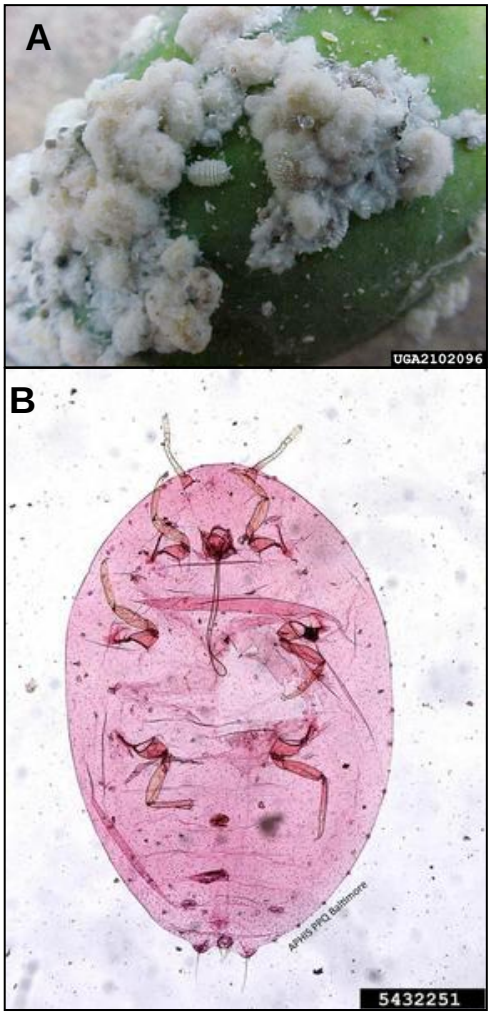

Figure 1. *Planococcus minor.* Photos courtesy of (A) Joel E. Miles, Office of Environmental Response and Coordination, and (B) James D. Young, USDA APHIS PPQ, www.bugwood.org.

Pest Description
Planococcus minor is a small scale-like sucking insect coated with a powdery, mealy, waxy secretion that gives it a cottony appearance. Females are oval, 1.3 to 3.2 mm (0.051 to 0.126 in.) long, and 0.8 to 1.9 mm (0.031 to 0.075 in.) wide (Venette and Davis, 2004). The insect body is distinctly segmented, yellow to pink in color, and covered with powdery wax with the appearance of “having been rolled in flour” (Fig. 1A) (CAPS, 2010). The margin of the body has a complete series of 18 pairs of cerarii, each cerarius with two conical setae (except for preocular cerarii which may have one or two setae) (Venette and Davis, 2004; CAPS, 2010). Legs are elongate.

It is assumed that this species is identical in appearance to *P. citri* as follows (CAPS, 2010): the body is oval and slightly rounded in lateral view. The insect is yellow when newly molted, but changes to pink or orange-brown when fully mature. The legs are brown-red. Mealy wax covers the body, but is not thick enough to hide the body color. There is a dorsomedial bare area on the dorsum that forms a central longitudinal stripe (more obvious than on *P. ficus*). The ovisac is ventral only, and may be two times longer

than body when fully formed. There are 17 or 18 lateral wax filaments, most are relatively short and often slightly curved. The posterior pair is slightly longer. The filaments anterior of the posterior pair are small; the posterior pair are about 1/8 the length of the body. The surface of lateral filaments is rough (Rung et al., 2007). *P. minor* primarily occurs on foliage of host. *P. minor* is oviparous, and the eggs are yellow.

Mealybugs produce honeydew, which is a liquid rich in sugar. Ants feed on honeydew, so some ants may defend the mealybugs by chasing away predators and parasitoids (CAPS, 2010). The ants also carry mealybugs around and therefore contribute to their distribution. *P. citri* was reported as a virus vector in cocoa, banana, and grape, but whether *P. minor* can serve as a vector is unknown (Jones and Lockhart, 1993; Canaleiro and Segura, 1997).

Symptoms/Signs

Mealybugs have piercing-sucking mouthparts. *Planococcus minor* is a phloem feeder, which can cause reduced yield, reduced plant or fruit quality, stunting, wilting, discoloration, and defoliation. Indirect or secondary damage is caused by sooty mold growth on honeydew excreted by the mealybug (CAPS, 2010).

Survey

CAPS-Approved Method*: Visual inspection is the approved method to survey for *P. minor*.

*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 and Davis, 2004)
Visual survey: Surveys for live mealybugs require “time-consuming and often laborious examination of plant material” (Millar et al., 2002). No simple, alternative techniques are available (Millar et al., 2002). In India, a regional survey for scales and mealybugs, including *P. minor*, was based on visually examining 25 branches or leaves on each of 15 plants collected from each of three field sites in 162 locations (25 x 15 x 3 x 162 = 182,250 leaves examined) (CAPS, 2010).

Researchers also use visual inspection to assess densities of *P. minor*. In a study of *P. minor* population dynamics, populations of the mealybug were evaluated by visual inspection of citrus leaves, specifically 10 to 15 leaves from 10 randomly selected plants (Bhuiya et al., 2000). Reddy et al. (1997) followed a similar protocol for coffee.

Surveys should be focused in areas that are most at risk for establishment of *P. minor*. The host range of *P. minor* includes a variety of plants grown in the United States, so this insect is able to establish populations that mirror the distribution of *P. citri* (CAPS, 2010). *P. citri* is present in the southern states and has been reported as far north as Ohio, Kansas, and Massachusetts. Venette and Davis (2004) estimate that approximately 52% of the continental United States would have a suitable climate for *P. minor*.

Planococcus minor was found in a site in Florida but appears to be causing little damage and is being controlled through natural enemies.

A recent host analysis by USDA-APHIS-PPQ-CPHST indicates that portions of Arkansas, Illinois, Indiana, Iowa, Minnesota, Missouri, Nebraska, Ohio, South Dakota, have the greatest risk for *P. minor* establishment based on host availability within the continental United States.

Trapping: A sex pheromone has been identified for *P. minor*. Ho et al. (2007) identified the sex pheromone as (*E*)-2-isopropyl-5-methyl-2,4-hexadienyl acetate. The (*Z*) isomer was found to be highly antagonistic. Millar (2008) describes the short and completely stereospecific synthesis of (*E*)-2-isopropyl-5-methyl-2,4-hexadienyl acetate. The availability of this pheromone is unknown at this time.

Key Diagnostics/Identification

CAPS-Approved Method*: Confirmation of *P. minor* is by morphological and molecular identification. A qualified taxonomist can identify adult females by using a series of morphological characters. Immatures are easily confused with other *Planococcus* species and other mealybug genera. For molecular analysis, it is important to maintain some specimens in 95 to 100% alcohol for DNA analysis. Final identification will be based on morphological identification of adult female followed by molecular analysis for confirmation (CAPS, 2010).

*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 and Davis, 2004)
Mealybugs are a quarantine problem on exported foliage and flowers. Species cannot be accurately identified outside of the lab, so inspectors/surveyors should treat all specimens as unknown species. There are a great number of endemic mealybug species in the United States, and identifications need to be done by a qualified taxonomic authority (CAPS 2010).

Planococcus species are not easily distinguishable from one another, especially when immature. A level of complexity is added with variable morphological characters in some species. Distinguishing morphological characters can change based upon environmental conditions, such as temperature (CAPS, 2010). Cox, (1981; 1983; and 1989) described distinguishable morphological characters of closely related mealybug species. A Lucid tool for scale insects has been recently developed, which contains a tool on mealybugs (see Rung et al., 2007, http://www.sel.barc.usda.gov/ScaleKeys/ScaleInsectsHome/ScaleInsectsMealybugs.html).

Planococcus citri and *P. minor* have been taxonomically confused and routinely misidentified because adults are similar in appearance and share similar hosts and

geographic range (Williams, 1985; Cox, 1989; Williams and Granara de Willink, 1992). Adults (females) can be identified based on close examination of morphological characters by a taxonomist. PPQ initiated a project to develop molecular diagnostics to separate *P. citri* from *P. minor*. A PCR-RFLP technique was developed to distinguish *P. citri*, *P. minor*, and a genetically distinct *Planoccocus* that is morphologically identical to *P. citri* from Hawaii (Rung et al., 2008; 2009).

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Plant Pathogens

Primary Pests of Cotton (Full Pest Datasheet)

Phytophthora boehmeriae

Scientific Name
Phytophthora boehmeriae Sawada

Synonyms:
None

Common Name(s)
Ramie Blight

Type of Pest
Fungal-like Pathogen

Taxonomic Position
Class: Oomycetes, **Order:** Pythiales, **Family:** Pythiaceae

Reason for Inclusion
National Threat

Pest Description
Based on Sawada (1927), the morphological characters of *P. boehmeriae* are as follows: Sporangiophores branch sympodially. Sporangia (Fig. 1A through 1D) are broadly ellipsoid to nearly spherical or obturbinate to obpyriform, 50 x 35 to 40 (up to 70 x 45) µm, deciduous, the pedicel is up to 3 µm long, and papilla and apical thickening are hemispherical or deeper. Chlamydospores are variable in number and 40 to 50 µm in diameter with a 2 µm thick wall. Oogonia (Fig. 1E & F) form readily in culture and are 27 to 40 µm. Antheridia are amphigynous (Fig. 1F) and 14 to 21 x 13 µm. Oospores (Fig. 1E & F) nearly fill the oogonia, which have walls up to 2 µm thick. Cultures are uniform, with dense aerial mycelium. Minimum temperature for development is 5 to 6ºC (41 to 43ºF), optimum 25ºC (77 ºF), and maximum 32ºC (90ºF) (Stamps, 1978).

Severe boll decay was observed on cotton in Larissa County, Greece, in August 1993 (Elena and Paplomatas, 1998; Paplomatas et al., 1995). The fungus-like organism was isolated from diseased cotton bolls and identified as *Phytophthora boehmeriae* based on the following characteristics by Paplomatas et al. (1995): sporangia ellipsoid to nearly spherical, mostly 39 to 48 x 29 to 35 µm, ratio 1.2:1.5, bearing papilla oogonia 29 t0 40 µm, antheridia mostly amphigynous, maximum growth temperature 33ºC (91ºF).

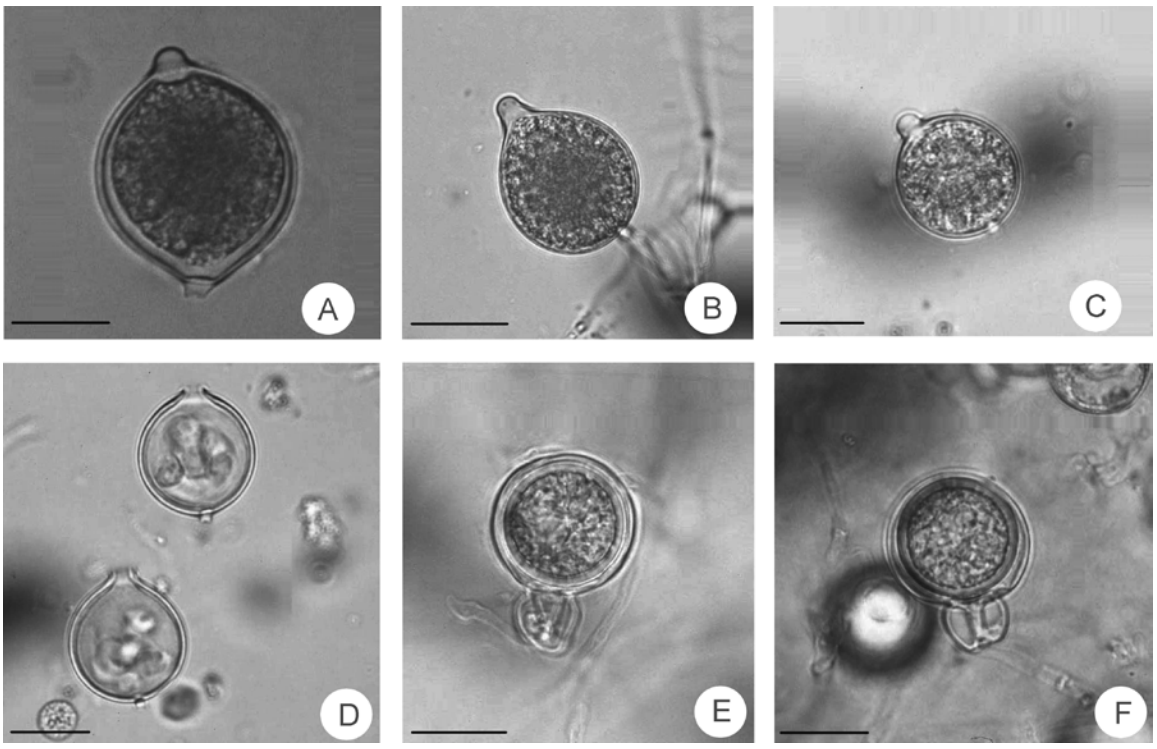

Figure 1. (A) Sporangium taken from fresh infected tissue; (B) Sporangium formed in water; (C) Sporangium produced on V8 juice agar; (D) Sporangia formed on agar medium and indirectly germinated; (E) Oogonium and antheridium with a pleurotic oospore; (F) Oogonium with more conspicuously amphigynous antheridium. Scale bars = 20 µm. Photos courtesy of Byung-Soo Kim, Kyungpook National University (Kim and Kim, 2004).

Kim and Kim (2004) isolated a species of *Phytophthora* from the diseased *Ailanthus altissiman* (tree of heaven) plant parts and identified this species as *P. boehmeriae*. They describe the fungus-like organism as follows (Fig. 1): Oogonia are spherical, 19.5 to 42.9 µm in diameter with an average of 29.4 ± 4.2 µm. Antheridia are amphigynous, round to ovoid, and measure 11.3 to 15.0 µm long and 12.5–14.5 µm wide. Oospores in the oogonia are spherical, 26.1 to 29.0 µm in diameter. Sporangia that formed in water were spherical to sub-spherical with a conspicuous papilla and measured 19.5 to 56.6 x 15.6 to 44.9 µm with an average of 44.0 ± 8.7 X 32.7 ± 6.3 µm. The mean length/breadth (l/b) ratio was 1.35. Papillae were 3.9 to 11.7 µm high and 3.9 to 9.8 µm wide. Sporangia formed slowly on V8 juice agar medium when cultured under fluorescent light at 12-hour alternation. The sporangia that formed on the agar medium were more spherical and measured 26.5 to 39.0 X 23.4 to 35.1 µm with an average of 33.6 ± 3.4 X 28.2 ± 3.2 µm and length to breath ratio of 1.19 (Kim and Kim, 2004).

Antheridia of *P. boehmeriae* are amphigynous and almost spherical in shape. They have been shown to change to paragynous, however, on media with low nutrient levels (Gao et al., 1998).

Biology and Ecology

P. boehmeriae is homothallic and oospores form abundantly in host tissues. Oospores in infected tissues (leaves, bolls, etc.) are released into the soil when the diseased tissues decompose. Under suitable conditions, they germinate by germ tubes to form sporangia or mycelia, which may produce sporangia. Temperature is the most important factor to affect the germination mode of sporangia. It has been observed that at 18 to 20ºC (64 to 68ºF), all sporangia germinated indirectly to produce zoospores, whereas at 22 to 24ºC (72 to 75 ºF) most sporangia germinated directly and terminated with secondary sporangia capable of releasing zoospores (CABI, 2010; USDA, 2010).

P. boehmeriae is commonly spread by soil and water (Ormsby, 2008). Rainwater or drainage water transports infested soil and spores along rows, terraces and ditches and into disease-free fields, drainage ponds, or streams. When water from these infested sources is used for irrigation, it may carry the oospores to new locations and cause disease. The spores may be dispersed by rain splashing or by wind during wet weather (Ormsby, 2008). When the spores lodge (encyst) against a stem or root of a certain host (such as ramie or pine), or splash onto a leaf or fruit of a suitable host plant (such as cotton), they germinate to produce appressoria or mycelia that infect the plant through the cuticle or via stomata or wounds. The role of the chlamydospore in the lifecycle is not clear. Some scientists suggest that the fungus-like organism may overwinter as chlamydospores, while others suggest that chlamydospores are seldom present in *P. boehmeriae* and that oospores can overwinter and survive in the soil (CABI, 2010; USDA, 2010).

The fungal-like pathogen prefers high humidity and warm temperatures, so it frequently occurs in low, wet areas in rainy years. The optimum temperature for mycelial growth in culture is 25 to 30ºC (77 to 91ºF), with minimum 9ºC (48 ºF) and maximum 34.5ºC (94ºF). The minimum temperatures for sporangium and oospore formation are 25ºC (77ºF) and 25 to 28ºC (77 to 82ºF), respectively. Darkness stimulates mycelial growth and oospore formation, whereas illumination inhibits both (CABI, 2010; USDA, 2010).

There is evidence that *P. boehmeriae* is seed-borne in cotton and can be disseminated by seeds (CABI, 2010; USDA, 2010).

Symptoms/Signs

In nature, *P. boehmeriae* can infect, damage, and blight the seedlings, leaves, and bolls of cotton, the seedlings, leaves and stems of ramie, the leaves of paper mulberry (*Broussonetia papyrifera*), the fruits of citrus, the leaves of Chinese wingnut, and the roots of pine (Ho and Lu, 1997; USDA, 2010).

Cotton: On seedlings, *P. boehmeriae* produces round or irregular water-soaked dark-green spots or lesions on cotyledons or true leaves, resulting in premature leaf fall or wilting of some or all leaves under cold, humid conditions. On the roots and root-stem transition zones, the disease first appears as brown streaks and then as a brown rot, resulting in wilting and death of whole seedlings (USDA, 2010).

On bolls, dark-green, water-soaked lesions generally form at basal cracks or tips, causing rotting of the tissues within, with a layer of fungal growth on the surface under humid conditions (Fig. 2). Sporangia may be present on the surface of rotten bolls and oospores may be present on the cotton lint and internal carpel surface of infected bolls. As the spots develop, entire diseased bolls may rot away. Infected tissues may turn almost black (USDA, 2010). Infection is restricted to the lower two-thirds of the cotton plant (Elena and, Paplomatas 1998).

Figure 2. Early stage of boll rot caused by *Phytophthora boehmeriae* (top), Infected cotton boll after a rain (bottom). Institute of Plant Protection (IPP), Chinese Academy of Agricultural Sciences (CAAS).

Ramie: The pathogen attacks leaves and stems. The leaf spots are round or irregular, water-soaked, and light green at first, then dark brown or dark green. At later stages, the spots turn yellowish brown or gray at the center with a brown margin. Diseased leaves tend to fall early. Elliptical, dark brown stem lesions are present, mainly at the bases, resulting in rotting of whole bases (USDA, 2010).

Paper mulberry and Chinese wingnut: Leaf spots similar to those found on cotyledons or leaves of cotton are observed. Root rot is also observed on Chinese wingnut (Ho and Lu, 1997; Gao, 2007).

Citrus: The pathogen causes brown fruit rot and root stock gummosis (USDA, 2010).

Pine: The pathogen causes root rot on pine (Oxenham and Winks, 1963).

Geraldton waxplant: Plants show progressive yellowing of the branches throughout the stems. Leaves of diseased plants become grayish green, then yellow, and finally straw-colored. Leaves remain attached to the branches after the plants die. Root and stem discoloration was observed and the root cortex sloughed off (Wolcan and Lori, 2001).

Black wattle: Disease caused by *P. boehmeriae* is evident by dark lesions at the trunk base without gum exudation up to 10 m (32.8 ft.) in height (Dos Santos et al*.,* 2006).

Tree of heaven: Major symptoms were water-soaking and subsequent blighting of leaflets, petioles, and rachises of pinnate leaves and young shoots of plants growing in shade under taller and more aggressive trees such as acacia (*Robinia pseudo-acacia*) (Fig. 3) (Kim and Kim, 2004).

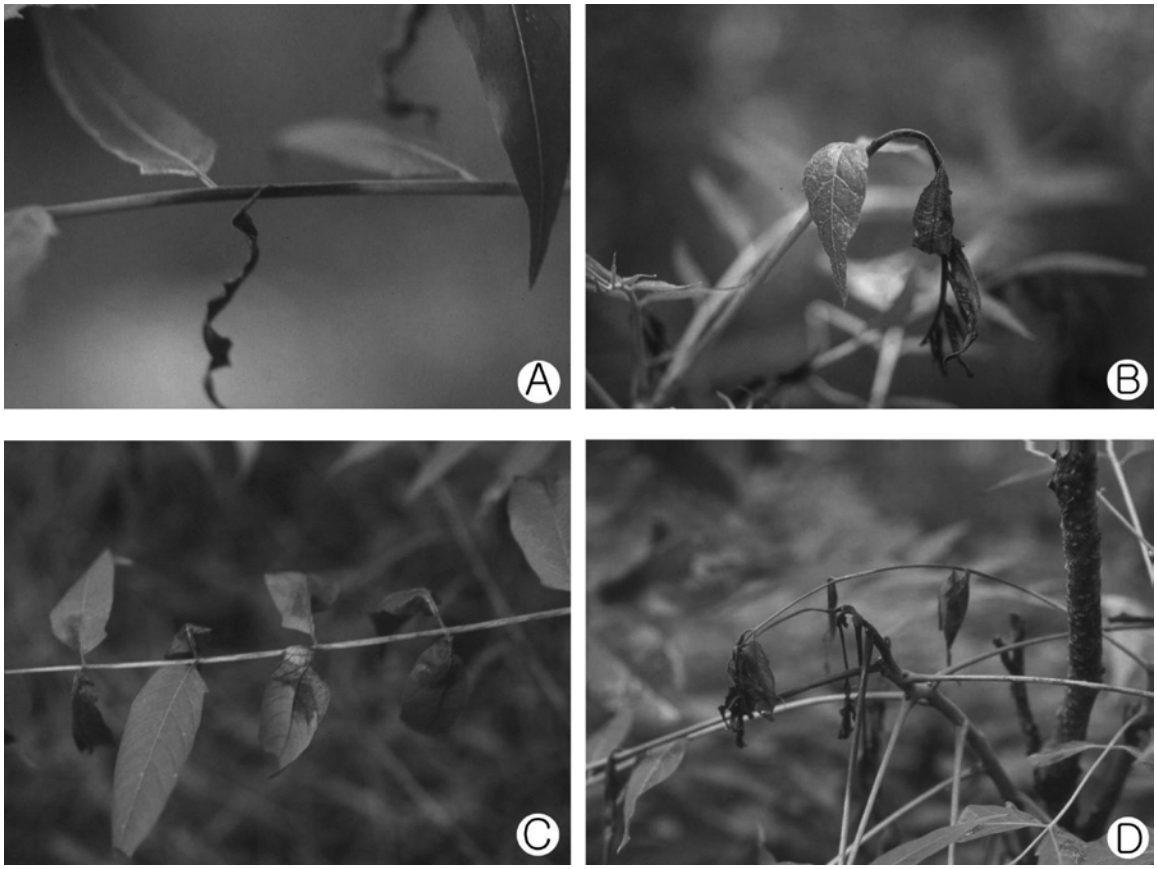

Figure 3. Symptoms on *Ailanthus altissiman* caused by *Phytophthora boehmeriae*. (A through C) Blight of leaflets, petioles, and rachises; (D) Shoot blight. Photos courtesy of Byung-Soo Kim, Kyungpook National University (Kim and Kim, 2004).

Pest Importance

Phytophthora boehmeriae has been reported to damage ramie, cotton, citrus, pine, paper mulberry (*Broussonetia papyrifera*), and Chinese wingnut (*Pterocarya stenoptera*) (Ho and Lu, 1997; Gao, 2007; USDA ,2010). In China, *P. boehmeriae* is the principal agent causing cotton blight and ramie blight (Gao, 2007). Cotton blight, including cotton seedling blight and cotton boll blight, is one of the main diseases in cotton in mainland China (Ho and Lu, 1997; Shen et al., 2005; Gao, 2007). Cotton seedling blight damages leaves, stems and roots resulting in lesions, premature leaf fall, wilting, and death of whole seedlings. A severe attack can kill 30 to 50% of seedlings in the field in cool, wet weather (Gao, 2007). In addition to yield loss, the disease affects cotton quality resulting in reduced fiber length and decreased ginning outturn (Li et al., 1992; Li et al., 1999; Gao, 2007). In Greece, *P. boehmeriae* boll rot has been reported as a new threat to cotton cultivation (Elena and Paplomatas, 1998).

Ramie blight, also called leaf spot of ramie, damages leaves and stems resulting in premature leaf fall and rotting of leaves and stems (Chen, 1986; Zhou, 1984; He et al., 1993; Gao, 2007).

In Argentina, *P. boehmeriae* was reported to cause basal rot in geraldton waxplant (*Chamelaucium uncinatum*) (Wolcan and Lori, 2001). In Australia it has been recorded to attack citrus and pine, causing rot of fruit and roots, respectively (Gerrettson-Cornell, 1989). *P. boehmeriae* was reported as one of the causal agents of the gummosis complex in black wattle in South Africa (TPCP, 2004) and Brazil (Dos Santos et al., 2006) and is now of quarantine importance for the citrus industry.

Known Hosts

From USDA (2010):
Acacia mearnsii (black wattle), *Ailanthus altissima* (tree-of-heaven), *Araucaria hetrophila* (Norfolk Island Pine), *Avicennia* spp. (mangrove), *Boehmeria frutescens* var. *concolor* (nakai), *Boehmeria nivea* (ramie), *Broussonetia papyrifera* (paper mulberry), *Cedrus deodara* (Deodar cedar), *Chamelaucium uncinatum* (Geraldton waxplant), *Citrus sinensis* (orange), *Citrus* spp., *Eucalyptus dunnii*, *Eucalyptus grandis*, *Eucalyptus macarthurii*, *Eucalyptus pilularis*, *Eucalyptus smithii*, *Ficus* spp., *Gossypium hirsutum*, *Gossypium* spp., *Malus domestica* (apple), *Malus sylvestris* (apple), *Persea americana* (avocado), *Persoonia longifolia* (long-leaf Personnia), *Pinus patula* (Mexican yellow pine), *Pinus* spp., *Pterocarya stenoptera* (Chinese wingnut), and *Solanum melogena* (eggplant) are hosts for *P. boehmeriae*.

Experimental hosts for *P. boehmeriae*: *Allium fistulosum* (Japanese bunching onion), *Benincasa hispida* (Chinese waxgourd), *Capsicum annuum* (bell pepper), *Cephalonoplos segetum* (common cephalanoplos), *Chenopodium album* (fathen), *Convolvulus arvensis* (field bindweed), *Corchorus capsularis* (white jute), *Cucumis sativus* (cucumber), *Cucurbita moschata* (butternut squash), *Ipomoea batatas* (sweet potato), *Ixeris denticulata* (stebbins), *Ixeris laevignata* (stebbins), *Lycopersicon esculentum* (tomato), *Nicotiana rustica* (tobacco), *Nicotiana tabacum* (tobacco), *Phaseolus vulgaris* (bean), *Portulaca oleracea* (little hogweed), *Pyrus* spp. (pear), *Rehmannia glutinosa* (Chinese-foxglove), *Ricinis communis* (castorbean), *Solanum tuberosum* (potato), *Taraxacum mongolicum* (Mongolian dandelion), *Vicia bungei* (vetch), and *Xanthium sibericum* (Siberian cocklebur).

Known Vectors (or associated organisms)

P. boehmeriae is not a known vector and does not have any associated organisms. In wet weather conditions, *P. boehmeriae* is usually the primary pathogen. It attacks the cotton plants and can allow other pathogens, such as *Pythium* spp., *Fusarium* spp., *Rhizoctonia,* and others, access to the plant (Shen et al., 2005).

Known Distribution

Africa: South Africa. **Asia:** China, India, Japan, and Korea. **Europe:** Greece. **North America:** Mexico. **Oceania:** Australia, Papua New Guinea, and New Zealand. **South**

America: Argentina and Brazil (Paplomatas et al., 1995; D'Souza et al., 1997; Ho and Lu, 1997; Dos Santos et al., 2006; USDA, 2010).

Potential Distribution within the United States

There is an herbarium record of *P. boehmeriae* from the United States in 1946. However, no locality or host information is provided. *P. boehmeriae* is not believed to be present in the United States (USDA, 2010). Based on the wide host range and wide range of optimal temperatures, this pathogen should have no problem finding a suitable host and environment in the United States.

Survey

CAPS-Approved method*: Conduct visual inspection for symptoms associated with *P. boehmeriae*. Soil sampling is also acceptable for this *Phytophthora* spp., particularly if sampling woody hosts. If a suspect is found, affected plant tissues should be collected and sent to a trained plant pathologist for confirmation of the presence of *P. boehmeriae.*

*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 visual inspection for symptoms associated with *P. boehmeriae*. Symptom recognition is based on the irregular, water soaked, dark-green spots or lesions on cotyledons or true leaves and brown streaks or brown rot of seedlings, and dark-green water soaked lesions at basal cracks or tips on bolls (USDA, 2010). Sporangia may be present on the surface of rotten bolls and oospores may be present on the cotton lint and internal carpel surface of infected bolls (USDA, 2010). Infection is restricted to the lower half to two-thirds of the cotton plant (Elena and Paplomatas, 1998; Paplomatas et al., 1995). Identification may be complicated by the similarity of symptoms caused by other *Phytophthora* species and the infection of *P. boehmeriae* in complex with other pathogens (Shen et al., 2005).

From USDA (2010): *Phytophthora boehmeriae* has been isolated from direct plating of symptomatic plant material and from soil. Linde et al. (1994) took soil and root samples from four eucalyptus and/or pine trees per site. One soil sample was sampled in the rhizosphere of each tree. The top 5 cm (2 in.) of soil was removed and ~1 kg of soil sampled at a depth of 5 to 25 cm (2 to 9.8 in.). Root samples consisted of diseased sections of adventitious roots with feeder roots. PDA, CMA, CMA +A, 3P, PVPH, PARP, PARPH, NARPH were used by various authors to culture *P. boehmeriae*.

Key Diagnostics/Identification

CAPS-Approved Method*:

1) Serological: An ELISA test is available for *Phytophthora* at the genus level for primary screening. A positive does not indicate *P. boehmeriae*

ID must be confirmed by other methods.

2) Morphological: Samples of affected tissues must be collected and sent to a laboratory where a trained plant pathologist can confirm the presence of *P. boemeriae.* Sawada (1927) published the original species description, but the description is not in English. Erwin and Ribeiro (1996) summarize the Sawada paper and others*. P. boehmeriae* can be recognized by the production of abundant oogonia and oospores in single culture, amphigynous antheridia, and broadly ovoid to subspherical conspicuously papillate sporangia.

*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:
Morphological: Symptomatology can be combined with morphology to identify *P. boehmeriae*. *P. boehmeriae* differs from *P. parasitica* and *P. citrophthora* by production of oospores in single culture and from *P. megasperma* by production of papillate sporangia.

Biochemical: Analysis of α-esterase isozymes revealed unique banding patterns for isolates of *P. boehmeriae* compared with those of *P. cactorum* and *P. parasitica,* which are morphologically similar *Phytophthora* species (Elena and Paplomatas, 1998).

Molecular: Shen et al. (2005) reported a PCR assay for the molecular detection of *P. boehmeriae* in infected cotton. They developed a pair of primers (PB1 and PB2) specific to *P. boehmeriae* from the internal transcribed spacer regions of ribosomal DNA, which produced a PCR product of approximately 750 bp. Using primers PB1 and PB2, *P. boehmeriae* could be detected in tissue one day after inoculation, prior to the appearance of symptoms. Using primers PB1 and PB2, detection sensitivity was approximately 10 fg DNA/µl. This method provides an accurate and sensitive way to detect *P. boehmeriae* in cotton tissue.

Note: SSCP analysis of PCR-amplified ribosomal DNA internal transcribed region I, as described above for *P. alni*, has now been used to identify and provide molecular fingerprints for *P. boehmeriae* (Gallegly and Hong, 2008).

Easily Confused Pests
As several *Phytophthora* species have been reported to infect cotton and the infection of *P. boehmeriae* may cause other pathogens access the diseased plants, disease diagnosis of *P. boehmeriae* is difficult on cotton. It is also difficult to identify the species when sporangia or other characteristic morphological structures are absent (Shen et al., 2005).

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Appendix A: Diagnostic Resource Contacts

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Appendix B: Glossary of Terms

Aedeagus: The male copulatory organ; the distal part of the phallus.

Aestivation: Dormancy in summer during periods of continued high temperatures, or during a dry season; also known as summer diapause.

Allopatric: Occurring in separate, non-overlapping geographic areas. Often used to describe populations of related organisms unable to crossbreed because of geographic separation.

Amphigynous: In Oomycota, when the oogonium grows through a single antheridium, which is then attached like a collar at the base of the oogonium. Compare to *paragynous.*

Antheridia: The male sexual structure of an Oomycete.

Albedo: The spongy white tissue on the inside of the rind of citrus fruit.

Appressoria: A specialized structure in fungi that attaches the germinating spore to the leaf surface using a sticky substance.

Blight: Sudden, severe, and extensive spotting, discoloration, wilting, or destruction of leaves, flowers, stems, or entire plants.

Bolls: The spherical fruits of cotton and flax.

***Bt*:** *Bacillus thuringiensis*. A gram-positive, soil-dwelling bacterium, commonly used as a pesticide.

***Bt* Cotton:** Genetically modified cotton that has been modified by the insertion of a gene or genes from *Bacillus thuringiensis*, a common soil bacterium.

Cambium layer: The layer of actively dividing cells between the bark and wood or xylem and phloem of a woody plant.

Cankers: Sunken stem lesions caused by plant pathogens.

Carpel: The ovule and seed-producing reproductive organ in flowering plants. One or more carpels make up the pistil.

Cerarii: These are characteristic of mealybugs and consist of groups of large setae, usually conical, on the lateral margins of the body.

Chlamydospore: The large, thick-walled resting spore of fungi.

Chlorotic: Abnormal condition of plants in which the green tissue loses its color or turns yellow as a result of decreased chlorophyll production due to disease or lack of light or oxygen.

Clavus: The oblong or triangular portion of the forewing (Hemiptera).

Corium: The elongate, usually thickened, basal part of the forewing (Hemiptera).

Costa: Any elevated ridge that is rounded at its crest; the thickened anterior margin of any wing, but usually the forewing of an insect.

Costal vein: A longitudinal wing vein usually forming the anterior margin of the wing.

Cotyledon: An embryonic leaf of a seed bearing plant.

Coxa (pl., coxae): The basal segment of the leg of an insect, by means of which it is articulated with the body.

Cremaster: A single hook or a series of smaller hooks that attach a pupa to twig or other structure.

Cuticle: The external skeletal structure of an arthropod.

Defoliate: Loss of leaves from a plant, whether normal or premature.

Degree Days: Development of poikilothermic ("cold-blooded") organisms such as insects, fungi, and plants, is regulated by environmental temperatures. Development to particular stages in the life cycles of these organisms is largely controlled by how much heat they experience, where heat is considered as a function of temperature and time. Degree-days are an estimate of the amount of heat accumulated over a 24-hr period. They are calculated using lower and upper developmental thresholds unique to a particular organism and, typically, some approximation of the 24-hour temperature pattern derived from minimum and maximum daily temperatures (which are commonly available from local weather-recording stations). Only those temperatures falling between the lower and upper thresholds are included in the calculations. Degree-day values may be positive or equal zero (all temperatures above or below thresholds), but are never negative. Degree-days are calculated for each day and are then summed to provide cumulative (total) degree-days. Starting points for calculating cumulative degree-days are usually arbitrary, typically January 1 but often later (e.g. April 1) in areas with cold winter temperatures. Based on experimental data, cumulative degree-days are linked to specific development events of interest (e.g. adult insect emergence). Thus, a pest manager can anticipate or predict an event of interest based on local temperature data and an appropriate degree-day based developmental model.

Deutonymph: The third instar of a mite.

Diapause: A period of arrested development and reduced metabolic rate, during which growth, differentiation, and metamorphosis cease; a period of dormancy not immediately referable to adverse environmental conditions.

Ductus: The oviduct of a female insect

Elytron (pl., elytra): A thickened forewing (Coleoptera, Dermaptera, and some hemiptera).

Epicranium: The upper part of an insect head; from the face to the neck.

Filiform: Hair-like or thread-like, refers to antennae.

Frass: Insect excrement.

Gummosis: A secretion of liquid from a woody plant used as part of defense against infection of pathogens.

Hemelytron (pl., *hemelytra*): The forewing in Hemiptera.

Homothallic: A condition in which sexual reproduction occurs with a single thallus; self-fertile.

Isomers: Compounds with the same molecular formula but different structural formulas.

Juxta: An organ in the males of most Lepidoptera that supports the aedeagus.

Labrum: The upper lip of an insect, directly below the clypeus.

Mandible: The jaw of an insect; one of the anterior pair of paired mouthpart structures.

Metathorax: The third segment of the thorax.

Mycelium: The vegetative part of a fungus, consisting of a network of threadlike, branching hyphae.

Obpyriform: Inversely pear shaped; pear shaped with narrow end at bottom.

Obturbinate: Having the shape of an up-side down top.

Ocelli: A simple eye of an insect or other arthropod.

Oogonia: Female gametangium of Oomycetes, containing one or more gametes.

Oospore: Thick-walled, sexually-derived resting spore of Oomycetes.

Ostium bursae: The copulatory opening in female moths.

Palp: A segmented process born by the maxillae or labium of an insect.

Papilla: A small nipple-like elevation.

Paragynous: Antheridia attached to the side of an oogonium.

Pedicel: The second segment of the antenna.

Pinaculum (pl., pinacula): A small, flat or very slightly elevated chitinized area bearing from one to four setae. In caterpillars, an enlarged seta-bearing papilla forming a flat plate.

Pronotum: The dorsal sclerite of the prothorax.

Prothorax: The first thoracic segment.

Protonymph: The second instar of a mite nymph.

Pygophore: The large upper sclerite of the genitalia in Hemiptera (Gordh et al., 2003).

Pygopod: A rudimentary hind leg.

Rachise: The principle axis of an inflorescence or of a compound leaf.

Sclerite: A hardened body-wall plate.

Scutellum: A sclerite of the thoracic notum.

Seta (pl., setae): A bristle or hair.

Sporangium: A fragile, sac-like structure in which oospores are produced in most Oomycetes.

Sporangiophores: Specialized, branched, treelike hyphal structures that aid in the air dispersal of sporangia.

Stoma (pl., Stomata): Minute pores in the epidermis of plants that allow for gas exchange.

Sympodial: Having or involving the formation of an apparent main axis from successive secondary axes.

Tarsus (pl., tarsi): The leg segment immediately beyond the tibia, sometimes consisting of more than one “segments” or subdivisions.

Tibia (pl., tibiae): The fourth segment of the leg, between the femur and the tarsus.

Teneral: A recently molted, pale, soft bodied individual.

Tubercle: A small knotlike or rounded protuberance.

Urogomphus (pl., urogomphi): Fixed or mobile cercus-like processes on the last segment of a beetle larva.

Vesica: In Lepidoptera, the penis, or terminal part of the aedeagus. The vesica is membranous and eversible, typically held within the tubular part of the aedeagus, but everted and inflated during copulation.

Whorl: Spiral pattern.

Zoospore: Asexual reproductive structures with flagella that are produced in some oomycetes.

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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

Version 10.2 5/1/09

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	Veronicelladae 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. cartiana*, *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 xyloteanus*	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 sleckmanni*	Wingless Weevil																	■																	1
34	*Oxycarenus hyalinipennis*	Cotton Seed Bug														■				■					■	■	■			■	■			■	■	9
47	*Panolis 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*	Pacific 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
8	*Spodoptera litura*	Cotton Cutworm	▲	■	▲	■		▲	▲	■	■	■	▲	▲	▲	▲		■	▲	▲	▲			■	■	▲	▲	▲	■	▲	▲		■	▲	■	28
39	*Tetranychus roseus*	No common name, a spider mite																																		0
36	*Thaumatotibia leucotreta*	False Codling Moth							▲						▲							■		▲		■		▲		■	▲			■	▲	10

2012 Commodity Matrix for the AHP Priority Pest List **Appendix D**

5	*Thaumetopoea processionea*	Oak Processionary Moth																										▲		■	■	3
6	*Tomicus destruens*	Pine Shoot Beetle																										■				1
4	*Tremex fuscicornis*	Tremex Wood Wasp																												▲	▲	2
27	*Tuta absoluta*	Tomato Leaf Miner																				▲	■									2
11	*Unaspis yanonensis*	Arrowhead Scale						▲																								1
21	*Veronicellia* spp.	Veronicellid Slugs			■			■	■	■	■	■				■		■				■										9
45	*Xanthomonas oryzae, X. oryzae pv oryzicola*	Bacterial Leaf Streak, Bacterial Blight															▲															1
	Total Pests Per Commodity:		6	4	9	4	3	11	15	9	9	6	9	8	11	7	2	9	15	9	11	1	13	14	14	9	7	6	12	10	15	12	25	27

▲ = Primary host
■ = Other host

■**Other Softwood Trees**: *Abies, Casuarina, Cupressus, Juniperus, Larix, Picea, Pseudotsuga, Tsuga*
■**Soft Hardwood Trees**: *Acacia, Albizia, Alnus, Asimina, Castanea, Catalpa, Celtis, Elaeagnus, Fraxinus, Liquidambar, Magnolia, Melaleuca, Persea, Platanus, Populus, Paulownia, Salad, Salix, Tamarix, Tilia, Ulmus*
■**Hardwood Trees**: *Acer, Aleurites, Amelanchier, Betula, Carpinus, Carya, Castanopsis, Cornus, Crataegus, Diospyros, Eucalyptus, Fagus, Ilex, Juglans, Lithocarpus, Malus, Melia, Morus, Prunus, Quercus, Sapium, Sorbus, Vaccinium*