SOUTHERN CHINCH BUG, *Blissus insularis* BARBER
(HETEROPTERA: BLISSIDAE), MANAGEMENT IN ST. AUGUSTINEGRASS

By

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A THESIS PRESENTED TO THE GRADUATE SCHOOL
OF THE UNIVERSITY OF FLORIDA IN PARTIAL FULFILLMENT
OF THE REQUIREMENTS FOR THE DEGREE OF
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This thesis is dedicated to an extraordinary woman, my grandmother, Margaret Congdon.

TABLE OF CONTENTS

LIST OF TABLES

LIST OF FIGURES

Abstract of Thesis Presented to the Graduate School
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The southern chinch bug, *Blissus insularis* Barber, is the most destructive insect

pest of St. Augustinegrass (*Stenotaphrum secundatum* [Walt.] Kuntze). The purpose of

this research was to examine the effectiveness of different components of an IPM

program for *B. insularis*, including sampling techniques, host plant resistance, biological,

chemical, and cultural controls.

The optimal sampling method for *B. insularis* in a greenhouse experiment was

flotation, compared to using a hand-held vacuum or berlese funnel. The large

blower/vacuum was nearly as accurate at extracting *B. insularis*, so it was used to collect

field populations for use in experiments and flotation was used to recover insects from

test plants in the laboratory. In a no-choice test, more than 62% of the *B. insularis*

successfully survived on both 'Floratine' and 'FHSA-115'. During the 3 wk experiment

feeding, mating, and molting were observed. The different life stages of *B. insularis* had

similar mortality after feeding on either variety, suggesting that neither variety was

resistant to *B. insularis*. Fifth instar and adult *Geocoris punctipes* (Say) consumed a similar number of *B. insularis*. When pooled together, fifth instar and adult *G. punctipes* consumed significantly more first-third instar (57%) *B. insularis* than fourth-fifth instar or adult stages (17.2 and 16.0% respectively).

This study evaluated the efficacy of professional and non-professional (homeowner) products. In the laboratory insecticide assay, diazinon (100 ppm) killed significantly more *B. insularis* adults on sprigs than any other diazinon or Bioblitz treatment 2 and 4 days after treatment. Homeowner products containing bifenthrin, carbaryl, deltamethrin, and λ-cyhalothrin achieved over 80% control 1 wk post-treatment in the field test. All of the professional products were statistically different from the control 1 wk after treatment, although carbaryl was the only treatment to kill all *B. insularis* at 1 wk.

Stunted growth from *B. insularis* feeding was observed from grass clippings collected after 1 wk. Dry weights of grass clippings collected from the different *B. insularis* densities were different from one another except during weeks two, six, and seven. During this time, the dry weights of grass clippings recovered from treatments containing 200 *B. insularis* were lower than dry weights of grass clippings collected from treatments containing 0 or 30 *B. insularis*. *Blissus insularis* feeding in densities of 30 and 200 lowered root weight. At five weeks, dry weight of grass clippings was lowest in treatments with low irrigation. At week seven, dry weights of grass clippings collected from treatments with low irrigation were lower than clippings from treatments with high irrigation levels. Irrigation did not impact root growth. Irrigation and *B. insularis* density did not interact to affect plant growth.

CHAPTER 1
LITERATURE REVIEW

Turfgrass production and maintenance are valuable parts of the Florida economy

(Hodges et al. 1994). In 1992, residential lawns accounted for 75% of the 4.4 million

acres of turfgrass used and maintained throughout the state. Consumers spent $5 billion

on turfgrass maintenance or roughly $1,200 per acre. Sales from products and services

by turfgrass producers and commercial distributors totaled $6.5 billion. The demand for

high quality turfgrass is growing with Florida's increasing annual population of almost

2% (U.S. Census Bureau 2003). Common grasses used in Florida include bahiagrass

(*Paspalum notatum* Flugge), bermudagrass [*Cynodon dactylon* (L.) Pers], centipedegrass

[*Eremochloa ophiuroides* (Munro) Hack], seashore paspalum (*Paspalum vaginatum*

Swartz), and zoysiagrass (*zoysia* spp). However, of the >6.3 million residences in Florida

(U.S. Census Bureau 2000), the most widely used grass is St. Augustinegrass

(*Stenotaphrum secundatum* [Walt.] Kuntze).

St. Augustinegrass is a warm-season, coarse-textured, aggressive, and stoloniferous

grass that is widely used in lawns in the coastal regions of the United States (Turgeon

1996). Most St. Augustinegrass cultivars have good salt and shade tolerance and

establish easily from sprigs or sod. Its aggressive growth habit gives it good recuperative

capability, but it is prone to thatch buildup (Potter 1998). Cultivars of St. Augustinegrass

that are commonly marketed include 'Floratam', 'Palmetto', 'Delmar', 'Floratine', and

'Seville' (White and Busey 1987, McCarty and Cisar 1997, Trenholm et al. 2000).

Several insect pests attack St. Augustinegrass in Florida including tropical sod webworms [*Herpetogramma phaeopteralis* (Guenee)], armyworms (*Spodoptera* spp.), grass loopers (*Mocis* spp.), mole crickets (*Scapteriscus* spp.), rhodesgrass mealybugs [*Antonina graminis* (Maskell)], and southern chinch bugs (*Blissus insularis* Barber). Sod webworms feed primarily at night. Newly hatched larvae skeletonize the grass blades, while older larvae chew on grass blades near the soil surface (Buss and Caldwell 2001). Armyworms first skeletonize the grass blades and later create bare spots. Mature larvae feed during the day and night (Buss 2001). Grass loopers chew on the grass blades, resulting in ragged, yellow to brown patches of grass (Sprenkel 2003). Mole crickets damage turfgrass by their tunneling, which uproots and dries out grass plants. Feeding from rhodesgrass mealybugs discolors grass and heavy infestations give the appearance that excess fertilizer has 'caked' around the grass nodes (Buss 2001). The southern chinch bug is the most destructive insect pest of St. Augustinegrass and can cause extensive damage or kill entire lawns (Reinert and Kerr 1973, Bruton et al. 1983).

Chinch Bugs

Many *Blissus* spp. are important pests that attack plants in the family Graminae. *Blissus* spp. feed on the phloem and xylem near the base of the plant where the grass meristem occurs (Painter 1928). Salivary sheaths are deposited along feeding tracks within the tissues, which is characteristic of feeding on phloem tissue bundles (Backus et al. 1988). Their feeding can cause wilting, chlorosis, stunting, and death through clogging of vascular transport systems (Painter 1928, Negron and Riley 1990, Spike et al. 1991). In particular, *B. insularis* prefer open sunny areas of St. Augustinegrass, especially areas with abundant thatch (Reinert and Kerr 1973), where they suck fluids from the crown and stem of grasses with their needle-like mouthparts. As the grass dies,

the insects feed at the edge of the dead spots, thus enlarging the damaged area. *Blissus insularis* activity begins between March and April (Kerr 1966) and continues until October. Damage is usually first noticed in drought-stressed areas along sidewalks, pavements, or in poorly irrigated areas.

At least 15 *Blissus* species occur in the United States and Canada; however, only four are considered damaging to turfgrass, including the buffalograss chinch bug, (*B. occiduus* Barber); common chinch bug, [*B. leucopterus leucopterus* (Say)]; hairy chinch bug, (*B. l. hirtus* Montandon); and southern chinch bug, (*B. insularis*) (Brandenburg and Villani 1995, Sweet 2000). *Blissus occiduus* is a serious pest of buffalograss in Arizona, California, Colorado, Kansas, Montana, Nebraska, New Mexico, Oklahoma, and parts of Canada (Baxendale et al. 1999, Vittum et al. 1999, Carstens 2003). *Blissus leucopterus leucopterus* has caused severe damage to wheat, spelt, corn, millet, pearl millet, rye, barley, sorghum, grain sorghum, rice, and Sudan grass (Sweet 2000). *Blissus leucopterus leucopterus* can be found in Colorado, the Great Plains, New Mexico, and Nebraska eastward to the Atlantic Ocean (Webster 1907, Leonard 1966, Khuhro 1994). *Blissus leucopterus hirtus* is a major pest of cool-season grasses throughout the Northeast, parts of the Midwest, into the mid-Atlantic states as far south as Virginia, and west to Minnesota (Vittum et al. 1999). *Blissus insularis* occurs in Florida, Alabama, Georgia, Louisiana, Mississippi, North and South Carolina, Texas, California, and Mexico (Henry and Froeschner 1988, Sweet 2000).

Adult *B. insularis* are small and somewhat elongate insects measuring about 3 mm long and 1 mm wide (Leonard 1968). Adult females are slightly larger than males. The head is usually narrower than the posterior margin of the pronotum (Miller 1971). Wings

are white with a distinctive triangular-shaped black marking in the middle of the outer edge of each wing and are folded flat over the back causing the tips to overlap. Populations may consist mostly of long-winged forms (macropterous), short-winged forms (brachypterous), or both (Webster 1907). In Florida, macroptery is greatest during the summer and fall (Cherry 2001a). Young nymphs are as small as 1.0 mm, are reddish-orange with a white band across the dorsal side of the abdomen, and become black in color as they mature. Eggs are laid singly or a few at a time in sheaths, soft soil, or in other protected areas. The eggs are white when first laid but turn bright orange just before hatching. Development from egg to adult requires about 13 wk at 21°C and 5 wk at 28°C (Potter 1998). All life stages are present year-round in most of the state with three to four generations occurring in northern Florida and seven to ten in southern Florida each year (Kerr 1966, Reinert and Kerr 1973).

Blissus insularis move between lawns mainly by walking and large numbers have been observed crawling across sidewalks and driveways bordering heavily infested lawns (Kerr 1966). All life stages are distributed vertically through the turf thatch and into the upper organic layer of the soil. Densities of 500-1,000 *B. insularis*/0.1 m^2 are common and infestations of more than 2,000/0.1 m^2 have been reported (Reinert and Kerr 1973). Light to moderate infestations are extremely aggregated in small areas in the lawn rather than evenly distributed (Cherry 2001b). *Blissus insularis* also attack other lawn grasses including bahiagrass, bermudagrass, centipedegrass, and zoysiagrass, but most of the injury to these has occurred near heavily infested St. Augustinegrass (Kerr 1966). Other hosts include crabgrass, torpedograss, and Pangolagrass (Slater and Baranowski 1990, Brandenburg and Villani 1995).

Management Practices

Chemical Control

Control of *B. insularis* has historically been achieved using insecticides such as tobacco dust, calcium cyanide, nicotine sulfate, DDT, parathion, dieldrin, aldrin, chlordane, chlorpyrifos, and diazinon (Watson and Bratley 1929a and b; Kelsheimer 1952, Wolfenbarger 1953, Kerr 1956, Brogdon and Kerr 1961). Up to six insecticidal applications per year may be needed on Florida lawns to control damaging populations of *B. insularis* (Reinert 1978). With such reliance on chemical use, this insect has developed resistance to organophosphates and organochlorines (Kerr 1958, 1961; Reinert & Niemczyk 1982; Reinert 1982; Reinert and Portier 1983). Currently, pyrethroids are most commonly available to homeowners and turfgrass professionals for controlling *B. insularis* outbreaks. A lack of rotation with other chemical formulations increases the risk of *B. insularis* developing resistance to pyrethroids as well. Some alternatives to chemicals are biological control, host-plant resistance, and cultural control.

Biological Control

Parasites, predators, and pathogens are found in association with *Blissus* spp. The parasitic wasp, *Eumicrosoma benefica* Gahan (Hymenoptera: Scelionidae), attacks the eggs of *B. leucopterus* (Say) in Kansas, reportedly reducing 'broods' of chinch bugs by as much as 50% (McColloch and Yuasa 1914). *Eumicrosoma benefica* also parasitized *B. insularis* eggs in Florida and field tests showed an average abundance of 34.5 wasps/0.1 m^2 in lawns containing *B. insularis* populations of 90.0/0.1 m^2 (Reinert 1972a). Along with *E. benefica*, predatory insects [e.g., *Pagasa pallipes* Stal (Heteroptera: Nabidae), *Xylocoris vicarius* (Reuter) (Heteroptera: Anthocoridae), *Lasiochilus pallidulus* Reuter (Heteroptera: Anthocoridae), *Sinea* spp. (Heteroptera: Reduviidae), *Labidura riparia*

Pallas, *Geocoris bullatus* (Say) and *Geocoris uliginosus* (Say) (Heteroptera: Geocoridae)] were observed feeding on *B. insularis* in Florida (Reinert 1978). The native fire ant, *Solenopsis geminata* (F.), and a spider, *Lycosa* sp., were also observed attacking *B. insularis* (Reinert 1978). However, the red imported fire ant, *Solenopsis invicta* Buren, was unable to suppress *B. insularis* populations (Cherry 2001c). *Beauveria globulifera* (Spegazzini) Picard and *Metarhizium anisopliae* (Metch.) Sorokin were unsuccessful in controlling *B. insularis* in field tests (Kerr 1958). *Beauveria bassiana* (Balsamo) Vuillemin was pathogenic on all life stages of *B. insularis*; however, success appeared limited to high humidity, *B. insularis* populations, and moisture levels (Reinert 1978).

Big-eyed bugs

Big-eyed bugs, *Geocoris* spp., are very abundant and widely distributed (Readio and Sweet 1982). These generalist predators attack a wide range of prey species, including various insect and mite pests of agricultural crops such as cotton, soybean, strawberry, peanut, turfgrass, vegetable, sugarbeet, alfalfa, and tobacco (McGregor and McDonough 1917, Knowlton 1935, York 1944, Champlain and Sholdt 1967a and b, Mead 1972, Crocker and Whitcomb 1980). They specifically have been observed attacking aphids, plant bugs, eggs and larvae of the bollworm [*Heliothis zea* (Boddie)], flea beetles (*Phyllotreta* spp.), false chinch bugs [*Nysius ericae* (Schilling)], southern green stink bugs [*Nezara viridula* (L.)], and green peach aphids [*Myzus persicae* (Sulzer)] (Bell and Whitcomb 1964, Crocker and Whitcomb 1980). *Geocoris* spp. will feed on eggs, immatures, or adults of relatively small or passive prey. They attack by inserting their proboscis into the head, thorax, or abdomen and tend to walk about with the prey suspended on the labium (Sweet 2000).

Although *Geocoris* spp. are predators, they also require water from plant sources (York 1944). In laboratory colonies, *Geocoris* spp. can temporarily be maintained on plant food alone, but prey are required for proper development and fecundity (Stoner 1970). *Geocoris bullatus*, once thought to be a turfgrass pest in Connecticut, feeds on lawn insects such as *B. l. hirtus* (Dunbar 1971). *Geocoris uliginosus* had densities as high as $16.0/0.1 \text{ m}^2$ when high *B. insularis* populations were present and fed on an average of 9.6 ± 3.3 (mean $\pm$ SEM) nymphs in 5 d under laboratory conditions (Reinert 1978). The most common species in Florida are *G. uliginosus*, *G. bullatus*, and *G. punctipes* (Say) (Mead 1972).

Geocoris punctipes

Geocoris punctipes are small oblong Lygaeids having the head broader than long, large prominent eyes that curve backward and overlap the front of the pronotum, and tylus that has a longitudinal groove (Mead 1972). They have relatively short antennae that are slightly enlarged at the tip. *Geocoris punctipes* adults are about 0.5 cm long and are silver-gray in color. Nymphs resemble adults but lack fully developed wings. Cigar-shaped eggs are white to tan in color with a distinctive red spot and are laid singly on leaves and stems of many crops. Eggs hatch in approximately 1 wk. *Geocoris* spp. have five instars, each of which lasts from 4 to 6 d. Adults live for about 1 mo and a female can lay up to 300 eggs during her lifetime. Several generations may occur throughout the season. Adult and immature *G. punctipes* feed by sucking juices from their prey through needle-like mouthparts. They are primarily active in the morning (Dumas et al. 1962). These insects are often confused for *Blissus* spp. *Geocoris* spp. are commonly found throughout the southern United States.

Host Plant Resistance

Host resistant grasses are used to suppress pest damage. There are three types of resistance that can occur: antibiosis, antixenosis, or tolerance. Antibiosis affects the biology of the pest insect and often results in increased mortality, or reduced oviposition and longevity. With antixenosis, the pest shows non-preference for a resistant grass compared to a susceptible one. When a host resistant grass shows tolerance, the grass is able to withstand or recover from damage caused by an insect. Floratam was released as a chinch bug resistant cultivar in 1973 by the University of Florida and Texas A&M. Floratam is the most widely produced and used cultivar of St. Augustinegrass and accounts for 80% of sod production in Florida (Haydu et al. 1998). It has a very coarse leaf texture, poor cold and shade tolerance, is resistant to the St. Augustine decline virus, and successfully minimized *B. insularis* problems for years (Busey 1979, Trenholm et al. 2000). The mode of resistance was antibiosis (Reinert and Dudeck 1974). However, *B. insularis* can now survive and reproduce on this variety (Busey and Center 1987; Busey 1990a and b). Research is being conducted to develop new cultivars of St. Augustinegrass that are resistant to *B. insularis*.

Cultural Control

Cultural practices may influence the susceptibility of St. Augustinegrass to *B. insularis*. Over-fertilizing, over-watering, and improper mowing can cause lawn grasses to develop a thick thatch layer (Trenholm et al. 2000). Thatch is a layer of accumulated dead leaf blades, stolons, and roots between the live plant and soil. Accumulation of thatch often makes the lawn more susceptible to pests (Trenholm et al. 2001), possibly by protecting them from predation and environmental stress. The abundance of *B. l. hirtus* was found to be closely linked to thatch thickness in lawns (Davis and Smitley 1990).

Excessive thatch (exceeding 2.5 cm) may need to be professionally removed by vertical mowing (Trenholm et al. 2000). Heavy fertilization may cause increased growth, thatch, susceptibility to pests such as *B. insularis*, and reduced tolerance to environmental stresses (Kerr 1966, Busey and Snyder 1993, Trenholm et al. 2001). Excess irrigation may lead to problems such as a shallow root system, increased thatch, and increased pest problems (Trenholm et al. 2003). Reduced fertilization and proper irrigation practices may help to reduce *B. insularis* problems (Potter 1998, Sweet 2000). St. Augustinegrass should be mowed to a height of 8 - 10 cm (Trenholm et al. 2000). Proper cultural practices promote healthy grass, which may be able to better tolerate chinch bug damage.

Objectives

An integrated pest management program (IPM) is needed for the southern chinch bug to reduce the turfgrass industry's sole reliance on pesticides. The purpose of this study was to evaluate the effectiveness of different components of an IPM program for *B. insularis* including: 1) sampling techniques, 2) the potential of 'FHSA-115' as a chinch bug resistant variety of St. Augustinegrass, 3) *B. insularis* predation by *G. punctipes*, 4) several homeowner, professional, and experimental insecticides labeled for chinch bug control, and 5) St. Augustinegrass growth response to three levels of irrigation and *B. insularis* density.

CHAPTER 2
SAMPLING METHODS FOR THE SOUTHERN CHINCH BUG, *Blissus insularis*
BARBER (HETEROPTERA: BLISSIDAE)

Introduction

A successful integrated pest management (IPM) program largely depends on the

development of an effective and efficient sampling technique (Hutchins 1994). The

proper use of such techniques provides a means by which to make informed decisions,

such as establishing more accurate economic and/or aesthetic thresholds (Southwood

1978), evaluating product trials, and designing innovative monitoring or management

strategies. However, the researcher must have an understanding of the biology of the

insect population and environmental factors (i.e. temperature, humidity, light), which

may affect activity periods of the insect and critical thresholds (Horn 1988). Effective

sampling also depends on insect distribution and behavior (Törmäla 1982, Standon

2000).

Insects with clumped distributions that live in dense plant material, such as the

southern chinch bug, *Blissus insularis*, may be somewhat difficult to accurately sample

(Cherry 2001b). *Blissus insularis* is one of the most important pests of St.

Augustinegrass (Kerr 1966, Crocker 1993, Cherry and Nagata 1997). Infestations usually

occur in open, sunny, and drought-stressed areas near sidewalks and driveways. Nymphs

and adults live in the thatch and suck fluids from the grass with their needle-like

mouthparts, resulting in yellow to brown patches in the lawn. Newly hatched *B. insularis*

nymphs often wedge themselves in between the grass blades at the nodes to feed (Kerr

1966), making it difficult to recover them without physically pulling the blades back to expose the insects. Techniques used to extract turfgrass insects include flotation, irritant sampling, and vacuuming. Although some studies have examined the effectiveness of various techniques used in sampling turfgrass pests, including chinch bugs (Short & Koehler 1979, Majeau et al. 2000), vacuum extraction was not included. Our objective for this study was to determine the most accurate and efficient method for specifically extracting *B. insularis* from St. Augustinegrass.

Materials and Methods

Blissus insularis were mass-collected from St. Augustinegrass lawns in Gainesville, FL (Alachua Co.), using a modified blower/vacuum (Electrolux Home Products, Augusta, GA) and transported in a mesh-covered bucket to the laboratory. Insects were provided fresh cuttings of Palmetto St. Augustinegrass as needed and maintained in the laboratory <10 d until being aspirated into plastic vials (2.5×10.2 cm) containing a cone-shaped, moistened, 70 mm Whatman filter paper to prevent being damaged from aspiration. Vials were closed using a foam cap to prevent insect escape.

Twenty 15.2 cm diameter clay pots of Palmetto St. Augustinegrass were established in Arrendondo fine sand (loamy, siliceous, hypothermic, Grossarenic Paleudalt) and maintained under ambient conditions in a greenhouse at the University of Florida (Gainesville, FL). Plants were fertilized every 2 wk and watered as needed. The thatch layer was <0.6 cm. Plants were maintained at a 7.6 cm height. Twenty fourth-fifth instars and five adult *B. insularis* were transferred from vials using a camel-hair brush into the center of each pot and caged on 13 May 2003 (Fig. 2-1). Insects were maintained in caged pots for 7 d. Plants were arranged in a complete randomized design

(CRD). After 7 d, four sampling methods (flotation; large, gas - powered vacuum; small, battery operated vacuum; or berlese funnel) were used to extract the insects (n = 5 pots per treatment).

The first method involved floating *B. insularis* from the plant material. *Blissus insularis* were forced from plant debris using a modified version of the flotation technique used in extracting *Solenopsis invicta* colonies from soil (Banks et al. 1981). Potted plants were placed into 19 liter buckets, water was added to each bucket with 1.3 cm diameter PVC tubing, brass connectors, and 7.6 cm long × 0.6 cm diameter rubber tubing (Fig. 2-2 A). Warm water (~38°C) slowly filled up to the edge of each pot, was reduced to a fast drip, and was turned off after about half of the grass blades were submerged. Insects were collected as they crawled up the grass blades, were placed into vials of 80% EtOH, and counted. Pots were submerged for ~2 h.

The second method used was a large, gas-powered blower/vacuum (Electrolux Home Products, Augusta, GA) equipped with a 0.8 m long hose attachment (Fig. 2-2 B). Insects were collected into a 12.7 cm diameter knitting-ring with a chiffon mesh that covered the vacuum attachment's intake hose. Each pot of grass was vacuumed for 1 min, samples were placed into plastic bags, and the number of chinch bugs collected were counted in the laboratory.

A smaller, hand-held, modified Black & Decker Dust-Buster™ vacuum (Bioquip Products, Rancho Dominguez, CA) equipped with a 12.7 cm long × 3.8 cm diameter hose attached to a removable collecting chamber (12.7 cm long × 5.1 cm diameter) was used as the third sampling method (Fig. 2-2 C). The vacuum was powered by a portable, 12-

volt DC battery pack (Bioquip Products, Rancho Dominguez, CA). Each pot was vacuumed as described above.

The last method involved placing infested plants into berlese funnels using 40 wattage bulbs. Potted plants were placed sideways into the berlese funnels and were left in the pots to minimize debris falling into the collection containers (Fig. 2-2 D). It was assumed that any living chinch bugs would fall into the funnel and container of 80% EtOH as the soil and plants dried within 48 h.

The total number of *B. insularis* collected using each method was counted. The mean number of chinch bugs recovered was analyzed using an analysis of variance (ANOVA, $P \leq 0.05$) and treatment means were compared using the Tukey-Kramer HSD multiple comparison test (Jmp®, SAS Institute Inc. 2001).

Results and Discussion

Flotation was the most effective technique for extracting a known number of *B. insularis* from pots of grass (Table 2-1). Different versions of this method are commonly used in *B. insularis* experiments. Under field conditions, a large metal cylinder is placed 3–5 cm into the soil near damaged turfgrass, filled with water, and any chinch bugs present float to the surface within 3–10 min (Kerr 1966; Reinert 1972b, 1982). For laboratory or greenhouse studies, cores or pots of grass are submerged in buckets of water, insects crawl up the grass blades, and are collected (Nagata and Cherry 1999, Richmond and Shetlar 2000). In this experiment, *B. insularis* were forced to escape slowly increasing water levels and could float on the water if they fell off the plant material. Even if completely submerged for considerable periods (≤ 4 h), chinch bugs were expected to survive (Britcher 1903, Janes et al. 1935). However, this method did

not recover all of the insects. Some mortality may have occurred during the experiment
or some insects may have remained in the plant material. Because soil particles were
heavy and fell to the bottom of the bucket, very little debris interfered with observation of
B. insularis movement on the plants or water. Although flotation was somewhat labor-
intensive, it extracted the highest number of *B. insularis* and was the only method where
variability among treatments was low.

The number of *B. insularis* recovered by flotation compared to the large
blower/vacuum was not statistically significant (22.0 and 15.2 respectively, Table 2-1).
Vacuums similar to the one used in this experiment have successfully been used for
collecting chinch bugs for laboratory assays and from field insecticide plots (Crocker and
Simpson 1981, Crocker 1993, Nagata and Cherry 1999). Standen (2000) compared the
effectiveness of pitfall traps and a D-vac suction trap combined with a lightweight swish
net and reported the highest number of Hemipterans being collected in D-vac samples.
Several different vacuums (whereby insects are sucked into a layer of fabric that is
stretched across an intake hose and released into a different container) or D-vac machines
are used to collect insects (Crocker and Simpson 1981, Crocker 1993, Nagata and Cherry
1999, Cherry 2001b). Vacuum extraction of insects may be faster and at least as
effective as the flotation method (Crocker 1993).

The number of *B. insularis* recovered by the large blower/vacuum compared to the
smaller, hand-held vacuum was not significant (Table 2-1). However, the smaller hand-
held vacuum recovered significantly fewer *B. insularis* than the flotation method (10.0
and 3.4 respectively). Additional sampling time may be required for the small, hand-held
vacuum to be effective in recovering known numbers of *B. insularis*. However, for its

convenient size, the hand-held vacuum might work well for rapid detection of chinch bugs in infested lawns. For the berlese funnel method, soil or grass samples may be placed into funnels and as actively moving insects escape the light and heat of low-wattage bulbs, they drop into containers of alcohol (Niemczyk et al. 1992, Heng-Moss et al. 2002). The lowest recovery of *B. insularis* occurred with the berlese funnel (Table 2-1). Additional testing and improved methods (i.e. different bulb wattage, plant arrangement, funnel design) for the berlese funnel technique may improve its effectiveness in recovering *B. insularis*.

As part of developing an IPM program, several of these techniques would be useful under the proper circumstances. The flotation method and both vacuums tested in this study could be used to detect the presence of *B. insularis* in the lawn. However, when conducting experiments using a known number of *B. insularis*, the flotation method or large blower/vacuum may be best to use since these techniques recovered the highest number of insects from infested pots of St. Augustinegrass.

Figure 2-1. St. Augustinegrass grown in 15.2 cm diameter clay pot enclosed in a chiffon
mesh cage.

Figure 2-2. Equipment used to recover *Blissus insularis* from test plants. A. Flotation
method. B. Large blower/vacuum. C. Hand-held vacuum. D. Berlese
funnel.

Table 2-1. Mean (± SEM) number and percentage of *Blissus insularis* recovered from 15.2 cm pots of 'Palmetto' St. Augustinegrass. [1]

Sampling method	Mean (± SEM) number of *B. insularis* recovered [2]	Percentage of *B. insularis* recovered
Flotation	22.0 ± 1.0 c	88.0
Large blower/vacuum	15.2 ± 2.6 bc	61.0
Hand-held vacuum	10.0 ± 2.6 ab	40.0
Berlese funnel	3.40 ± 1.3 a	14.0

[1] 25 *B. insularis* were placed into each pot.
[2] Means ± SEM followed by the same letter were not significantly different ($P \leq 0.05$) by the Tukey-Kramer HSD multiple comparison test. ANOVA statistics: n = 5 reps; F = 15.16; df = 3, 19; $P < 0.0001$.

CHAPTER 3
SOUTHERN CHINCH BUG, *Blissus insularis* (HETEROPTERA: BLISSIDAE),
INTEGRATED PEST MANAGEMENT (IPM)

Introduction

St. Augustinegrass is the most widely used turfgrass in the >6.3 million lawns in
Florida (U.S. Census Bureau 2000). Its primary insect pest is the southern chinch bug,
Blissus insularis, which sucks fluid from the crown and stems of the grass. Control of
this pest has historically been achieved by use of up to six insecticide applications per
year (Reinert 1978, Cherry 2001c). Organophosphates, such as chlorpyrifos and
diazinon, were routinely used to control and prevent outbreaks, but are no longer
available because of the Food Quality Protection Act. The primary insecticides
remaining for urban turfgrass use are pyrethroids. Given the history of *B. insularis*
resistance to organophosphates (Kerr 1958, 1961; Reinert and Niemczyk 1982; Reinert
1982; Reinert and Portier 1983), eventual resistance to pyrethroids is possible with
repeated use without rotation with another pesticide class. Other problems from overuse
or misuse of pesticides include drift, run-off, ground water contamination, and non-target
effects. An integrated pest management program (IPM) is needed for this pest.

Biological control of turfgrass pests has been underutilized in the United States,
but both parasitism and predation of *B. insularis* have been observed (Beyers 1924,
Wilson 1929, Kerr 1966). *Eumicrosoma benefica* are chinch bug egg parasitoids in
Florida (Reinert 1972a). Reinert (1978) also observed *Geocoris* spp. (Heteroptera:
Lygaeidae) feeding on *B. insularis*. *Geocoris* spp. are generalist predators that occur in

grass systems, and feed on other chinch bug species, such as the buffalograss chinch bug,

Blissus occiduus (Heng-Moss et al. 1998, Carstens 2003). However, little is known about

the effectiveness of these biological control agents in suppressing *B. insularis*

populations.

Host plant resistance has been an effective tool against *B. insularis*, but new

resistant cultivars are needed. Floratam, a cultivar of St. Augustinegrass, was released in

1973 by the University of Florida and Texas A&M (Busey 1979, Trenholm et al. 2000).

It successfully minimized *B. insularis* problems for years and is still the most widely

produced cultivar of St. Augustinegrass (Haydu et al. 1998). The mode of resistance was

antibiosis (Reinert and Dudeck 1974). However, *B. insularis* can now survive and

reproduce on this variety (Busey and Center 1987; Busey 1990a and b).

The objective of this study was to examine several components of an IPM

program including host plant resistance, predation by *G. punctipes*, and the efficacy of

several professional and non-professional (homeowner) insecticides.

Materials and Methods

Insect Collection and Colony Maintenance

Blissus insularis used in the following experiments were collected from St.

Augustinegrass lawns in Gainesville, Alachua Co., FL., using a modified Weed Eater

Barracuda blower/vacuum (Electrolux Home Products, Augusta, GA) and transported in

a mesh-covered bucket to the laboratory. Insects were provided fresh cuttings of

Palmetto St. Augustinegrass as needed and maintained in the laboratory 7-14 d, until

being aspirated into plastic vials (2.5×10.2 cm) containing a cone-shaped, moistened, 70

mm Whatman filter paper, and a foam cap.

Host Plant Resistance

Blissus insularis survival on an experimental variety (FHSA-115) and susceptible

cultivar of St. Augustinegrass (Floratine) was tested in a laboratory assay. Terminal

sprigs of FHSA-115 and Floratine St. Augustinegrass were cut from established plots at

the G.C. Horn Memorial Turfgrass Field Laboratory at the University of Florida

(Gainesville, FL), transported on ice to the laboratory, and refrigerated (<12 h). All

sprigs were between 5.0-6.4 cm in length, with three leaflets and one node.

Lab assay. The arena consisted of a cone-shaped, moistened, 70 mm Whatman

filter paper at the bottom of a plastic vial (2.5×10.2 cm), one grass sprig (either FHSA-

115 or Floratine), and a foam cap to prevent insects from escaping. Ten *B. insularis*

(either first-second instar, third-fourth instar, fifth instar, or adult) were transferred into

each vial using a camel-hair brush. Certain instars were grouped because of the difficulty

separating and distinguishing between life stages. One uninfested sprig of each variety

served as a control to ensure that grasses did not die from factors other than *B. insularis*

feeding. Sprigs were replaced every 7 d. There were 10 replicates in a complete

randomized design (CRD). All vials were kept at 80°F (± 2°) and a photoperiod of 11:13

(L:D). The number of dead *B. insularis* were counted daily for 3 wk.

An analysis of variance (ANOVA) was conducted to determine *B. insularis*

mortality differences between grasses. Treatment means were analyzed using Tukey's

Studentized Range (HSD) test (SAS Institute Inc. 2000).

Predation Assay

Geocoris punctipes were acquired from a laboratory colony maintained by

Entomos, LLC. (Gainesville, FL), and immediately separated to avoid cannibalism.

Individuals were transferred with a camel-hair brush to petri dishes containing four

Entomos food beads, a piece of moistened cotton ball, and one-fourth of an 11 cm

Whatman filter paper. After 24 h of feeding on the food beads, *G. punctipes* were starved

24 h before bioassay, but were provided moistened cotton balls. *Blissus insularis* were

collected as previously described.

   Bioassay. A cone-shaped, moistened, 70 mm Whatman filter paper was placed at

the bottom of each plastic vial (2.5 × 10.2 cm) and one Palmetto St. Augustinegrass sprig

was added to the vial. Sprigs of Palmetto St. Augustinegrass were cut from established

pots maintained in the Landscape Entomology greenhouse at the University of Florida

(Gainesville, FL) and refrigerated until use (<12 h). All sprigs were between 5.0-6.4 cm

in length, with three leaflets and one node. Twenty *B. insularis* from three different age

groups (either first-third instar, fourth-fifth instar, or adult) were transferred into each vial

using a camel-hair brush. Certain instars were grouped because of the difficulty

separating and distinguishing between life stages. Insects were allowed to acclimate for

24 h before the bioassay.

   Either one adult or one fifth-instar *G. punctipes* was transferred into each vial using

a camel-hair paintbrush. Control vials containing first–third *B. insularis* and no *G.*

punctipes were set up to ensure *B. insularis* survival. Vials were kept in the laboratory at

80°F and 12:12 h (L:D). There were ten replicates in a CRD. After 24 h, *G. punctipes*

were removed from vials. The number of *B. insularis* live, injured, or dead were counted,

and percentage mortality were determined.

   Percent mortality was square root arcsine transformed. An analysis of variance

(ANOVA) was conducted to determine if mean mortality of a particular age group was

greater when compared to others. Treatment means were analyzed using Tukey's

Studentized Range (HSD) test (SAS Institute, Inc. 2000).

Insecticidal Control

Lab assay. Different concentrations of an experimental biorational product

(Bioblitz, Jentree Canada, INC.) and an industry standard (diazinon, Spectracide Group,

Division of United Industries Corporation) were tested in the laboratory. All treatments

were applied on 27 Nov 2002 and replicated ten times in a CRD. Floratine St.

Augustinegrass sprigs were cut from established plots at the G.C. Horn Memorial

Turfgrass Field Laboratory in Gainesville, FL, and dipped in 1, 10, or 100 ppm Bioblitz

or diazinon. Control sprigs were dipped in water. For the bioassay, a cone-shaped,

moistened, 70 mm Whatman filter paper was placed at the bottom of each plastic vial

$(2.5 \times 10.2$ cm), and one dry, treated sprig was added to the vial. All sprigs were

between 5.0-6.4 cm in length, with three leaflets and one node. *Blissus insularis* were

collected and maintained as previously described. Ten healthy *B. insularis* adults were

transferred into each vial using a camel-hair brush. All vials were kept at 80°F ($\pm 2°$) and

a photoperiod of 12:12 (L:D). The number of live *B. insularis* were counted 2 and 4 d

after treatment (DAT).

Percent mortality was square root arcsine transformed. An analysis of variance

(ANOVA) was conducted to determine *B. insularis* mortality differences between

treatments. Treatment means were analyzed using Tukey's Studentized Range (HSD)

test (SAS Institute, Inc. 2000).

Field tests. The efficacy of several professional and non-professional (homeowner)

insecticides were field-tested against *B. insularis*. Two schedule 80 PVC rings (15 cm

diameter × ca. 20 cm high) were placed 10 cm into the greenest sections of established 1 m^2 Floralawn St. Augustinegrass plots at the G.C. Horn Memorial Turfgrass Field Laboratory at the University of Florida, Gainesville, FL (Fig. 3-1 A). Grass was maintained at a height of 7.6 cm and vacuumed to ensure that no *B. insularis* were present before ring establishment. Twenty fourth-fifth instar and ten adult *B. insularis* were placed in the center of each ring and allowed to acclimate for 24 h. Rings were covered with chiffon mesh to prevent insect escape and predation during the experiment.

Homeowner products were applied in June 2003 [n = 4, CRD] and professional products were applied in September 2003 (n = 5, CRD design). Homeowner products included bifenthrin (Scotts MaxGuard® Insect Protection with Turf Builder Fertilizer 24-3-10, The Scotts Company, Marysville, OH), carbaryl (GardenTech Sevin® Concentrate Bug Killer, GardenTech™, Lexington, KY), cyfluthrin (Bayer Advanced Lawn® and Garden Multi-Insect Killer Ready to Use Spray, Bayer Advanced LLC, Birmingham, AL), deltamethrin (Southern® Ag Mole Cricket and Chinch Bug Lawn Insect Control, Southern Agricultural Insecticides, Inc., Palmetto, FL), λ-cyhalothrin (Spectracide® Triazicide™ Soil & Turf Insect Killer Granules, Realex Corporation, Spectrum Brands, St. Louis, MO), and permethrin (Real Kill Multi-Purpose Insect Killer Concentrate®, Realex Corporation, Spectrum Brands, St. Louis, MO). Professional products included bifenthrin (Talstar® F, FMC Corp., Philadelphia, PA), carbaryl (Sevin® SL, Bayer Environmental Science, Montvale, NJ), cyfluthrin (Tempo® SC Ultra, Bayer Environmental Science, Montvale, NJ), cypermethrin (Demon® TC, Syngenta Crop Protection, Inc., Greensboro, NC), deltamethrin (Deltagard® T & O, Bayer Environmental Science, Montvale, NJ), λ-cyhalothrin (Scimitar® CS, Syngenta Crop

Protection, Inc., Greensboro, NC), and permethrin (Astro®, FMC Corp., Philadelphia,

PA). Control plots were untreated. All treatments were applied at the label rate. An

80% active non-ionic surfactant (Amway APSA® 80, Amway Phils., L.L.C.) was added

to Scimitar® CS and Demon® TC according to label recommendation. Liquid

formulations were sprayed uniformly onto the grass using a hand-held spray bottle and

granular insecticides were applied by hand and watered according to label

recommendations. During the course of the experiment, irrigation was applied every

morning in amounts corresponding to average monthly evapotranspiration (ET) rates for

Florida. June treatments received 0.42 cm and September treatments received 0.31 cm.

Soil cores were removed at 1 and 4 wk post-treatment and surviving insects were

removed by flotation (Fig. 3-2 B).

Percent mortality was square root arcsine transformed. An analysis of variance

(ANOVA) was conducted to determine *B. insularis* mortality differences between

treatments. Treatment means were analyzed using Tukey's Studentized Range (HSD)

test (SAS Institute, Inc. 2000).

Results and Discussion

More than 62% of the *B. insularis* successfully survived on both Floratine and

FHSA-115 during the 3 wk experiment with feeding, mating, and molting observed. The

different life stages of *B. insularis* had similar mortality after feeding on either variety

(Table 3-1), suggesting that neither variety was resistant to *B. insularis*.

Fifth instar and adult *G. punctipes* consumed a similar number of *B. insularis* ($F =$

0.15; df $= 1, 78$; $P = 0.7$). When pooled together, fifth instar and adult *G. punctipes*

consumed significantly more first-third instar *B. insularis* compared with the other chinch

bug life stages. These results are similar to reports of *G. uliginosus* feeding on *B. occiduus*, where feeding was highest in first–fourth instars (Carstens 2003). After 24 h, the mean percentage (± SEM) of first–third instar, fourth–fifth instar, or adult *B. insularis* killed by *G. punctipes* were 57.0 ± 0.4, 17.2 ± 0.3, and 16.0 ± 0.4, respectively (n = 20; F = 37.89; df = 2, 57; $P < 0.0001$). A small percentage (5.3%) of *B. insularis* died in the controls, possibly from handling.

In the laboratory insecticide assay, diazinon (100 ppm) killed significantly more *B. insularis* adults on sprigs than any other diazinon or Bioblitz treatment 2 and 4 DAT (Table 3-2). Diazinon at the 1 and 10 ppm rates and Bioblitz at the 100 ppm rate were also statistically different from the untreated control at 4 DAT (Table 3-2). However, *B. insularis* mortality was likely too low for industry standards. Some mortality occurred in the untreated control, possibly from handling. No phytotoxicity was observed.

Homeowner products containing bifenthrin, carbaryl, deltamethrin, and λ-cyhalothrin achieved over 80% control 1 wk post-treatment in the field test (Table 3-3). Carbaryl killed significantly more *B. insularis* than any other treatment 4 wk after treatment. Although plots treated with bifenthrin, deltamethrin, and permethrin had less than 10% *B. insularis* survival, these treatments did not differ from the control (Table 3-2). All of the professional products were statistically different from the control 1 wk after treatment, although carbaryl was the only treatment to kill all *B. insularis* at 1 wk (Table 3-4). Though most product labels recommended watering in after application, the label for carbaryl did not. The effectiveness of carbaryl may be reduced by irrigation and/or rainfall within 24 h of application. Plots treated with bifenthrin had few live *B. insularis* at 1 wk and complete mortality at 4 wk post-treatment.

The results from this research provide groundwork for the development of an IPM program for *B. insularis*. An IPM program utilizing biological, host plant resistance, and cultural control could help reduce the amount of insecticides used to control *B. insularis*.

Table 3-1. Mean percent mortality of *Blissus insularis* (± SEM) after feeding on
Floratine or FHSA - 115 St. Augustinegrass.

	% Mortality ± SEM [1]	
Life Stage	Floratine	FHSA - 115
1st-2nd instar	15.4 ± 7.3*	23.4 ± 4.7*
3rd-4th instar	23.0 ± 4.7*	37.7 ± 8.1*
5th instar	21.0 ± 4.1*	32.0 ± 4.2*
Adult	23.0 ± 5.8*	32.3 ± 5.1*

[1] Percent ± SEM followed by * are not significantly different ($P \leq 0.05$) by ANOVA (n = 20 *B. insularis* of each respective age per vial; F = 2.02; df = 7, 72; P = 0.06).

Table 3-2. Mean percent mortality of *Blissus insularis* (± SEM) killed by different rates of Bioblitz and diazinon at 2 and 4 days after treatment.

Treatment	Rate	% Mortality [1]	
		2 DAT	4 DAT
Untreated control	----	12.0 ± 0.7	23.0 ± 0.6
Bioblitz	1 ppm	23.0 ± 0.7	33.0 ± 0.3
	10 ppm	19.0 ± 0.9	34.0 ± 0.4
	100 ppm	18.0 ± 0.7	38.0 ± 0.2 *
Diazinon	1 ppm	14.0 ± 0.8	40.0 ± 0.3 *
	10 ppm	16.0 ± 0.8	54.0 ± 0.3 *
	100 ppm	82.0 ± 0.5 *	97.0 ± 0.08 *

[1] Percent ± SEM followed by * are significantly different ($P \leq 0.05$) using Tukey's Studentized Range (HSD) test (n = 10 *B. insularis* of each respective age per vial; 2 DAT: $F = 8.28$; df = 6, 63; $P < 0.0001$; 4DAT: $F = 23.0$; df = 6, 63; $P < 0.0001$).

Table 3-3. Mean percent survival of *Blissus insularis* (± SEM) at 1 and 4 wk post
treatment.

Treatment	Rate	1 wk	4 wk
Control	----	76.7 ± 3.6	18.0 ± 8.3
Bifenthrin	152.5 kg/ha	18.0 ± 8.3* [1]	6.7 ± 4.1
Carbaryl	38.2 L/ha	3.3 ± 1.4*	0*
Cyfluthrin	19.1 L/ha	55.9 ± 12.6	20.8 ± 5.7
Deltamethrin	146.4 kg/ha	2.5 ± 1.6*	5.0 ± 1.7
λ-cyhalothrin	146.4 kg/ha	9.0 ± 2.1*	21.6 ± 4.4
Permethrin	19.1 L/ha	39.0 ± 10.4	8.3 ± 5.0

[1] Percent ± SEM followed by * are significantly different ($P \leq 0.05$) using Tukey's
Studentized Range (HSD) test (n = 4 reps. 1 wk: $F = 13.23$; df = 6, 21; $P < 0.0001$; 4
wk: $F = 4.30$; df = 6, 21; $P = 0.006$).

Table 3-4. Mean percent survival of *Blissus insularis* (± SEM) at 1 and 4 wk post treatment.

Treatment	Rate	1 wk	4 wk
Control	---	76.0 ± 5.5	26.6 ±11.4
Bifenthrin	1.6 L/ha	1.0 ± 0.7 * [1]	0 *
Carbaryl	18.78 L/ha	0 *	0 *
Cypermethrin	2.1 L/ha	1.0 ± 0.7 *	0.7 ± 1.0 *
Cyfluthrin	0.86 L/ha	34.0 ± 11.2 *	9.3 ± 4.0
Deltamethrin	146 kg/ha	3.0 ± 2.0 *	1.3 ± 1.0 *
λ-cyhalothrin	1.5 L/ha	14.0 ± 3.3 *	4.5 ± 3.2 *
Permethrin	2.5 L/ha	7.3 ± 4.4 *	2.7 ± 2.7 *

[1] Percent ± SEM followed by * are significantly different ($P \le 0.05$) using Tukey's Studentized Range (HSD) test (n = 5 reps. 1 wk: $F = 26.28$; df = 7, 32; $P < 0.0001$; 4 wk: F = 6.37; df = 7, 32; $P < 0.001$).

Figure 3-1. Methods used in pesticide field trials. A. PVC rings used in pesticide field trials. B. Flotation of St. Augustinegrass post-treatment.

CHAPTER 4
ST. AUGUSTINEGRASS GROWTH RESPONSE TO THREE LEVELS OF IRRIGATION AND *Blissus insularis* DENSITY

Introduction

Blissus insularis, an important pest of St. Augustinegrass, has been reported to cause more damage and be more abundant in sunny, open, drought-stressed areas of lawns (Kuitert and Nutter 1952, Reinert and Kerr 1973). Particularly susceptible areas include turf near sidewalks, pavements, or in poorly irrigated areas. The nymphs and adults live in the thatch and suck fluids from the crown and stem of grasses. This feeding results in brown, dead patches of turf that are aesthetically displeasing and allow weed encroachment. Because *B. insularis* populations are aggregated with up to 2,000/0.1 m^2 (Reinert and Kerr 1973), turfgrass managers attempt to prevent outbreaks with frequent insecticide applications. Currently, 20-25 *B. insularis*/0.1 m^2 is considered enough to warrant a treatment (Short et al. 1982).

Moisture has been reported to have a "marked but paradoxical" effect on *B. insularis* (Kerr 1966). Adequately irrigated turf may be attractive to *B. insularis* or easier to feed on. But rapidly growing grass may withstand the effects of feeding, and excess moisture may actually suppress populations (Braman 1995). However, little is known about the response of St. Augustinegrass to *B. insularis* feeding and the interaction with irrigation. The objective of this research was to quantify St. Augustinegrass growth response to three levels each of irrigation and *B. insularis* densities.

Materials and Methods

Insect Collection and Maintenance

Blissus insularis were collected from St. Augustinegrass lawns in Ocala, FL

(Marion Co.), using a modified Weed Eater Barracuda blower/vacuum (Electrolux Home

Products, Augusta, GA) and transported in mesh-covered buckets containing fresh

cuttings of Palmetto St. Augustinegrass to the laboratory. Grass was replaced as needed

and buckets were maintained at 80°F and 13:11 (L:D) for <2 wk. Just before bioassay, *B.*

insularis were aspirated into plastic vials (2.5×10.2 cm) containing a cone-shaped,

moistened, 70 mm Whatman filter paper and a foam cap.

Bioassay. Forty-five sewer polyvinyl chloride (PVC) pipes (15.2 cm diameter $\times$

43.2 cm long) fitted with sewer caps were filled with 7.6 cm of river rock, a piece of #4

Whatman filter paper, and Arrendondo fine sand up to 1.3 cm from the top (Fig. 4-1).

Holes were drilled into the sewer caps to allow drainage and the filter paper layer

prevented root and soil migration into the rock layer. Soil was allowed to settle for 24 h.

Forty-five 15.2 cm diameter plugs of Palmetto St. Augustinegrass were transplanted from

pots onto the top of the lysimeters on 14 Aug 2003 and lysimeters were placed on

reinforced metal platforms in a climate-controlled greenhouse at the G. C. Horn

Memorial Turfgrass Field Laboratory at the University of Florida in Gainesville, FL.

Daytime and nighttime temperatures were 27°C and 24°C, respectively. Plants were

fertilized with 16-4-8 water soluble complete N source NH_4NO_3 at 0.5 lb N/1000 ft^2 and

allowed to establish for 1 mo before bioassay.

Treatments included irrigation [low (30%), medium (60%), or high (100%)

saturation] and either 0, 30, or 200 fourth-fifth instar *B. insularis*. There were five

replicates in a CRD. A mesh cage was placed 5.0 cm into the soil and around the plants, such that the grass blades could be maintained at 7.6 cm height. Lysimeters were completely saturated with 1,500 ml of water and allowed to drain for 24 h. After 24 h, insects were transferred from vials using a camel-hair brush and cages were closed with nylon. Each lysimeter was weighed to determine its initial saturation.

Amounts of water to apply at respective irrigation levels were determined by replacing some fraction of the water used in evapotranspiration (ET). This was determined gravimetrically by the following:

$$ET = W_{max} - W_{min}$$

$$W_{needed} = \text{deficit irrigation level} \times ET_{control}$$

$$W_{max} = W_{min} + W_{needed}$$

W_{min} and W_{max} were the lysimeter weights before and after water was applied. W_{needed} represented the water amount applied to lysimeters. $ET_{control}$ represented the mean of 100% irrigation levels of ET. ET rates were measured and plants were irrigated weekly.

After 2 mo, *B. insularis* were removed from lysimeters using a small, hand-held Black & Decker Dust-Buster™ vacuum (Bioquip Products, Rancho Dominguez, CA) equipped with a 12.7 cm long $\times$ 3.8 cm diameter hose attached to a removable collecting chamber (12.7 cm long $\times$ 5.1 cm diameter). The vacuum was powered by a portable 12-volt DC battery pack (Bioquip Products, Rancho Dominguez, CA). Each lysimeter was vacuumed for 2 min, samples were placed into plastic bags, and the number of *B. insularis* collected were counted in the laboratory.

To collect root data, lysimeters were emptied onto a screen table and the roots were removed just below the crown. Crown contents were placed into buckets, covered

with mesh, and brought to the laboratory. Remaining *B. insularis* in the crown material were removed by flotation and counted. Soil was washed off roots, and roots were transported to the laboratory in paper bags. Roots were rinsed again in the laboratory using a #20 standard testing sieve (Fisher Scientific Co.) and non-root debris were removed. Wet and dry weights of roots were recorded. For initial dry weights, roots were dried for 48 h in a Blue M Stabil-Therm mechanical convection oven with Pro-Set II control at 55°C. The dry ash procedure was done to obtain the percent organic matter of the roots. Dry roots were placed in 150 ml Erlenmeyer beakers and baked at 450°C in a Fisher Scientific Isotemp Muffle furnace (model #550-58). Dry ash weights were subtracted from initial dry weights to obtain the percent organic matter of roots.

Grass blades were cut weekly to a 7.6 cm height, and clippings were collected and transported to the laboratory in paper bags in a cooler to obtain fresh and dry weights. For dry weights, grass blades were dried for 24 h in a Blue M Stabil-Therm mechanical convection oven with Pro-Set II control at 55°C.

An analysis of variance (ANOVA) was conducted to evaluate differences between treatment means in root weights and weekly grass clippings. Data were analyzed as a two-way factorial complete randomized design with irrigation and *B. insularis* as main factors using the Student-Newman-Keuls test (SAS Institute, Inc. 2000).

Results and Discussion

Stunted growth from *B. insularis* feeding was observed from grass clippings collected after 1 wk (Table 4-1). Dry weights of grass clippings collected from the different *B. insularis* densities were different from one another except during weeks two, six, and seven (Table 4-1). During weeks two, six, and seven, the dry weight of grass

clippings recovered from treatments containing 200 *B. insularis* were lower than dry weights of grass clippings collected from treatments containing 0 or 30 *B. insularis*. *Blissus insularis* feeding from densities of 30 and 200 lowered root weight as shown in Table 4-2. Research on other *Blissus* spp. reports their feeding can cause wilting, chlorosis, stunting, and death through clogging of vascular transport systems (Painter 1928, Negron and Riley 1990, Spike et al. 1991). Beyer (1924) observed that large numbers of *B. insularis* created a 'dwarfed condition' of St. Augustinegrass, eventually leading to plant death.

The dry weights of grass clippings collected from treatments with different irrigation levels were different during weeks five and seven. At week five, the dry weight of grass clippings collected from treatments with low irrigation were lower than the dry weights of grass clippings collected from treatments containing medium or high irrigation (Table 4-1). At week seven, dry weights of grass clippings collected from treatments with low irrigation were lower than clippings from treatments with high irrigation levels. Irrigation did not affect root weight (Table 4-2). The interaction between irrigation and *B. insularis* was not significant for grass clipping ($F = 0.75$; df = 4, 39; $P = 0.57$) or root weight data ($F = 0.54$; df = 4, 39; $P = 0.71$). It is possible that *B. insularis* populations are found first in drought-stressed areas in the lawn because of an increased chance of survival. Heavy rainfall and irrigation have been observed to drown early instar *Blissus* spp. (Luginbill 1922, Beyer 1924, Wilson 1929, Kuitert and Nutter 1952). Also, environmental factors such as temperature and light may play a role in where *B. insularis* outbreaks occur first in the lawn.
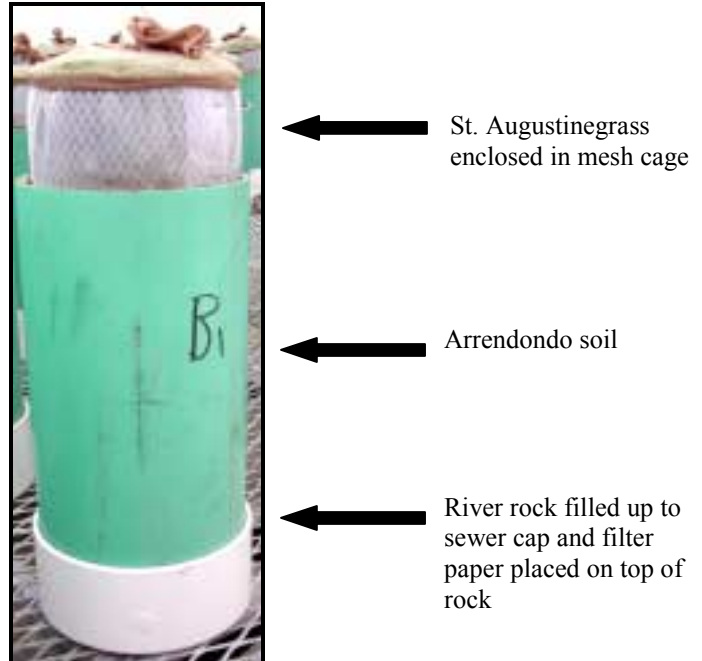
Figure 4-1. Lysimeter used in experiment.

Table 4-1. Mean dry weight (mg) (± SEM) of grass clippings by week. [1]

Main Effect	Level	1 wk	2 wk	3 wk	4 wk	5 wk	6 wk	7 wk	8 wk
B. insularis	0	400.0 ± 20.0^a	320.0 ± 20.0^a	270.0 ± 60.0^a	180.0 ± 30.0^a	170.0 ± 30.0^a	130.0 ± 20.0^a	110.0 ± 20.0^a	110.0 ± 20.0^a
	30	330.0 ± 20.0^b	280.0 ± 20.0^a	190.0 ± 60.0^b	140.0 ± 40.0^b	140.0 ± 50.0^b	110.0 ± 30.0^a	100.0 ± 20.0^a	80.0 ± 20.0^b
	200	230.0 ± 20.0^c	140.0 ± 10.0^b	60.0 ± 10.0^c	40.0 ± 3.00^c	20.0 ± 4.00^c	30.0 ± 6.00^b	20.0 ± 3.00^b	20.0 ± 4.00^c
F-value		17.98	34.78	59.14	37.89	72.86	38.25	30.61	26.03
P-value		< 0.0001	< 0.0001	< 0.0001	< 0.0001	<0.0001	<0.0001	<0.0001	<0.0001
Irrigation	low	330.0 ± 30.0^a	240.0 ± 20.0^a	160.0 ± 10.0^a	110.0 ± 0^a	90.0 ± 1.00^b	70.0 ± 0^a	60.0 ± 0.30^b	60.0 ± 2.00^a
	med	310.0 ± 20.0^a	250.0 ± 30.0^a	180.0 ± 10.0^a	120.0 ± 5.00^a	120.0 ± 10.0^a	100.0 ± 3.00^a	80.0 ± 3.00^{ba}	70.0 ± 10.0^a
	high	320.0 ± 20.0^a	250.0 ± 20.0^a	190.0 ± 10.0^a	130.0 ± 4.00^a	130.0 ± 4.00^a	100.0 ± 6.00^a	90.0 ± 3.00^a	70.0 ± 4.00^a
F-value		0.39	0.12	2.12	1.55	6.88	2.90	3.18	26.0
P-value		0.68	0.88	0.13	0.23	0.003	< 0.07	< 0.05	0.10

[1] Means ± SEM within a column followed by the same letter are not significantly different (P ≤ 0.05) by the Student-Newman-Kuels test.

Table 4-2. Mean root weight (± SEM) of St. Augustinegrass after 8 wk.

Treatment	Level	Mean root wt (mg)
B. insularis	0	2100.0 ± 140.0
	30	1600.0 ± 170.0 * [1]
	200	1300.0 ± 130.0 *
Irrigation	low	1700.0 ± 200.0
	med	1400.0 ± 130.0
	high	1800.0 ± 150.0

[1] Mean root weight ± SEM followed by * are significantly different ($P \leq 0.05$) using Student-Newman-Kuels test. (*B. insularis*: $F = 7.33$; df = 2, 42; $P = 0.002$. Irrigation: $F = 1.81$; df = 2, 42; $P = 0.18$).

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BIOGRAPHICAL SKETCH

Julie Cara Congdon was born on October 25, 1970, in St. Petersburg, Florida. Spending most of her childhood in Gainesville, she moved to Elma, Washington, and attended Elma High School. After enjoying 10 years in Washington, she returned to Gainesville to pursue a college degree. She enrolled at Santa Fe Community College and after taking several honors courses developed an interest in entomology. In 1998, Cara entered the University of Florida as an undergraduate entomology major. While at the University of Florida, Cara gained practical experience in both pest control and research by working for the Florida Pest Control and Chemical Company, the University of Florida's Entomology and Nematology Department (urban entomology laboratory), United States Department of Agriculture (USDA), and FMC Corporation. She is a research associate for the Division of Plant Industry and is a member of the Entomological Society of America, Florida Entomological Society, Florida Turfgrass Association, Certified Pest Control Operators of Florida, Entomology and Nematology Student Organization (ENSO) and the Urban Entomological Society (UES). Cara plans to pursue a Ph.D. in entomology at the University of Florida.