Biology and Management of the Green Stink Bug

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Stink bugs (Hemiptera: Pentatomidae) are significant economic pests of many agricultural crops and are frequently one of the most difficult pests to control in crops such as soybean [Glycine max (L.) Merr.], cotton (Gossypium hirsutum L.), tomato (Solanum lycopersicum L.), and many fruit crops. The green stink bug, Acrosternum hilare (Say) (Fig. 1), is native to North America and is a pest found throughout the United States. It is one of several important stink bug species that inflict serious economic injury to agricultural commodities.

Identification. The green stink bug is 1.3–2.0 cm long and 0.8 cm wide. It is usually bright green and may have a yellow border around the margin of the abdomen, head, and thorax (Underhill 1934). Although rare, an orange color morph of this insect exists (Fig. 1b).

Green stink bug adults are similar in appearance to the southern green stink bug, Nezara viridula (L.), but can be differentiated by black bands on the antenna and a pointed abdominal spine, instead of red anten- nal bands with a rounded abdominal spine (Fig. 2) (Miner 1966, McPherson 1982, McPherson and McPherson 2000). The nymphs of the green stink bug are easily distinguishable from those of the southern green stink bug. Southern green stink bug nymphs have two rows of white dots on their abdomen, whereas the green stink bug nymphs do not. Color morphs are found in both species of nymphs, and both dark and light colorations are commonly exhibited by green stink bug nymphs (Figs. 3 and 4).

Taxonomy. The green stink bug first was described by Thomas Say as Pentatoma hilaris in 1832 (Say 1832). Its scientific name was changed several times, and eventually in 1915, it was changed to its current name, A. hilare (Parshley 1915). However, it has been suggested recently that the green stink bug is misnamed and should be classified as Chinavia hilaris (Say) (D. Rider, personal communication). According to their findings, the genus Acrosternum belongs to smaller Palearctic species of stink bugs that live in arid environments. However, because the Entomological Society of America has not officially changed the species name of the green stink bug, this paper will use the nomenclature A. hilare (ESA 2012).

ABSTRACT. The green stink bug, Acrosternum hilare [Say] [Chinavia hilaris [Say]] (Hemiptera: Pentatomidae) is one of the most damaging native stink bug species in the United States. It is a pest of economic importance in a variety of commodities, including cotton (Gossypium hirsutum L.), soybeans [Glycine max (L.) Merr.], tomatoes (Solanum lycopersicum L.), and peaches [Prunus persica (L.) Batsch]. Stink bug feeding can result in cosmetic damage as well as reduced quality and yield. Acrosternum hilare and other stink bugs have become a major challenge to integrated pest management systems because control options are basically limited to the application of broad-spectrum insecticides such as organophosphates, carbamates, and pyrethroids. However, neonicotinoids are generally effective for control of this stink bug and may be less disruptive to natural enemies. Further options for stink bug management that are being explored include the use of trap crops and enhancing beneficial parasitoid populations.

Life Cycle and Seasonal Biology. The green stink bug overwinters as an adult, preferring to do so in leaf litter and deciduous woodlands (Underhill 1934). It emerges from diapause when temperatures surpass 18°C (McPherson 1982). Adults are most active when temperatures exceed 24°C, and are more prone to flight when temperatures exceed 27°C (Underhill 1934).

After emerging from diapause, the adult females are not reproductively mature and require a preovipositional period to develop (Kamminga et al. 2009a, Nielsen and Hamilton 2009). The ovipositional period of overwintered adults in eastern North America begins in the middle of June and ends the first week of September, with peak egg laying occurring the third week of July (Underhill 1934, Jawahery 1990). The largest populations of first generation egg masses are found near the woods where adults have overwintered. This generation continues to feed on weedy hosts and migrates into nearby crops once they become attractive as a food source (i.e., budding or fruiting structures). Miner (1966) reported that the second generation remains in the cropping system throughout its life stages.

The green stink bug has a single generation in northern areas such as Canada (Javahery 1990), but in more favorable southern conditions there may be two generations (Sailer 1953, Kamminga et al. 2009a). It is thought that the differences in reproductive cycles may reflect differences in climate. Wilde (1969) reported that photoperiod is the primary determinant of generation number, as opposed to temperature.

Newly eclosed females begin copulating after ~22 d, and lay their first egg masses about 3 wk later (Miner 1966). Females can lay a new egg cluster every 8–10 d. These eggs are deposited vertically in clusters of 1–72 (Underhill 1934) with an average of 32 eggs per egg mass (Miner 1966). However, >130 eggs per cluster have been observed under laboratory conditions (Miner 1966, Jawahery 1990). Time between egg mass depositions typically decreases after the first mass is laid (Underhill 1934, Nielsen et al. 2008).

Eggs are usually adhered to the underside of leaves. The eggs are barrel-shaped and change from light green to yellow (Fig. 5), and then...
the egg stage depends on temperature. Shorter durations occur in warmer temperatures, and longer durations in cooler temperatures (Underhill 1934, Capinera 2001). Upon hatching, they undergo five instars before becoming adults (Underhill 1934, Miner 1966). The first instars do not feed and remain clustered together around the egg mass. Second instars are less gregarious and begin to feed. Third instars behave in a similar manner to second instars, but are slightly larger in size. Feeding by the fourth and fifth instars can result in as much economic damage to the plants as from adults (Barbour et al. 1988). Development time of the nymphs can vary based on the host and temperature, and development of each instar occurs most rapidly at 27°C (Simmons and Yeargan 1988). Adulthood is reached in ≈36 d (Underhill 1934, Miner 1966). A complete description of each life stage is available by Miner (1966).


Crop Damage

The green stink bug is one of several species of agriculturally important stink bugs in the United States. Similar to other stink bugs, green stink bug populations reach their peak in late summer. Crop injury is accrued when the feeding stylet penetrates plant tissue. The economic injury potential of green stink bug feeding varies by crop.

**Tomato.** Several species of stink bugs, including green stink bug, are reported as economic pests of the tomato (Lye et al. 1988, Zalom et al. 1997, McPherson and McPherson 2000, Nault and Speese 2002). Stink bugs inject a toxin into the fruit when they feed, which can result in a spongy white area. This renders it unmarketable for processing and fresh market tomatoes (Fig. 6) (Kennedy et al. 1983). Stylet insertion also leads to premature ripening and smaller fruit (Lye et al. 1988). Nault and Speese (2002) reported stink bugs as causing more damage to spring tomatoes in Virginia than either thrips or lepidopterans. In addition, Kennedy et al. (1983) stated that the green stink bug is one of the most damaging pests of tomato in North Carolina.

**Peach.** Stink bug feeding on fruit trees can result in extensive damage, and feeding injury in the early developmental stage of the fruit results in the most damage (McPherson and McPherson 2000). In peaches, feeding injury results in blemishes on the skin, yield loss, misshapen fruit, or catfacing (Fig. 7) (Rings 1957, McPherson and McPherson 2000). Catfacing occurs because the site of the feeding puncture does not grow, although the areas around it continue to grow. This results in bumpy areas on the surface of the fruit, making it unmarketable (Rings 1957). This same dimpling and damage to the
surface of the peach can also occur later in the development of the fruit (Rings 1957, McPherson and McPherson 2000).

Historically, green stink bug was considered an economically important stink bug pest of peaches along with the brown stink bug, *Euschistus servus* (Say), and the dusky stink bug, *Euschistus tristigmus* (Say), in the eastern United States (Hogmire 1995). More recently, the brown marmorated stink bug, *Halyomorpha halys* (Stål), has caused the greatest economic loss to peaches in the mid-Atlantic states (Leskey and Hamilton 2010).

Soybean. Stink bug feeding preference varies with the developmental progress of the crop. The insects typically move into a soybean field after flowering and remain through plant maturity. A survey of soybean and cotton in southeastern Virginia determined that brown and green stink bugs were the most common species in 2005 and 2006 (Kamminga 2008). More recently, the brown marmorated stink bug has become a dominant stink bug in many agricultural commodities in the mid-Atlantic, including areas of New Jersey, Maryland, Delaware, Pennsylvania, West Virginia, and Virginia. It was reported that the brown marmorated stink bug outnumbered all native species in soybean fields in Pennsylvania (Nielsen et al. 2011).

Stink bug feeding on soybean can cause green stem syndrome, a plant response that results in green stems past maturity. Feeding also results in reduced seed quality (Fig. 8); decreased yield (Underhill 1934, McPherson and McPherson 2000); and less oil than undamaged beans (Daugherty et al. 1964). Feeding punctures from green stink bug stylets may also increase the possibility of infection by a pathogen. The insect has been shown to transmit the causative agent of yeast-spot disease (McPherson and McPherson 2000). Importers may reject seeds if too much stink bug damage is present (Chyen et al. 1992), resulting in even greater economic loss.

Cotton. Stink bugs have been considered to be pests of cotton since the early 1900s (Morrill 1910). Historically, stink bugs were controlled by broad-spectrum insecticide sprays, intended to control other
pests such as the boll weevil, *Anthonomus grandis grandis* Boheman (Coleoptera: Curculionidae), and the bollworm, *Helicoverpa zea* (Boddie) (Lepidoptera: Noctuidae) (Greene et al. 1997). Recently, stink bug populations in cotton have increased. This is in part because of fewer insecticide sprays targeting boll weevil, resulting from a widely successful eradication program throughout most of the southern United States. In addition, *B. t.*,* Bacillus thuringiensis* Berliner, crops have increased in acreage and provide control over many lepidopteran pests without the need to spray insecticides. These advancements have decreased the number of broad-spectrum insecticide sprays on cotton, resulting in an increase in stink bugs.

A complex consisting of the four primary stink bugs, the green, southern green, redbanded, *Piezodorus guildinii* (Westwood), and brown stink bug are found in cotton throughout the southeast. Reay-Jones et al. (2009, 2010) reported the green stink bug as being the primary stink bug collected during sampling in cotton in South Carolina and Georgia in 2008 and 2007. In addition, surveys in Virginia also determined that the green stink bug is a prominent stink bug in the cotton system (Kamminga 2008).

Stink bugs feed on young, tender cotton bolls resulting in injured seeds and lint, and lower yield (Fig. 9) (Barbour et al. 1988, 1990). Researchers have investigated an association between external feeding punctures (sunken lesions) and internal injury; however, it is still difficult to accurately correlate the two (Blinka et al. 2010).

**Monitoring and Management Options**

**Sweep Nets and Beat Sheets.** Sampling of insects on crops is traditionally performed to assess the pest complex in relation to economic thresholds. Sweep net and beat sheet sampling methods are recommended for monitoring stink bugs in field crops (Todd and Herzog 1980). For fruits and vegetables, beat sheets are more common (Nielsen and Hamilton 2009).

Stink bug thresholds vary according to the crop. For example, the threshold for tomatoes is one stink bug per three beat sheet samples (Brust and Zalom 2005). For soybeans, recommendations for stink bug thresholds can also vary by state (Parker 2012). In many southern states, a beat sheet sample containing above one stink bug per 0.3-row meter may require an insecticide application (Parker 2012). However, thresholds can also vary by maturity (Parker 2012) and seed spacing (Herbert 2012). Current practices in cotton recommend a dynamic threshold based on the age of the boll and the percent internal damaged bolls per field. Moreover, Herbert (2012) indicates that one stink bug per 25 sweeps or one stink bug per two-row meters may be above economic threshold.

Establishing accuracy of the sampling methods is complex, as this can depend on the life stage and density of both crops and insects, as well as time of day (Reay-Jones et al. 2009). Sampling using sweep nets and beat sheets can help determine the seasonality and life stage of stink bugs, but they may drop to the ground or fly away when disturbed. Other monitoring methods, like pheromone and blacklight traps, may offer additional information as to the species, relative abundance, and flight activity of stink bugs on farms.

**Blacklight Trapping.** Many insects, including stink bugs, can be monitored using blacklight traps (Fig. 10). Blacklight trap catch may be useful for improving the timing of scouting and management methods for stink bugs (Kamminga et al. 2009a, Nielsen and Hamilton 2009). In addition, flight activity and distribution can be determined for native and possible invasive species.
Pheromone Trapping. Pheromone traps have been used to monitor green stink bug populations in peach, Prunus persica (L.) Batsch; apple, Malus domestica Borkh. (Leskey and Hogmire 2005); and pecan, Carya illinoinensis (Wangenh.) K. Koch, orchards (Mizell and Tedders 1995). A yellow base pyramid trap with a pheromone lure is commonly used to monitor green stink bug populations (Fig. 11) (Leskey and Hogmire 2005, Hogmire and Leskey 2006). The aggregation pheromone blend for the green stink bug has been identified as a 95:5 cis:trans blend of (4S)-cis-Z-bisabolene epoxide and (4S)-trans-Z-bisabolene epoxide (Aldrich et al. 1989, 1993; McBrien et al. 2001). Cross attractiveness of the green stink bug to the southern green stink bug pheromone also has been documented (Tillman et al. 2010) and may occur because of the similarities in their chemical composition (Aldrich et al. 1989, 1993). However, synthesis of the aforementioned pheromone is expensive and currently inhibits widespread use of this technique.

Chemical Control. Broad-spectrum insecticides, such as organophosphates and pyrethroids, are the most frequently applied insecticides for stink bug management (Kuhar et al. 2006, Herbert 2012), and both classes provide efficacy against Acrosternum spp. (Willrich et al. 2003). Dicrotophos, an organophosphate, has been cited as having a high toxicity to various stink bug species (Tillman and Mullinix 2004, Snodgrass et al. 2005), and it is suggested that it provides consistent control of the green stink bug (Willrich et al. 2003, Snodgrass et al. 2005). The green stink bug also has been reported as having a lower LC50 (lethal concentration required to kill 50% of the insects tested) value to pyrethroids and organophosphates than the brown stink bug (Greene et al. 2001, Snodgrass et al. 2005). In addition, Kamminga et al. (2009c) performed efficacy trials on stink bug nymphs and adults in southeastern Virginia, and found that the green stink bug was especially susceptible to all of the pyrethroids tested. Neonicotinoids also were effective against green stink bug adults. In particular, thiamethoxam, dinotefuran, and imidacloprid were found to be efficacious against green stink bug adults; whereas dinotefuran and clothianidin were efficacious against green stink bug nymphs. In a similar study, Tillman (2006b) reported that acetamiprid and thiamethoxam exhibited some residual contact and ingestion activity on nymphs, but not adults, of the southern green stink bug.

Differences in the susceptibility of stink bug life stages to insecticides are common (McPherson et al. 1979; Willrich et al. 2002, 2003; Kamminga et al. 2009c). Willrich et al. (2003) reported that brown stink bug nymphs were more susceptible to the pyrethroid lambda-cyhalothrin than the adults. Conversely, southern green stink bug nymphs were less susceptible to the pyrethroids cypermethrin and lambda-cyhalothrin than the adults. McPherson et al. (1979) and Willrich et al. (2002) reported that the LD50 (lethal dose required to kill 50% of the insects tested) value to pyrethroids and organophosphates than the brown stink bug (Greene et al. 2001, Snodgrass et al. 2005). In addition, Kamminga et al. (2009c) performed efficacy trials on stink bug nymphs and adults in southeastern Virginia, and found that the green stink bug was especially susceptible to all of the pyrethroids tested. Neonicotinoids also were effective against green stink bug adults. In particular, thiamethoxam, dinotefuran, and imidacloprid were found to be efficacious against green stink bug adults; whereas dinotefuran and clothianidin were efficacious against green stink bug nymphs. In a similar study, Tillman (2006b) reported that acetamiprid and thiamethoxam exhibited some residual contact and ingestion activity on nymphs, but not adults, of the southern green stink bug.

Very little research has been done to test the efficacy of Organic Materials Review Institute-certified (organic) insecticides against stink bugs. The research that has been published primarily investigates the effects of the extract azadirachtin from the neem tree, Azadirachta indica (A. Juss), on stink bug nymphs. Azadirachtin acts as an anti-feedant (Seymour et al. 1995, Abudulai et al. 2003, Kamminga et al. 2009b), but mixed results have been reported in its efficacy against stink bugs (Kamminga et al. 2009b). Azadirachtin is also an insect growth regulator and has been reported as disrupting the molting of stink bug nymphs (Abudulai et al. 2003, Riba et al. 2003), but research also has indicated that it is not always efficacious against stink bug nymphs that were fed an azadirachtin-treated food source (Overall 2008, Kamminga et al. 2009b).

Pyrethrins and an organic formulation of spinosad have been found to be effective against stink bug nymphs (Overall 2008, Kamminga et al. 2009b). Pyrethrins were reported as repelling stink bugs, whereas spinosad attracted them (Kamminga et al. 2009b). Spinosad also was determined to have a lower LC50 on harlequin bug nymphs, Morgania histrionica (Hahn), than either pyrethrins or azadirachtin (Overall 2008). In addition, in behavioral bioassays, stink bugs were found to feed more on spinosad-treated tomatoes than untreated-, azadirachtin-, and pyrethrin-treated tomatoes. Spinosad caused high mortality when green and brown stink bug adults were fed treated green beans, but mixed results were obtained when tested against nymphs (Kamminga et al. 2009b).

Cultural Control. Trap crops are designed to intercept and allow for the removal of a pest before it moves into a cash crop by providing a preferred food source. This method has been found to be an effective management technique for stink bugs (Todd and Schumann 1988, Hokkanen 1991). For example, sorghum, Sorghum bicolor L. Mo-
Host Plant Resistance. Stink bug resistant varieties of soybean have been shown to have a lower population of pentatomids than other varieties (Gilman et al. 1982, Jones and Sullivan 1982). More recent studies have indicated that certain breeding lines of soybean may be possible genetic material for stink bug resistant soybeans (McPherson et al. 2007, Campos et al. 2010). These resistant lines may decrease damage caused by stink bug feeding and insecticide applications for stink bug control in soybeans.

Biological Control. Biological control, which relies upon natural enemies of the pest to manage the pest population and reduce commodity damage, is a part of stink bug pest management. Stink bugs are vulnerable to multiple predators, parasitoids, and pathogens. Meristhen nematodes have been reported as infesting stink bug adults and nymphs (Fuxa et al. 2000, Ribiero and Castiglioni 2008). Lacewing larvae; spined soldier bugs, Podisus maculiventris (Say); and birds are common predators of stink bugs (Underhill 1934, McPherson 1982, McPherson and McPherson 2000). Most stink bug parasitoids are tachinid flies that oviposit on the abdomen of the host. A common example is Trichopoda pennipes (F.) (Diptera: Tachinidae) (Fig. 12) (Capierno 2001). The female fly deposits one or multiple small grayish eggs on the abdomen of adults or nymphs. The eggs hatch in ~30 h and the larvae then burrow into the body of the host. After ~16 d, a single larva will emerge as a maggot from the host. This maggot pupates and an adult fly emerges in 2–4 wk (Worthley 1924). Overwintering larvae remain in their host throughout the winter and emerge in the spring.

Other egg parasitoids include wasps of the genera Trissolcus, Anastatus, and Telenomus (Fig. 13). Trissolcus basalis Wollaston (Hymenoptera: Scelionidae) (Fig. 14) has shown potential against southern green stink bugs in soybean (Ehler 2002). Inoculative releases of 15,000 T. basalis adults per hectare in a trap crop of early maturing soybean reduced the stink bug density an average of 58% in the main crop of late planted soybeans (Corrêa-Ferreira and Moscardi 1996), maintaining it below economic threshold. Koppel et al. (2009) reported 47% green stink bug egg parasitism during a survey of crops in the mid-Atlantic United States. Orr et al. (1986) reported an egg parasitization rate of 14–22% in Louisiana soybeans. The Louisiana and Virginia egg parasitoid surveys determined that green stink bug egg parasitoids were parasitized by Trissolcus euschisti Ashmead, T. basalis, Trissolcus edessae Fouts, and Telenomus podisi Ashmead (Orr et al. 1986, Koppel et al. 2009). An additional parasitoid species, Telenomus cristatus Johnson, also was identified as parasitizing green stink bug eggs in Louisiana (Orr et al. 1986).

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