Management of Palmer Amaranth in Glufosinate-Resistant Cotton and Cogongrass Eradication in the Southern United States

By

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Abstract

The Southeastern US is struggling with cogongrass (*Imperata cylindrica*) and glyphosate-resistant Palmer amaranth (*Amaranthus palmeri*)–two of the most difficult-to-manage weeds of non-crop and row-crop areas, respectively. We evaluated herbicide treatments at spring, summer, and fall application timings for cogongrass patch eradication. Herbicide treatments included glyphosate at 4.5 kg ai ha^{-1}, imazapyr at 0.84 kg ai ha^{-1} and a glyphosate plus imazapyr tank mixture at the same rates. Cogongrass response to treatments varied by location but by 36 months after initial treatment (MAIT), cogongrass rhizome eradication was achieved with: 1) glyphosate + imazapyr treatment at any application timing; 2) imazapyr treatment in August or October; and 3) glyphosate treatment applied in May and October each year. An additional field study evaluated the growth dynamics of six cogongrass ecotypes (‘Auburn’, ‘Mobile’, ‘Florida’, ‘Louisiana’, ‘Mississippi’ and ‘Red Baron’) and their sensitivity to glyphosate across a historic fertility gradient. There was significant variation among ecotypes for tiller number, spread diameter, shoot and rhizome biomass, rhizome depth, and total nonstructural carbohydrates levels. Glyphosate (3.36 kg ae ha^{-1}) completely controlled aboveground growth, but all ecotypes grew back by 12 months after glyphosate treatment. A dose-response relationship for different ecotypes indicated ‘Florida’, ‘Louisiana’, and ‘Mississippi’ as most sensitive; ‘Auburn’ and ‘Mobile’ as moderately sensitive; and ‘Red Baron’ as least sensitive to glyphosate.

For Palmer amaranth management, two field studies were conducted to evaluate: 1) the role of soil-inversion, cover crops and herbicide regimes for Palmer amaranth management in glufosinate-resistant cotton; 2) the role of soil-inversion, cover crops and secondary tillage methods for Palmer amaranth management in glufosinate-resistant cotton. In both studies the main plots were two soil-inversion treatments: fall inversion tillage (IT) and non-inversion tillage (NIT). The subplots were three cover crop treatments: crimson clover, cereal rye and winter fallow. In the first study, the sub-subplots were four herbicide regimes: PRE (single preemergence application of pendimethalin at 0.84 kg ae ha^{-1} plus fomesafen at 0.28 kg ai ha^{-1}) alone, POST (single postemergence application of glufosinate at 0.60 kg ai ha^{-1} plus *S*-metolachlor at 0.54 kg ai ha^{-1}) alone, PRE + POST (combination of prior two components) and a no-herbicide check. In the second study the sub-subplots were four spring tillage methods: disk followed by chisel plow (DCH), disk followed by field cultivator (DCU), disk followed by disk (DD), and a no-tillage check (NT). One or two POST blanket applications were made two and four weeks after planting depending on the production year. Results from the first study indicated > 96 % reduction in Palmer amaranth density two and six weeks after cotton planting by PRE and PRE + POST herbicide regimes in both IT and NIT over the three years. Furthermore, the PRE, POST and PRE + POST produced three times more cotton than the no-herbicide check. In the second study, Palmer amaranth was controlled > 90% in DD regardless of soil inversion and cover crop. Furthermore, the DD tillage method produced maximum cotton (2251 kg ha^{-1}) regardless of cover crop. Additionally, the IT (2133 kg ha^{-1}) produced 21% more cotton than NIT (1766 kg ha^{-1}).

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Introduction and Literature Review-Part I

1.1. Palmer Amaranth Management in Glufosinate-Resistant Cotton

1.1.1. Introduction

Palmer amaranth [*Amaranthus palmeri* (L.) S.Wats, family *Amaranthaceae*] is a highly invasive and aggressive weed in southeastern, Mississippi delta, and Great Plains row crop production areas (Fernald 1950). In addition, several species of the genus *Amaranthus* occur throughout these regions (Horak et al. 1994; McGregor 1986). Compared to other pigweed species such as common waterhemp [*Amaranthus rudis* (L.) Sauer], redroot pigweed (*Amaranthus retroflexus* L.), and tumble pigweed (*Amaranthus albus* L.), Palmer amaranth produced the maximum dry weight and leaf area and height (Horak and Loughin 2000). It has C4 photosynthetic mechanism and hence has high photosynthetic efficiency (Ehleringer 1983). Palmer amaranth is a dioecious plant with tremendous seed production potential and rapid seed germination (Horak and Loughin 2000; Steckel et al. 2004). A single female plant can produce more than 600,000 seeds, depending upon density and other environmental factors that have an average diameter of 1.0 mm (Keeley et al. 1987). Additionally, Palmer amaranth has exceptional drought tolerance and can endure dry conditions very well (Ehleringer 1983; Place et al. 2008; Wright et al. 1999). Palmer amaranth can perform very well under low light conditions such as dense crop canopies (Jha et al. 2008). Palmer amaranth is a highly problematic plant-pest

interfering with the economical production of several crops such as cotton, corn, cucurbits, grain sorghum, peanut, potato, soybean, sweet potato and several vegetable crops (Bensch et al. 2003; Burke et al. 2007; Klingaman and Oliver 1994; Massinga et al. 2001; Menges 1987; Meyers et al. 2010; Morgan et al. 2001; Monks and Oliver 1988; Moore et al. 2004; Rowland et al. 1999; Smith et al. 2000; Webster 2005). Several researchers have reported Palmer amaranth to be one of the most competitive weeds in cotton (*Gosypium hirsutum* L.) (Askew and Wilcut 2002; Bensch et al. 2003; Rowland et al. 1999; Smith et al. 2000). Morgan et al. (2001), reported a linear decrease in cotton yields from 13 to 54% for 1 to 10 Palmer amaranth plants/9.1 m of row. In Oklahoma, Rowland et al. (1999) documented 5.9 to 11.5% cotton lint yield decrease with each increment of one Palmer amaranth plant/10 m of row. Additionally, yield reductions have been documented in other row crops. Klingaman and Oliver (1994), in Arkansas, documented high yield losses in soybean [*Glycine max* (L.) Merr. Lloyd] ranging from 17 to 68% for Palmer amaranth densities of 0.33 to 10 plants/m of row, respectively. Similarly in Kansas, Bensch et al. (2003) reported Palmer amaranth to be more competitive than redroot pigweed (*Amaranthus retroflexus* L.) and common waterhemp (*Amaranthus rudis* Sauer) causing a yield reduction of 78.7% at a density of 8 plants/m of row. Menges (1988) reported allelopathic inhibition of grain sorghum [*Sorghum bicolor* (L.) Moench] and cabbage (*Brassica oleracea* var. *capitata* L.) growth by Palmer amaranth. Allelochemicals released from Palmer amaranth residue resulted in 49 and 68% decrease in carrot (*Daucus carota* L. var. *sativa*) and onion (*Allium cepa* L.) growth, respectively (Menges 1987).

1.1.2. Tillage System

Effect of tillage systems on seedling emergence patterns, composition and abundance of weed species have been the focus of many previous studies (Ball and Miller 1993; Dyer 1995; Forcella et al. 1993; Froud-Williams 1988; Froud-Williams et al. 1983a and1983b; Mulugeta and Stoltenberg 1997; Webster et al. 1998; Yenish et al. 1992 and 1996a). Tillage can alter weed seedling emergence patterns by modifying seed burial depth, dormancy, predation and mortality. Furthermore, it modifies the environmental factors crucial for germination, such as temperature, moisture, and oxygen (Benech-Arnold et al. 2000; Forcella et al. 2000; Leon and Owen 2004; Mohler 1993).

Historically, cotton was grown in a conventional tillage environment using primary and secondary tillage such as mold board plowing, disking and or cultivating. A conventional tillage system has been defined, by USDA-NRCS, as "a tillage system that involve primary tillage utilizing moldboard plows or heavy disks followed by one or more secondary tillage, planting and row cultivation operations that bury nearly all previous crop residue". The advantages associated with conventional tillage include; destruction of weeds, incorporation of manures and fertilizers, improved seed and soil contact, improved soil aeration and nutrient availability, exposure of harmful insects and plant pathogens to solar radiation and incorporation of preplant soil applied herbicides (FAO, 2013). Common detrimental-effects associated with conventional tillage are; soil compaction, increased cost of crop production due to high fuel, time and labor inputs, decline in soil fertility and productivity, increased soil erosion and environmental pollution through increased fossil fuel combustion (FAO 2013). As a consequence of increasing economic inputs, low commodity prices, and concerns for declining soil organic matter and

subsoil compaction, the cotton producers readily adopted the conservation tillage systems such as no-till and strip-tillage production systems (Troeh et al. 1991; Wauchope et al. 1985). Conservation tillage, according to the USDA-NRCS definition, is “any tillage system that leaves at least 30% of the soil surface covered with residue at the time of planting the main crop”. Benefits of conservation tillage include reductions in soil and wind erosion, savings in time, fuel and labor required to prepare fields for planting, increased water infiltration into the soil profile, improved beneficial soil biological activity and in some instances, reductions in disease, insects, and weed pests (Cantonwine et al. 2006; Doran 1980; Edward and Lofty 1982; Fawcett et al. 1994; Feng et al. 2003; Frye 1984; Johnson et al. 2001; Kay 1990; Lal 1997; Lal et al. 1994; Price et al. 2007; Rasmussen and Collins 1991;Unger and Jones 1998). However, inadequate weed control has been identified as a major production constraint associated with the conservation-tillage system (McWhorter and Jordan 1985).

Weersink et al. (1992a, 1992b) and Yiridoe et al. (1994) also perceived that changes in farming practices, such as the adoption of conservation tillage, may present unknown risks that can delay implementation by farmers. Clements et al. (1994) noted that changes in farm management systems will influence weed species diversity. Several cases of shift in weed species have already been reported, globally, due to adoption of conservation tillage system. Spreading dayflower (*Commelina diffusa* Burm. f.) and purple nutsedge (*Cyperus rotundus* L.) were recorded as the dominant weed species in no till rice in India (Singh et al. 2005). Further, wind disseminated species, annual and perennial grasses, and perennial dicot weeds increase with zero-till and minimum tillage in temperate regions of the world (Froud-Williams et al. 1981). Triplett (1985) attributed the abundance of perennial weed species with minimum and zero-till systems to the lack of disturbance of the root systems of established perennial weeds.

In conservation-agriculture, weeds seed germination is often higher because most of the weed seeds lie on the soil surface where germination conditions are more favorable (Banting 1966; Ball 1992; Chauhan et al. 2006; Chauhan and Johnson 2009; Chauhan and Johnson 2010a; Clements et al. 1996; Yenish et al. 1992). Therefore, small seeded weed species such as Palmer amaranth and horseweed have become highly prevalent in reduced tillage production systems (Buhler 1995). The reduction of tillage or seed burial is beneficial for weed seed germination and emergence (Egley and Williams 1991). However, an increase in weed seed predation has also been reported under conservation tillage production systems. Hulme (1994) reported that the weed seeds are most vulnerable to surface-dwelling seed predators when on the soil surface. Similarly, Chauhan et al. 2010b also reported more than 87% post dispersal weed seed predation from the soil surface in no-till rice production. Nevertheless, the infestation of small seeded annual weeds such as Palmer amaranth has often been attributed to the conservation tillage systems that preclude burial of weed seed.

The effect of tillage on weed seedling emergence depends on several factors such as type of tillage system, timing, depth, and speed of the tillage equipment. Different tillage implements disturb the soil differently which in turn affect weed population dynamics (Liebman et al. 2001). For instance, moldboard plows invert the soil and bury weeds with relatively little weed seed injury. Discs and rotary tillers mix weeds and crop residues into the soil profile. Disc plows disturb the soil minimally than compared with field cultivator or chisel plow (Chauhan et al. 2006). Therefore, it is expected that vertical seed distribution caused by different tillage implements may vary. Information on the vertical seed distribution may help predict the seedling recruitment (Grundy and Mead 1998).

Moldboard plowing with soil inversion to the depth of 30 cm (12 in) has been shown to reduce glyphosate-resistant Palmer amaranth emergence 46% to 60% because many of the weed seeds are placed at a depth where emergence cannot occur (Culpepper et al. 2009 and 2010). However, Price et al. (2011), perceiving the threat to the practice of conservation tillage, advocated the integration of traditional and alternative weed control strategies, such as utilization of crop and herbicide rotation and integration of high residue cereal cover crops in order to sustain conservation tillage practices and soil quality attributes.

1.1.3. Cover Crops

Cover crops have multifarious benefits associated with them; the important ones include weed suppression, soil erosion control, and restoration of soil fertility and reduction in nitrate leaching. Because of these virtues their cultivation is gaining momentum and, perhaps, is a judicious farming decision in today's conservation tillage system. While live cover crops offer competition to weeds for light, space, nutrients and moisture (Shilling et al. 1986; Lehman and Blum 1997) their residues physically suppress weeds through production of mulch (Bhowmik and Inderjit 2003). In addition, many release certain chemicals that work to the detriment of other plants growing in immediate vicinity. These chemicals are either directly released out of their root system when the covers are alive or may be released by microbial decomposition when cover residues decompose or are incorporated in soil.

As previously mentioned, conservation tillage systems are highly efficient in reducing soil erosion, conserving water, improving soil organic matter and reducing nitrate leaching but they are prone to increased weed infestations when chemical weed control methods are insufficient due to selectivity or improper application (Cardina et al. 2002; Sosnoskie et al.

2006). In such a scenario, the cover crops may form an important component in weed management. In modern day reduced and no tillage systems, the weed suppression potential of certain legume and grass covers is being increasingly capitalized and it has produced encouraging outcomes. Cover crops may include: winter rye (*Secale cereale* L.), barley (*Hordeum vulgare* L.), buckwheat (*Fagopyrum esculentum*), sorghum-sudangrass (*Sorghum bicolor* L. x *S. sudanense* L.), german millet (*Setaria italica*) pearlmillet (*Pennisetum glaucum*), Japanese millet (*Enchinochloa frumentacea*), crimson clover (*Trifolium incarnatum* L.), cowpea (*Vigna unguiculata*), Sunhemp (*Crotolaria juncea*), sweet clover (*Melilotus alba* Desr.), velvetbean (*Mucuna deeringiana*) and hairy vetch (*Vicia villosa*) etc. are some of the popular leguminous covers.

Recently, the inclusion of cover crops in conservation-tillage system is being promoted due to the ability of some cover crops to suppress early-season weed density and growth either through direct competition from cover crop biomass (Ateh and Doll 1996; Bauer et al. 2003; Collins et al. 2007; Reddy 2001; Teasdale et al. 1991; Teasdale and Mohler 2000; Yenish et al. 1996b) or through allelopathy (Barnes and Putnam 1986; Barnes et al.1987; Dhima et al. 2006; White et al. 1989).

1.1.3.1. Crimson Clover

Crimson clover, also known as scarlet clover, is an important winter season legume cover crop. It is believed to be native to Atlantic and southern Europe, Caucasus and Transcaucasus (Duke 1981). The maximum crimson clover height can be 18 inches but usual range is of 12 to 16 inches (Finch and Sharp 1983). The root system is typically a taproot, and well nodulated for nitrogen. It tolerates a wide range of climatic and soil conditions and is well-adapted to the

climate of the southeastern USA (Knight, 1985; Hargrove, 1986). Knight 1985) accentuates the need of timely establishment of crimson clover by at least 6 weeks before the average date of the first frost. It thrives in regions receiving 35 inches or more annual rainfall and cannot endure drought (McLeod, 1982). Knight (1985) has enlisted five most popular crimson clover cultivars used as cover crop as: 'Dixie,' 'Auburn,' 'Autauga,' 'Chief,' and 'Talladega'. Subsequently several new cultivars such as 'AU Robin' and 'AU Sunrise' and 'AU Sunup' have been released by Auburn University.

The early season weed suppression properties of crimson clover and several other legume species such as hairy vetch, and subterranean clover (*Trifolium subterraneum* L.) have been investigated by many researchers (Reddy 2001, 2003; Teasdale and Daughtry 1993; Teasdale et al. 1991; Yenish et al.1996b).In addition, it has been documented that the residue of leguminous crops is of higher forage quality than grasses because of lower carbon to nitrogen (C: N) ratio. In a conservation tillage setting, they decompose quicker and add more N to the soil than grasses thus helping contribute to increasing subsequent soil quality (Ranells and Wagger, 1996). Legume cover crops improve nitrogen availability to subsequent crops and reduce leaching of nitrogen into the immediate environment (Glasener et al. 2002; Sainju and Singh 1997; Varco et al.1999).

1.1.3.2. Cereal Rye

Cereal rye has been well documented for both high biomass potential and allelopathic properties by several researchers (Barnes and Putnam 1986; Burgos and Talbert 2000; Dhima et al. 2006; Price et al. 2006, Reeves et al. 2005). It is characterized by rapid growth, winter hardy nature, and huge biomass production potential that can be ≥10,000 pounds/acre (Sattell 1998). Its

extensive root system helps alleviate soil compaction. Further, its weed suppression and allelopathic properties have been well recognized. Kimber (1973) observed growth inhibition in wheat with the extract of slightly green rye decomposed over 21 days. Similarly, Barnes and Putnam (1983) noticed that spring -planted winter rye resulted in reduction in early season biomass by 98% in common lambsquarter (*Chenopodium album*), 42% in large crabgrass (*Digitaria sanguinalis* (L.) Scop.) and 90% in common ragweed (*Ambrosia artemisiifolia* L.) over fallow control. Further, the residues of fall planted rye reduced the biomass of barnyard grass and redroot pigweed by 74% and 55%, respectively as compared to poplar excelsior (*Populus spp*.) control mulch. Reeves et al. (2005) reported better weed control with a cereal rye cover crop than with wheat in conservation-tillage cotton, and control with rye plus a preemergence (PRE) herbicide was equal to control with a high-input herbicide system. Several studies on cover crops have reported an excellent early-season weed control that can preclude the use of PRE herbicides in crops (Ateh and Doll 1996; Fisk et al. 2001; Isik et al. 2009; Molin 2006; Reddy 2001; Reeves et al. 2005; Teasdale 1996; Teasdale et al. 1991, 2005; Vasilakoglou et al.2006; Yenish et al. 1996b; Zasada et al. 1997).

Although the success of a cover crop in early-season weed suppression is determined by the biomass production potential (Ateh and Doll 1996; Brennan and Smith 2005; Mohler and Teasdale 1993; Teasdale 1996**),** biomass production varies with year, location and management practices (Schomberg et al. 2006). Daniel et al. (1999) reported a higher rye biomass than crimson clover (*Trifolium incarnatum* L.), hairy vetch (*Vicia villosa* Roth), wheat, or white lupine. Culpepper et al. (2010), observed that rye residue alone were effective in reducing the glyphosate-resistant Palmer amaranth emergence by 94% in the row middle and 50% within the row. Price et al. (2008), reported that the use of high residue cover crops in conjunction with

chemical and cultural weed control tactics could provide effective Palmer amaranth control in established glyphosate-resistant populations as well as help prevent the development of resistance in current glyphosate-susceptible populations.

Some of the adverse effects associated with the cover crops include; depletion of soil moisture needed by the succeeding crop, low soil temperatures, high pest pressure, interference with planting operations and poor soil to seed contact **(**Bowman et al. 1998; Fernandez et al. 2008; Price et al. 2007). Conversely, improved storage of excessive soil moisture during periods of high rainfall was reported by Fernandez et al. 2008. Additionally, reduction in efficacy of soil applied herbicides through interception and sorption has been indicated in high residue production systems (Banks and Robinson 1982, 1984; Ehrback and Lovely 1975; Johnson et al. 1989; Gaston et al. 2003; Lowder and Weber 1979).

Nevertheless, the benefits of use of cover crops would likely outweigh their adverse impacts. Therefore, the inclusion of cover crops in conservation tillage system have the potential to provide weed control benefits similar to those realized from deep plowing under conventional tillage system.

1.1.4. Glufosinate-Resistant Technology

Higher weed control efficiency is an important virtue of the herbicide-resistant crops (Burnside 1992; Radosevich et al. 1992**).** Other benefits are simplification of weed management and enhancement in adoption of conservation tillage practices (Duke 1999; Martinez-Ghersa et al. 2003). The advent of glyphosate-resistant soybean, cotton, and corn in 1996, 1997, and 1998, respectively, has radically impacted the row crop production in the United States. This

technological breakthrough not only increased area under glyphosate-resistant crops but also greatly boosted the adoption of conservation tillage.

Glyphosate is a non-selective, systemic herbicide that works by inhibition of 5-enolpyruvylshikimate-3-phosphate synthase (EPSPS; 2.5.1.26), an enzyme in the shikimate biosynthetic pathway that produces the essential aromatic amino acids (tryptophan, tyrosine, and phenylalanine) and subsequently phenolics, lignins, tannins, and other phenylpropanoids (Duke 1992). This herbicide was commercialized in 1974 and by 1995 the volume of sales reached 4.5 million kg in the United States. With the introduction of glyphosate-resistant cotton varieties in 1997, the use of glyphosate in cotton increased dramatically; from a 700,000 kg in 1997 to 3870,000 kg yr^{-1} in 2006 (Young 2006). Further, reduction in glyphosate prices due to expiration of key patents resulted in more than 10- fold increase in glyphosate consumption. Therefore, the extensive adoption of glyphosate-resistant cotton technology virtually replaced the conventional weed control technology consisting of preplant incorporated (PPI), preemergence (PRE) and postemergence (POST) and postdirected (PDS) grass and broadleaf herbicides (Young 2006).

Until recently, the glyphosate has been very efficacious on several weeds including Palmer amaranth (Aulakh et al. 2011; Corbett et al. 2004; Culpepper and York 1998; Parker et al. 2005; Price et al. 2011). However, the sole reliance on glyphosate herbicide in glyphosate-resistant cotton resulted in selection for resistant biotypes of Palmer amaranth. Ever since the first reported case of occurrence of glyphosate resistance in Palmer amaranth from Macon County in Georgia (Culpepper et al. 2006), more resistance cases are being reported from other states in the southeastern United States. As of 2012, glyphosate-resistant Palmer amaranth populations have been confirmed in 20 states (Alabama, Arizona, Arkansas, California, Delaware, Florida, Georgia, Illinois, Kansas, Louisiana, Michigan, Missouri, Mississippi, North

Carolina, New Mexico, North Carolina, Ohio, South Carolina, Tennessee, and Virginia) (Heap 2013). In the wake of this recent glyphosate-resistant phenomenon, the cotton growers are considering other weed management options such as inversion tillage, use of cover crops and adoption of glufosinate-resistant cotton technology.

Glufosinate-resistant (GR) cotton was commercialized in 2004 and was tolerant to topical applications of glufosinate from crop emergence until the early bloom stage (Miller et al. 2012). Previous research has shown that POST glufosinate treatment at rates up to 3.3 kg ai/ha did not harm cotton and had no effect on plant height at maturity, total number of bolls, bolls per plant, and boll positions of 'Coker 312' (Blair-Kerth et al. 2001). Glufosinate is a non-selective herbicide that can effectively control glyphosate-resistant Palmer amaranth. Glufosinate inhibits glutamine synthetase (EC 6.3.1.2), an enzyme that catalyzes the synthesis of glutamine from glutamate and ammonium (Bellinder et al. 1987; Dekker and Duke 1995; Devine et al. 1993a). Glutamine synthetase inhibition leads to the accumulation of toxic levels of ammonia within the cell which eventually results in plant death (Coetzer and Al-Khatib 2001). Glufosinate resistance in cotton was bred by incorporation of the bialaphos resistance (BAR) gene from the fungus *Streptomyces viridochromogenes* that encodes for high levels of phosphinothricin acetyltransferase (pat). The pat enzyme converts the active molecule of glufosinate (L-phosphinothricin) into nontoxic N-acetyl-L-phosphinothricin (Devine et al. 1993a 1993b; Droge et al. 1992).

Glufosinate is highly effective against a broad spectrum of weeds that are commonly found in cotton, corn, and soybean (Bradley et al. 2000; Culpepper and York 1998; Culpepper et al. 2006; York and Culpepper 2004). However, as with glyphosate, glufosinate does not have any soil residual activity. Additionally, weed control efficacy of glufosinate varies with the age or

growth stage of weed (Corbett et al. 2004; Shaw and Arnold 2002). The lack of soil residual activity of glufosinate and occurrence of several weed flushes over the crop growing season necessitate multiple herbicide applications (Reddy and Norsworthy 2010). However, multiple applications would impose selection pressure on the weeds as has been the case with the evolution of glyphosate-resistant Palmer amaranth. Therefore, the use of residual herbicides as PRE, mid-POST (MPOST), and LAYBY postdirected applications would likely reduce the over reliance on glufosinate applications to minimize the selection pressure on weeds. Additionally, residual herbicides with diverse modes of actions will not only prevent or delay weed shifts and evolution of glufosinate-resistant weeds but also enhance the control of weeds that are not effectively controlled by glufosinate alone (Martinez-Ghersa et al. 2003).

Management of glyphosate-resistant Palmer amaranth in cotton is a major production issue in the southeastern United States. Inversion tillage is being readopted by many farmers in Georgia who have severe infestation of glyphosate-resistant Palmer amaranth on their farms. Although inversion tillage can improve control of glyphosate-resistant Palmer amaranth, increased input costs and potential soil erosion are significant challenges for growers. However the inclusion of cover crops and adoption of glufosinate-resistant cotton technology seem to be vindicated in the light of these economic and environmental considerations. Therefore, keeping in view the current Palmer amaranth management perceptions of cotton growers, field studies were conducted from fall 2008 through 2011 with the following objectives:

1. To evaluate the role of soil inversion, cover crops and herbicide system components for Palmer amaranth management in glufosinate-resistant cotton technology.

2. To evaluate the role of soil inversion, cover crops and secondary tillage methods for Palmer amaranth management in glufosinate-resistant cotton technology.

1.1.5. Literature Cited

Askew, S.D. and J.W. Wilcut. 1999. Cost and weed management with herbicide programs inglyphosate-resistant cotton (*Gossypium hirsutum*). Weed Technol. 13, 308-313.

Ateh, C.M. and J.D. Doll. 1996. Spring-planted winter rye as a living mulch to control weeds in soybean. Weed Technol. 10: 347-353.

Aulakh, J.S., Price, A.J., K.S. Balkcom. 2011. Weed management and cotton yield under two row spacings in conventional and conservation tillage systems utilizing conventional, glufosinate- and glyphosate-based weed management systems. Weed Technol. 25: 542–547.

Ball, D. A. 1992. Weed seedbank response to tillage, herbicides, and crop rotation sequence. Weed Sci. 40:654–659.

Ball, D. A. and S. D. Miller. 1993. Cropping history, tillage and herbicide effects on weed flora composition in irrigated corn. Agron. J. 85:817–821.

Banks, P.A., and E.L. Robinson. 1982. The influence of straw mulch on the soil reception and persistence of metribuzin. Weed Sci. 30:164-168.

Banks, P.A., and E.L. Robinson. 1984. The fate of oryzalin applied to straw-mulched and non-mulched soils. Weed Sci. 32:269-272.

Banting, J. D. 1966. Studies on the persistence of *Avena fatua*. Can. J. Plant Sci. 46:129-140.

Barnes, J. P. and A. R. Putnam. 1986. Evidence for allelopathy by residues and aqueous extracts of rye (*Secale cereale L.*). Weed Sci. 34:384–390.

Barnes, J. P., A. R. Putnam, B. A. Burke, and A. J. Aasen. 1987. Isolation and characterization of allelochemicals in rye herbage. Phytochemistry 26:1385–1390.

Barnes, J.P., and A.R. Putnam. 1983. Rye residues contribute weed suppression in no-tillage cropping systems. J. of Chem. J. Ecol. 9:1045-1057.

Bauer, P.J., D.W. Reeves, R. M. Johnson, and J. M. Bradow. 2003. Cover crop, tillage, and N rate effects on cotton growth in ultra-narrow rows. Crop Manage. Available online at http://www.plantmanagementnetwork.org/pub/cm/research/2003/ultranarrow (verified 1/21/ 2013).

Bellinder, R.R., R.E. Lyons, S.E. Scheckler, and H.P. Wilson. 1987. Cellular alterations resulting from foliar applications of HOE-39866. Weed Sci. 35:27-35.

Benech–Arnold, R. L., R. A. Sanchez, F. Forcella, B. C. Kruk, and C. M.Ghersa. 2000. Environmental control of dormancy in weed seed banks in soil. Field Crops Res. 67:105–122.

Bensch, C. N., M. J. Horak, and D. Peterson. 2003. Interference of redroot pigweed (*Amaranthus retroflexus*), Palmer amaranth (*Amaranthus palmeri*), and common waterhemp (*Amaranthus rudis*) in soybean. Weed Sci. 51:37–43.

Bhowmik P.C. and Inderjit. 2003. Challenges and opportunities in implementing allelopathy for natural weed management. J. Crop Prot. 22: 661-671.

Blair-Kerth, L.K., P.A. Dotray, J.W. Keeling, J.R. Gannaway, M.J. Oliver, and J.E. Quisenberry. 2001. Tolerance of transformed cotton to glufosinate. Weed Sci. 49:375-380.

Bowman, G., C. Shirley, and C. Cramer. 1998. Managing cover crops profitably. 2nd ed. Washington, D.C.: Sustainable Ag. Network.

Bradley, P. R., W. G. Johnson, S. E. Hart, M. L. Buesinger, and R. E. Massey.2000. Economics of weed management in glufosinate-resistant corn (*Zea mays* L.). Weed Technol. 14:495–501.

Brennan, E.B., and R.F. Smith. 2005. Winter cover crop growth and weed suppression on the central coast of California. Weed Technol. 19:1017–1024.

Buhler, D. D. 1995. Influence of tillage systems on weed population dynamics and management in corn and soybean in the central USA. J. Crop Sci. 35:1247–1258.

Burgos, N. R. and R. E. Talbert. 2000. Differential activity of allelochemicals from *Secale cereale* in seedling bioassays. Weed Sci. 48:302-310.

Burke, I. C., M. Schroeder, W. E. Thomas, and J. W. Wilcut. 2007. Palmer amaranth interference and seed production in peanut. Weed Technol. 21:367–371.

Burnside, O. C. 1992. Rationale for developing herbicide-resistant crops. Weed Technol. 6:621–625.

Cantonwine, E.G., A.K. Culbreath, K.L. Stevenson, R.C. Kemerait, Jr., T.B. Brenneman, N.B. Smith, and B.G. Mullinix, Jr. 2006. Integrated Disease Management of Leaf Spot and Spotted Wilt of Peanut. Plant Dis. 90:493-500.

Cardina, J., C.P. Herms, and D.J. Doohan. 2002. Crop rotation and tillage system effects on weed seedbanks. Weed Sci. 50:448–460.

Chauhan, B., G. Gill, and C. Preston. 2006. Influence of tillage system on vertical distribution, seedling recruitment and persistence of rigid ryegrass (*Lolium rigidum*) seed bank. Weed Sci. 54:669-676.

Chauhan, B.S. and D.E. Johnson. 2009. Influence of tillage systems on weed seedling emergence pattern in rainfed rice. Soil Till. Res. 106: 15–21.

Chauhan, B.S. and D.E. Johnson. 2010. The role of seed ecology in improving weed management strategies in the tropics. Adv. Agron. 105: 221–262.

Chauhan, B.S., T. Migo, P.R. Westerman, and D.E.Johnson. 2010. Post-dispersal predation of weed seeds in rice fields. Weed Res. 50: 553–560.

Clements, D. R., S. F. Weise, and C. J. Swanton. 1994. Integrated weed management and weed species diversity. Phytoprotection 75:1–18.

Clements, D.R., D.L. Benoit, and C.J. Swanton. 1996. Tillage effects on weed seed return and seedbank composition. Weed Sci. 44: 314–322.

Coetzer, E., K. Al-Khatib, and D.E. Peterson. 2002. Glufosinate efficacy on Amaranthus species in glufosinate-resistant soybean (*Glycine max*).Weed Technol. 16:326-331.

Collins, H.P., J.A. Delgado, A.K. Alva, and R.F. Follett. 2007. Use of nitrogen-15 isotopic techniques to estimate nitrogen cycling from a mustard cover crop to potatoes. Agron. Journal 99 (1):27-35.

Corbett, J. L., S. D. Askew, W. E. Thomas, and J. W. Wilcut. 2004. Weed efficacy evaluations for bromoxynil, glufosinate, glyphosate, pyrithiobac, and sulfosate. Weed Technol. 18:443–453.

Culpepper, A. S. and A. C. York. 1998. Weed management in glyphosate-tolerant cotton. J. Cotton Sci. 2:174–185.

Culpepper, A. S., T. L. Grey, W. K. Vencill, J. M. Kichler, T. M. Webster, S. M. Brown, A. C. York, J. W. Davis, and W. W. Hanna. 2006. Glyphosate-resistant Palmer amaranth (*Amaranthus palmeri*) confirmed in Georgia. Weed Sci. 54:620–626.

Culpepper, A. S., A. C. York, and M. W. Marshall. 2009. Glyphosate-resistant Palmer amaranth in the Southeast. Pages in T. M. Webster, ed. Proceedings of the Southern Weed Science Society. Orlando, FL: Southern Weed Science Society, 62:371.

Culpepper, A.S., J. Kichler, L. Sosnoskie, A. York, D. Sammons, and R. Nichols. 2010. Integrating cover crop residue and moldboard plowing into glyphosate-resistant Palmer amaranth management programs, 1531. Proceedings of Beltwide Cotton Conference Jan 4-7 New Orleans, LA.

Daniel, J.B., A.O. Abaye, M.M. Alley, C.W. Adcock, J.C. and Maitland.1999. Winter annual cover crops in Virginia no-till cotton production system. II. Cover crop and tillage effects on soil moisture, cotton yield, and cotton quality. J. Cotton Sci. 3, 84–91.

Dekker J. and Duke S.O. 1995. Herbicide-resistant field crops. *Advan.Agron.* **54**, 69–116.

Devine, M., S. O. Duke and C. Fedtke. 1993a. Herbicide effects on lipid synthesis. p.225- 242. *In.* Physiology of herbicide action. Prentice Hall, Englewood Cliffs, NJ.

Devine, M., S. O. Duke and C. Fedtke. 1993b. Inhibition of Amino Acid Biosynthesis p.251-294. *In.* Physiology of herbicide action. Prentice Hall, Englewood Cliffs, NJ.

Dhima, K.V., I.B. Vasilakoglou, I.G. Eleftherohorinos and A.S. Lithourgidis 2006. Allelopathic potential of winter cereals and their cover crop mulch effect on grass weed suppression and corn development. Crop Sci. 46:345–352.

Doran, J.W. 1980. Soil microbial and biochemical changes associated with reduced tillage. Soil Sci. Soc. Am. J. 44:765-771.

Droge, W., I. Broer, and A. Pulher. 1992. Transgenic plants containing the phosphinothricin-N-acetyltransferase gene metabolize the herbicide L-phosphinothricin (glufosinate) differently from untransformed plants. Planta 18:142-151.

Duke J.A. 1981. *Trifolium incarnatum* L. In: Duke J.A. (ed.) Handbook of Legumes of World Economic Importance, pp. 245-248. New York: Plenum Press.

Duke, S. O. 1992. Modes of action of herbicides used in cotton. Pages 403–437 in C. G. McWhorter and J. L. Abernathy, eds. Weeds of Cotton. Memphis,TN: The Cotton Foundation.

Duke, S. 1999. "Weed Management: Implications of Herbicide Resistant Crops". Publications from USDA-ARS / UNL Faculty Paper 481. Available online at: http://digitalcommons.unl.edu/cgi/viewcontent.cgi?article=1487&context=usdaarsfacpub. (Verified on 2/15/2013).

Dyer, W. 1995. Exploiting weed seed dormancy and germination requirements through agronomic practices. Weed Sci. 43:498–503.

Edwards, C.A., and J.R. Lofty. 1982. The Effect of direct drilling and minimal cultivation on earthworm populations. J. Appl. Ecol. 19:723-734.

Egley, G.H., and R.D. Williams. 1991. Emergence periodicity of six summer annual weed species. Weed Sci. 39:340-344.

Ehleringer, J. 1983. Ecophysiology of *Amaranthus palmeri*, a Sonoran Desert summer annual. Oecologia 57:107–112.

Ehrbach, D.C., and W.G. Lovely. 1975. Effect of plant residue on herbicide performance in no-tillage corn. Weed Sci. 23:512-515.

FAO, 2013: Conservation of natural resources for sustainable agriculture, Available online at: http://www.fao.org/ag/ca/Training_Materials/CD27-English/sf/soil_fertility.pdf. (Verified, 2/ 15/2013).

Fawcett, R.S., B.R. Christensen, and D.P. Tierney. 1994. The impact of conservation tillage on pesticide runoff into surface water: A review and analysis. J. Soil Water Cons. 49:126-135.

Feng, Y., A.C. Motta, D.W. Reeves, C.H. Burmester, E. van Santen, and J.A. Osborne. 2003. Soil microbial communities under conventional-till and no-till continuous cotton systems. Soil Biol. Biochem. 35:1693-1703.

Fernald, M. L. 1950. Gray's Manual of Botany. 8th ed. New York, American Book Co. 602.

Fernandez, R., A. Quiroga, E. Noellemeyer, D. Funaro, J. Montoya, B. Hitzmann, and N. Peinemann. 2008. A study of the effect of the interaction between site-specific conditions, residue cover and weed control on water storage during fallow. Agricultural Water Manage. 95: 1028-1040.

Finch, C. V., and C. W. Sharp. 1976. Cover crops in California orchards and vineyards.U.S.Dep.Agric. , Soil Conservation Service, Washington, DC.

Fisk J.W., Hesterman O.B., Shrestha A., Kells J.J., Harwood R.R., Squire J.M. et al. 2001.Weed suppression by annual legume cover crops in no tillage corn. Agron. J. 93: 319–325.

Forcella, F., N. Colbach, and G.O. Kegode. 2000. Estimating seed production of three Setaria species in row crops. Weed Sci. 48:436–444.

Forcella, F.F., K. Eradat-Oskouim, and S.W. Wagner. 1993. Applications of weed seedbank ecology to low-input crop management. Ecological Applications 3:74-83.

Froud-Williams, R.J., R.J. Chancellor, and D.S.H. Drennan. 1981. Potential changes in weed floras associated with reduced-cultivation systems for cereal production in temperate regions. Weed Res. 21:99-109.

Froud-Williams, R.J., R.J. Chancellor, and D.S.H. Drennan. 1983a. Influence of cultivation regime upon buried weed seeds in arable cropping systems. Appl. Ecol. 20:199-208.

Froud-Williams, R. J., D.S.H. Drennan, and R. J. Chancellor. 1983b. Influence of cultivation regime on weed floras of arable cropping systems. J. Appl. Ecol. 20:187–197.

Froud-Williams, R. J. 1988. Changes in weed flora with different tillage and agronomic management systems. Pages 213–236 in M. A. Altieri and M. Liebman, eds. Weed management in agroecosystems: Ecological approaches. Boca Raton, FL: CRC Press.

Frye, W.W. 1984. Energy requirements in no-tillage, p. 127-151. In R.E. Phillips and S.H. Phillips (ed.). No-tillage agriculture: Principles and Practices, van Nostrand Reinhold, New York.

Gaston, L.A., D.J. Boquet, and M.A. Bosch. 2003. Pendimethalin wash-off from cover crop residues and degradation in a Loessial soil. Communications in Soil Science and Plant Analysis. 34, 2515-2527.

Glasener, K. M., M.G. Wagger, C.T. MacKown, and R.J. Volk. 2002. Contributions of shoot and root nitrogen-15 labeled legume nitrogen sources to a sequence of three cereal crops. Soil Sci. Soc. Am. J. 66:523–530.

Grundy, A.C. and A. Mead.1998. Modelling the effects of seed depth on weed seedling emergence. Aspects Appl. Biol. 51, 75–82.

Grundy, A.C., Mead, A., 1998. Modeling the effects of seed depth on weed seedling emergence. Aspects Appl. Biol. 51, 75–82.

Hargrove, W.L. 1986. Winter legumes as a nitrogen source for no-till grain sorghum. Agron. J. 78:70-74.

Heap, I. 2013. The international survey of herbicide resistant weeds. Available online at http:// www.weedscience.org. (Verified, 1/ 21/2013).

Horak, M. J. and T. M. Loughin. 2000. Growth analysis of four Amaranthus species. Weed Sci. 48:347–355.

Horak, M. J., D. E. Peterson, D. J. Chessman, and L. M. Wax. 1994. Pigweed Identification: A Pictorial Guide to the Common Pigweeds of the Great Plains. Manhattan, KS: Kansas State University. 12 p. http://digitalcommons.unl.edu/usdaarsfacpub/481. (Verified on 1/23/2013).

Hulme, P.E. 1994. Post-dispersal seed predation in grassland: its magnitude and sources of variation. Ecol. 82: 645-652.

Isik, D., Kaya, E., Ngouajio, M., Mennan, H., 2009. Weed suppression in organic pepper (*Capsicum annuum* L.) with winter cover crops. Crop Prot. 28, 356– 363.

Jha, P., J. K. Norsworthy, M. B. Riley, D. G. Bielenberg, and W. Bridges, Jr. 2008. Acclimation of Palmer amaranth (*Amaranthus palmeri*) to shading. Weed Sci. 56:729–734.

Johnson, M.D., D.L. Wyse, and W.E. Lueschen. 1989. The influence of herbicide formulation on weed control in four tillage systems. Weed Sci. 37, 239-249.

Johnson, W. Carroll III, T.B. Brenneman, S.H. Baker, A.W. Johnson, D.R. Sumner, and B.G. Mullinix, Jr. 2001. Tillage and pest management considerations in a peanut-cotton rotation in the Southeastern coastal plain. Agron. J. 93:570-576.

Kay, B.D. 1990. Rates of change of soil structure under different cropping systems. Advances in Soil Science. 12:1-52.

Keeley, P. E., C. H. Carter, and R. M. Thullen. 1987. Influence of planting date on growth of Palmer amaranth (*Amaranthus palmeri*). Weed Sci. 35:199–204.

Kimber, RWL. 1973. Phytotoxicities form plant residues. I. The influence of rotted wheat straw on seedling growth. Aust. J. Agric. Res. 18: 361-374.

Klingaman, T.E. and L.R. Oliver. 1994. Palmer amaranth (*Amaranthus palmeri*) interference in soybeans (*Glycine max*). Weed Sci. 42:523–527.

Knight, W. E. 1985. Crimson clover. Univ. of Calif. SAREP Cover Crops Resource Page. www.sarep.ucdavis.edu/ccrop. (Verified on 10/23/2012).

Lal, R. 1997. Residue management, conservation tillage and soil restoration for mitigating greenhouse effect by CO_2 enrichment. Soil & Till. Res. 43:81-107.

Lal, R., A. A. Mahboubi, and N.R. Fausey. 1994. Long-term tillage and rotation effects on properties of a central Ohio soil. Soil Sci. Soc. Am. J. 58:517-522.

Lehman M. E. and U. Blum.1997. Cover crop debris effects on weed emergence as modified by environmental factors. J. Allelopathy 4: 69-88.

Liebman, M., C.L. Mohler, and C.P. Staver. 2001. Ecological management of agricultural weeds. Cambridge, U.K.: Cambridge University Press. 532 p.

Lowder, S.W., and J.B. Weber. 1979. Atrazine retention by crop residues in reduced-tillage systems. Proc. South. Weed Sci. Soc. 32:303-307.

Martinez-Ghersa, M. A., C. A. Worster, and S. R. Radosevich. 2003. Concerns a weed scientist might have about herbicide-tolerant crops: a revisitation. Weed Technol. 17:202–210.

Massinga, R. A., R. S. Currie, M. J. Horak, and J. Boyer. 2001. Interference of Palmer amaranth in corn. Weed Sci. 49:202–208.

McGregor, R. L. 1986. Amaranthaceae, the pigweed family. In Flora of the Great Plains. Lawrence, KS: University Press of Kansas. pp. 179–184.

McLeod, E. 1982. Feed the Soil. Organic Agriculture Research Institute, Graton, CA. p. 209.

McWhorter, C. G. and T. N. Jordan. 1985. Limited tillage in cotton production. Pages 61-75 in A. F. Wiese ed., weed control in limited-tillage systems. Champaign, IL,Weed Sci. Soc. Am.

Menges, R.M. 1987. Allelopathic effects of Palmer amaranth (*Amaranthus palmeri*) and other plant residues in soil. Weed Sci 35:339–347.

Menges, R.M. 1988. Allelopathic effects of Palmer amaranth (*Amaranthus palmeri*) on seedling growth. Weed Sci 36:325–328.

Meyers, S. L., K.M. Jennings, J.R. Schultheis, and D.W. Monks.2010.Evaluation of Flumioxazin and *S*-metolachlor Rate and Timing for Palmer Amaranth (*Amaranthus palmeri*) Control in Sweetpotato. Weed Technol. 24: 495-503.

Miller, D.K., D.O. Stephenson IV, and D. C. Blouin. 2012. Glufosinate-resistant cotton tolerance to combinations of glufosinate with insecticides and mepiquat chloride. J. Cotton Sci. 16: 125-128.

Mohler, C. L. and J. R. Teasdale. 1993. Response of weed emergence to rate of *Vicia villosa* Roth and *Secale cereale* L. residue. Weed Res. 33: 487–499.

Mohler, C.L., 1993. A model of the effects of tillage on emergence of weed seedlings. Ecol. Appl. 3, 53–73.

Molin, W. T. 2006. Contributions of tillage, rye cover crop and herbicide programs to weed control in glyphosate-tolerant cotton. Pages 171–173 in R. C. Schwartz, R. L. Baumhardt and J. M. Bell, eds. Proceedings of the 28th Southern Conservation Tillage Systems Conference. Bushland, TX.

Monks, D. M. and L. R. Oliver. 1988. Interactions between soybean (*Glycine max*) cultivars and selected weeds. Weed Sci. 36:770–774.

Moore, J. W., D. S. Murray, and R. B. Westerman. 2004. Palmer amaranth (*Amaranthus palmeri*) effects on the harvest and yield if grain sorghum (*Sorgum biclor*). Weed Technol. 18: 23-29.

Morgan, G. D., P. A. Baumann, and J. M. Chandler. 2001. Competitive impact of Palmer amaranth (*Amaranthus palmeri*) on cotton (*Gossypium hirsutum*) development and yield. Weed Technol. 15:408–412.

Mulugeta, D., and D.E. Stoltenberg. 1997. Weed and seedbank management with integrated methods as influenced by tillage. Weed Sci. 45:706–715.

Parker, R. G., A. C. York, and D. L. Jordan. 2005. Comparison of glyphosate products in glyphosate-resistant cotton (*Gossypium hirsutum*) and corn (*Zea mays*). Weed Technol.19:796–802.

Place, G., D. Bowman, M. Burton, and T. Rufty. 2008. Root penetration through a high bulk density soil layer: differential response of a crop and weed species. Plant and Soil 307:179–190.

Price, A.J., D.W. Reeves, and M.G. Patterson. 2006. Evaluation of weed control provided by three winter cereals in conservation-tillage soybean. Ren. Agric. and Food Sys. 21:159-164.

Price, A.J., D.W. Reeves, M.G. Patterson, B.E. Gamble, K.S. Balkcom, F.J. Arriaga, and C.D. Monks. 2007. Weed control in peanut grown in a high-residue conservation-tillage system. Peanut Sci. 34:59-64.

Price, A.J., M.E. Stoll, J.S. Bergtold, F.J. Arriaga, K.S. Balkcom, T.S. Kornecki, and R.L. Raper. 2008. Effect of cover crop extracts on cotton and radish radicle elongation. Comm. Biometry Crop Sci. 3:60-66.

Price, A.J., K.B. Balkcom, A.S. Culpepper, J.A. Kelton, R.L. Nichols, and H.H. Schomberg. 2011. Glyphosate-resistant Palmer amaranth: A threat to conservation tillage. J. Soil Water Conserv. 66:265-275.

Radosevich, S. R., C. M. Ghersa, and G. Comstock. 1992. Concerns a Weed Scientist might have about herbicide tolerant crops. Weed Technol. 6: 635–639.

Ranells, N.N., M.G. Wagger. 1996. Nitrogen release from grass and legume cover crop monocultures and bicultures. J. Agron. 88: 777–782.

Rasmussen, P.E. and H.P. Collins. 1991. Long-term impacts of tillage, fertilizer, and crop residue on soil organic matter in temperate semiarid regions. Adv. Agron. 45:93-134.

Reddy, K.N. 2001. Effects of cereal and legume cover crop residues on weeds, yield, and net return in soybean (*Glycine max*). Weed Technol. 15:660–668.

Reddy, K. N. 2003. Impact of rye cover crop and herbicides on weeds, yield, and net return in narrow-row transgenic and conventional soybean (*Glycine max*). Weed Technol. 17:28-35.

Reddy, K.N. and J.K. Norsworthy. 2010. Glyphosate-resistant crop production systems: Impact on weed species Shifts. pp. 165-184 in Vijay K. Nandula. Eds. Glyphosate resistance in crops and weeds: History, development, and management. John Wiley & Sons, Inc., New York, New York.

Reeves, D.W., A.J. Price, and M.G. Patterson. 2005. Evaluation of three winter cereals for weed control in conservation-tillage nontransgenic cotton. Weed Technol. 19: 731-736.

Rowland, M. W., D. S. Murry, and L. M. Verhalen. 1999. Full-season Palmer amaranth (*Amaranthus palmeri*) interference with cotton (*Gossypium hirsutum*). Weed Sci. 47:305–309.

Sainju, U.M., and B.P. Singh. 1997. Winter cover crops for sustainable agricultural systems: Influence on soil properties, water quality, and crop yields. Hort. Sci. 32:21-28.

Sattell, R. 1998. Using cover crops in Oregon. EM 8704. Oregon State University Extension Service, Corvallis, OR.

Schomberg, H.H., D.M. Endale, A. Calegari, R. Peixoto, M. Miyazawa, and M.L. Cabrera. 2006. Influence of cover crops on potential nitrogen availability to succeeding crops in a Southern Piedmont soil. Biology and Fertility of Soils 42:299-307.

Shaw, D. R. and J. C. Arnold. 2002. Weed control from herbicide combinations with glyphosate. Weed Technol. 16:1–6.

Shilling D.G., L.A. Jones, A.D. Worsham, C.E. Parker, and R.F. Wilson.1986. Isolation and identication of some phytotoxic compounds from aqueous extracts of rye (*Secale cereale* L.). J. Agril. and Food Chem. 34: 633-638.

Singh, V.P., Singh, G., Singh, S.P., Kumar, A., Singh, Y., Johnson, D.E., M. Mortimer. 2005. Effect of rice wheat establishment methods and weed management in irrigated rice–wheat production system. In: Workshop on ‘‘Direct Seeded Rice in the Rice–wheat System of the Indo-Gangetic Plains, 1 February to 2 February 2005, G.B. Pant University of Agriculture and Technology, Pantnagar, U.S. Nagar, Uttaranchal, India, p. 12.

Smith, D. T., R. V. Baker, and G. L. Steele. 2000. Palmer amaranth (*Amaranthus palmeri*) impacts on yield, harvesting, and ginning in dryland cotton (*Gossypium hirsutum*). Weed Technol. 14:122–126.

Sosnoskie, L.M., C.P. Herms and J. Cardina. 2006. Weed seedbank community composition in a 35-yr-old tillage and rotation experiment. Weed Sci. 54, 263- 273.

Steckel, L. E., C. L. Sprague, E. W. Stoller, and L. M. Wax. 2004. Temperature effects on germination of nine Amaranthus species. Weed Sci. 52:217–221.

Teasdale, J. R., C. E. Beste, and W. E. Potts. 1991. Response of weeds to tillage and cover crop residue. Weed Sci. 39:195–199.

Teasdale, J. R. & C. S. T. Daughtry. 1993. Weed suppression by live and desiccated hairy vetch (*Vicia villosa*). Weed Sci. 41:207–212.

Teasdale, J. R. 1996. Contribution of cover crops to weed management in sustainable agricultural systems. J. Prod. Agric. 9:475–479.

Teasdale, J. R. and C. L. Mohler. 2000. The quantitative relationship between weed emergence and the physical properties of mulches. Weed Sci. 48:385–392.

Teasdale, J. R., P. Pillai and Ronald T. Collins. 2005 Synergism between cover crop residue and herbicide activity on emergence and early growth of weeds. Weed Sci. 53:521-527.

Triplett, G.B., Jr.; G.D. Lytle. 1972. Control and ecology of weeds in continuous corn without tillage. Weed Sci. 20: 453–457.

Troeh, F. R., J. A. Hobbs, and R. L. Donahue. 1991. Soil and water conservation. New Jersey: Prentice Hall. 232 p.

Unger, P.W. and Jones, O.R. 1998. Long-term tillage and cropping systems affect bulk density and penetration resistance of soil cropped to dry land wheat and grain sorghum. Soil and Till. Res. 45: 39-57.

Varco, J.J., W.W. Frye, M.S. Smith, and C.T. Mackown. 1989. Tillage effects on nitrogen recovery by corn from a nitrogen-15 labeled legume cover crop. Soil Sci. Soc. Am. J.53:822-827.

Vasilakoglou , I., K. Dhima, I. Eleftherohorinos and A. Lithourgidis. 2006. Winter Cereal Cover Crop Mulches and Inter-Row Cultivation Effects on Cotton Development and Grass Weed Suppression. Agron. J. 98: 1290-1297.

Wauchope, R. D., L. L. McDowell, and L. J. Hagen. 1985. Environmental effects of limited tillage. In Weed Control in Limited Tillage Systems. Champaign, IL, Weed Sci. Soc. Am. Pages 266-281.

Webster, T. M. 2005. Weed survey—southern states. Proc. South. Weed Sci. Soc. 58:291–306.

Webster, T. M., J. Cardina, and H. M. Norquay. 1998. Tillage and seed depth effects on velvetleaf (*Abutilon theophrasti*) emergence. Weed Sci. 46:76–82.

Weersink, A., M. Walker, C. Swanton, and J. Shaw. 1992a. Economic comparison of alternative tillage systems under risk. Can. J. Agric. Econ. 40:199–217.

Weersink, A., M. Walker, C. Swanton, and J. E. Shaw. 1992b. Costs of conventional and conservation tillage systems. J. Soil Water Conserv.47:328–334.

White, R.H., A.D. Worsham, and U. Blum. 1989. Allelopathic potential of legume debris. Weed Sci. 37:674–679.

Wright, S. R., M. W. Jennette, H. D. Coble, and T. W. Rufty. 1999. Root morphology of young *Glycine max, Senna obtusifolia,* and *Amaranthus palmeri*. Weed Sci. 47:706–711.

Yenish, J. P., J. D. Doll, and D. D. Buhler. 1992. Effects of tillage on vertical distribution and viability of weed seed in soil. Weed Sci. 40: 429–433.

Yenish, J.P., Fry, T.A., Durgan, B.R., Wyse, D.L., 1996a. Tillage effects on seed distribution and common milkweed (*Asclepias syriaca*) establishment. Weed Sci. 44, 815–820.

Yenish, J.P., A.D. Worsham, and A.C. York. 1996b. Cover crops for herbicide replacement in no-tillage corn (*Zea mays*). Weed Technol. 10:815-821.

Yiridoe, E. K., A. Weersink, C. J. Swanton, and R. C. Roy. 1994. Risk efficient choice of bean-winter wheat rotation, cover crop and tillage system on light textured soils. J. Prod. Agric. 7:374–380.

York, A.C., and A.S. Culpepper. 2009. Weed Management in Cotton. p. 68-125. In 2009 Cotton Information. Publ. AG-417. North Carolina Coop. Ext. Serv., Raleigh, NC.

Young, B. G. 2006. Changes in herbicide use patterns and production practices resulting from glyphosate-resistant crops. Weed Technol. 20:301–307.

Zasada, I. A, H. M. Linker, and H. D. Coble. 1997. Initial weed densities affect no-tillage weed management with a rye (*Secale cereale*) cover crop. Weed Technol. 11:473-477.

Introduction and Literature Review-Part II

1.2. Cogongrass Patch Eradication

1.2.1. Introduction

Cogongrass (*Imperata cylindrica* (L.) Beauv. var. major tribe Andropogoneae) is a highly invasive, perennial, rhizomatous grass and a serious weed problem in over 73 countries across the globe (MacDonald 2004). It has been ranked as the seventh most troublesome weed worldwide (Falvey 1981; Holm et al. 1977).In the United States, cogongrass is a weed of non-agricultural area but in several tropical and subtropical countries elsewhere, it is a major constraint to production of more than 35 agricultural crops (Akobundo and Ekeleme 2000; Chikoye et al. 2000; Chikoye et al. 2005; Coile and Shilling 1993; Udensi et al.1999; Holm et al. 1977). International trade and movement of humans during the last century have been the vectors of accidental or intentional introduction of nonnative invasive species in many parts of the world (Rejmanek, 1996; Starfinger et al. 1998; Mooney and Hobbs, 2000). The advent of cogongrass into the United States dates back to 1911 when it was accidentally shipped in, along with Satsuma orange plants, as seed via Grand Bay, AL (Dickens, 1974; Patterson et al. 1983; Tabor, 1952). Subsequently, it was purposefully imported and planted at several places in Alabama, Florida and Mississippi to evaluate its potential as forage and soil stabilizer (Dickens and Buchanan 1975; Tabor, 1949; Tabor, 1952). In the southeastern United States it has invaded a wide range of habitats ranging from dry sand dunes to wet swamps and river margins (Chikoye

et al. 1999; Garrity et al. 1996). Globally, cogongrass infestation is spread across more than 500 million ha, of which 200 million hectares are in Asia alone (Dickens 1974; Falvey 1981; Holm et al. 1977).

At present it occupies more than 500,000 ha of land in the United States and classified as a noxious weed in Alabama, Florida, Georgia, Louisiana, Mississippi, Oregon, South Carolina, Texas and Virginia. (Bryson and Carter 1993; Byrd and Bryson 1999, Faircloth et al. 2005; Patterson et al.1980 and 1983; Van Loan et al. 2002; Willard et al. 1988 and 1990). Cogongrass is well recognized for its adverse economic, ecological and ecosystem impacts. Additionally, consideration of the economics of the pine industry and the risks cogongrass may pose to human life and property via ignition of wildfires might magnify the benefit cost ratio (Jose et al. 2002; Miller, 2000). Lippincott (2000) described cogongrass as a powder keg for forest fires. Cogongrass competitively displaces native vegetation and strongly alters wildlife habitats, feeding and breeding grounds that could eventually lead to tremendous loss of biodiversity.

1.2.2. Distribution and Biology of Cogongrass

Typically, cogongrass grows in tropical, subtropical, and some temperate regions of the world (Akobundo and Agyakwa 1998; Bryson and Carter 1993) and is present in all continents except Antarctica (Holm et al. 1977; Hubbard 1944). It is found along the Mediterranean Sea in Europe and northern Africa to the Middle East. It is most wide spread in Asia infesting over 70 million acres (Garrity et al.1993). It also occurs throughout Southeast Asia, Indonesia and the Pacific Islands and is estimated to occupy 8.5 to more than 64 million hectares of land in Indonesia (Garrity et al.1996; Suryanta and McIntosh, 1980). Large and solid cogongrass stands in these areas are often called mega-grasslands, imperata savannas or sheet imperata. In African

continent, cogongrass is prevalent in the West African countries, and in the eastern countries of Egypt, Sudan, Ethiopia (Holm et al. 1977). It is also found in western South America whereas its very close relative, Brazilian satintail (*Imperata brasiliensis* Trin.) is found in central South America. In the United States it is a federal noxious weed in several southeastern states.

Cogongrass is characterized by C_4 photosynthesis mechanism, wide environmental and edaphic adaptability, rapid vegetative growth, high genotypic variability and tremendous reproductive potential from rhizomes as well as seeds (Bryson et al. 2010; Capo-chichi et al. 2008; Sajise, 1972; Vergara et al. 2008). The vegetative propagation via rhizomes, however, is the predominant mode of regeneration (Dickens and Moore, 1974; Hubbard et al. 1944; Sajise, 1976; Wilcut et al. 1988a; Wilcut et al. 1988b; Willard, 1988). Cogongrass is best adapted to full sun light but can also thrive under moderate shade (Hubbard et al. 1944). Gaffney (1996) and Ramsey et al. (2003) recorded a light compensation point of 32 to 35 mol m^{-2} s^{-1} indicating the ability to survive as an understory species. In a greenhouse experiment cogongrass was documented to tolerate an 89% reduction in light levels and resume rapid growth when exposed to full sunlight (Patterson 1980). Cogongrass shoots arise from subsoil, scaly rhizomes and reach lengths of 1.2 to 3 m (Brown 1944; Gabel 2003; Holm et al. 1977). Eussen (1980) observed that one rhizome of cogongrass can produce over 350 shoots in 6 weeks and cover up to 4 m^{-2} in 11 weeks. Soerjani (1970) reported that over 2 million shoots of cogongrass could be present per acre. Cogongrass leaf blades are up to 1.5 m long and range from 1.0 to 28.0 mm wide with an off-centre mid vein and inflorescences are fluffy, silvery white ranging from 5.7 to 52.0 cm long (Gabel 2003). Single plant can produce over 3000 air-borne seeds that are capable of natural long-distance dispersal, and a common method of initiating new sites (Hubbard et al. 1944). Cogongrass seeds lack dormancy and remain viable for up to a year with the viability decreasing

sharply with the age. The wind wafted seeds are documented to travel long distances usually over 15 miles (Holm et al.1977). Wilcut et al. (1988a) attributed the long distance distribution and spread of cogongrass in Alabama from 1973 to 1985, along rights-of-way bordering interstate 65, to the seed dispersal by north-easterly winds blowing off the Gulf of Mexico. Similarly, King and Grace (2000) reported successful invasion via seeds in wet pine savannas in Grand Bay National Wildlife Refuge in Mississippi. However, the success of establishment from seed is very rare due to dependence upon soil disturbance (Kushwaha et al. 1983).

Cogongrass spread within patches is primarily through the rhizomes (Willard et al. 1990). Ayeni (1985) reported a positive correlation of regeneration from rhizome with the age, weight, length, thickness and number of visible buds. Rhizomes are chiefly concentrated in the top 6-8 inch soil and form more than 80% of total plant biomass. Terry et al. (1997) suggested that fresh weight of rhizomes can be as high as 40 tons per hectare which indicates strong regeneration potential from rhizomes. Tominga (1993) categorized cogongrass rhizomes into three types; pioneer rhizomes, secondary colonizing rhizomes and tillering rhizomes. Of these three types, the ‘‘pioneer’’ rhizomes are the largest (extend up to 25 cm from parent shoots) and thickest (3 to 4 mm diameter) and are crucial for increase in patch size. Gaffney (1996) observed the presence of apical dominance in cogongrass rhizome and pointed out the inability of lateral buds to send out new shoots. Cogongrass has a wide shoot-to-root ratio ranging from 0.40 to 1.4 (Sajise, 1972; Saxena and Ramakrishnan 1981). It effectively competes with native vegetation for nutrients, light, and water, and cause physical injury by piercing the roots of other plants (Boonitee and Ritdhit, 1984; Eussen and Soerjani, 1975; Jagoe, 1938). It is highly efficient in extracting moisture from shallow soil depths compared with other perennial grasses and is therefore more competitive (Dozzier et al 1998). It also exerts adverse effect on the surrounding

vegetation via allelopathy (Casini et al. 1998; Eussen 1979; Koger and Bryson 2004; Koger et al. 2004). Cogongrass can produce several different phenolic compounds such as caffeic, ferulic, p-hydroxybenzoic, p-coumaric, vanillic, chlorogenic and syringic acids that exert allelopathic action to inhibit neighboring plants' growth (Hussain and Abidi 1991). Phenolic compounds present in foliage, roots, and rhizomes of cogongrass may be responsible for the allelopathic inhibition of germination and seedling development of surrounding plant species (Inderjit and Dakshini, 1991).

1.2.3. Ecological Importance

The increasing invasion of nonnative and invasive species is being perceived as a major threat to the biodiversity and stability of natural ecosystems (Sandlund et al. 1999; Callaway and Aschehoug, 2000; Smith and Knapp, 2001). Adverse impacts of nonnative invasive species in natural ecosystems have been well established. Invasive species have been documented to alter fire regimes (Brooks et al. 2004; Lippincott 2000), nutrient cycling (Ashton et al. 2005; Ehrenfeld 2003; Windham and Ehrenfeld 2003), disturbance regimes (Mack and D'Antonio 1998) and numerous other ecosystem level properties (Gordon 1998; Vitousek and Walker 1989; Vitousek et al. 1987).

Cogongrass invasion has been documented to reduce forest productivity (Daneshgar et al. 2008), alter nutrient availability (Daneshgar and Jose 2009a), and decrease native species biodiversity (Collins et al. 2007). Cogongrass has been reported to reduce available nutrients such as nitrogen and potassium (Collins and Jose 2008), and is recognized as a superior competitor for phosphorus as well (Brewer and Cralle 2003). In addition, Daneshgar and Jose (2009a) documented a 63% reduction in total nitrogen in loblolly pine seedlings grown in

association with cogongrass compared to seedlings grown in association with native understory plants in northwest Florida. Same study reported 39% more storage of nitrogen in below ground rhizome or root biomass of cogongrass compared to native understory species. While cogongrass invasion has been well recognized to decrease vegetation diversity, however, the native species richness does not affect the ability of cogongrass to invade native communities (Collins et al. 2007; Daneshgar and Jose (2009a). Several native and non-native desirable plant species are highly vulnerable to cogongrass invasion. Bahiagrass (*Paspalum notatum* L.), a turf species is very susceptible to cogongrass invasion and is being infested and displaced by cogongrass in Florida (Busey and Myers 1979; Shilling et al. 1997; Watson and Burson 1985; Willard and Shilling 1990). Cogongrass invasion poses a serious threat to endangered species such as the gopher tortoise (*Gopherus polyphemus* Daudin) and Camp Shelby burrowing crayfish (*Fallicambarus gordonii* Itzpatrick) due to habitat displacement. Apprehensions are that the habitats of whitetail deer, turkey, dove, squirrel, quail, and rabbit will be destroyed by vast cogongrass expanses that tend to eliminate their foraging and breeding grounds (Miller 2007). Being a pyrogenic species, cogongrass highly benefits from fire hazards for establishing monoculture stands and thereby, reduces the diversity of the native vegetation (Brewer 2008; Lippincott, 2000). Cogongrass thatch fires can destroy most other aboveground native vegetation which creates huge monoculture expanses, maintains vegetative dominance, creates openings for cogongrass seedling establishment and alters natural ecosystems (Bryson and Carter 1993; Dozier et al. 1998; Eussen and Wirjahardja 1973; Garrity et al. 1996; Lippincott 2000; Shilling et al. 1995; Soerjani and Soemarwoto 1969). Typically, Cogongrass fires are 15 to 20% hotter and more intense than natural fires in pine-based ecosystems in the Southern U.S.

1.2.4. Cogongrass Management

For invasive species management, early detection and eradication are the keys to success than trying to manage large, well-established infestations (DiTomaso 2000; Mullin et al. 2000; Rejma´nek and Pitcairn 2004; Westbrooks 2004). Choice of a management option depends upon a number of factors that include habitat type, nature and extent of infested area, efficacy of available methods, environmental safety and availability of funds etc. Management options often used in developing countries to control cogongrass include slashing, hand-weeding, burning, deep tillage, use of cover crops, and herbicides (Akobundo and Ekeleme 2000; Chikoye et al. 1999; Feuillette et al. 1994; Ivens 1980). Chikoye et al. (1999) indicated that only 3% of farmers use herbicides to control cogongrass in West Africa due to various economical and technical reasons. However, such non-chemical control methods have limited success and cannot provide a sustainable control of cogongrass (Anoka et al. 1991). Further, due to its vigorously growing and extensive rhizome system, cogongrass is highly difficult to manage even with herbicides (Bryson et al. 2007; Dickens and Buchanan 1975; Faircloth et al. 2003; Johnson et al. 1999; Miller 2000; Tominga 1993; Udensi et al. 1999). Additionally, the control may vary depending on the age and rhizome mat density and depth (Faircloth 2004; Johnson 1999).

Mechanical weed management options such as disking and mowing have been shown to temporarily reduce cogongrass biomass, (Faircloth 2004; Gaffney 1996; Johnson 1999b; Willard et al. 1996). However, repeated deep tillage has been reported as an effective method to control cogongrass (Willard et al. 1996). Burning alone tends to promote cogongrass infestation, however, early spring burning is recommended, where feasible, to get rid of dead thatch and

promote maximum growth for herbicide to come into contact with (Johnson 1999a; Lippincott 2000, Yager 2007).

Several biological control options have also been attempted for cogongrass management. A wide variety of natural enemies of cogongrass, including over 80 pathogens, 90 insects, and several nematodes and mites, have been documented world-wide (Van Loan et al. 2002). A gall midge (*Orsioliella javanica* Kieffer), has been reported as a specific pest of cogongrass (Soerjani, 1970; Mangoendiharjo, 1980). Yandoc et al. (1999) discovered an isolate of *Bipolaris sacchari* that causes leaf lesions and severe foliar blighting on cogongrass. Some researchers have suggested that dense and abundant woody vegetation along forest edges serves as biotic resistance to invasion by creating a vegetative barrier to dispersal, growth, or establishment of invasive species (Brothers and Springarn 1992; Cadenasso and Pickett 2001). Yager et al. (2010) observed that clonal growth of cogongrass was reduced in pine forests having dense shrub mid-story and also noticed that cogongrass growth was inversely related to shrub cover.

Chemical control of cogongrass has been shown to be cheaper and more effective (Akobundu 1993; Chikoye et al. 2001). Several herbicides are tested for the control of cogongrass; chemicals such as dalapon, glyphosate, imazapyr, glufosinate, and sulfometuron etc have been reported to give varied control of cogongrass, depending on the rate of application, climate, and soil type (Akobundu 1992; Willard et al. 1997). The most effective herbicides for cogongrass management include glyphosate and imazapyr (Akobundo 1993; Dozier et al. 1998; Udensi et al. 1999). Both glyphosate and imazapyr are systemic herbicides that translocate throughout the plant. However, they differ in their mechanisms-of-action. Imazapyr is an imidazolinone herbicide that works by binding to the acetolactate synthase enzyme, thereby preventing synthesis of the branched-chain amino acids leucine, isoleucine, and valine (Shaner

and O'Connor, 1991). Glyphosate works by binding to the EPSPS enzyme thereby preventing the synthesis of aromatic amino acids tryptophan, tyrosine and phenylalanine (Devine et al. 1993).

Imazapyr has a long soil residual effect and prevent reestablishment of most of the desirable native vegetation which is critical for re-habitation process (Byrd and Bryson 1999; Johnson et al. 1999a). Higher rates of these herbicides provide partial control of cogongrass up to 1 yr after application (Miller 2000), therefore, multiyear applications are required to achieve satisfactory control of cogongrass (Johnson et al. 1999a). Single application of soil sterilant herbicides such as bromacil or diuron + imazapyr has been shown to provide acceptable but expensive cogongrass control (Dickens and Buchanan 1975; Johnson et al. 1999a). Gaffney (1996) evaluated influence of chemical herbicides and two plant species, bermudagrass (*Cynodon dactylon* L.) and hairy indigo (*Indigofera hirsuta* Harvey), in suppressing cogongrass and preventing its re-infestation. Results indicated that the application of herbicides and the presence of the two plant species suppressed cogongrass for up to 2 yr after seeding.

Time of application of herbicide has also been reported to influence the efficacy of herbicides on cogongrass. Fall-season application (September to November) of glyphosate or imazapyr have been shown to be more effective due to better translocation of herbicides to the underground rhizomes since the photosynthates are being channelized towards rhizomes(Faircloth et al. 2005; Johnson 1999a; Miller 2007:Shilling et al. 1997; Tanner et al. 1992). Similar results were obtained with fall applications of herbicides for controlling other weed species including leafy spurge (*Euphorbia esula* L.) (Lym and Messersmith, 1990), redvine (*Brunnichia ovate* L.) (Shaw and Mack, 1991), and torpedograss (*Panicum repens* L.) (Smith et al.1993).

1.2.5. Cogongrass Eradication

The term ‘eradication’ is often misinterpreted, especially by public policy makers; it is not synonymous with the term ‘control’ (Zamora at al.1989). Eradication is broadly defined as the destruction of every propagule of a species from an area surrounded by natural or manmade barriers sufficiently effective to prevent reinvasion except at man’s intervention (Newsom 1978, Zamora et al. 1989). Bomford and O’Brien (1995) define it as ‘complete and permanent elimination of all wild populations from a defined area by a time-limited campaign’. Despite the existence of evidence of successful eradication of several species, there is still a widespread view that eradication is not feasible. The fundamental reason for this view may be reverse hype, i.e., unsuccessful events are always much touted and widely criticized while the success stories are often under reported (Simberloff 2009). In the past, several invasive invertebrate species have been eradicated successfully (Klassen and Curtis 2005; Simberloff 2002a; Wittenberg and Cock 2001). However, the eradication of invasive plants is often regarded as impossible owing to the existence of a soil seed bank for many species (Rejmanek and Pitcairn 2002). Contrarily, several weed eradication projects have been successful; eradication of sandbur (*Cenchrus echinatus*) from Laysan (Flint and Rehkemper 2002) and Bassia scoparia (*Kochia scoparia* L.) from a large area in Western Australia (Randall 2001). Several small scale eradications have also been cited by Mack and Lonsdale (2002) in Australia, New Zealand, and the United States. The success of an eradication project is largely governed by five crucial features: early detection and prompt action, knowledge of biology and ecology of the target species, a blanket eradication approach, unflagging motivation and availability of resources (Myers et al. 2000; Mack and Lonsdale 2002; Simberloff 2002b; Mack and Foster 2004). Successful eradication of the Pacific alga *Caulerpa*

taxifolia from southern California (Merkel & Associates. 2006) is an outstanding example of the significance of quick and decisive action; whereas, the failure to eradicate this species in Europe owes to the lack of timely and decisive action (Meinesz 2001). Although early detection and prompt action are not absolutely necessary, the eradication of populations confined to a small area is much more cost-effective and may have a high probability of success (Simberloff 2009; Soria et al. 2002). Eradication of witchweed (*Striga asiatica* M. Vahl) is an example of a large scale campaign against a long established and widespread invasion, which has cost over 100 million dollars to date (Eplee 2001). As a result of this mega scale effort, witchweed infestations in the United States have been reduced from 200,000 ha in the early 1970s (Eplee 2001) to approximately 900 ha by the end of 2007. Clearly, the costs and rigors of invasive species eradication can be tremendous. However, invasive weed management decisions cannot be based solely upon benefit cost analyses of agricultural and forestry production areas. Therefore, if eradication is feasible, it should be attempted since it may be more cost-effective than any other control method (Wittenberg and Cock 2001). Bomford and O'Brien (1995) determined six criteria for assessing whether eradication is technically possible and preferable to continuing control. These six criteria include: 1) Rate of removal exceeds rate of increase at all population densities; 2) Immigration is zero; 3) All reproductive individuals must be at risk; 4) Target species can be detected at low densities; 5) Discounted benefit-cost analysis favors eradication over control and; 6) There must be a suitable socio-political environment.

Often a management plan is specifically aimed at eradication of a species, but the methods are the same as those that would be used to reduce a population to an economically or ecologically acceptable level (Simberloff 1997). Recently, Alabama spent almost 6.3 million dollars on control and localized eradication of cogongrass infestations (Schelhas 2011). In the

context of cogongrass, the term eradication specifically insinuates complete elimination of all living propagules, both sexual (seed) as well as asexual (rhizomes) within a defined boundary. The current cogongrass eradication approach largely resembles the wild fire management paradigm which encompasses prevention, early detection, quick initial attack, integrated control and site rehabilitation (Dewey et al. 1995). Similar to wildfires, new invasive weed infestations frequently arise as a single plant or small patch followed by a slow growth lag phase which ultimately lead to exponential spread. The movement of seed or rhizomes by various dispersal agents exacerbates the problem as new 'spot fire' patches establish beyond the main infestation.

Since the goals of eradication are much more rigid than weed control, eradication treatments must have extremely high efficacy (Zamora et al. 1989). Historically, several chemical and non-chemical methods have been tested for the control of cogongrass (Akobundu 1992, 1993; Akobundu et al. 2000; Byrd and Bryson 1999; Dickens and Buchanan 1975; Gaffney 1996; Johnson et al. 1999; MacDonald et al. 2001 and 2002; Miller 2000; Ramsey et al. 2003; Terry et al. 1997; Willard et al. 1996 and 1997). Glyphosate and imazapyr have been identified as the most effective herbicides for cogongrass management (Dozier et al. 1998; Udensi et al. 1999). Repeated applications of these herbicides over multiple years have been reported to provide greater than 95% control (Miller 2007). Fall-season applications (September to November) of glyphosate or imazapyr have been shown to be more effective due to better translocation of herbicides to the underground rhizomes as photosynthates are directed towards rhizomes in the fall (Faircloth et al. 2005; Johnson 1999 and 2000; Miller 2007; Shilling et al. 1997). However, the focus of all of these previous research efforts was only to attain satisfactory control of cogongrass, not eradication. To date, no research has been published on methods suitable for the eradication of cogongrass patches. Since cogongrass seeds are very short lived in

the environment (MacDonald 2004), eliminating the dense, rhizome mat is very likely the limiting factor for successful eradication in most situations. Previous researchers have observed low spikelet fill (0 to 40 %), short seed viability (< 16 months), and poor seedling survival (Sajise, 1972; Santiago, 1974; Dickens, 1976; Santiago, 1980; Kushwaha et al. 1983; Hopkins and Graham, 1984). Additionally, glyphosate and imazapyr herbicides reduce cogongrass cover and seedhead production more than 80 % and 97 %, respectively, up to a year following treatment (Enloe et al. 2012). Byrd (2007) reported reduction in number of viable seeds following treatment with several herbicides. In light of these views, we assumed non-existence of a viable cogongrass seed bank in the treated plots. Therefore, our primary objective in this study was to determine the feasibility of cogongrass patch eradication while targeting elimination of the entire rhizome layer. Our overall research questions were as follows:

1) Can glyphosate and/or imazapyr be effective for cogongrass patch eradication?

2) Does the combination of glyphosate and imazapyr improve cogongrass control over either herbicide alone?

3) Does the timing of repeated herbicide applications (spring, summer, or fall) influence the effectiveness of glyphosate and imazapyr for cogongrass patch eradication?

4) Do repeated herbicide treatments influence energy levels or depth of surviving rhizomes?

1.2.6. Literature Cited

Akobundu, I. O. 1992. Integrated weed management techniques to reduce soil degradation. Proc. Intern. Weed Cont. Cong. Melbourne, Australia 1:278-288.

Akobundu, I. O. 1993. Chemical control of cogongrass (*Imperata cylindrica* (L.) Raeuschel) in arable fields. Proc. Weed Sci. Soc. Am. 33:6.

Akobundo, I. O. and C. W. Agyakwa. 1998. A Handbook of West African Weeds. 2nd ed. Ibadan, Nigeria: International Institute of Tropical Agriculture. 496 p.

Akobundu, I.O. and F.E. Ekeleme. 2000. Effect of method of *Imperata cylindrica* management on maize grain yield in the derived savanna of south-western Nigeria. Weed Res. (Oxford) 40:335-341.

Akobundu, I.O., U.E. Udensi, and D. Chikoye. 2000. Velvetbean (*Mucuna spp*.) suppresses speargrass (*Imperata cylindrica* (L.) Raeuschel) and increases maize yield. Int. J. Pest Manage. 46:103-108.

Amy, A.C. 1932. Variations in the organic reserves in underground parts of five perennial weeds from late April to November. Minn. Agr. Exp. Sta. Tech. Bull. 84

Anoka, U.A., I.O. Akobundu and S.N.C. Okonkwo. 1991. Effect of *Gliricidia sepium* (Jacq.) Steud. and *Leucaena leucocephala* (Lam.) deWit on growth and development of *Imperata cylindrica* (L.) Raeuschel. Agroforest. Syst. 16:1-12.

Ashton, I.W., L. A. Hyatt, K.M. Howe, J. Gurevitch and M. T. Lerdau. 2005. Invasive species accelerate decomposition and litter nitrogen loss in a mixed deciduous forest. Ecological Appl. 15: 1263–1272.

Ayeni, A.O. 1985. Observations on the vegetative growth patterns of speargrass [*Imperata cylindrica* (L.) Beauv.]. Agr. Ecosyst. Environ. 13:301-307.

Becker, R. L. and R. S. Fawcett. 1998. Seasonal carbohydrate fluctuations in hemp dogbane (*Apocynum cannabinum*) crown roots. Weed Sci. 46: 358–365.

Bomford, M. and P. O'Brien. 1995. Eradication or control for vertebrate pests? Wildland Soc. Bull. 23:249–255.

Boonitee, A. and P. Ritdhit. 1984. Allelopathic effects of some weeds on mungbean plants (*Vigna radiata*). Proc. of the 1st Tropical Weed Sci. Conference, Hat Yai, Songkla, Thailand 2:401-406.

Brewer J. S. and S.P. Cralle. 2003. Phosphorus addition reduces invasion of longleaf pine savanna (Southeastern USA) by an non-indigenous grass (*Imperata cylindrica*). Plant Ecol. 167:237-245.

Brewer, S. 2008. Declines in plant species richness and endemic plant species in longleaf pine savannas invaded by *Imperata cylindrica*. Biol. Invas. 10:1256- 1264.

Brooks, M. L., C. M. D'antonio, D. M. Richardson, J. B. Grace,J. E. Keeley, J. M. Ditomaso, R. J. Hobbs, M. Pellant, and D. Pyke. 2004. Effects of invasive alien plants on fire regimes. BioScience. 54: 677–688.

Brothers, T.S. and A. Springarn. 1992. Forest fragmentation and alien plant invasion of central Indiana old-growth forests, Conserv. Biol. 6: 91-100.

Brown, D. 1944. Anatomy and reproduction in *Imperata cylindrica*. Joint Publ. 7:15–18. UK: Imperial Agricultural Bureaux. 66 p.

Bryson, C.T., and R. Carter. 1993. Cogongrass, *Imperata cylindrica*, in the United States. Weed Technol. 7:1005- 1009.

Bryson, C.T., C.H. Koger, and J. Byrd. 2007. Effects of Temperature and Exposure Period to Heat on Cogongrass (*Imperata Cylindrica*) Viability. Weed Technology 21:141-144.

Bryson, C.T., L. J. Krutz, G. N. Ervin, K.N. Reddy, and J. D. Byrd, Jr. 2010. Ecotype variability and edaphic characteristics for cogongrass (*Imperata cylindrica*) populations in Mississippi. Invasive Plant Sci. Manag. 3:199–207.

Busey, P. and B. J. Myers. 1979. Growth rates of turfgrass propagated vegetatively. Agron. J. 71:817–821.

Byrd, J. D., Jr. and C. T. Bryson. 1999. Biology, ecology, and control of cogongrass [*Imperata cylindrica* (L.) Beauv.]. Mississippi State, MS: Bureau of Plant Industry, Mississippi Department of Agriculture and Commerce Fact Sheet 1999-01. 2 p.

Byrd, J. D., Jr. and C. T. Bryson. 1999. Biology, ecology, and control of cogongrass [*Imperata cylindrica* (L.) Beauv.]. Mississippi State, MS: Bureau of Plant Industry, Mississippi Department of Agriculture and Commerce Fact Sheet 1999-01. 2 p.

Cadenasso, M.L. and S.T.A. Pickett 2001. Effect of edge structure on the flux of species into forest interiors. Conserv. Biol. 15:91-97.

Callaway, R.M. and E.T. Aschehoug. 2000. Invasive plants versus their new and old neighbors: a mechanism for exotic invasion. *Sci.* 290 521-523.

Capo-chichi, L. J. A., W. H. Faircloth, A. G. Williamson, M. G. Patterson, J. H. Miller, and E. van Santen. 2008. Invasion dynamics and genotypic diversity of cogongrass (*Imperata*

cylindrica) at the point introduction in the southeastern United States. Invasive Plant Sci. Manag. 1:133–141.

Casini, P., V. Vecchio, and I. Tamantini. 1998. Allelopathic interference of itchgrass and cogongrass: Germination and early development of rice. Trop. Agric 75:445–451.

Chikoye, D., F. Ekeleme and U. Udensi. 2001. Cogongrass suppression by intercropping cover crops in corn/cassava systems. Weed Sci. 49:658-667.

Chikoye, D., F. Ekeleme, and J. T. Ambe. 1999. Survey of distribution and farmers' perceptions of speargrass [*Imperata cylindrica* (L.) Raeuschel] in cassava-based systems in West Africa. Int. J. Pest Manag. 45:305–311.

Chikoye, D., U.E. Udensi, and S. Ogunyemi. 2005. Integrated management of cogongrass [*Imperata cylindrica* (L.) Rauesch.] in corn using tillage, glyphosate, row spacing, cultivar and cover cropping. Agron. J. 97:1164-1171.

Chikoye, D., V.M. Manyong, and F. Ekeleme. 2000. Characteristics of speargrass (*Imperata cylindrica*) dominated fields in West Africa: crops, soil properties, farmer perceptions and management strategies. Crop Prot. 19:481-487.

Coile, N. C. and D. G. Shilling. 1993. Cogongrass, (*Imperata cylindrical* (L.) Beauv.): A Good Grass Gone Bad! Gainesville, FL: Florida Department of Agriculture and Consumer Services, Division of Plant Industry, Botany Circular, Volume 28. 3 p.

Collins, A. R. and S. Jose. 2008. Cogongrass invasion alters soil chemical properties of natural and planted forestlands in the southeastern United States. In Invasive Plants and Forest Eco-systems, Kohli R., S. Jose, D. Batish. and H. Singh (eds.). CRCPress, Boca Raton, FL.

Collins, A.R., Jose, S., Dansehgar, P. and Ramsey, C. 2007. Elton's hypothesis revisited: *Imperata cylindrica* invasion in the southeastern United States. Biological Invasions 9:433-443.

Daneshager, P. and Jose, S. 2009. *Imperata cylindrica*, an alien invasive grass, maintains control over nitrogen availability in an establishing pine forest. Plant Soil. 320: 209-218.

Daneshgar, P., Jose, S., Collins, A., and Ramsey, C. 2008. Cogongrass (*Imperata cylindrica*), an alien invasive grass, reduces survival and productivity of an establishing pine forest. Forest Sci. 54: 579-586.

Devine, M., S. O. Duke, and C. Fedtke. 1993. Inhibition of amino acid biosynthesis. Pages 251-294 in Physiology of Herbicide Action. P T R Prentice Hall, New Jersey.

Dewey, S.A., Jenkins, M.J., Tonioli, R.C., 1995. Wildfire suppression—a paradigm for noxious weed management. Weed Technol. 9: 621–627.

Dickens, R. 1974. Cogongrass in Alabama after sixty years. Weed Sci. 22:177-179.

Dickens, R. and G. A. Buchanan. 1975. Control of cogongrass with herbicides. Weed Sci. 23:194-197.

Dickens, R. and G. M. Moore. 1974. Effects of light, temperature, KNO3, and storage on germination of cogongrass. Agron. J. 66:187-188.

DiTomaso, J. M. 2000. Invasive weeds in rangelands: species, impacts, and management. Weed Sci. 48:255–265.

Dozier, H., J. H. Gaffney, S. K. McDonald, E.R.R.L. Johnson, and D. G. Shilling.1998. Cogongrass in the United States: history, ecology, impacts, and management. Weed Technol. 12:737-743.

Ehrenfeld, J.G. 2003. Effects of exotic plant invasions on soil nutrient cycling processes. Ecosystems 6: 503–523.

Enloe, S. F., J. Belcher, N. Loewenstein, J. S. Aulakh and E. van Santen. 2012. Cogongrass control with aminocyclopyrachlor in pastures. Forage and Grazinglands. doi:10.1094/FG-2012-0828-02-RS.

Eplee, R.E. 2001. Coordination of witchweed eradication in the USA. In: Wittenberg R, Cock MJW (eds.) Invasive alien species: a toolkit of best prevention and management practices. CAB International, Wallingford, p 36.

Eussen, J. H. H. 1979. Some competition experiments with alang-alang [*Imperata cylindrica* (L.) Beauv.] in replacement series. Oecologia 40:351–356.

Eussen, J. H. H. 1980. Biological and ecological aspects of alang–alang [*Imperata cylindrica* (L.) Beauv.]. In: Proceedings of BIOTROP workshop on alang–alang in Bogor, 27–29 July 1976. Biotropica Special Pub. No. 5 Bogor, Indonesia, pp 15–22.

Eussen, J. H. H. and M. Soerjani. 1975. Problems and control of alang-alang [*Imperata cylindrica* (L.) Beauv.] in Indonesia. Proc. of 5th Ann. Conf. of Asian-Pacific Weed Sci. Soc. 5:58-64.

Eussen, J. H. H. and S. Wirjahardja. 1973. Studies of an alang-alang, *Imperata cylindrica* (L.) Beauv. vegetation. Biotropica Bull. No. 6, 24 pp.

Faircloth, W. H. 2004. Integrated vegetation management of cogongrass in Alabama. Ph.D. Dissertation, Auburn University. 156 p.

Faircloth, W. H., M. G. Patterson, J. H. Miller, and D. H. Teem. 2005. Wanted dead or alive: cogongrass. Auburn, AL: Alabama Cooperative Extension Publication ANR-1241. 4 p.

Faircloth, W. H., M.G. Patterson, D.H. Teem and J. H. Miller. 2003. Cogongrass (*Imperata cylindrica*): Management tactics on rights-of-way. Proc. South. Weed Sci. Soc. 55:162.

Falvey, J. L. 1981. *Imperata cylindrica* and animal production in southeastern Asia: a review. Trop. Grassl. 15:52–56.

Feuillette, B., P. Marnotte, and T. Le-Bourgeois. 1994. The control of *Imperata cylindrica*. Agric. Dev. 3:47–48.

Flint, E. and C. Rehkemper. 2002. Control and eradication of the introduced grass, *Cenchrus echinatus*, at Laysan Island, Central Pacific Ocean. Pages 110–115 in C. R. Veitch and M. N. Clout, eds. Turning the Tide: The Eradication of Invasive Species. Auckland, New Zealand: Invasive Species Specialist Group of the World Conservation Union (IUCN).

Gabel, M.L. 1982. A biosystematic study of the genus Imperata (Gramineae: Andropogoneae). Ph.D. dissertation. Iowa State University, Ames, Iowa USA. 94 p.

Gaffney, J.F. 1996. Ecophysiological and technical factors influencing the management of cogongrass (*Imperata cylindrica*). Ph.D. dissertation, University of Florida, Gainesville, FL, USA. 111 p.

Garrity, D.P. (Ed.). 1997. Special Issue: Agroforestry innovations for *Imperata* grassland rehabilitation. Agroforestry Syst. 36:1-284.

Garrity, D.P. D.M. Kummer and E.S. Guiang. 1993. Country Profile: The Philippines, In: Sustainable Agriculture and the Environment in the Humid Tropics. National Academy of Science, Washington, DC.

Garrity, D.P., M. Soekardi, M. Van Noordwijk, R. De La Cruz, P.S. Pathak, H.P.M. Gunasena, N. Van So, G. Huijun, and N.M.Majid. 1996. The *Imperata* grasslands of tropical Asia: area, distribution, and typology. Agroforest. Syst. 36:3-29.

Gordon, D. R. 1998. Effects on invasive, non-indigenous plant species on ecosystem processes: Lessons from Florida. Ecological Monographs 8: 975-989.

Greenfield, S.B. and D. Smith. 1974. Diurnal variations of nonstructural carbohydrates in the individual parts of switchgrass shoots at anthesis. J. Range Mgmt. 27:466-469.

Harding, V. J. and Downs, C. E. 1933. Notes on a Shaffer-Somogyi copper reagent. J. biol. Chem. IOI, 487.

Hodgson, J. M. 1968. The nature, ecology, and control of Canada thistle. United States Department of Agriculture, Technical Bulletin 1386, 32 pp.

Holm, L. G., D. L. Pucknett, J. B. Pancho, and J. P. Herberger. 1977. The World's Worst Weeds. Distribution and Biology. Univ. Press of Hawaii, Honolulu, HI. 609 p.

Hubbard, C. E. 1944. *Imperata cylindrica*. Taxonomy, Distribution, Economic Significance, and Control. Imp. Agric. Bur. Joint Publ. No. 7, Imperial Bureau Pastures and Forage Crops, Aberystwyth, Wales. Great Britton. 53 p.

Hussain, F. and N. Abidi. 1991. Allelopathy exhibited by *Imperata cylindrica* (L) Beauv. Pak. J. Bot. 23:15–25.

Inderjit and K. M. M. Dakshini. 1991. Investigations on some aspects of chemical ecology of cogongrass, *Imperata cylindrica* (L.) Beauv. J. Chem. Ecol. 17:343–352.

Ivens, G.W. 1980. *Imperata cylindrica* (L.) Beauv. in West African agriculture. In: Proceedings of BIOTROP workshop on alang-alang in Bogor, 27-29 July 1976. pp. 149-156. Biotropica Special Pub. No. 5 Bogor, Indonesia.

Jagoe, B. B. 1938. The effect of alang alang grass (*Imperata cylindrica*) on the growth of coconut palms. Malay. Agr. J., 26:369-376.

Johnson, E. R. R. L. 1999a. Management of the non-native weed cogongrass (*Imperata cylindrica* (L.) Beauv.) utilizing an integrated management program. M.S. Thesis, the University of Florida. 87 p.

Johnson, E.R.R.L., D.G. Shilling, G.E. MacDonald, J.F. Gaffney and B.J. Brecke. 2000. Time of year, rate of herbicide application, and revegetation: Factors that influence the control of cogongrass [*Imperata cylindrica* (L.) Beauv.] control. Proc. South. Weed Sci. Soc. 53:70.

Johnson, E.R.R.L., J.F. Gaffney and D.G. Shilling. 1999. The influence of discing on the efficacy of imazapyr for cogongrass [*Imperata cylindrica* (L.) Beauv.] control. Proc. South. Weed Sci. Soc. 52:165.

Jose, S., J. Cox, D. L. Miller, D.G. Shilling and S. Merritt. 2002. Alien plant invasions: The story of cogongrass in southeastern forests. J. Forest. 100:41-44.

King S. M., J. B. Grace. 2000. The effects of gap size and disturbance type on invasion of wet pine savanna by cogongrass, *Imperata cylindrica* (Poaceae). Am. J. Bot. 87:1279–1286.

Klassen, W., Curtis, C. F.2005. History of the sterile insect technique. In Dyck V.A., Hendrichs J., Robinson A.S. (eds) Sterile insect technique. Principles and practice in area-wide integrated pest management. Springer, Dordrecht, The Netherlands. pp 3–36

Koger, C. H., C. T. Bryson, and J. D. Byrd. 2004. Response of selected grass and broadleaf species to cogongrass (*Imperata cylindrica*) residues. Weed Technol. 18:353–357.

Koger, C.H. and C.T. Bryson. 2003. Effect of cogongrass (*Imperata cylindrica*) residues on bermudagrass (*Cynodon dactylon*) and Italian ryegrass (*Lolium multiflorum*). Proc. South. Weed Sci. Soc. 56:341.

Kushwaha, S.P.S., P. S. Chancellor, and R. S. Tripathi. 1983. Population dynamics of *Imperata cylindrica* var. major related to slash and burn agriculture Jhum in northeastern India. Proc. Indian Acad. Sci. Plant Sci. 92:313-322.

Lippincott, C.L. 2000. Effects of *Imperata cylindrica* (L.) Beauv. (cogongrass) invasion on fire regime in Florida sandhill. Nat. Area. J. 20:140-149.

Lym, R.G., and C.G. Messersmith. 1990. Effect of temperature on picloram absorption and translocation in leafy spurge (*Euphorbia esula*). Weed Sci. 38:471-474.

MacDonald, G.E. 2004. Cogongrass (*Imperata cylindrica*)— Biology, Ecology, and Management. Critical Reviews in Plant Sciences. 23:367-380.

MacDonald, G.E., D.G. Shilling, J. Meeker, R. Charudattan, M. Minno, A. VanLoan, J. DeValerio, C. Yandoc and E.R.R.L. Johnson. 2001. Integrated management of non-native invasive plants in southeastern pine forest ecosystems - cogongrass as a model system. Final Report. USDA Forest Service, Forest Health Technologies. 50 pp.

MacDonald, G.E., E.R.R.L. Johnson, D.G. Shilling, D.L. Miller and B.J. Brecke. 2002. The use of imazapyr and imazapic for cogongrass [*Imperata cylindrica* (L.) Beauv.] control. Proc. South. Weed Sci. Soc. 55:110.

Mack M. C. and C.M. D'Antonio. 1998. Impacts of biological invasions on disturbance regimes. Trends in Ecol. and Evol.13: 195–198.

Mack, R. N. and W. M. Lonsdale. 2002. Eradicating invasive plants: Hard-won lessons for islands. In: Veitch, C. R. and Clout, M. N. (eds.). Turning the tide: the eradication of island invasives, pp. 164-172. IUCN SSC Invasive Species Specialist Group. IUCN, Gland, Switzerland and Cambridge, UK.

Mack, R.N. and S.K. Foster. 2004. Eradication or control? Combating plants through a lump sum payment or on the installment plan. In: Sindel BM, Johnson SB (eds) Proceedings of the fourteenth Australian weeds conference. Weed Science Society of New South Wales, Sydney, pp 55–61

Mangoendiharjo, S. 1980. Some notes on the natural enemies of alang-alang (*Imperata cylindrica*) in Java. In: Proceedings of BIOTROP workshop on alang-alang in Bogor, 27-29 July 1976. pp. 47-55. Biotropica Special Pub. No. 5 Bogor, Indonesia.

Meinesz, A. 2001. Killer algae, 2nd edn. University of Chicago Press, Chicago.

Merkel & Associates, Inc. 2006. Final report on the eradication of the invasive seaweed *Caulerpa taxifolia* from Agua Hedionda Lagoon and Huntington Harbour, California.Southern California Caulerpa Action Team, San Diego.

Miller, J. H. 2007. What research has found about establishing loblolly pines in cogongrass infestations. In Loewenstein, N. J., and J. H. Miller, (eds) Proceedings of the regional

cogongrass conference: a cogongrass management guide. Alabama Cooperative Extension System, Auburn University, Mobile, AL, pp 43–47.

Miller, J.H. 2000. Refining rates and treatment sequences for cogongrass (*Imperata cylindrica*) control with imazapyr and glyphosate. Proc. South. Weed Sci. Soc. 53:131-132.

Minogue, J. P., J. H. Miller, D. K. Lauer. 2012. Use of glyphosate and imazapyr for Cogongrass (*Imperata cylindrica*) management in southern pine forests. South. J. Appl. For. 36: 19-25.

Mooney H.A. and R.J. Hobbs (eds) 2000. Invasive species in a changing world. Island Press, Washington, D.C., U.S.A.

Mullin, B. H., L.W.J. Anderson, J. M. DiTomaso, R. E. Eplee, and K. D. Getsinger. 2000. Invasive plant species. Ames, IA: Council for Agricultural Science and Technology Issue Paper 13.

Myers, J. H., D. Simberloff, A. M. Kuris, J. R. Carey. 2000. Eradication revisited: dealing with exotic species. Trends Ecol. Evol. 15:316–320.

Newsom, L. D. 1978. Eradication of plant pests-con. Bull. Entomol. Soc. Am. 24:35-40.

Omezine, A. and Fethia Skiri-Harzalla.(2009). Regenerative capacity of speargrass (*Imperata cylindrica* (L.) P. Beauv. Pak. J. Weed. Sci. Res. 15:53-69.

Patterson, D.T. and C.G. McWhorter. 1980. Distribution and control of cogongrass (*Imperata cylindrica*) in Mississippi. Proc. South. Weed Sci. Soc. 33:251.

Patterson, D.T., E.E. Terrell and R. Dickens. 1983. Cogongrass in Mississippi. Miss. Agric. For. Exp. Stn. Res. Rep. 46:1-3.

Potter, R. L., J. L. Petersen, and D.N. Ueckert. 1986. Seasonal trends of total nonstructural carbohydrates in Lindheimer pricklypear (*Opuntia lindheimeri*). Weed Sci. 34: 361-365.

Ramsey, C.L., S. Jose, D.L. Miller, J. Cox, K.M. Portier, D.G. Shilling and S. Merritt. 2003. Cogongrass [*Imperata cylindrica* (L.) Beauv.] response to herbicides and disking on a cutover site in a mid-rotation pine plantation in southern USA. Forest Ecol. Manage. 179:195-209.

Randall, R. 2001. Eradication of a deliberately introduced plant found to be invasive. In: Wittenberg R, Cock, M.J.W. (eds) 2001. Invasive alien species: a toolkit of best prevention and management practices. CAB International Wallingford, Oxon, p 174.

Reineke, L.H. 1933.Perfecting a stand-density index for even-aged forests. J. Agric. Res. 46:627-638.

Rejma´nek, M. and M. J. Pitcairn. 2002. When is eradication of exotic pest plants a realistic goal? Pages 249–253 in C. R. Veitch and M. N. Clout eds. Turning the Tide: The Eradication of Invasive Species. Auckland, New Zealand: Invasive Species Specialist Group of the World Conservation Union (IUCN).

Rejma´nek, M. and M. J. Pitcairn. 2004. When is eradication of exotic pests a realistic goal? Pages 249–253 in C. R. Veitch and M. N. Clout, eds. Turning the tide: the eradication of invasive species. Gland, Switzerland and Cambridge, UK: SSC Invasive Species Specialist Group, International Union for the Conservation of Nature.

Rejmánek, M. 1996. Species richness and resistance to invasions. In: Diversity and Processes in Tropical Forest Ecosystems. (eds G.H. Orians, R. Dirzo & J.H. Cushman), pp. 153–72.

Sajise, P.E. 1972. Evaluation of Cogon (*Imperata cylindrica* L.) Beauv.) as a Seral Stage in Philippine Vegetational Succession: I. The Cogonal Seral Stage and Plant Succession. II. Autecological Studies on Cogon. Ph.D. dissertation. Cornell University, Ithaca, NY.

Sajise, P.E. 1976. Evaluation of cogon [*Imperata cylindrica*(L.) Beauv.] as a serial stage in Philippine vegetational succession. 1. The cogonal seral stage and plant succession. 2. Autecological studies on cogon. Dissertation Abstracts International B (1973) 3040-3041. from: Weed Abstracts 1976, No. 1339.

Saldivar, A. J., W. R. Ocumpaugh, R. R. Gildersleeve, and G. M. Prine. 1992. Total nonstructural carbohydrates and nitrogen of 'Florigraze' rhizoma peanut. Agron. J. 84:439-44.

Sandlund O. T., Schei P. J. & Vilken A., eds. 1999. Invasive Species and Biodiversity Management. Kluwer, Dordrecht.

Saxena, K. G. and P. S. Ramakrishnan. 1983. Growth and allocation strategies of some perennial weeds of slash and burn agricutlure (Jhum) in northerastern India. Can. J. Bot. 61: 1300-1306.

Schelhas, J. 2011. A socioeconomic assessment of Forest Service Recovery Act projects: the Alabama Cogongrass Control Center. General Technical Report - Pacific Northwest Research Station, USDA Forest Service 2011 No. PNW-GTR-831 pp. 5-22.

Shaner, D. L. and S. L. O'Connor. 1991. Absorption and translocation of imidazilinone herbicides. pp. 53-69. In M. Baker (ed.) The Imidazilinone Herbicides. CRC Press, Boca Raton, FL.

Shaw D.R. and R.E. Mack. 1991. Application timing of herbicides for the control of redvine (*Brunnichia ovata*).Weed Technol. 5, 125–129.

Shilling, D.G., T.A. Bewick, J.F. Gaffney, S.K. McDonald, C.A. Chase, and E.R.R.L. Johnson. 1997. Ecology, Physiology, and Management of Cogongrass (*Imperata cylindrica*). Final Report. Florida Institute of Phosphate Research. 128 p.

Simberloff, D. 2002a. Today Tiritiri Matangi, tomorrow the world! —are we aiming too low in invasives control? In: Veitch CR, Clout MN (eds.) Turning the tide: the eradication of invasive species. IUCN Species Survival Commission, Gland, pp 4–12.

Simberloff, D. 2002b. Why not eradication? In: Rapport D.J, Lasley W.L, Ralston D.E, Nielsen N.O, Qualset C.O, Damania A.B (eds) Managing for healthy ecosystems. CRC/ Lewis Press, Boca Raton, pp 541–548.

Simberloff, D. 2009. We can eliminate invasions or live with them. Successful management projects. Biol. Invas. 11: 149-157.

Smith, B. E., D. G. Shilling, W. T. Haller, and G. E. MacDonald. 1993. Factors influencing the efficacy of glyphosate on torpedograss (*Panicum repens* L.). J. Aquat. Plant Manage. 31:199-202.

Smith, M. D. and Knapp, A. K. 2001. Size of the local species pool determines invasibility of a C_4-dominated grassland. Oikos 92: 55-61.

Soerjani, M. 1970. Alang-alang *Imperata cylindrica* (L.) Beav., pattern of growth as related to its problem of control. Biol. Trop. Bull. 1:88-96.

Soria, M., Gardener, M.R., and Tye, A. 2002. Eradication of potentially invasive plants with limited distributions in the Galapagos islands. In Veitch, C.R. and Clout, M.N. (eds.).

Turning the tide: the eradication of invasive species, pp 287–292. IUCN SSC invasive species specialist group. IUCN, Gland, Switzerland and Cambridge, UK.

Springer-Verlag, Berlin.

Stamm Katovich, E. J., R. L. Becker, and B. D. Kinkaid. 1996. Influence of nontarget neighbors and spray volume on retention and efficacy of triclopyr in purple loosestrife (*Lythrum salicaria*). Weed Sci. 44:143–147.

Starfinger, U., Edwards, K., Kowarik, I., Williamson, M., 1998. Plant Invasions: Ecological Mechanisms and Human Responses. Backhuys Publishers, Leiden.

Suryatna, E.S. and J.L. McIntosh. 1980. Food crops production and control of *Imperata cylindrica* (L.) Beauv. on small farms, pp. 135-147. In: Proceedings of BIOTROP workshop on alang-alang in Bogor, 27-29 July 1976. pp. 15- 22. Biotropica Special Pub. No. 5 Bogor, Indonesia.

Tabor, P. 1949. Cogongrass, *Imperata cylindrica* (L.) Beauv., in the southeastern United States. Agron. J. 41:270.

Tabor, P. 1952. Comments on cogon and torpedograsses: A challenge to weed workers. Weeds. 1:374-375.

Tanner, G.W., J. M. Wood, and S.A. Jones. 1992. Cogongrass (*Imperata cylindrica*) control with glyphosate. Florida Scient. 55:112-115.

Terry, P.J., G. Adjers, I.O. Akobundu, A.U. Anoka, M.E. Drilling, S. Tjitrosemito, and M. Utomo. 1997. Herbicides and mechanical control of *Imperata cylindrica* as a first step in grassland rehabilitation. Agroforest. Syst. 36: 151-179.

Tominaga, T. 1993. Rhizome systems and sprouting pattern of shoots in *Imperata cylindrica*. Jpn. J. Trop. Agric 37:120–123.

Udensi, U.E., I.O. Akobundu, A.O. Ayeni, and D. Chikoye. 1999. Management of cogongrass (*Imperata cylindrica*) with velvetbean (*Mucuna pruriens* var. utilis) and herbicides. Weed Technol. 13:201-208.

Van Loan, A.N., J.R. Meeker and M.C. Minno. 2002. Cogongrass, In: Biological control of invasive plants in the eastern United States, 413 p., Van Driesche, R., Ed., USDA Forest Service Publication FHTET-2002-04.

Vergara, R., M. C. Minno, M. Minno, S. E. Soltis, and P. S. Soltis. 2008. Preliminary study using ISSRs to differentiate Imperata taxa (Poaceae: Andropogoneae) growing in the US. Southeast. Nat. 7: 267–276.

Vitousek, P.M. and L.R. Walker. 1989. Biological invasion by *Myrica faya* in Hawai'i: plant demography, nitrogen fixation, ecosystem effects. Ecological Monographs 59, 247–265.

Vitousek, P.M., Walker, L.R., Whiteaker, L.D., Mueller-Dombois, D., Matson, P.A., 1987. Biological invasion by *Myrica faya* alters ecosystem development in Hawaii. Science 238, 802–804.

Watson, V. H. and B. L. Burson. 1985. Bahiagrass, Carpetgrass, and Dallisgrass in Forages: The Science of Grassland Agriculture. Ames: Iowa State University Press. Pp. 255–257.

Westbrooks, R. G. 2004. New approaches for early detection and rapid response to invasive plants in the United States. Weed Technol. 18:1468–1471.

Wilcut, J. W., B. Truelove, D. E. Davis, and J. C. Williams. 1988b. Temperature factors limiting the spread of cogongrass (*Imperata cylindrica*) and torpedograss (*Panicum repens*). Weed Sci. 36:49-55.

Wilcut, J. W., R. R. Dute, B. Truelove, and D. E. Davis. 1988a. Factors limiting distribution of cogongrass, *Imperata cylindrica*, and torpedograss, *Panicum repens*. Weed Sci. 36:577-582.

Willard, T. R. and D. G. Shilling. 1990. The influence of growth stage and mowing on competition between *Paspalum notatum* and *Imperata cylindrica*. Trop. Grassl. 24:81–86.

Willard, T. R., D. W. Hall, D. G. Shilling, J. A. Lewis, and W. L. Currey. 1990. Cogongrass (*Imperata cylindrica*) distribution on Florida highway rights-of-way. Weed Technol. 4:658-660.

Willard, T.R. 1988. Biology, Ecology and management of cogongrass [*Imperata cylindrica* (L.) Beauv.]. Ph.D. dissertation, University of Florida, Gainesville, FL, USA. 113 p.

Willard, T.R., D.G. Shilling, J.F. Gaffney and W.L. Currey. 1996. Mechanical and chemical control of cogongrass (*Imperata cylindrica*). Weed Technol. 10:722-726.

Willard, T.R., J.F. Gaffney, and D.G. Shilling. 1997. Influence of herbicide combinations and application technology on cogongrass (*Imperata cylindrica*) control. Weed Technol. 11:76-80.

Windham, L. and J. G. Ehrenfeld. 2003. Net impact of a species invasion on nitrogen-cycling processes within a brackish tidal marsh. Ecol. Appl. 13: 883–897.

Wittenberg, R. and M. J. W. Cock, eds. 2001. Invasive Alien Species: A Toolkit of Best Prevention and Management Practices. Wallingford, UK: CAB International.

Yager, L. Y. 2007. Watching the grass grow: effects of habitat type, patch size, and land use on cogongrass (*Imperata cylindrica* (L.) Beauv.) spread on Camp Shelby Training Site Mississippi. Ph.D Dissertation, Misissippi State University. 178 p.

Yager, L. Y., D. L. Miller, and J. Jones. 2010. Susceptibility of longleaf pine forest associations in south Mississippi to invasion by cogongrass [*Imperata cylindrica* (L.) Beauv.]. Nat. Areas J. 30:226–232.

Yandoc, C., R. Charudattan, and D.G. Shilling. 1999. Enhancement of efficacy of *Bipolaris sacchari* (E.Butler) Shoem., a bioherbicide agent of cogongrass [*Imperata cylindrica* (L.) Beauv.], with adjuvants. Weed Sci. Soc. America Abstracts. 39:72.

Yoda, K., T. Kira, H. Ogawa, and K.Hozumi. 1963. Self-thinning in crowded pure stands under cultivated and natural conditions. J. Biol. Osaka city Univ. 14:107-129.

Zamora, D. L., D. C. Thill, and R. E. Eplee. 1989. An eradication plan for plant invasions. Weed Technol. 3:2–12.

Chapter 2

Integrated Palmer amaranth Management in Glufosinate-resistant Cotton: I. Soil-inversion, High Residue Cover Crops and Herbicide Regimes

2.1. Abstract

A three year field experiment was conducted to evaluate the role of soil-inversion, cover crops and herbicide regimes for Palmer amaranth between-row (BR) and within-row (WR) management in glufosinate-resistant cotton. The main plots were two soil-inversion treatments: fall inversion tillage (IT) and non-inversion tillage (NIT). The subplots were three cover crop treatments: crimson clover, cereal rye and winter fallow; and sub subplots were four herbicide regimes: preemergence (PRE) alone, postemergence (POST) alone, PRE + POST and a no herbicide check (None). The PRE herbicide regime consisted of a single application of pendimethalin at 0.84 kg ae ha^{-1} plus fomesafen at 0.28 kg ai ha^{-1}. The POST herbicide regime consisted of a single application of glufosinate at 0.60 kg ai ha^{-1} plus *S*-metolachlor at 0.54 kg ai ha^{-1} and the PRE + POST regime combined the prior two components. At 2 weeks after planting (WAP) cotton, Palmer amaranth density, both BR and WR, was reduced ≥ 90% following all cover crop treatments in the IT. In the NIT, crimson clover reduced Palmer amaranth density > 65% and 50% compared to winter fallow and cereal rye covers, respectively. At 6 WAP, the

PRE and PRE + POST herbicide regimes in both IT and NIT reduced BR and WR Palmer amaranth density > 96% over the three years. Additionally, the BR density was reduced ≥ 59% in no-herbicide (None) following either cereal rye or crimson clover when compared to no-herbicide in the winter fallow. In IT, PRE, POST and PRE + POST herbicide regimes controlled Palmer amaranth > 95% 6 WAP. In NIT, Palmer amaranth was controlled ≥ 79% in PRE and ≥ 95% in PRE + POST herbicide regimes over three years. POST herbicide regime following NIT was not very consistent. Averaged across three years, Palmer amaranth controlled ≥ 94% in PRE and PRE + POST herbicide regimes regardless of cover crop. Herbicide regime effect on cotton yield was highly significant; the maximum cotton yield was produced by the PRE + POST herbicide regime. Averaged over three years, the PRE, POST and PRE + POST cotton yields were about three times higher than no herbicide regime. In a conservation tillage production system, a PRE + glufosinate POST herbicide based regime coupled with a cereal rye cover crop may effectively control Palmer amaranth and maximize cotton yields.

2.2. Introduction

Palmer amaranth (*Amaranthus palmeri* S.Wats) is a highly aggressive dioecious row crop weed in the Southeastern US [1]. It has several unique weedy characteristics including rapid growth of > 5 inches a day during peak growth and can attain a final height of ≥ 2 m [2]. It is a prolific seed producer; a single female plant can produce more than 600,000 seeds, which have an average diameter of 1.0 mm [3]. In addition, Palmer amaranth has exceptional drought–tolerance and can endure moisture stress very well [4–7]. Palmer amaranth is highly problematic and interferes with the production of crops such as cotton, corn (*Zea mays* L.), cucurbits, grain sorghum (*Sorghum bicolor* L. Moench), peanut (*Arachis hypogea* L.), potato (*Solanum*

tuberosum L.), soybean (*Glycine max* L.), sweet potato (*Ipomoea batatas* L.) and several vegetable crops [8–21].

The occurrence of glyphosate resistance in Palmer amaranth has challenged cotton-weed managers in the Southeastern US. Until 2005, glyphosate had been very efficacious on Palmer amaranth [22–24]. However, the extensive adoption of glyphosate-resistant technology virtually replaced the conventional weed control technology consisting of preplant incorporated (PPI), preemergence (PRE), postemergence (POST) and post-directed (PDS) applied herbicides [25]. The sole reliance on glyphosate has resulted in selection for resistant biotypes of Palmer amaranth. Glyphosate resistant in Palmer amaranth was first documented in Macon County Georgia, in 2006 [26, 27]. As of 2010, glyphosate-resistant Palmer amaranth populations have been confirmed in 10 states [28]. Additionally, the Palmer amaranth populations resistant to dinitroaniline herbicides have been also reported [28, 29].

Historically, cotton was grown in conventional tillage utilizing primary and secondary tillage including moldboard plowing, disking and cultivation. However, increasing input costs, low commodity prices, and concerns for declining soil organic quality, and in some regions subsoil compaction, necessitated the adoption of alternative tillage options such as strip-tillage production systems that utilize a within row subsoiler to disrupt soil compaction under the crop row and minimizes surface residue disturbance [30,31]. Consequently, inadequate weed control has been reported in some conservation-tillage cotton production [32]. The infestation of small-seeded annual weeds such as Palmer amaranth has often been attributed to conservation tillage systems that preclude burial of weed seed. Moldboard plowing with soil inversion to the depth of 30 cm (12 in) has been shown to reduce glyphosate-resistant Palmer amaranth emergence 46% to 60% because many of the weed seeds are placed at depths which prevent emergence [33, 34].

However, the return to conventional tillage to control glyphosate-resistant Palmer amaranth threatens to reduce conservation tillage practices. Some researchers [35, 36] advocate the integration of traditional and alternative weed control strategies, such as the utilization of crop and herbicide rotation and integration of high residue cereal cover crops in order to sustain conservation tillage practices.

Over the last decade, the inclusion of cover crops in conservation-tillage systems has been researched due to the ability of some cover crops to suppress early-season weed density and growth either through direct competition from cover crop biomass [37–41] or through allelopathy [42–47]. Cereal rye (*Secale cereale* L.) has been well documented for both high biomass potential and allelopathic properties by several researchers [42, 43, 48–52]. Several studies on cover crops have reported excellent early-season weed control that can preclude the use of preemergence herbicides in crops [39, 41, 52–59]. However, the success of a cover crop in early-season weed suppression is determined by the biomass production potential which varies with year, location and management practices [37, 48, 53, 56, 60]. It has been observed that cereal rye residue alone was effective in reducing the glyphosate-resistant Palmer amaranth emergence by 94% in the row middle and 50% within the row [34]. The use of high residue cover crops in conjunction with chemical and cultural weed control tactics could provide effective Palmer amaranth control in established glyphosate-resistant populations as well as help prevent the development of resistance in the remaining glyphosate-susceptible populations [61]. Thus, the inclusion of cover crops in conservation tillage system may provide weed control benefits similar to those realized from inversion tillage in conventional tillage system.

With the widespread appearance of glyphosate–resistant Palmer amaranth, cotton producers are considering other weed management options such as inversion tillage, surface

tillage, and increased integration of soil active herbicides, cover crops and adoption of alternative GMO herbicide-crop systems such as glufosinate resistant cotton technology. Although inversion tillage can improve control of glyphosate-resistant Palmer amaranth, increased input costs and potential soil erosion are significant challenges. However the integration of cover crops and glufosinate-resistant cotton technology are possible viable alternatives. Therefore, a field study was conducted to evaluate the role of soil inversion, cover crops and herbicide regimes for Palmer amaranth management in glufosinate-resistant cotton.

2.3. Materials and Methods

A three year field experiment was conducted from fall 2008 through 2011 at the E.V. Smith Research Center, Field Crops Unit near Shorter, AL on a Compass sandy loam soil (coarse-loamy, siliceous, subactive, thermic Plinthic Paleudults) with 1.9% to 2.1% organic matter and pH 6.2 to 6.4. The experiment occupied a site that had been in continuous strip-tillage for the previous six years prior to experiment establishment, infested with a mixed population of resistant and susceptible Palmer amaranth, and subsequent treatments remained in the same location for three years without re-randomization of treatments. The experimental design consisted of a split-split plot treatment restriction in a randomized complete block design with three replicates. The main plots (43.9 by 9.1 m) were two soil-inversion treatments: fall inversion tillage (IT) and non-inversion tillage (NIT). After establishment, all IT plots reverted to NIT in future years. The subplots (14.6 by 9.1 m) were three different cover crops: crimson clover, cereal rye and winter fallow. The sub subplots (3.6 by 9.1 m) were four different herbicide regimes: preemergence (PRE) alone, postemergence (POST) alone, PRE + POST, and a no-herbicide check (None). A schedule of operations performed each year is given in Table 2.1.

2.3.1. Soil inversion, Cover Crops, and Cover Crop Management

In the fall 2008, approximately 28 million native glyphosate susceptible Palmer amaranth seeds were broadcast per hectare to ensure a sizeable seedbank. Half of each replicate was subjected to fall inversion tillage (IT) by moldboard plowing (30 cm) immediately fb one pass each of a disk and field cultivator, and half was under non-inversion tillage (NIT) using a within-row subsoiler equipped with pneumatic tires only to close the subsoiling slot. Subsequently each year in the fall, cereal rye (var. 'Elbon' in 2009 and 2010 and 'Wrens Abruzzi' in 2011) and crimson clover (*Trifolium incarnatum* L.) var. 'Dixie' cover crops were planted using 101 and 28 kg ha^{-1} seed, respectively in both the IT and NIT. Different cereal rye varieties were planted due to seed availability; Wrens Abruzzi has been shown to be more allelopathic [62]. In 2009 and 2010, frequent rain delayed both the harvesting of cotton and subsequent planting of cover crops [36]. Cereal rye cover was fertilized using 34 kg ha^{-1} of a 33-0-0 fertilizer. A winter fallow control was also included as check.

Cover crops were rolled with a three section straight bar roller/crimper (Bigham Brothers, Inc., Lubbock, TX, USA) in late April or early May using a JD 7730 equipped with an AutoSteer GPS. Cover crop rolling was immediately followed by an application of glyphosate (Roundup Weathermax®, Monsanto Company, St. Louis, MO, USA) at 0.84 kg ae ha^{-1} plus glufosinate (Ignite®, Bayer Crop Science, Research Triangle Park, NC, USA) at 0.49 kg ae ha^{-1}. The mixture was needed to enhance crimson clover termination. Cover crop biomass samples were taken prior to desiccation and oven dry biomass was recorded. The entire experimental area was sub-soiled in May using the previously described equipment to remove hardpan induced interactions; thus, no hardpans existed throughout the experimental area which could likely bias the yield

results. Subsoiling was followed by planting of glufosinate-resistant cotton (FM 1845 LLB2 in 2009, and FM 1735 LL, in 2010 and 2011, Bayer Crops Science, Research Triangle Park, NC). Each year, cotton was fertilized using 211 kg ha^{-1} of 16-16-16 fertilizer at the time of planting.

2.3.2. Herbicide Regimes

Four herbicide regimes constituted the sub-sub plot treatments. The PRE herbicide regime consisted of a single application of pendimethalin (Prowl®, BASF Ag. Products, Research Triangle Park, NC, USA) at 0.84 kg ae ha^{-1} plus fomesafen (Reflex®, Syngenta Crop Protection, Inc., Greensboro, NC, USA) at 0.28 kg ai ha^{-1}. The POST herbicide regime consisted of a single application of glufosinate at 0.60 kg ai ha^{-1} plus S-metolachlor (Dual II Magnum®, Syngenta Crop Protection, Inc., Greensboro, NC, USA) at 0.54 kg ai ha^{-1} and the PRE + POST regime consisted of both the aforementioned PRE and POST regimes. PRE herbicides were applied with a CO_2-pressurized backpack sprayer calibrated to deliver 145 L ha^{-1} with 8002 flat-fan nozzles. POST herbicides were applied to 3 to 4 lf Palmer amaranth between 15 and 20 days after planting cotton with an ATV-mounted sprayer delivering 145 L ha^{-1} with 8002 flat-fan spray nozzles. A last application (LAYBY) directed spray consisting of a prometryn (Caporal®, Syngenta Crop Protection, Inc., Greensboro, NC, USA) at 0.84 kg ai ha^{-1} + MSMA (Drexel Chemical Company, Memphis, TN) at 1.4 kg ai ha^{-1} was applied. Sethoxydim (Poast Plus®, Bayer AG. Products, Research Triangle Park, NC, USA) was applied at 0.28 kg ai ha^{-1} as needed to maintain grass control.

2.3.3. Herbicide Palmer Amaranth Sampling and Control Ratings

Palmer amaranth density was recorded once before the application of POST and again before the LAYBY application. Between-row (BR) Palmer amaranth density was recorded as number of plants in a quadrat (0.25 m^{-2}) randomly placed at 4 different positions between the 2nd and 3rd row of a four-row cotton plot. Similarly, the within-row (WR) Palmer density was recorded from a quadrant (0.25 m^{-2}) randomly placed at 4 different positions within the 2nd and 3rd rows. Palmer amaranth control was assessed visually at weekly intervals, starting a week after application of PRE until LAYBY application. A 0 to100 scale was used where 0 and 100 indicate no control and complete control, respectively. Each year, the Palmer amaranth was hand removed from all the plots before application of LAYBY to facilitate harvest. Therefore, Palmer amaranth was 100% controlled in each plot after LAYBY until cotton harvest. Cotton yields were recorded by mechanically harvesting two center 9 m rows within each four-row plot with a spindle picker.

2.3.4. Statistical Analysis

Three years data were subjected to combined ANOVA using Proc GLIMMIX in SAS (version 9.2, SAS Institute, Inc., Cary, NC, USA). Year, soil-inversion, cover crop, herbicide regime and their interactions were treated as fixed effects, while replication, replication x year, replication × soil-inversion, and replication × soil-inversion × cover crop were treated as random effects. When year and its interaction with other factors were significant, data were analyzed and presented by year. Palmer amaranth visual control data were arcsine-transformed and Palmer amaranth density data were square root transformed. However, the original and transformed data

analyses gave similar results, thus non-transformed data are presented. Multiple mean comparisons were made using the 'adj = simulate' option in the statistical analysis system at the 5% significance level.

2.4 Results

2.4.1 Cover Crop Biomass

Analysis of the three year data revealed significant effect of type of cover crop only. Averaged over three years, the maximum cover crop biomass was produced by cereal rye (4047 kg ha^{-1}) fb crimson clover (3570 kg ha^{-1}) that was significantly more than and winter fallow (1253 kg ha^{-1}).

2.4.2 Palmer Amaranth Density

Palmer amaranth density at 2 WAP revealed significant year by treatment interactions. Therefore data are presented by year. A soil-inversion by cover crop interaction was observed for both BR and WR density at 2 WAP in 2008–2009 and 2010–2011 while only the IT main effect was significant in 2009–2010. The highest BR and WR density of 49 and 35 plants m^{-2}, respectively, occurred in winter fallow following NIT in 2009–2010 (Table 2.2). In 2008–2009 and 2010–2011, crimson clover reduced Palmer amaranth density by as much as 96% BR and 82% WR in NIT. Similar reductions in Palmer amaranth and other weed densities by cover crop residues have been reported [63, 64–66]. Both BR and WR density was reduced > 90% following all cover crops in the IT. Each year with IT both BR and WR density of Palmer amaranth, was ≥ 90% lower than with NIT (Table 2.2). Furthermore each year at 2 WAP, Palmer

amaranth was 100% controlled by the PRE and PRE + POST herbicide regimes that received a PRE application within two days of planting (data not shown).

At 6 WAP, the effect of year and its interactions with other factors were not significant. However, both the BR and WR density demonstrated strong interaction of soil-inversion by herbicide regime. Additionally, a cover crop by herbicide regime interaction ($P < 0.0001$) was detected, for BR density only. The BR and WR density was markedly reduced (≤1 plant m^{-2}) under PRE, POST and PRE + POST herbicide regimes following IT and PRE and PRE + POST herbicide regimes following NIT (Table 2.3).

The Palmer amaranth density, both BR and WR, was reduced ≥ 77% in no-herbicide regime (None) following IT when compared to no-herbicide regime following NIT. The PRE and PRE + POST herbicide regimes in both IT and NIT reduced BR and WR density ≥ 96%. With the cover crop by herbicide regime interaction, the BR density was reduced ≥ 55% in no-herbicide (None) following either cereal rye or crimson clover when compared to no-herbicide in the winter fallow (Table 2.4).

PRE, POST and PRE + POST herbicide regimes' Palmer amaranth density was similar but lower than the no-herbicide (None) regime following any cover crop. However, the PRE and PRE + POST herbicide regimes were very consistent in reducing Palmer amaranth density (>95%) following all the cover crops. Earlier research also indicated the need of either a PRE or PRE + POST herbicide regime to supplement partial weed control obtained following different cover crops in a conservation tillage system [63]. Previous researchers also reported similar cover crop by herbicide interaction effect [67]. Excellent control of Palmer amaranth with a combination of pendimethalin and fomesafen has been reported [68].

2.4.3. Palmer Amaranth Visual Control

Palmer amaranth visual percent control at 6 WAP reflected significant year by treatment interactions. Analysis by year also indicated significant two way interactions between soil-inversion and herbicide regime and cover crop by herbicide regime each year. Additionally, a soil-inversion by cover crop interaction was highly significant in 2010–2011 ($P = 0.0007$). All main effects were also highly significant each year ($P < 0.0001$). In 2008–2009, Palmer amaranth was controlled 38%, 79%, and 95% in POST, PRE and PRE + POST herbicide regimes, respectively, following NIT while all herbicide regimes following IT provided ≥ 91% control (Table 2.5).

In 2009–2010 and 2010–2011, both PRE and PRE + POST herbicide regimes controlled Palmer amaranth ≥ 93% regardless of the soil-inversion treatment. In 2009–2010, the POST herbicide regime controlled Palmer amaranth 84 and 97% in NIT and IT, respectively. However in 2010–2011, POST herbicide regime controlled Palmer amaranth 100% in both soil-inversion treatments. The reason for poor performance of the POST herbicide regime in NIT in 2008–2009 is likely attributed to the oversized (> 10 cm) Palmer amaranth plants at the time of application.

Cover crop by herbicide regime interaction over the years revealed ≥ 94% control of Palmer amaranth in PRE and PRE + POST herbicide regimes regardless of type of cover crop (Table 2.6). The POST herbicide regime following both winter fallow and cereal rye provided 83% control of Palmer amaranth and was similar to the POST following crimson clover. However, Palmer amaranth control varied from 36% to 63% in no-herbicide (None) regime following different cover crops. Analysis revealed a soil-inversion by cover crop interaction in 2010–2011; Palmer amaranth control following different cover crops varied from 75% to 82% in

NIT and 89% to 100% in IT, respectively (Figure 2.1). Both cereal rye and crimson clover in IT gave significantly higher Palmer amaranth control (≥ 97%) than winter fallow in both IT and NIT and crimson clover and cereal rye in NIT. Previous research indicates the need to utilize residual herbicides throughout the season to aid in management of glyphosate resistant Palmer amaranth [69–72].

2.4.4. Cotton Yield

Analysis of the yield data revealed significant effect of herbicide regimes only. Although the cotton yield differences were not significant between PRE, POST and PRE + POST herbicide regimes, the maximum cotton yield was produced by the PRE + POST herbicide regime (1878 kg ha^{-1}) followed by POST (1658 kg ha^{-1}) and PRE (1532 kg ha^{-1}) alone regimes. PRE, POST and PRE + POST herbicide regimes' cotton yields were ≥ 2.7 times higher than no-herbicide (None) regime (Table 2.7).

2.5. Discussion and Conclusions

Recent evolution of herbicide resistance in Palmer amaranth has revealed that an urgent restructuring of weed management tactics is needed. Consequently, integration of various weed management approaches such as IT, cover crops, crop rotations, competitive cultivars, herbicide rotation, soil residual chemistries and tank mixture of herbicides with different modes of action could diversify the weed control practices and thereby preclude the selection pressure for herbicide resistance. Furthermore, the longevity of herbicide resistant technology itself necessitates the inclusion of multiple tactics in weed management systems.

Fall-inversion tillage offers improved Palmer amaranth control by allowing the deep burial of seed. Considering the rapid loss of Palmer amaranth seed viability with time, IT would help reduce the amount of viable seed near the surface [73]. However, IT is well known to deplete soil quality parameters such as soil organic matter while simultaneously increasing soil erosion. An occasional rotation with IT immediately followed by a cover crop conservation-tillage system could diversify weed management systems and prevent soil erosion. There is a great need of practical weed management solutions on farms severely impacted by glyphosate resistant Palmer amaranth [74].

Our research evaluated soil-inversion, cover crops and alternative herbicide regimes as an integrated approach to managing Palmer amaranth**.** Results indicate that IT alone resulted in ≥ 77% control of Palmer amaranth 6 WAP due to Palmer amaranth seed burial; addition of winter cover crops further increased Palmer amaranth control. Cover crops alone in NIT provided ≤50% control of Palmer amaranth; thus indicating the need in both IT and NIT to integrate other effective weed management practices to protect cotton yields. In an IT-cereal rye cover crop situation, a PRE or POST alone herbicide regime was as effective as a PRE + POST regime due to lower Palmer amaranth densities. However, with NIT, an effective and timely PRE + POST herbicide regime was necessary to control the higher Palmer amaranth densities present in this situation. Overall, the PRE + POST herbicide regime resulted in the maximum Palmer amaranth control and higher cotton yields in both soil-inversion treatments (Table 8).

The highest Palmer amaranth densities, regardless of soil-inversion treatment and herbicide regime, were consistently recorded in the winter fallow situation. Therefore, in a conservation tillage production system, a PRE + glufosinate POST herbicide based regime

coupled with a cover crop may effectively control Palmer amaranth and maximize cotton yields (Table 8).

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2.7. Literature Cited

1 Fernald, M.L. *Gray's Manual of Botany*, 8th ed.; American Book Co.: New York, NY, USA, 1950; p. 602.

2 Horak, M.J.; Loughin, T.M. Growth analysis of four *Amaranthus* species. *Weed Sci.* 2000, *48*, 347–355.

3 Keeley, P.E.; Carter, C.H.; Thullen, R.M. Influence of planting date on growth of Palmer amaranth (*Amaranthus palmeri*). *Weed Sci.* 1987, *35*, 199–204.

4 Ehleringer, J. Ecophysiology of *Amaranthus palmeri*, a Sonoran Desert summer annual. *Oecologia* 1983, *57*, 107–112.

5 Jha, P.; Norsworthy, J.K.; Malik, M.S.; Bangarwa, S.K.; Oliveira, M.J. Temporal emergence of Palmer amaranth from a natural seedbank. *Proc. South. Weed Sci. Soc.* 2006, *59*, 177.

6 Place, G.; Bowman, D.; Burton, M.; Rufty, T. Root penetration through a high bulk density soil layer: Differential response of a crop and weed species. *Plant Soil* 2008, *307*, 179–190.

7 Wright, S.R.; Jennette, M.W.; Coble, H.D.; Rufty, T.W. Root morphology of young *Glycine max, Senna obtusifolia,* and *Amaranthus palmeri*. *Weed Sci.* 1999, *47*, 706–711.

8 Bensch, C.N.; Horak, M.J.; Peterson, D. Interference of redroot pigweed (*Amaranthus retroflexus*), Palmer amaranth (*A. palmeri*), and common waterhemp (*A. rudis*) in soybean. *Weed Sci.* 2003, *51*, 37–43.

9 Burke, I.C.; Schroeder, M.; Thomas, W.E.; Wilcut, J.W. Palmer amaranth interference and seed production in peanut. *Weed Technol.* 2007, *21*, 367–371.

10 Klingaman, T.E.; Oliver, L.R. Palmer amaranth (*Amaranthus palmeri*) interference in soybeans (*Glycine max*). *Weed Sci.* 1994, *42*, 523–527.

11 Massinga, R.A.; Currie, R.S.; Horak, M.J.; Boyer, J. Interference of Palmer amaranth in corn. *Weed Sci.* 2001, *49*, 202–208.

12 Menges, R.M. Allelopathic effects of Palmer amaranth (*Amaranthus palmeri*) and other plant residues in soil. *Weed Sci.* 1987, *35*, 339–347.

13 Meyers, S.L.; Jennings, K.M.; Schultheis, J.R.; Monks, D.W. Evaluation of Flumioxazin and *S*-metolachlor rate and timing for Palmer amaranth (*Amaranthus palmeri*) control in Sweet potato. Weed Technol. 2010, *24*, 495–503.

14 Monks, D.M.; Oliver, L.R. Interactions between soybean (*Glycine max*) cultivars and selected weeds. *Weed Sci.* 1988, *36*, 770–774.

15 Moore, J.W.; Murray, D.S.; Westerman, R.B. Palmer amaranth (*Amaranthus palmeri*) effects on the harvest and yield of grain sorghum (*Sorghum biclor*). *Weed Technol.* 2004, *18*, 23–29.

16 Morgan, G.D.; Baumann, P.A.; Chandler, J.M. Competitive impact of Palmer amaranth (*Amaranthus palmeri*) on cotton (*Gossypium hirsutum*) development and yield. *Weed Technol.* 2001, *15*, 408–412.

17 Norsworthy, J.K.; Griffith, G.M.; Scott, R.C.; Smith, K.L.; Oliver, L.R. Confirmation and control of glyphosate-resistant Palmer amaranth (*Amaranthus palmeri*) in Arkansas. *Weed Technol.* 2008, *21*, 108–113.

18 Norsworthy, J.K.; Smith, K.L.; Scott, R.C.; Gbur, E.E. Consultant perspectives on weed management needs in Arkansas cotton. *Weed Technol.* 2007, *21*, 825–831.

19 Rowland, M.W.; Murry, D.S.; Verhalen, L.M. Full-season Palmer amaranth (*Amaranthus palmeri*) interference with cotton (*Gossypium hirsutum*). *Weed Sci.* 1999, *47*, 305–309.

20 Smith, D.T.; Baker, R.V.; Steele, G.L. Palmer amaranth (Amaranthus palmeri) impacts on yield, harvesting, and ginning in dryland cotton (*Gossypium hirsutum*). *Weed Technol.* 2000, *14*, 122–126.

21 Webster, T.M. Weed survey—Southern states. *Proc. South. Weed Sci. Soc.* 2005, *58*, 291–306.

22 Corbett, J.L.; Askew, S.D.; Thomas, W.E.; Wilcut, J.W. Weed efficacy evaluations for bromoxynil, glufosinate, glyphosate, pyrithiobac, and sulfosate. *Weed Technol.* 2004, *18*, 443–453.

23 Culpepper, A.S.; York, A.C. Weed management in glyphosate-tolerant cotton. *J. Cotton Sci.* 1998, *2*, 174–185.

24 Parker, R.G.; York, A.C.; Jordan, D.L. Comparison of glyphosate products in glyphosate-resistant cotton (*Gossypium hirsutum*) and corn (*Zea mays*). *Weed Technol.* 2005, *19*, 796–802.

25 Young, B.G. Changes in herbicide use patterns and production practices resulting from glyphosate-resistant crops. *Weed Technol.* 2006, *20*, 301–307.

26 Culpepper, A.S.; Grey, T.L.; Vencill, W.K.; Kichler, J.M.; Webster, T.M.; Brown, S.M.; York, A.C.; Davis, J.W.; Hanna, W.W. Glyphosate-resistant Palmer amaranth (*Amaranthus palmeri*) confirmed in Georgia. *Weed Sci.* 2006, *54*, 620–626.

27 Culpepper, A.S.; Whitaker, J.R.; MacRae, A.W.; York, A.C. Distribution of glyphosate-resistant Palmer amaranth (*Amaranthus palmeri*) in Georgia and North Carolina during 2005 and 2006. *J. Cotton Sci.* 2008, *12*, 306–310.

28 Heap, I. The international survey of herbicide resistant weeds. Available online: www.weedscience.org (accessed on 11 March 2012).

29 Gosset, B.J.; Murdock, E.C.; Toler, J.E. Resistance of Palmer amaranth (*Amaranthus palmeri*) to the dinitroaniline herbicides. *Weed Technol.* 1992, *6*, 587–591.

30 Troeh, F.R.; Hobbs, J.A.; Donahue, R.L. *Soil and Water Conservation*; Prentice Hall: Englewood Cliffs, NJ, USA, 1991; p. 232.

31 Wauchope, R.D.; McDowell, L.L.; Hagen, L.J. Environmental effects of limited tillage. In *Weed Control in Limited Tillage Systems*, Proceedings of Weed Science Society of America, Champaign, IL, USA, 14-16 January, 1985; pp. 266–281.

32 McWhorter, C.G.; Jordan, T.N. Limited tillage in cotton production. In *Weed Control in Limited-Tillage Systems*; Wiese, A.F., Ed.; Weed Science Society of America: Champaign, IL, USA, 1985; pp. 61–75.

33 Culpepper, A.S.; York, A.C.; Marshall, M.W. Glyphosate-resistant Palmer amaranth in the Southeast. *Proc. South. Weed Sci. Soc. 2009*, 62, 371.

34 Culpepper, A.S.; Kichler, J.; Sosnoskie, L.; York, A.; Sammons, D.; Nichols, R. Integrating cover crop residue and moldboard plowing into glyphosate-resistant Palmer amaranth management programs. In *Proceedings of Beltwide Cotton Conference*, New Orleans, LA, USA, 4–7 January 2010.

35 Price, A.J.; Balkcom, K.B.; Culpepper, A.S.; Kelton, J.A.; Nichols, R.L.; Schomberg, H.H. Glyphosate-resistant Palmer amaranth: A threat to conservation tillage. *J. Soil Water Conserv.* 2011, *66*, 265–275.

36 Price, A.J.; Balkcom, K.S.; Duzy, L.M.; Kelton, J.A. Herbicide and cover cop residue integration for *Amaranth* control in conservation agriculture cotton. *Weed Technol.* 2012, *26*, 490–498.

37 Brennan, E.B.; Smith, R.F. Winter cover crop growth and weed suppression on the central coast of California. *Weed Technol.* 2005, *19*, 1017–1024.

38 Collins, H.P.; Delgado, J.A.; Alva, A.K.; Follett, R.F. Use of nitrogen-15 isotopic techniques to estimate nitrogen cycling from a mustard cover crop to potatoes. *Agron. J.* 2007, *99*, 27–35.

39 Reddy, K.N. Effects of cereal and legume cover crop residues on weeds, yield, and net return in soybean (*Glycine max*). *Weed Technol.* 2001, *15*, 660–668.

40 Teasdale, J.R.; Mohler, C.L. The quantitative relationship between weed emergence and the physical properties of mulches. *Weed Sci.* 2000, *48*, 385–392.

41 Yenish, J.P.; Worsham, A.D.; York, A.C. Cover crops for herbicide replacement in no-tillage corn (*Zea mays*). *Weed Technol.* 1996, *10*, 815–821.

42 Barnes, J.P.; Putnam, A.R. Evidence for allelopathy by residues and aqueous extracts of rye (*Secale cereale*). *Weed Sci.* 1986, *34*, 384–390.

43 Barnes, J.P.; Putnam, A.R.; Burke, B.A.; Aasen, A.J. Isolation and characterization of allelochemicals in rye herbage. *Phytochemistry* 1987, *26*, 1385–1390.

44 Burgos, N.R.; Talbert, R.E. Differential activity of allelochemicals from *Secale cereale* in seedling bioassays. *Weed Sci.* 2000, *48*, 302–310.

45 Dhima, K.V.; Vasilakoglou, I.B.; Eleftherohorinos, I.G.; Lithourgidis, A.S. Allelopathic potential of winter cereals and their cover crop mulch effect on grass weed suppression and corn development. *Crop Sci.* 2006, *46*, 345–352.

46 Vasilakoglou, I.; Dhima, K.; Eleftherohorinos, I.; Lithourgidis, A. Winter cereal cover crop mulches and inter-row cultivation effects on cotton development and grass weed suppression. *Agron. J.* 2006, *98*, 1290–1297.

47 White, R.H.; Worsham, A.D.; Blum, U. 1989. Allelopathic potential of legume debris. *Weed Sci.* 1989, *37*, 674–679.

48 Mohler, C.L.; Teasdale, J.R. Response of weed emergence to rate of *Vicia villosa* Roth and *Secale cereale* L. residue. *Weed Res.* 1993, *33*, 487–499.

49 Nagabhushana, G.G.; Worsham, A.D.; Yenish, J.P. Allelopathic cover crops to reduce herbicide use in sustainable agricultural systems. *Allelopathy J.* 2001, *8*, 133–146.

50 Price, A.J.; Balkcom, K.S.; Arriaga, F.J. Rye biomass amount affects weed suppression levels in conversation-tillage cotton. In *Proceedings of the Beltwide Cotton Conference*, New Orleans, LA, USA, 4–7 January, 2005; pp. 2921–2923.

51 Price, A.J.; Arriaga, F.J.; Raper, R.L.; Balkcom, K.S.; Kornecki, T.S.; Reeves, D.W. Comparison of mechanical and chemical winter cereal cover crop termination systems and cotton yield in conservation agriculture. *Cotton Sci.* 2009, *13*, 238–245.

52 Reeves, D.W.; Price, A.J.; Patterson, M.G. Evaluation of three winter cereals for weed control in conservation-tillage non-transgenic cotton. *Weed Technol.* 2005, *19*, 731–736.

53 Ateh, C.M.; Doll, J.D. Spring-planted winter rye as a living mulch to control weeds in soybean. *Weed Technol.* 1996, *10*, 347–353.

54 Fisk, J.W.; Hesterman, O.B.; Shrestha, A.; Kells, J.J.; Harwood, R.R.; Squire, J.M.; Sheaffer, C.C. Weed suppression by annual legume cover crops in no tillage corn. *Agron. J.* 2001, *93*, 319–325.

55 Isik, D.; Kaya, E.; Ngouajio, M.; Mennan, H. Weed suppression in organic pepper (*Capsicum annuum* L.) with winter cover crops. *Crop Prot.* 2009, *28*, 356–363.

56 Teasdale, J.R. Contribution of cover crops to weed management in sustainable agricultural systems. *J. Prod. Agric.* 1996, *9*, 475–479.

57 Teasdale, J.R.; Beste, C.E.; Potts, W.E. Response of weeds to tillage and cover crop residue. *Weed Sci.* 1991, *39*, 195–199.

58 Teasdale, J.R.; Pillai, P.; Collins, R.T. Synergism between cover crop residue and herbicide activity on emergence and early growth of weeds. *Weed Sci.* 2005, *53*, 521–527.

59 Zasada, I.A.; Linker, H.M.; Coble, H.D. Initial weed densities affect no-tillage weed management with a rye (*Secale cereale*) cover crop. *Weed Technol.* 1997, *11*, 473–477.

60 Schomberg, H.H.; Endale, D.M.; Calegari, A.; Peixoto, R.; Miyazawa, M.; Cabrera, M.L. Influence of cover crops on potential nitrogen availability to succeeding crops in a Southern Piedmont soil. *Biol. Fertil. Soil* 2006, *42*, 299–307.

61 Price, A.J.; Stoll, M.E.; Bergtold, J.S.; Arriaga, F.J.; Balkcom, K.S.; Kornecki, T.S.; Raper, R.L. Effect of cover crop extracts on cotton and radish radicle elongation. *Comm. Biometry Crop Sci.* 2008, *3*, 60–66.

62 Reberg-Horton, S.; Burton, J.; Danehower, D.; Ma, G.; Monks, D.; Murphy, J.; Ranells, N.; Williamson, J.; Creamer, N. Changes over time in the allelochemical content of ten cultivars of rye (*Secale cereale* L.). *J. Chem. Ecol.* 2005, *31*, 179–193.

63 Price, A.J.; Reeves, D.W.; Patterson, M.G. Evaluation of weed control provided by three winter cereals in conservation-tillage soybean. *Renew. Agric. Food Syst.* 2006, *21*, 159–164.

64 Price, A.J.; Reeves, D.W.; Patterson, M.G.; Gamble, B.E.; Balkcom, K.S.; Arriaga, F.J.; Monks, C.D. Weed control in peanut grown in a high-residue conservation-tillage system. *Peanut Sci.* 2007, *34*, 59–64.

65 Reddy, K.N. Impact of rye cover crop and herbicides on weeds, yield, and net return in narrow-row transgenic and conventional soybean (*Glycine max*). *Weed Technol.* 2003, *17*, 28–35.

66 Saini, M.; Price, A.J.; van Santen, E. Cover crop residue effects on early-season weed establishment in a conservation-tillage corn-cotton rotation. In *Proceedings of the 28th Southern Conservation Systems Conference*, Amarillo, TX, USA, 26–28 June 2006.

67 Weston, L. Cover crop and herbicide influence on row crop seedling establishment in no-tillage culture. *Weed Sci.* 1990, *38*, 166–171.

68 Timothy, L.G.; Sosnoskie, L.; Webster, T.M. Palmer amaranth control as affected by herbicide, method of application, and winter cover crop. Available online: http://commodities.caes.uga.edu/fieldcrops/cotton/rerpubs/2009/p049RER2009.pdf (accessed on 22 July 2012).

69 Steckel, L. Cotton Weed Control. In *Weed Control Manual for Tennessee*; Technical Report PB 1580-6M-12/08 for University of Tennessee Agriculture Extension Service: Knoxville, TN, USA, 2008; pp. 9–83.

70 Stephenson, D.; Stewart, S.; Vidrine, R. *Herbicide Resistance Management in Roundup Ready Cotton*; Technical Report for Louisiana State University Agricultural Center: Baton Rouge, LA, USA, 2008.

71 Whitaker, J.R.; York, A.C.; Jordan, D.L.; Culpepper, A.S.; Sosnoskie, L.M. Residual herbicides for Palmer amaranth control. *Cotton Sci.* 2011, *15*, 89–99.

72 York, A.C.; Culpepper, A.S. Weed Management in Cotton. In *2009 Cotton Information*; North Carolina Cooperative Extension Service: Raleigh, NC, USA, 2009; pp. 68–125.

73 Sosnoski, L.M.; Webster, T.M.; Culpepper, A.S. Reductions in Palmer amaranth seed viability over time. Available online: http://commodities.caes.uga.edu/fieldcrops/cotton/rerpubs/2009/p055RER2009.pdf (accessed on 11 March 2012).

74 Shaw, D.R.; Culpepper, A.S.; Owen, M.; Price, A.J.; Wilson, R. Herbicide-resistant weeds threaten soil conservation gains: Finding a balance for soil and farm sustainability, 2012. Available online: http://www.cast-science.org (accessed on 11 March 2012).

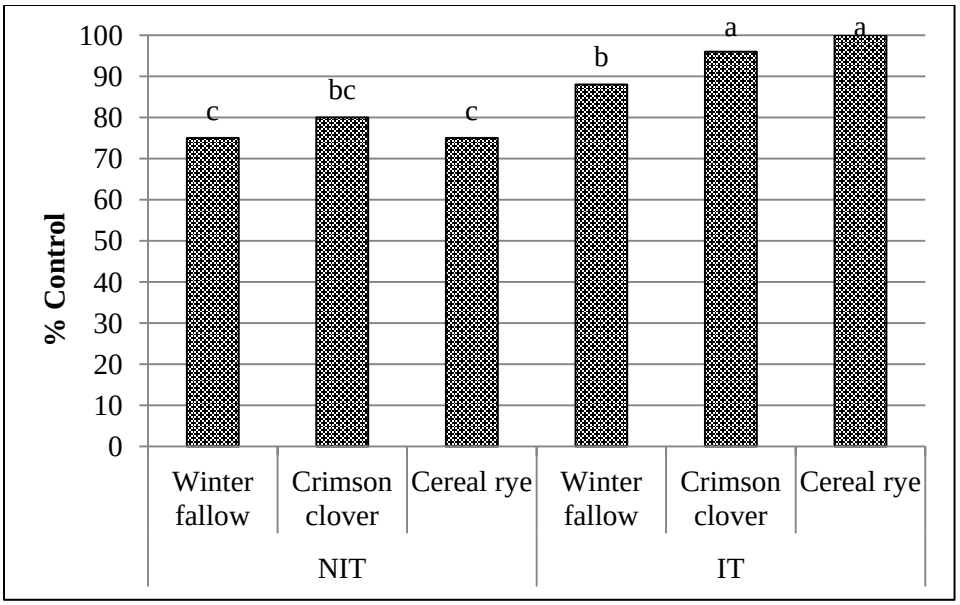

Figure 2.1. Soil inversion by cover crop interaction on Palmer amaranth control in 2011. Different letters indicate significant differences at P = 0.05.
*Abbreviations: IT, fall–inversion tillage; NIT, non-inversion tillage.

Table 2.1. Schedule of operations performed during the experiment

Operations	Experiment years		
	2008–2009	2009–2010	2010–2011
Broadcasting Palmer amaranth seed	19 Nov	–	–
Fall inversion tillage	19 Nov	–	–
Planting of cover crops	20 Nov	6 Jan	2 Dec
Rolling and termination of cover crops	22 Apr	18 May	19 Apr
Subsoiling	23 Apr	24 May	26 Apr
Cotton planting	1 Jun	27 May	5 May
Fertilization (16-16-16)	1 Jun	27 May	5 May
PRE application	3 Jun	27 May	6 May
POST application	16 Jun	16 Jun	24 May
Graminicide application (Poast Plus® + COC)	13 July	8 July	6 July
LAYBY application	14 Aug	16 Aug	19 July
Cotton defoliation	26 Oct	14 Oct	13 Sep
Cotton harvesting	9 Nov	20 Oct	30 Sep

Table 2.2. Influence of soil-inversion and cover crop on Palmer amaranth density at 2 WAP over three production years

Experimental variable		Palmer amaranth density (plants m^{-2})					
Soil-inversion	Cover crop	2008–2009		2009–2010		2010–2011	
		BR *	WR	BR	WR	BR	WR
Non-inversion (NIT)	Winter fallow	10 a **	11 a	49 a	35 a	22a	17 a
	Crimson clover	3 b	2 c	29 ab	26 a	1 c	6 c
	Cereal rye	9.0 a	7 b	18 b	31 a	10 b	12 b
	Mean	7.3 A	6.7 A	32.0 A	30.7 A	11.0 A	11.7 A
Fall-inversion (IT)	Winter fallow	1 c	0 c	2 c	2 c	2 c	1 d
	Crimson clover	0 c	0 c	2 c	0 c	0 c	0 d
	Cereal rye	1 c	1 c	3 c	2 c	0 c	1 d
	Mean	0.7 B	0.3 B	3.7 B	1.3 B	0.7 B	0.7 B

* Abbreviations: WAP, weeks after cotton planting; BR, between row; WR, within row;
** Means within a column followed by the same letter are not significant (P = 0.05).

Table 2.3. Influence of soil–inversion and herbicide regime on Palmer amaranth density at 6 WAP * and cotton yield with cover crop and three production years' data combined

Experimental variable		Palmer amaranth density (plants m^{-2})		Cotton yield (kg ha^{-1})
Soil-inversion	Herbicide regime	BR *	WR	
Non-inversion (NIT)	None	26 a **	23 a	105 c
	PRE *	1 b	1 b	1520 a
	POST	4 b	3 b	1423 a
	PRE + POST	1.0 b	1 b	1716 a
	Mean	8.0 A	7.0 A	1191 B
Fall-inversion (IT)	None	6 b	4 b	976 b
	PRE	1 b	0 b	1544 a
	POST	1 b	1 b	1893 a
	PRE + POST	0 b	0 b	2041 a
	Mean	2.0 B	1.3 B	1613 A

* Abbreviations: WAP, weeks after cotton planting; BR, between row; WR, within row; PRE, only preemergence; POST, only postemergence; PRE + POST, both preemergence and postemergence; ** Means within a column followed by same letter are not significant (P = 0.05).

Table 2.4. Influence of cover crop and herbicide regime on BR * Palmer amaranth density at 6 WAP * and cotton yield with soil-inversion and three production years' data combined.

Experimental variable		Palmer amaranth density	Cotton yield
Cover crop	Herbicide regime	(plants m^{-2})	(kg ha^{-1})
Winter fallow	None	27 a **	141 b
	PRE *	1 c	1506 a
	POST	4 c	1449 a
	PRE + POST	1c	1869 a
	Mean	11.0 A	1242 A
Crimson clover	None	10 b	711 b
	PRE	1 c	1544 a
	POST	1 c	1918 a
	PRE + POST	1 c	2047 a
	Mean	4.0 B	1555 A
Cereal rye	None	11 b	768 b
	PRE	1 c	1546 a
	POST	3 c	1606 a
	PRE + POST	0 c	1720 a
	Mean	5. 0 B	1410 A

* Abbreviations: BR, between row; WAP, weeks after cotton planting; PRE, only preemergence; POST, only postemergence; PRE + POST, both preemergence and postemergence; ** Means within a column followed by same letter are not significant (P = 0.05).

Table 2.5. Influence of soil-inversion by herbicide regime on Palmer amaranth control at 6 WAP * with cover crop data combined in three production years.

Experimental variable		Year (% control)		
Soil-inversion	Herbicide regime	2008–2009	2009–2010	2010–2011
Non-inversion (NIT)	None	27 c **	15 c	9 c
	PRE *	79 b	93 a	100 a
	POST	38 c	84 b	100 a
	PRE + POST	95 a	98 a	100 a
	Mean	60.0 B	72.0 B	77.0 B
Fall-inversion (IT)	None	91 a	77 b	81 b
	PRE	99 a	98 a	100 a
	POST	95 a	97 a	100 a
	PRE + POST	100 a	100 a	100 a
	Mean	96.0 A	93.0 A	95.0 A

* Abbreviations: WAP, weeks after cotton planting; PRE, only preemergence; POST, only postemergence; PRE + POST, both preemergence and postemergence; ** Means within a column followed by same letter are not significant (P = 0.05).

Table 2.6. Cover crop by herbicide regime interaction effect on Palmer amaranth control at 6 WAP * with soil-inversion and three production years' data combined

Experimental variable		Palmer amaranth control	Cotton yield
Cover crop	Herbicide regime	(%)	(kg ha^{-1})
Winter fallow	None	36 d **	141 b
	PRE *	95 ab	1506 a
	POST	83 b	1449 a
	PRE + POST	98 ab	1869 a
	Mean	78 B	1242 A
Crimson clover	None	63 c	711 b
	PRE	96 ab	1544 a
	POST	91 ab	1918 a
	PRE + POST	99 a	2047 a
	Mean	87 A	1555 A
Cereal rye	None	52 c	768 b
	PRE	94 ab	1546 a
	POST	83 b	1606 a
	PRE + POST	99 a	1720 a
	Mean	82 AB	1410 A

* Abbreviations: WAP, weeks after cotton planting; PRE, only preemergence; POST, only postemergence; PRE + POST, both preemergence and postemergence; ** Means within a column followed by same letter are not significant (P = 0.05).

Table 2.7. Influence of herbicide regimes on cotton yield with cover crop, soil-inversion, and three production years' data combined

Herbicide regime	Herbicides	Cotton yield (kg ha^{-1})
None	LAYBY consisting of prometryn + MSMA	560 b **
PRE *	Pendimethalin + fomesafen fb LAYBY	1532 a
POST	Glufosinate + S-metolachlor fb LAYBY	1658 a
PRE + POST	Pendimethalin + fomesafen (PRE) fb Glufosinate + S-metolachlor (POST) fb LAYBY	1878 a

* Abbreviations: PRE, only preemergence, POST; only postemergence; PRE + POST, both preemergence and postemergence; fb, followed by; ** Means followed by same letter are not significant (P = 0.05).

Table 2.8. Palmer amaranth density and visual percent control at 6 WAP * and cotton yield from selected treatments with data combined over three production years

Experimental variable			Palmer amaranth density (plants m^{-2})		Palmer amaranth Control (%)	Cotton yield (kg ha^{-1})
Soil-inversion	Cover crop	Herbicide regime	BR *	WR		
Non-inversion (NIT)	Crimson clover	PRE + POST *	1	<1	98	1931
		PRE	2	<1	92	1439
		POST	2	1	82	1652
	Cereal rye	PRE + POST	1	<1	98	1620
		PRE	2	<1	90	1433
		POST	3	1	68	1425
	Winter fallow	PRE + POST	2	1	96	1699
		PRE	1	1	90	1597
		POST	5	4	72	1185
Fall-inversion (IT)	Crimson clover	PRE + POST	0	0	100	2163
		PRE	<1	<1	99	1650
		POST	<1	<1	99	2185
	Cereal rye	PRE + POST	0	0	100	1820
		PRE	<1	0	99	1667
		POST	2	<1	97	1780
	Winter fallow	PRE + POST	0	0	100	2139
		PRE	<1	<1	99	1315
		POST	1	1	95	1713
The following treatments were the best in terms of both Palmer amaranth control and cotton yield.						
Non-inversion (NIT)	Crimson clover	PRE + POST	1	0	98	1931

Fall-inversion (IT)	Cereal rye	PRE + POST	1	<1	98	1620
	Winter fallow	PRE + POST	2	1	96	1699
	Crimson clover	PRE + POST	0	0	100	2163
	Cereal rye	PRE + POST	0	0	100	1820
	Winter fallow	PRE + POST	0	0	100	2139

* Abbreviations: WAP, weeks after cotton planting; PRE, only preemergence; POST, only postemergence; PRE + POST, both preemergence and postemergence.

Chapter 3

Integrated Palmer Amaranth Management in Glufosinate-Resistant Cotton: II. Primary, Secondary and Conservation Tillage

3.1. Abstract

A three year field experiment was conducted to evaluate the role of soil inversion, cover crops and spring tillage methods for Palmer amaranth between-row (BR) and within-row (WR) management in glufosinate-resistant cotton. Main plots were two soil inversion treatments: fall inversion tillage (IT) and non-inversion tillage (NIT). Subplots were three cover treatments: crimson clover, cereal rye or none (i.e., winter fallow); and the sub subplots were four secondary spring tillage methods: disking followed by (fb) cultivator (DCU), disking fb chisel plow (DCH), disking fb disking (DD) and no tillage (NT). Averaged over years and soil inversion, the crimson clover produced maximum cover biomass (4390 kg ha^{-1}) fb cereal rye (3698 kg ha^{-1}) and winter fallow (777 kg ha^{-1}). Two weeks after planting (WAP) and before the postemergence (POST) application, Palmer amaranth WR and BR density were two- and four-times less, respectively, in IT than NIT. Further, Palmer amaranth WR and BR density were reduced two-fold following crimson clover and cereal rye than following winter fallow at 2 WAP. Without IT, early season Palmer amaranth densities were 40% less following DCU, DCH and DD, when compared with IT. Following IT, no spring tillage method improved Palmer amaranth control. The timely application of glufosinate + *S*-metolachlor POST tank mixture greatly improved Palmer amaranth control in both IT and NIT systems. The highest cotton yields were obtained with DD following cereal rye (2251 kg ha^{-1}), DD following crimson clover (2213 kg ha^{-1}) and DD

following winter fallow (2153 kg ha^{-1}). On average, IT cotton yields (2133 kg ha^{-1}) were 21% higher than NIT (1766 kg ha^{-1}). Therefore, from an integrated weed management standpoint, an occasional fall IT could greatly reduce Palmer amaranth emergence on farms highly infested with glyphosate-resistant Palmer amaranth. In addition, a cereal rye or crimson clover cover crop can effectively reduce early season Palmer amaranth emergence in both IT and NIT systems. For effective and season-long control of Palmer amaranth, one or more POST applications of glufosinate + residual herbicide as tank mixture may be needed in a glufosinate-based cotton production system.

3.2. Introduction

Palmer amaranth [*Amaranthus palmeri* (L.) S. Wats] is one of the several pigweed species that are problematic in row crops in the southeastern United States. Compared to other pigweed species, such as common waterhemp [*Amaranthus rudis* (L.) Sauer], redroot pigweed (*Amaranthus retroflexus* L.) and tumble pigweed (*Amaranthus albus* L.), Palmer amaranth produced the highest dry weight, leaf area and height [1]. Palmer amaranth grows relatively quickly and can attain a height of 2 m or more [1]. It is a dioecious plant with tremendous seed production potential and rapid seed germination [1–3]. A single female plant can produce more than 600,000 seeds, depending upon density, which have an average diameter of 1.0 mm [2]. It has exceptional drought tolerance [4–7]. Additionally, Palmer amaranth can grow under low light conditions, such as dense crop canopies [8]. Palmer amaranth interference and subsequent yield losses have been documented in several crops, such as cotton, corn, cucurbits, grain sorghum, peanut, potato, soybean, sweet potato and several vegetable crops [9–23].

Until recently, glyphosate-resistant cotton production systems were very effective for managing a broad spectrum of weeds, including Palmer amaranth [24, 25]. However, the evolution of glyphosate resistant Palmer amaranth has forced cotton producers to explore other management options and integrated approaches. These included inversion tillage and adoption of glufosinate-resistant varieties [26]. Additionally, resistance to dinitroaniline herbicides has also been reported in some Palmer amaranth populations [26, 27].

The role of tillage in altering the distribution, abundance, composition of species, as well as seedling emergence patterns, has been well documented [28–36]. In conservation tillage systems, where soil incorporation is minimized, seeds accumulate near soil surface. Contrarily, the soil disturbance resulting from various tillage practices places weed seeds at different depths that vary in availability of moisture, diurnal temperature fluctuation, light exposure and activity of predators [37]. Moldboard plowing buries weed seeds deeply in the soil; however, deeper burial may lead to long-term weed problems, because of increased seedbank longevity [38].

The type of tillage implement used to till the soil greatly influences the vertical distribution and density of seeds within the soil profile [28, 30, 36, 39–42]. Inversion tillage implements bury a large proportion of the weed seed, while non-inversion tillage implements leave more of the seed near the soil surface [28]. Previous research demonstrated that more than 60% of weed seedbank was concentrated in top 5 cm following either a no-till or chisel plow [41]. Therefore, considering the inability of small Palmer amaranth seed to emerge from depths greater than 7.5 cm and a light requirement for germination, moldboard plowing followed by a conservation system may reduce the Palmer amaranth populations to manageable levels.

Currently, an integrated weed control system utilizing high-residue cover crops as a weed management tool is gaining popularity [43–54]. Cover crops provide early season weed control

by reducing light transmission and quality, altering soil temperature, physically suppressing weed emergence and allelopathy [55–58]. Cereal rye (*Secale cereale* L.) has been documented as having high biomass potential, early season weed suppression and allelopathic properties by several researchers [44, 46–48, 59–62]. It has also been observed that cereal rye residue alone was effective at reducing glyphosate-resistant Palmer amaranth emergence by 94% in the row middle and 50% within the cotton row [63]. Others reported that the use of high residue cover crops in conjunction with chemical and cultural weed control tactics provided effective control of established glyphosate-resistant Palmer amaranth, while helping to prevent the development of resistance in glyphosate-susceptible populations [64].

Considering the magnitude of current herbicide resistance problems, inversion tillage can likely improve control of glyphosate-resistant Palmer amaranth. However, increased input costs and potential soil erosion are significant challenges for growers. Integration of cover crops and glufosinate-resistant cotton technology may be viable alternatives in the light of these economic and environmental considerations. Therefore, with the current Palmer amaranth management challenges, a field study was conducted to evaluate the role of primary inversion tillage, cover crops and secondary tillage methods for Palmer amaranth management in glufosinate-resistant cotton.

3.3. Materials and Methods

3.3.1. Experimental Design and Establishment

A three year field experiment was conducted from fall 2008 through harvest 2011 at the E.V. Smith Research Center, Field Crops Unit near Shorter, AL, on a Compass sandy loam soil (coarse-loamy, siliceous, subactive, thermic Plinthic Paleudults) with 1.9 to 2.1% organic matter

and pH 6.2 to 6.4. The experiment occupied a site that had been in continuous strip-tillage for previous six years. The entire experimental area was infested with glyphosate-susceptible Palmer amaranth prior to experiment establishment, and the subsequent treatments remained in the same location for three years without re-randomization of treatments. Treatments consisted of a factorial arrangement of two levels of soil inversion—fall inversion tillage (IT) and non-inversion tillage (NIT)—three levels of winter cover crops—cereal rye, crimson clover (*Trifolium incarnatum* L.) and none (*i.e.*, winter fallow)—and four different spring tillage methods, resulting in a 24-treatments test. The four spring tillage methods were disk followed by (fb) chisel plow (DCH), disk fb field cultivator (DCU), disk twice (DD) and a no-tillage control (NT). The experimental design consisted of a split-split plot treatment restriction in a randomized complete block design with three replicates. Soil inversion, cover crop and spring tillage were assigned to the main plots, sub plots and sub-sub plots, respectively. The size of the main, sub and sub-sub plots were 43.9 m by 9.1 m, 14.6 by 9.1 m and 3.6 by 9.1 m, respectively. A schedule of operations performed each year is given in Table 1.

In the fall 2008, approximately 28 million native glyphosate-susceptible Palmer amaranth seeds were broadcast per hectare to ensure a sizeable seedbank of this weed. Prior to broadcasting, Palmer amaranth seed germination was tested by placing 25 seeds on commercial germination paper in four petri dishes at 35 °C. Seeds were kept moist with tap water inside closed petri dishes. Seeds were considered germinated when the radicle emerged 1 mm. Germination percentage was calculated as the number of germinated seeds divided by the total number of seeds multiplied by 100. Two weeks after initiation, 87% of the seeds germinated. One half of each block was subject to fall inversion tillage (IT) by moldboard plowing (30 cm) immediately fb one pass each of a disk and field cultivator; the other half was under non-

inversion tillage (NIT) using a within-row subsoiler equipped with pneumatic tires to close the subsoiling slot. During the fall of each year, cereal rye (cv. 'Elbon' in 2009 and 2010 and 'Wrens Abruzzi' in 2011) and crimson clover (*Trifolium incarnatum* L. cv. 'Dixie') cover crops were seeded at rates of 101 and 28 kg seed ha^{-1}, respectively, in both IT and NIT. Different cereal rye cultivars had to be used due to seed availability; Wrens Abruzzi has been shown to be more allelopathic [65]. In 2009 and 2010, frequent rains delayed both the harvesting of cotton and subsequent planting of cover crops. Cereal rye cover was fertilized using 34 kg ha^{-1} of a 33-0-0 fertilizer. A winter fallow control was also included as check.

3.3.2. Cover Crop Management

Cover crops were rolled with a three section straight bar roller (Bigham Brothers Inc., Lubbock, TX, USA; Mention of a trade name, proprietary product, or specific equipment does not constitute a guarantee or warranty by the USDA or Auburn University and does not imply endorsement of a product to the exclusion of others that may be suitable) in late April or early May using a JD 7730 equipped with an AutoSteer GPS. Cover crop rolling was immediately followed by an application of glyphosate (Roundup Weathermax®, Monsanto Company, St. Louis, MO, USA) at 0.84 kg ae ha^{-1} plus glufosinate (Ignite®, Bayer Crop Science, Research Triangle Park, NC, USA) at 0.49 kg ai ha^{-1}; the mixture was needed to enhance crimson clover termination. Cover crop biomass samples were taken prior to desiccation, and dry biomass was recorded. The entire experimental area was sub-soiled in May to 45 cm depth to break hardpans. A within-row subsoiler equipped with pneumatic tires only, to close the subsoiling slot, was used. Sub-soiling was followed by planting of glufosinate-resistant cotton (cvs. FM 1845 LLB2 in 2009 and FM 1735 LL in 2010 and 2011, Bayer Crops Science, Research Triangle Park, NC,

USA). Each year, cotton was fertilized using 211 kg ha^{-1} of 16-16-16 fertilizer at the time of planting.

3.3.3. Secondary Tillage and Weed Management

The DCH tillage consisted of a single pass of 3 m disk fb a single pass of 1.8 m chisel plow, DD consisted of double pass of 3 m disk and DCU was a single pass of 3 m disk fb a single pass of 4.1 m field cultivator. A single postemergence (POST) application of a tank mixture of glufosinate at 0.60 kg ai ha^{-1} (Ignite®, Bayer Crops Science, Research Triangle Park, NC, USA) plus *S*–metolachlor (Dual II Magnum®, Syngenta Crop Protection, Inc., Greensboro, NC, USA) at 0.54 kg ai ha^{-1} tank mixture was made to 2 to 3 lf Palmer amaranth, between 15 and 20 days after planting cotton, with an ATV-mounted sprayer delivering 145 L ha^{-1} with flat-fan spray tips. In 2011, an additional POST application of glufosinate at 0.60 kg ai ha^{-1} plus *S*–metolachlor at 0.54 kg ai ha^{-1} was carried out three weeks after the first POST application. A last directed POST application (LAYBY) of prometryn (Caporal®, Syngenta Crop Protection, Inc., Greensboro, NC, USA) at 0.84 kg ai ha^{-1} plus MSMA (Drexel Chemical Company, Memphis, TN, USA) at 1.4 kg ai ha^{-1} was carried out approximately 2 months after the first POST application. Sethoxydim (Poast Plus®, Bayer Ag. Products, Research Triangle Park, NC, USA) was applied at 0.28 kg ai ha^{-1} as needed to maintain grass control.

3.3.4. Palmer Amaranth Sampling and Visual Control Ratings

Palmer amaranth density was determined both between (BR) and within (WR) the cotton rows before the POST application and again before the LAYBY application. BR Palmer amaranth density was recorded as number of plants in a quadrat (0.25 m^{-2}) randomly placed at four different positions between the second and third row of a four-row cotton plot. Similarly,

WR Palmer amaranth density was recorded from a quadrat (0.25 m^{-2}) randomly placed at four different positions within the second and third rows. Palmer amaranth control was also assessed visually, for the entire plot, at weekly intervals on a 0 to 100 scale, where 0 and 100 indicate no control and complete control, respectively. Palmer amaranth was hand-pulled from all the plots before the LAYBY application, but following density counts and control ratings to facilitate cotton harvest. Cotton yields were determined by mechanically harvesting the two central rows within each four-row plot with a spindle picker.

3.3.5. Statistical Analysis

Data were analyzed using generalized linear mixed models or linear mixed models methodology as implemented in SAS® PROC GLIMMIX based on the underlying design, which was a randomized complete block design (r = 3) with a split-split-split plot in time restriction on randomization. Soil inversion, cover crop, spring tillage method, year and all their interactions were treated as fixed effects. Block and Block × treatment factors were treated as random effects. The split plot nature of the experiment requires five different residual terms: (1) block × soil inversion as the appropriate error term for soil inversion; (2) block × soil inversion × cover crop as the appropriate error term for cover crop and its interaction with soil inversion; (3) block × soil inversion × cover crop × spring tillage method as the appropriate error term for spring tillage and its interaction with soil inversion and cover crop; (4) block × year as the appropriate error term for year; and (5) the residual error term as the appropriate error term for all interactions effects of year with the remaining factors. The factor year is of a repeated measures nature that induces a covariance relationship because of the lack of re-randomization. All the standard covariance models were evaluated, but none improved the AICC fit statistic, which is a penalized

-2log likelihood. However, grouping the residual variance by year using the 'random _residual/group = year' option in SAS gave a slightly improved fit. Fit was improved by creating variance groups, even though the maximum *F*-test of residuals among the three years did not detect heterogeneous variances. Palmer amaranth density data were analyzed using a lognormal distribution function, and back transformed means along with 95% confidence intervals are reported. Palmer amaranth control rating data at three and six weeks after application were arcsine-transformed, and back transformed means along with 95% confidence intervals are reported. No transformation was required for cover crop biomass and cotton yield data. Multiple means' comparisons of significant effects were made using the 'Adj = simulate' option in SAS PROC GLIMMIX at the 5% significance level.

3.4. Results

3.4.1. Cover Crop Biomass

Cover crop biomass was significantly affected by the type of cover crop. Averaged over three production years and soil inversion, crimson clover (4390 kg ha^{-1}) produced significantly higher biomass than both cereal rye (3698 kg ha^{-1}) and winter fallow (777 kg ha^{-1}).

3.4.2. Palmer Amaranth Density

At 2 WAP, the main effects of soil inversion, cover crop and spring tillage method were highly significant for both BR and WR density ($p < 0.0001$). Palmer amaranth density BR and WR was four and two-fold, respectively, less in IT than NIT (Table 2).

Of the cover crops, both cereal rye and crimson clover resulted in a two-fold reduction in BR and WR density than winter fallow. With regard to spring tillage methods, BR density was significantly less in DD than NT. However, DCH and DCU did not result in a significant reduction in BR and WR density compared with NT. The WR density was significantly less in DD than DCH and DCU. Furthermore, the WR density in DCH, DCU and NT was similar. Previous researchers also observed two-fold higher early season pigweed density in a winter fallow conservation tillage systems compared to the similar conventional tillage systems [64]. Additionally, the effect of cover crop residue on inhibition of weed seed germination and seedling emergence has been well documented [55, 62, 63, 65, 66].

At 6WAP, a soil inversion by spring tillage method interaction revealed significant differences in BR density ($p < 0.05$). Following IT, the BR density was reduced ≥90% in DCH, DCU and DD spring tillage methods compared to NT following NIT (Table 3).

The NT following IT resulted in a 69% reduction in BR density compared to the NT following NIT, but was similar to the DCU, DCH and DD in the NIT. Previous researchers observed a similar reduction in weed density with the comparable tillage practices [28, 36, 40, 67–69]. With regard to WR density at 6 WAP, soil inversion and spring tillage method main effects were strongly significant ($p < 0.0001$). The Palmer amaranth WR density was two-fold less in IT (2 ± 1 plants m^{-2}) than NIT (5 ± 1 plants m^{-2}). Of spring tillage methods, DD (2 ± 1 plants m^{-2}) significantly reduced WR density compared to the NT (4 ± 1 plants m^{-2}). However, WR density was similar in DD, DCH (2 ± 2 plants m^{-2}) and DCU (2 ± 2 plants m^{-2}). Similarly, the WR density following DCH and DCU were not different from NT.

3.4.3. Palmer Amaranth Control

Three weeks after herbicide application (3 WAA), a soil inversion by cover crop by spring tillage method interaction was highly significant ($p < 0.05$). Following non-inversion tillage, Palmer amaranth was controlled more than 90% in both DD and DCU in crimson clover and DD in both cereal rye and winter fallow (Table 4).

With non-inversion tillage and crimson clover, Palmer amaranth control improved ≥16% in DCH, DCU and DD compared with NT. With non-inversion tillage and cereal rye, Palmer amaranth was controlled 14, 11 and 22%, more in DCH, DCU and DD, respectively, than NT. With non-inversion tillage and winter fallow, Palmer amaranth was controlled ≥74% in DCH, DCU and DD compared with NT. Furthermore, following non-inversion tillage, Palmer amaranth control was higher with all spring tillage methods in crimson clover than cereal rye and winter fallow. This may be attributed partly to the higher clover biomass, resulting in greater suppression of Palmer amaranth. Following fall inversion tillage, Palmer amaranth was controlled ≥80% in all spring tillage methods compared with NT following NIT. With fall inversion tillage, Palmer amaranth was controlled ≥91% in DCH, DCU and DD spring tillage methods regardless of type of cover crop. Furthermore, Palmer amaranth control in DCU and DD in both crimson clover and winter fallow and DD in cereal rye was significantly higher than in NT following winter fallow in IT. Previous research suggests supplementing the partial weed control obtained following different cover crops in a conservation tillage system [55]. It has been documented that cereal rye cover crop provided short-term weed control in no-till corn, but failed to provide season-long control [53]. Similarly, a 35 and 50% reduction in total weed density by wild radish and cereal rye cover crops, respectively, was recorded in sweet corn (*Zea mays* L.) at 4 WAP. However, weeds were controlled >95% when cover crops were grown in

conjunction with a half or full rate of atrazine and *S*–metolachlor [70]. Several researchers have emphasized the need of conjunction of cover crops with herbicides for effective control of weeds [61, 62, 71].

Six weeks after herbicide application (6 WAA) and before LAYBY application, again, a soil inversion by cover crop by spring tillage method interaction was highly significant; results were similar to those observed at 3 WAA (data not shown). However, the Palmer amaranth control decreased four to ten percent in all spring tillage methods following cereal rye and winter fallow in NIT. Similarly, following IT, Palmer amaranth control decreased one to three percent in all spring tillage methods, regardless of the type of cover crop. Additionally, a year by spring tillage method interaction was highly significant at 6 WAA (Table 5).

In 2009, Palmer amaranth was controlled 95% in DD that was significantly higher than in DCU and NT. Palmer amaranth control in DCH (83%) was not different from DCU and NT. In 2010 and 2011, Palmer amaranth control was significantly higher in DCH, DCU and DD than observed in NT. Furthermore, Palmer amaranth control in 2011 was higher than in 2009 and 2010 due to an additional POST application.

3.4.4. Cotton Yield

Data analysis revealed significant interaction between cover crop and spring tillage method ($p < 0.001$). The main effects of soil inversion, cover crop and spring tillage method were also highly significant ($p < 0.0001$). Cover crop by spring tillage method interaction indicated maximum cotton yields with DD, regardless of type of cover crop (Table 6).

Following cereal rye, the DD tillage method produced maximum cotton (2251 kg ha^{-1}), which was similar to DD following winter fallow and DCH, DCU and DD following crimson

clover. Following crimson clover, DCU and DD cotton yields were 13 and 18%, respectively, higher than NT cotton yields. However, the DCU and DCH cotton yields were similar under different cover crops. As expected, the NT following winter fallow produced the minimum cotton (1082 kg ha^{-1}), which was at least 42% less than the NT following either cereal rye or crimson clover. Averaging over cover crop and spring tillage method, the IT (2133 kg ha^{-1}) cotton yields were 21% higher than NIT (1766 kg ha^{-1}). Similar differences in cotton yields under conservation and conventional tillage systems have been reported by previous researchers [72–75].

3.5. Discussions and Conclusions

Our research evaluated soil inversion, cover crops and spring tillage methods as an integrated approach to managing Palmer amaranth in glufosinate-resistant cotton production. Results indicate that IT alone may result in about 60% reduction in early season Palmer amaranth density. Cover crops can also contribute to early season Palmer amaranth suppression. However, the amount of suppression likely varies with the quantity of biomass produced. Nevertheless, both crimson clover and cereal rye greatly reduced Palmer amaranth emergence compared to winter fallow. Without IT, early season Palmer amaranth densities were 40% less in DCU, DCH and DD when compared with IT. Following IT, no spring tillage method improved Palmer amaranth control. The timely application of glufosinate + *S*-metolachlor POST tank mixture greatly improved Palmer amaranth control in both IT and NIT systems. The timely application of glufosinate + *S*-metolachlor POST tank mixture greatly improved Palmer amaranth control in both IT and NIT. Therefore, from an integrated weed management standpoint, an occasional fall IT could greatly reduce Palmer amaranth emergence on farms

highly infested with glyphosate-resistant Palmer amaranth. For effective and season-long control of Palmer amaranth, one or more POST applications of glufosinate + residual herbicide as tank mixture may be needed in a glufosinate-based cotton production system.

3.6. Acknowledgements

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3.7. Literature Cited

1 Horak, M.J.; Loughin, T.M. Growth analysis of four Amaranthus species. *Weed Sci.* 2000, *48*, 347–355.

2 Keeley, P.E.; Carter, C.H.; Thullen, R.M. Influence of planting date on growth of Palmer amaranth (*Amaranthus palmeri*). *Weed Sci.* 1987, *35*, 199–204.

3 Steckel, L.E.; Sprague, C.L.; Stoller, E.W.; Wax, L.M. Temperature effects on germination of nine Amaranthus species. *Weed Sci.* 2004, *52*, 217–221.

4 Ehleringer, J. Ecophysiology of *Amaranthus palmeri*, a Sonoran Desert summer annual. *Oecologia* 1983, *57*,107–112.

5 Jha, P.; Norsworthy, J.K.; Malik, M.S.; Bangarwa, S.K.; Oliveira, M.J. Temporal emergence of Palmer amaranth from a natural seedbank. *Proc. South. Weed Sci. Soc.* 2006, *59*, 177.

6 Place, G.; Bowman, D.; Burton, M.; Rufty, T. Root penetration through a high bulk density soil layer: differential response of a crop and weed species. *Plant Soil* 2008, *307*, 179–190.

7 Wright, S.R.; Jennette, M.W.; Coble, H.D.; Rufty, T.W. Root morphology of young Glycine max, Senna obtusifolia and *Amaranthus palmeri*. *Weed Sci.* 1999, *47*, 706–711.

8 Jha, P.; Norsworthy, J.K.; Riley, M.B.; Bielenberg, D.G.; Bridges, W., Jr. Acclimation of Palmer amaranth (*Amaranthus palmeri*) to shading. *Weed Sci.* 2008, *56*, 729–734.

9 Bensch, C.N.; Horak, M.J.; Peterson, D. Interference of redroot pigweed (*Amaranthus retroflexus*), Palmer amaranth (*A. palmeri*) and common waterhemp (*A. rudis*) in soybean. *Weed Sci.* 2003, *51*, 37–43.

10 Burke, I.C.; Schroeder, M.; Thomas, W.E.; Wilcut, J.W. Palmer amaranth interference and seed production in peanut. *Weed Technol.* 2007, *21*, 367–371.

11 Klingaman, T.E.; Oliver, L.R. Palmer amaranth (*Amaranthus palmeri*) interference in soybeans (Glycine max). *Weed Sci.* 1994, *42*, 523–527.

12 Massinga, R.A.; Currie, R.S.; Horak, M.J.; Boyer, J. Interference of Palmer amaranth in corn. *Weed Sci.* 2001, *49*, 202–208.

13 Menges, R.M. Allelopathic effects of Palmer amaranth (*Amaranthus palmeri*) and other plant residues in soil. *Weed Sci.* 1987, *35*, 339–347.

14 Menges, R.M. Allelopathic effects of Palmer amaranth *(Amaranthus palmeri*) on seedling growth. *Weed Sci.* 1988, *36*, 325–328.

15 Meyers, S.L.; Jennings, K.M.; Schultheis, J.R.; Monks, D.W. Evaluation of flumioxazin and *S*-metolachlor rate and timing for Palmer Amaranth (*Amaranthus palmeri*) control in sweetpotato. *Weed Technol.* 2010, *24*, 495–503.

16 Monks, D.M.; Oliver, L.R. Interactions between soybean (*Glycine max*) cultivars and selected weeds. *Weed Sci.* 1988, *36*, 770–774.

17 Moore, J.W.; Murray, D.S.; Westerman, R.B. Palmer amaranth (*Amaranthus palmeri*) effects on the harvest and yield if grain sorghum (*Sorghum bicolor*). *Weed Technol.* 2004, *18*, 23–29.

18 Morgan, G.D.; Baumann, P.A.; Chandler, J.M. Competitive impact of Palmer amaranth (*Amaranthus palmeri*) on cotton (*Gossypium hirsutum*) development and yield. *Weed Technol.* 2001, *15*, 408–412.

19 Norsworthy, J.K.; Smith, K.L.; Scott, R.C.; Gbur, E.E. Consultant perspectives on weed management needs in Arkansas cotton. *Weed Technol.* 2007, *21*, 825–831.

20 Norsworthy, J.K.; Griffith, G.M.; Scott, R.C.; Smith, K.L.; Oliver, L.R. Confirmation and control of glyphosate-resistant Palmer amaranth (*Amaranthus palmeri*) in Arkansas. *Weed Technol.* 2008, *21*, 108–113.

21 Rowland, M.W.; Murry, D.S.; Verhalen, L.M. Full-season Palmer amaranth (*Amaranthus palmeri*) interference with cotton (*Gossypium hirsutum*). *Weed Sci.* 1999, *47*, 305–309.

22 Smith, D.T.; Baker, R.V.; Steele, G.L. Palmer amaranth (*Amaranthus palmeri*) impacts on yield, harvesting and ginning in dryland cotton (*Gossypium hirsutum*). *Weed Technol.* 2000, *14*, 122–126.

23 Webster, T.M. Weed survey—Southern states. *Proc. South. Weed Sci. Soc.* 2005, *58*, 291–306.

24 Culpepper, A.S.; Grey, T.L.; Vencill, W.K.; Kichler, J.M.; Webster, T.M.; Brown, S.M.; York, A.C.; Davis, J.W.; Hanna, W.W. Glyphosate-resistant Palmer amaranth (*Amaranthus palmeri*) confirmed in Georgia. *Weed Sci.* 2006, *54*, 620–626.

25 Culpepper, A.S.; Whitaker, J.R.; MacRae, A.W.; York, A.C. Distribution of glyphosate-resistant Palmer amaranth (*Amaranthus palmeri*) in Georgia and North Carolina during 2005 and 2006. *J. Cotton Sci.* 2008, *12*, 306–310.

26 Heap, I. The international survey of herbicide resistant weeds, 2012. Available online: www.weedscience.org (accessed on 11 March 2012).

27 Gosset, B.J.; Murdock, E.C.; Toler, J.E. Resistance of Palmer amaranth (*Amaranthus palmeri*) to the dinitroaniline herbicides. *Weed Technol.* 1992, *6*, 587–591.

28 Ball, D.A. Weed seedbank response to tillage, herbicides and crop rotation sequence. *Weed Sci.* 1992, *40*, 654–659.

29 Barberi, P.; Cozzani, A.; Macchia, M.; Bonari, E. Size and composition of the weed seedbank under different management systems for continuous maize cropping. *Weed Res.* 1998, *38*, 319–334.

30 Cardina, J.; Regnier, E.; Harrison, K. Long-term tillage effects on seedbanks in three Ohio soils. *Weed Sci.* 1991, *39*, 186–194.

31 Dyer, W. Exploiting weed seed dormancy and germination requirements through agronomic practices. *Weed Sci.* 1995, *43*, 498–503.

32 Mulugeta, D.; Stoltenberg, D.E. Weed and seedbank management with integrated methods as influenced by tillage. *Weed Sci.* 1997, *45*, 706–715.

33 Triplett, G.B., Jr.; Lytle, G.D. Control and ecology of weeds in continuous corn without tillage. *Weed Sci.* 1972, *20*, 453–457.

34 Webster, T.M.; Cardina, J.; Norquay, H.M. Tillage and seed depth effects on velvetleaf (*Abutilon theophrasti*) emergence. *Weed Sci.* 1998, *46*, 76–82.

35 Wicks, G.A.; Burnside, O.C.; Felton, W.L. Weed control in conservation tillage systems. In *Managing Agricultural Residues*; Unger, P.W., Ed.; Lewis Publishers: Boca Raton, FL, USA, 1994; pp. 211–244.

36 Yenish, J.P.; Doll, J.D.; Buhler, D.D. Effects of tillage on vertical distribution and viability of weed seed in soil. *Weed Sci.* 1992, *40*, 429–433.

37 Benech-Arnold, R.L.; Sanchez, R.A.; Forcella, F.; Kruk, B.C.; Ghersa, C.M. Environmental control of dormancy in weed seed banks in soil. *Field Crops Res.* 2000, *67*, 105–122.

38 Bond, W.; Grundy, A.C. Non-chemical weed management in organic farming systems. *Weed Res.* 2001, *41*, 383–405.

39 Buhler, D.D.; Kohler, K.A.; Thompson, R.L. Weed seed bank dynamics during five-year crop rotation. *Weed Technol.* 2001, *15*, 170–176.

40 Cardina, J.; Herms, C.P.; Doohan, D.J. Crop rotation and tillage system effects on weed seedbanks. *Weed Sci.* 2002, *50*, 448–460.

41 Clements, D.R.; Benoit, D.L.; Swanton, C.J. Tillage effects on weed seed return and seedbank composition. *Weed Sci.* 1996, *44*, 314–322.

42 Staricka, J.A.; Burford, P.M.; Allmaras, R.R.; Nelson, W.W. Tracing the vertical distribution of simulated shattered seeds as related to tillage. *Agron. J.* 1990, *82*, 1131–1134.

43 Ateh, C.M.; Doll, J. Spring-planted winter rye as a living mulch to control weeds in soybean. *Weed Technol.* 1996, *10*, 347–353.

44 Barnes, J.P.; Putnam, A.R. Evidence for allelopathy by residues and aqueous extracts of rye (*Secale cereale* L.). *Weed Sci.* 1986, *34*, 384–390.

45 Barnes, J.P.; Putnam, A.R.; Burke, B.A.; Aasen, A.J. Isolation and characterization of allelochemicals in rye herbage. *Phytochemistry* 1987, *26*, 1385–1390.

46 Burgos, N.R.; Talbert, R.E. Differential activity of allelochemicals from Secale cereale in seedling bioassays. *Weed Sci.* 2000, *48*, 302–310.

47 Collins, H.P.; Delgado, J.A.; Alva, A.K.; Follett, R.F. Use of nitrogen-15 isotopic techniques to estimate nitrogen cycling from a mustard cover crop to potatoes. *Agron. J.* 2007, *99*, 27–35.

48 Dhima, K.V.; Vasilakoglou, I.B.; Eleftherohorinos, I.G.; Lithourgidis, A.S. Allelopathic potential of winter cereals and their cover crop mulch effect on grass weed suppression and corn development. *Crop Sci.* 2006, *46*, 345–352.

49 Reddy, K.N. Effects of cereal and legume cover crop residues on weeds, yield and net return in soybean (*Glycine max*). *Weed Technol.* 2001, *15*, 660–668.

50 Teasdale, J.R. Contribution of cover crops to weed management in sustainable agricultural systems. *J. Prod. Agric.* 1996, *9*, 475–479.

51 Teasdale, J.R.; Beste, C.E.; Potts, W.E. Response of weeds to tillage and cover crop residue. *Weed Sci.* 1991, *39*, 195–199.

52 Teasdale, J.R.; Mohler, C.L. The quantitative relationship between weed emergence and the physical properties of mulches. *Weed Sci.* 2000, *48*, 385–392.

53 Yenish, J.P.; Worsham, A.D.; York, A.C. Cover crops for herbicide replacement in no-tillage corn (*Zea mays*). *Weed Technol.* 1996, *10*, 815–821.

54 White, R.H.; Worsham, A.D.; Blum, U. Allelopathic potential of legume debris. *Weed Sci.* 1989, *37*, 674–679.

55 Price, A.J.; Reeves, D.W.; Patterson, M.G. Evaluation of weed control provided by three winter cereals in conservation-tillage soybean. *Ren. Agric. Food Sys.* 2006, *21*, 159–164.

56 Price, A.J.; Reeves, D.W.; Patterson, M.G.; Gamble, B.E.; Balkcom, K.S.; Arriaga, F.J.; Monks, C.D. Weed control in peanut grown in a high-residue conservation-tillage system. *Peanut Sci.* 2007, *34*, 59–64.

57 Teasdale, J.R.; Daughtry, C.S.T. Weed suppression by live and desiccated hairy vetch (*Vicia villosa*). *Weed Sci.* 1993, *41*, 207–212.

58 Weston, L. Cover crop and herbicide influence on row crop seedling establishment in no-tillage culture. *Weed Sci.* 1990, *38*, 166–171.

59 Price, A.J.; Balkcom, K.S.; Arriaga, F.J. Rye biomass amount affects weed suppression levels in conversation-tillage cotton. In *Proceedings of the Beltwide Cotton Conference*, New Orleans, LA, USA, 4–7 January 2005, pp. 2921–2923.

60 Price, A.J.; Arriaga, F.J.; Raper, R.L.; Balkcom, K.S.; Kornecki, T.S.; Reeves, D.W. Comparison of mechanical and chemical winter cereal cover crop termination systems and cotton yield in conservation agriculture. *J. Cotton Sci.* 2009, *13*, 238–245.

61 Price, A.J.; Balkcom, K.B.; Culpepper, A.S.; Kelton, J.A.; Nichols, R.L.; Schomberg, H.H. Glyphosate-resistant Palmer amaranth: A threat to conservation tillage. *J. Soil Water Conserv.* 2011, *66*, 265–275.

62 Reeves, D.W.; Price, A.J.; Patterson, M.G. Evaluation of three winter cereals for weed control in conservation-tillage non-transgenic cotton. *Weed Technol.* 2005, *19*, 731–736.

63 Culpepper, A.S.; Kichler, J.; Sosnoskie, L.; York, A.; Sammons, D.; Nichols, R. Integrating cover crop residue and moldboard plowing into glyphosate-resistant Palmer amaranth management programs, 1531. In *Proceedings of Beltwide Cotton Conference*, New Orleans, LA, USA, 4–7 January 2010.

64 Price, A.J.; Stoll, M.E.; Bergtold, J.S.; Arriaga, F.J.; Balkcom, K.S.; Kornecki, T.S.; Raper, R.L. Effect of cover crop extracts on cotton and radish radicle elongation. *Comm. Biometry Crop Sci.* 2008, *3*, 60–66.

65 Reberg-Horton, S.; Burton, J.; Danehower, D.; Ma, G.; Monks, D.; Murphy, J.; Ranells, N.; Williamson, J.; Creamer, N. Changes over time in the allelochemical content of ten cultivars of rye (*Secale cereale* L.). *J. Chem. Ecol.* 2005, *31*, 179–193.

66 Saini, M.; Price, A.J.; van Santen, E. Cover crop residue effects on early-season weed establishment in a conservation-tillage corn-cotton rotation. In *Proceeding of the 28th Southern Conservation Tillage Conference*, Amarillo, TX, USA, 26–28 June 2006; Volume 28, pp. 175–178.

67 Childs, D.; Jordan, T.; Ross, M.; Bauman, T. *Weed Control in No-Tillage Systems*; Conservational Tillage Series CT-2; Purdue University Cooperative Extension Service: West Lafayette, IN, USA, 2001

68 Thullen, R.J.; Keeley, P.E. Control of ivyleaf morningglory (*Ipomoea hederacea*) in cotton (*Gossypium hirsutum*) by combinations of methods. *Weed Tech.* 1994, *8*, 772–776.

69 Wright, S.D.; Vargas, R.N. Integrating weed management tools in cotton and corn: GMO's and conventional herbicides. *Proc. Calif. Weed Sci. Soc.* 2003, *55*, 117–122.

70 Malik, M.S.; Norsworthy, J.K.; Culpepper, A.S.; Riley, M.B.; Bridges, W., Jr. Use of wild radish (*Raphanus raphanistrum*) and rye cover crops for weed suppression in sweet corn. *Weed Sci.* 2008, *56*, 588–595.

71 Price, A.J.; Balkcom, K.S.; Duzy, L.M.; Kelton, J.A. Herbicide and cover cop residue integration for Amaranth control in conservation agriculture cotton. *Weed Technol.* 2012, *26*, 490–498.

72 Aulakh, J.S.; Price, A.J.; Balkcom, K.S. Weed management and cotton yield under two row spacings in conventional and conservation tillage systems utilizing conventional, glufosinate- and glyphosate-based weed management systems. *Weed Technol.* 2011, *25*, 542–547.

73 Balkcom, K.S.; Price, A.J.; van Santen, E.; Delaney, D.P.; Boykin, D.L.; Arriaga, F.J.; Bergtold, J.S.; Kornecki, T.S.; Raper, R.L. Row spacing, tillage system and herbicide technology affects cotton plant growth and yield. *Field Crops Res.* 2010, *117*, 219–225.

74 Daniel, J.B.; Abaye, A.O.; Alley, M.M.; Adcock, C.W.; Maitland, J.C. Winter annual cover crops in a Virginia no-till cotton production system: II. Cover crop and tillage effects on soil moisture, cotton yield and cotton quality. *J. Cotton Sci.* 1999, *3*, 84–91.

75 Unger, P.W.; Vigil, M.F. Cover crops effects on soil water relationships. *J. Soil Water Cons.* 1998, *53*, 241–244.

Table 3.1. Schedule of operations performed during the experiment.

Operations	Experimental Years		
	2008–2009	2009–2010	2010–2011
Broadcasting of Palmer amaranth seed	19 Nov 2008	–	–
Fall inversion tillage	19 Nov 2008	–	–
Planting of cover crops	20 Nov 2008	6 Jan 2010	2 Dec 2010
Rolling and termination of cover crops	22 Apr 2009	18 May 2010	19 Apr 2011
Subsoiling	23 Apr 2009	24 May 2010	26 Apr 2011
Planting of cotton	1 Jun 2009	27 May 2010	5 May 2011
Fertilization (16-16-16)	1 Jun 2009	27 May 2010	5 May 2011
POST application	16 Jun 2009	16 Jun 2010	24 May 2011
Graminicide application (Sethoxydim + COC)	13 Jul 2009	8 Jul 2010	6 Jul 2011
LAYBY application	14 Aug 2009	16 Aug 2010	19 Jul 2011
Cotton defoliation	26 Oct 2009	14 Oct 2010	13 Sep 2011
Cotton harvesting	9 Nov 2009	20 Oct 2010	30 Sep 2011

Table 3.2. Soil inversion, cover crop and spring tillage method means for between-row and within-row Palmer amaranth density at 2 WAP *; data combined over three production years.

	Palmer amaranth density			
	Between-row		Within-row	
Experimental variable	Mean	95% CI	Mean	95% CI
Soil inversion **	plants m^{-2}			
Non-inversion tillage	8 ***	(6,11)	5	(4,7)
Fall inversion tillage	2	(1,3)	2	(2,4)
Cover crop **				
Cereal rye	3	(2,4)	3	(2,4)
Crimson clover	3	(2,4)	3	(2,3)
Winter fallow	7	(6,8)	6	(4,7)
Spring tillage method **				
DCH	4	(3,5)	4	(3,5)
DCU	4	(3,5)	5	(3,6)
DD	3	(3,4)	3	(2,3)
NT	6	(4,7)	3	(2,4)

* Abbreviations: DCH, disking followed by (fb) chisel plow; DCU, disking fb field cultivator; DD, disking fb disking; NT, no tillage; WAP, weeks after cotton planting; ** Soil inversion means averaged over cover crops and spring tillage methods; cover crop means averaged over soil inversion and spring tillage methods; spring tillage method means averaged over soil inversion and cover crops; *** Means, UL and LL columns represent back transformed data.

Table 3.3. Soil inversion by spring tillage method interaction means for between-row Palmer amaranth density at 6 WAP *; data combined over cover crops and production years.

Experiment variable		Between-row density	
Soil inversion	Spring tillage method	Mean	95% CI
		plants m^{-2}	
IT *	DCH *	2 **	(1,3) **
	DCU	2	(1,3)
	DD	2	(1,3)
	NT	6	(4,8)
NIT	DCH	4	(3,6)
	DCU	7	(5,10)
	DD	4	(2,5)
	NT	19	(11,27)

* Abbreviations: DCH, disking followed by (fb) chisel plow; DCU, disking fb field cultivator; DD, disking fb disking; IT, fall inversion tillage; NIT, non-inversion tillage; NT, no tillage; WAP, weeks after cotton planting; ** Means averaged over cover crops and years; means and 95% CI columns represent back transformed data.

Table 3.4. Soil inversion by cover crop by spring tillage method interaction means for Palmer amaranth control at 3 WAA *; data combined over production years.

		Spring tillage method							
Experimental	Variable	DCH *		DCU		DD		NT	
Soil inversion	Cover crop	Mean	95% CI	Mean	95% CI	Mean	95% CI	Mean	95% CI
					%				
IT *	Crimson clover	94 **	(85,99) **	99	(94,100)	99	(94,100)	88	(77,96)
	Cereal rye	96	(89,100)	94	(87,99)	99	(96,99)	94	(85,99)
	Winter fallow	91	(81,97)	97	(90,100)	97	(90,100)	80	(67,90)
NIT	Crimson clover	89	(78,96)	90	(84,98)	93	(83,98)	73	(59,85)
	Cereal rye	77	(63,89)	74	(60,86)	95	(87,99)	63	(49,77)
	Winter fallow	84	(72,93)	74	(60,86)	91	(81,98)	0	(0,2)

* Abbreviations: DCH, disking followed by (fb) chisel plow; DCU, disking fb field cultivator; DD, disking fb disking; NT, no tillage; IT, fall inversion tillage; NIT, non-inversion tillage; WAA, weeks after post application; ** Means averaged over years; means and 95% CI columns represent back transformed data.

Table 3.5. Year by spring tillage method interaction means for Palmer amaranth control 6 WAA *; data combined over soil inversion and cover crops.

	Spring tillage method							
	DCH *		DCU		DD		NT	
Year	Mean	95% CI	Mean	95% CI	Mean	95% CI	Mean	95% CI
				%				
2009	83 **	(72,92) **	69	(56,81)	95	(88,99)	65	(51,78)
2010	85	(78,90)	85	(79,91)	83	(77,89)	35	(27,42)
2011	99	(98,100)	99	(98,100)	99	(98,100)	85	(80,89)

* Abbreviations: DCH, disking followed by (fb) chisel plow; DCU, disking fb field cultivator; DD, disking fb disking; NT, no tillage; WAA, weeks after post application; ** Means averaged over soil inversion and cover crops; means and 95% CI columns represent back transformed data.

Table 3.6. Interaction effect of cover crop by spring tillage method on cotton yield; data combined over cover crop and production years.

	Spring tillage method			
Experimental variable	DCH *	DCU	DD	NT
Cover crop	kg ha^{-1}			
Crimson clover	2075 abc **	2126 ab	2213 ab	1877 c
Cereal rye	1867 c	1965 bc	2251 a	1956 bc
Winter fallow	1952 bc	1867 c	2153 ab	1082 d

* Abbreviations: DCH, disking followed by (fb) chisel plow; DCU, disking fb field cultivator; DD, disking fb disking; NT, no tillage; ** Means averaged over cover crops and years; multiple mean comparisons were made using 'adj = simulate' option in SAS PROC GLIMMIX; means followed by same letter are not significantly different (p = 0.05).

Chapter 4

Pushing Towards Cogongrass Patch Eradication: The Influence of Herbicide Treatment and Application Timing on Cogongrass Rhizome Elimination

4.1. Abstract

Cogongrass is an invasive grass native to Asia that has infested thousands of hectares in the southeastern US. While numerous studies have examined cogongrass control, no published studies have tested strategies for cogongrass eradication. Since cogongrass has a persistent, thick, rhizome mat and ephemeral seedbank, successful eradication methods must largely focus on the rhizome issue. A field study was conducted at two locations near Tillman's Corner and Bayou La Batre in southwestern Alabama to evaluate specific herbicide treatments at spring, summer, and fall application timings for cogongrass patch eradication. Herbicide treatments included glyphosate at 4.48 kg ai ha^{-1}, imazapyr at 0.84 kg ai ha^{-1} and a tank mix of glyphosate and imazapyr at the same rates. The three application times were May, August and October and treatments were applied at each timing for three consecutive years. Cogongrass visual control, vegetative cover, shoot and rhizome biomass, and rhizome depth and energy content were sampled over the course of the study. Cogongrass response to treatments varied by location but by 36 months after initial treatment (MAIT), complete elimination of cogongrass shoot and rhizome biomass, and 100 % visual control was achieved in several herbicide treatment timing combinations at both locations. These included glyphosate + imazapyr treatment at any application timing, imazapyr treatment in August or October, and glyphosate treatment applied in May and October of each year. Total nonstructural carbohydrate (TNC) levels of healthy

rhizomes were not affected by herbicide treatments but a seasonal pattern was observed. The maximum live-rhizome depth was not influenced by any treatment, indicating that herbicides were not preferentially leaving deeper surviving rhizomes. These results demonstrate for the first time that the entire rhizome layer of cogongrass can be eliminated with multiple treatment options within three years and that cogongrass patch eradication is possible for many land managers.

4.2. Introduction

The term ‘eradication’ is often misinterpreted, especially by public policy makers; it is not synonymous with the term ‘control’ (Zamora at al.1989). Eradication is broadly defined as the destruction of every propagule of a species from an area surrounded by natural or manmade barriers sufficiently effective to prevent reinvasion except at man’s intervention (Newsom 1978, Zamora et al. 1989). Bomford and O’Brien (1995) define it as ‘complete and permanent elimination of all wild populations from a defined area by a time-limited campaign’. Despite the existence of evidence of successful eradication of several species, there is still a widespread view that eradication is not feasible. The fundamental reason for this view may be reverse hype, i.e., unsuccessful events are always much touted and widely criticized while the success stories are often under reported (Simberloff 2009). In the past, several invasive invertebrate species have been eradicated successfully (Klassen and Curtis 2005; Simberloff 2002a; Wittenberg and Cock 2001). However, the eradication of invasive plants is often regarded as impossible owing to the existence of a soil seed bank for many species (Rejmanek and Pitcairn 2002). Contrarily, several weed eradication projects have been successful; eradication of sandbur (*Cenchrus echinatus*) from Laysan (Flint and Rehkemper 2002) and Bassia scoparia (*Kochia scoparia* L.) from a large

area in Western Australia (Randall 2001). Several small scale eradications have also been cited by Mack and Lonsdale (2002) in Australia, New Zealand, and the United States. The success of an eradication project is largely governed by five crucial features: early detection and prompt action, knowledge of biology and ecology of the target species, a blanket eradication approach, unflagging motivation and availability of resources (Myers et al. 2000; Mack and Lonsdale 2002; Simberloff 2002b; Mack and Foster 2004). Successful eradication of the Pacific alga *Caulerpa taxifolia* from southern California (Merkel & Associates. 2006) is an outstanding example of the significance of quick and decisive action; whereas, the failure to eradicate this species in Europe owes to the lack of timely and decisive action (Meinesz 2001). Although early detection and prompt action are not absolutely necessary, the eradication of populations confined to a small area is much more cost-effective and may have a high probability of success (Simberloff 2009; Soria et al. 2002). Eradication of witchweed (*Striga asiatica* M. Vahl) is an example of a large scale campaign against a long established and widespread invasion, which has cost over 100 million dollars to date (Eplee 2001). As a result of this mega scale effort, witchweed infestations in the United States have been reduced from 200,000 ha in the early 1970s (Eplee 2001) to approximately 900 ha by the end of 2007. Clearly, the costs and rigors of invasive species eradication can be tremendous. However, invasive weed management decisions cannot be based solely upon cost benefit analyses of agricultural and forestry production areas. Therefore, if eradication is feasible, it should be attempted since it may be more cost-effective than any other control method (Wittenberg and Cock 2001). Bomford and O'Brien (1995) determined six criteria for assessing whether eradication is technically possible and preferable to continuing control. These six criteria include: 1) Rate of removal exceeds rate of increase at all population densities; 2) Immigration is zero; 3) All reproductive individuals must be at risk; 4) Target

species can be detected at low densities; 5) Discounted benefit-cost analysis favors eradication over control and; 6) There must be a suitable socio-political environment.

Cogongrass [*Imperata cylindrica* (L.) Beauv. var. *major* tribe *Andropogoneae*] is a highly invasive, perennial, rhizomatous grass and a serious weed in over 73 countries around the world (MacDonald 2004). It has been ranked as the seventh most troublesome weed worldwide (Falvey 1981; Holm et al. 1977). At present it occupies more than 500,000 ha of land in the United States and classified as a noxious weed in Alabama, Florida, Georgia, Louisiana, Mississippi, Oregon, South Carolina, Texas and Virginia. (Bryson and Carter 1993; Byrd and Bryson 1999, Faircloth et al. 2005; Patterson et al.1980 and 1983; Van Loan et al. 2002; Willard et al. 1988 and 1990). Cogongrass is well recognized for its adverse economic, ecological and ecosystem impacts. Additionally, consideration of the economics of the pine industry and the risk cogongrass may pose to human life and property via ignition of wildfires might magnify the benefit cost ratio (Jose et al. 2002; Miller, 2000). Lippincott (2000) described cogongrass as a powder keg for forest fires. Cogongrass competitively displaces native vegetation and strongly alters wildlife habitats, feeding and breeding grounds that could eventually lead to tremendous loss of biodiversity. Southeast Asia is a glaring example of pseudo 'biological deserts' created by cogongrass where solid cogongrass stands known as 'Imperata sheets' or 'Imperata savannas' are the only vegetation growing on areas greater than 8,900 continuous hectares (Suryatna and McIntosh 1980).

Recently, Alabama spent almost 6.3 million dollars on control and localized eradication of cogongrass infestations (Schelhas 2011). In the context of cogongrass, the term eradication specifically insinuates complete elimination of all living propagules, both sexual (seed) as well as asexual (rhizomes) within a defined boundary. The current cogongrass eradication approach

largely resembles the wild fire management paradigm which encompasses prevention, early detection, quick initial attack, integrated control and site rehabilitation (Dewey et al. 1995). Similar to wildfires, new invasive weed infestations frequently arise as a single plant or small patch followed by a slow growth lag phase which ultimately lead to exponential spread. The movement of seed or rhizomes by various dispersal agents exacerbates the problem as new 'spot fire' patches establish beyond the main infestation.

Since the goals of eradication are much more rigid than weed control, eradication treatments must have extremely high efficacy (Zamora et al. 1989). Historically, several chemical and non-chemical methods have been tested for the control of cogongrass (Akobundu 1992, 1993; Akobundu et al. 2000; Byrd and Bryson 1999; Dickens and Buchanan 1975; Gaffney 1996; Johnson et al. 1999; MacDonald et al. 2001 and 2002; Miller 2000; Ramsey et al. 2003; Terry et al. 1997; Willard et al. 1996 and 1997). Glyphosate and imazapyr have been identified as the most effective herbicides for cogongrass management (Dozier et al. 1998; Udensi et al. 1999). Repeated applications of these herbicides over multiple years have been reported to provide greater than 95% control (Miller 2007). Fall-season applications (September to November) of glyphosate or imazapyr have been shown to be more effective due to better translocation of herbicides to the underground rhizomes as photosynthates are directed towards rhizomes in the fall (Faircloth et al. 2005; Johnson 1999 and 2000; Miller 2007; Shilling et al. 1997). However, the focus of all of these previous research efforts was only to attain satisfactory control of cogongrass, not eradication. To date, no research has been published on methods suitable for the eradication of cogongrass patches. Since cogongrass seeds are very short lived in the environment (MacDonald 2004), eliminating the dense, rhizome mat is very likely the limiting factor for successful eradication in most situations. Previous researchers have observed

low spikelet fill (0 to 40 %), short seed viability (< 16 months), and poor seedling survival (Sajise, 1972; Santiago, 1974; Dickens, 1976; Santiago, 1980; Kushwaha et al., 1983; Hopkins and Graham, 1984). Additionally, glyphosate and imazapyr herbicides reduce cogongrass cover and seedhead production more than 80 % and 97 %, respectively, up to a year following treatment (Enloe et al. 2012). Byrd (2007) reported reduction in number of viable seeds following treatment with several herbicides. In light of these views, we assumed non-existence of a viable cogongrass seed bank in the treated plots. Therefore, our primary objective in this study was to determine the feasibility of cogongrass patch eradication while targeting elimination of the entire rhizome layer. Our overall research questions were as follows:

1) Can glyphosate and/or imazapyr be effective for cogongrass patch eradication?

2) Does the combination of glyphosate and imazapyr improve cogongrass control over either herbicide alone?

3) Does the timing of repeated herbicide applications (spring, summer, or fall) influence the effectiveness of glyphosate and imazapyr for cogongrass patch eradication?

4) Do repeated herbicide treatments influence energy levels or depth of surviving rhizomes?

4.3. Materials and Methods

A field study was conducted from 2008 through 2011 at two locations near Tillman's Corner (30.50282^{O} N, -88.15251^{O} W) and Bayou La Batre (30.42500^{O} N, -88.28835^{O} W) in southwestern Alabama. Both locations were abandoned open fields with near monotypic stands of cogongrass that had not been managed for several years. Soil at the Tillman’s Corner site was a Benndale sandy loam (siliceous, subactive, thermic Typic Paleudults). Soil at Bayou La Batre

was Troup–Heidel loamy sand (siliceous, semiactive, thermic Typic Paleudults). Soils at both locations were well drained, greater than 60 inches deep and strongly acidic in reaction. The study was established at each location in a randomized complete block design with four replications. The plot size was 9.1 m by 9.1 m (30 by 30 ft) with a 3 m (10 ft) buffer around each plot that was maintained cogongrass free for the duration of the study to prevent rhizome growth between plots. Since a cogongrass patch can grow more than a meter in a year (Yager, 2007), wider buffers were made to eliminate the chance of encroachment from adjacent plots. Plot size was kept larger compared to 1.8 m by 4.7 m in previous studies (Willard et al. 1996 and 1997) for greater efficacy of treatment application and more valid estimates of research variables. The experiment consisted of a factorial arrangement of three herbicide treatments and three application timings, resulting in nine treatments. A nontreated control was also included. Herbicide treatments were glyphosate applied at 4.48 kg ai ha^{-1} (4.0 lb ai $acre^{-1}$) (Accord® Concentrate, Dow AgroSciences LLC, Indianapolis, IN), imazapyr applied at 0.84 kg ai ha^{-1} (0.75 lb ai $acre^{-1}$) (Arsenal® Powerline, BASF, Research Triangle Park, NC), and a combination of glyphosate and imazapyr at the same rates. The three application times were May, August and October. Each herbicide treatment and timing combination was applied in 2008, 2009, and 2010 to the same plots. Across all three years, the spring, summer and fall treatments were applied between May 16th to 20th, July 30th to August 10th, and October 5th to 12th, respectively. The May glyphosate-alone treatment was modified to also include a second glyphosate treatment at the same rate each October. However, both the August and October glyphosate treatments were applied once a year due to non-feasibility of second treatment within a year. Methylated seed oil at 1 % v/v (Destiny® HC, Winfield Solutions LLC, St. Paul, MN) was added to the imazapyr alone and glyphosate + imazapyr treatments, and a nonionic surfactant at 0.25 % v/v

(Timberland® 90, UAP, Loveland Products Inc, Loveland, CO) was added to the glyphosate treatment. At each application timing, treatments were applied at 187 liters ha^{-1} to green and actively-growing cogongrass with an ATV mounted boom sprayer fitted with AI 11002 nozzles operating at 50 psi. Cogongrass biomass at the time of each treatment varied over the course of the study. At the time of initial treatment, all plots had 80-100% green vegetative cover. However, cogongrass height and cover continuously declined across all herbicide treated plots over the duration of the study. At each location, temperature (Figure 4.1) and precipitation (Figure 4.2) patterns closely followed historic averages during the treatment application times. The mean air temperature at the time of treatment application ranged from 70 to 80 (F) for the May and October treatments and was in the low to mid 90s (F) for the August treatments. In August 2009, a precipitation event of about 1 cm rain occurred within 3 hours of treatment application. This was the only treatment time over all three years that may have been influenced by precipitation within 4 hours of treatment application.

Data were collected three times each year (May, August and October) on percent green cogongrass cover, visual percent control, cogongrass green shoot biomass, rhizome biomass, maximum live-rhizome depth, and rhizome total non-structural carbohydrate (TNC) content. Data on rhizome and shoot biomass, maximum live rhizome depth were recorded from a 0.25 m^2 quadrat randomly placed in each plot. Green cogongrass shoots were clipped at the ground level inside the quadrat and oven dried at 60 C for 72 h for shoot dry weight. Rhizome biomass was quantified by excavating a 50 by 50 cm pit beneath each quadrat to a depth of 30 cm. The bottom of each pit was closely inspected to verify that no rhizomes were present below the excavation point. Over the course of the study, with over 700 rhizome pits excavated, no cogongrass rhizomes were ever found below a 30 cm soil depth. Excavated rhizomes were separated from

the soil, washed and classified as alive or dead. Rhizomes were conservatively classified as dead only if they were completely desiccated or degraded with absolutely no live tissue remaining. All live rhizomes within each quadrat were oven dried at 60 C for 72 h and weighed. Visual assessments of cogongrass control were made for the entire plot on a 0 to 100 % scale with zero being equivalent to the vegetative cover and vigor of cogongrass in the nontreated control plots in each block and 100 being complete death of all cogongrass shoots in the treated plot. Visual estimates of green cogongrass cover were recorded to the nearest one percent from an additional three randomly placed 0.25 m^2 quadrats in each plot.

To quantify TNC content, 15 grams of healthy white rhizomes were collected from the rhizomes harvested from each pit, placed inside a plastic freezer bag and stored on dry ice inside a cooler to prevent respiration losses during transportation. The rhizome samples were collected from horizontal rhizome sections found between root crowns and new rhizome tips generally exhibiting curvature upward. These sections were previously demonstrated to have a higher TNC content than vertical sections (S.F. Enloe, unpublished data). The rhizome samples were kept in a freezer at -20^o C until analysis. TNC content was determined by using a modified version of the Shaffer-Somogyi method (Harding and Downs, 1933). The method consisted of digesting 0.20 to 0.25 g of finely ground (less than 1 mm) rhizome samples with 50 ml of 0.05 N H_2SO_4 and boiling for 15 minutes. The samples were then cooled in a shallow ice-water bath, and 2.5 to 3.0 ml 1.0 N NaOH solution was added. The pH of the samples was maintained at 4.5 ± 0.1 while stirring using 1.0 N and 0.1 N H_2SO_4 and 1.0 N and 0.1 N NaOH. One ml of glucoamaylase enzyme solution was then added and stirring was continued. The samples were then incubated for one hour and filtered in a 250 ml volumetric flask using 541 Whatman filter paper or glass wool. Ten ml of diluted sample was transferred to a test tube and 10 ml of Shaffer-Somogyi

solution were then added. Samples were subsequently boiled for 15 minutes and cooled immediately in an ice-bath. The cooled samples were treated with 2 ml of potassium iodide-oxalate, 10 ml of 1.0 N H_2SO_4, 0.2 ml of 'Fastbreak' defoamer (1:100) solution, and 1 ml of 1 % starch solution. Finally, the samples were titrated with 0.02 N sodium thiosulphate solution until a clear light blue end-point. The percentage of TNC in the rhizomes was calculated on a dry weight basis using following formula.

$$\text{Percent TNC} = \frac{\text{Sample TNC(mg)} - \text{Enzyme TNC (mg)} * 100}{\text{Dry sample weight (mg)}}$$

The sample TNC was determined by subtracting the sample titer value from the blank. Enzyme TNC was the mg of glucoamaylase enzyme used.

4.4. Statistical Analysis

Data were analyzed using generalized linear mixed models or linear mixed models methodology as implemented in SAS® PROC GLIMMIX based on a randomized complete block design (r= 4) with split plot in time restriction on randomization. Before statistical analysis, data on shoot and rhizome biomass and cogongrass cover were converted into a percentage reduction compared to the nontreated control to adjust for variation not associated with the treatments. Percent control data was analyzed without the nontreated control values. The rhizome depth and TNC data were analyzed including the nontreated control. Analyses were done using 12, 24, and 36 MAIT (months after initial treatment) data. Location, treatment, MAIT, and their interactions were treated as fixed effects while block (location), treatment by block (location), and MAIT by treatment by block (location), were treated as random effects. The factor MAIT was of a repeated measures nature that induced a covariance relationship because of the lack of re-randomization. A heterogeneous autoregressive covariance structure ARH (1) was used to model

the covariance relationship between observations taken from the same plot at 12, 24, and 36 MAIT. Where the location and/or location by treatment interaction were significant, the data are presented separately by location. Rhizome and shoot biomass, percent visual control and percent cover data were transformed with arcsine of the square root but data transformation did not change the results, therefore non-transformed means are presented. No transformations were required for % TNC and rhizome depth data. Multiple means' comparisons of significant effects were made using the 'Adj=simulate' option in SAS PROC GLIMMIX at the 5% significance level.

4.5. Results and Discussion

Mean monthly air temperature and cumulative precipitation data indicate fairly stable climatic conditions from year to year. The pattern of mean monthly air temperature during each year was quite similar to the 118–year average (Figure 4.1). Similarly, the precipitation pattern was relatively stable. However, the cumulative precipitation varied from year to year and from the 118-year average of 157 cm (Figure 4.2). Cumulative precipitation totaled 137, 193, 152, and 127 cm during 2008, 2009, 2010, and 2011, respectively (http://www.ncdc.noaa.gov). The fall of 2009 and winter of 2010 were slightly wetter than in other years, while the 2011 spring and the summer were drier than normal.

Analysis of the data revealed significant effects of location and location by treatment interaction. This suggested that the efficacy of certain treatments varied with the location. Within each location however, the 12, 24, and 36 MAIT data indicated a similar pattern of differences among treatments. Since the main objective of this research was the eradication of cogongrass,

the results from different response variables are discussed with a focus on 36 MAIT data within each location.

In the nontreated control plots within a location, significant differences occurred in percent cover, shoot biomass and TNC levels over the three treatment application months at 36 MAIT (Table 4.1). However, the rhizome depth was similar at both locations. At 36 MAIT, percent cover in nontreated control plots varied from 51 to 73 % at Tillman's Corner and from 58 to 80% at Bayou La Batre. At both locations, percent cover in October was significantly higher than in May. A similar trend was observed for shoot biomass. At Tillman's Corner, shoot biomass ranged from 156 g m^{-2}(1.56 tons ha^{-1}) in May to 356 g m^{-2} (3.56 tons ha^{-1}) in October. At Bayou La Batre, shoot biomass varied between 300 g m^{-2} (3.0 tons ha^{-1}) in May to 524 g m^{-2} (5.24 tons ha^{-1}) in October. No differences were observed in rhizome biomass over the three treatment application months at Tillman's corner. Previous research also documented similar seasonal variation in shoot biomass (Shilling et al. 1997).

At Tillman's Corner, rhizome biomass did not vary significantly over the sample periods, ranging from 6.2 to 6.7 tons ha^{-1}. At Bayou La Batre, biomass varied between 4.6 tons ha^{-1} in May to 8.7 tons ha^{-1} in October. Similar quantities of rhizome biomass have been reported by of previous researchers (Soerjani, 1970 and Omezine and Harzalla, 2009). Moreover, rhizome biomass levels as high as 40 tons ha^{-1} have also been recorded (Terry et al.1997). The minimum rhizome and shoot biomass in May could be attributed to the utilization of reserved photosynthates in rhizomes for new growth as well as flowering that in Alabama typically occurs in the spring. The maximum biomass in October resulted from the production of new rhizomes throughout the summer and fall. Additionally, some temporal and spatial variation in rhizome biomass may also occur due to density related mortality (Reineke 1933; Yoda et al. 1963).

Total nonstructural carbohydrate levels at both locations differed significantly over the three treatment application months. At Tillman's Corner, the May TNC levels were significantly lower than in October. However at Bayou La Batre, the lowest TNC levels occurred in August. Previous research indicates significant seasonal variations in the TNC content of other plants (Amy 1932; Becker and Fawcett 1998; Hodgson 1968; Potter et al 1986; Saldivar et al. 1992; Saxena and Ramakrishan 1983; Stam Katovich et al. 1998). In addition, diurnal variations in TNC content have also been reported (Greenfield and Smith 1974).

With regard to rhizome depth, no significant difference occurred over the sample periods in the nontreated control plots. At both locations average rhizome depth was 16-17 cm. Usually, rhizomes grow shallower (less than 20 cm) in fine textured soils and deeper (greater than 50 cm) in sandy soils (Omezine and Harzalla 2009). However, past research has documented cogongrass rhizomes to grow as deep as one meter (Holm et al. 1977; Bryson and Carter 1993; Gaffney 1996; Omezine and Harzalla 2009). Nevertheless, cogongrass is unlikely to send out shoots from rhizome located deeper than 8 cm (Wilcut et al. 1988a and 1988b).

At 36 MAIT at Tillman's Corner, complete eradication of cogongrass was achieved at Tilman's Corner with glyphosate applied in May and October, imazapyr applied in August or October, and glyphosate + imazapyr applied at any of the three timings. Collectively, zero percent cogongrass cover from the quadrat data, a 100% reduction in shoot and rhizome biomass, and 100% visual control (which encompassed a thorough search of the entire plot for any cogongrass shoots not detected in quadrat sampling), and a zero measurement of rhizome depth, data indicated the achievement of eradication by these treatments (Table 4.2). With the imazapyr treatment applied in May, cogongrass shoot and rhizome biomass was not detected within the sampling area (0.25 m^{-2}quadrat). However, percent cover in three additional quadrats and a

visual percent control assessment on the whole plot basis indicated a small percentage of cogongrass remaining which indicates eradication was not achieved. Glyphosate applied in either August or October completely failed in cogongrass eradication, as evidenced by all variables collected.

At 36 MAIT for each treatment timing at Bayou La Batre, complete eradication was achieved with all imazapyr and glyphosate + imazapyr treatments (Table 4.3). Glyphosate applied in May and October each year also resulted in eradication. However, glyphosate applied in either August or October did not eradicate cogongrass by 36 MAIT, although these treatments were more effective at Bayou La Batre than Tillman's Corner.

On the basis of these results, the following treatments are found to be highly potent for eradicating cogongrass; 1) Glyphosate applied in May and October; 2) imazapyr applied at August or October timings; 3) glyphosate + imazapyr applied at May, August or October timings. Over the duration of this study, the pattern of rhizome biomass depletion over time was the single most suitable criterion to elucidate the relative efficacy of treatments. However, the confirmation of eradication was done based on multiple criteria as previously described. Therefore, to demonstrate the relative speed of eradication with different treatments, the following discussion will focus on the rhizome biomass depletion trends in select treatments within a location.

4.5.1. Tillman's Corner

With the May + October glyphosate treatment, the rhizome biomass elimination pattern was not very consistent until 18 MAIT (Figure 4.3a). Six months after the first glyphosate treatment i.e., in October 2008 rhizome biomass showed significant recovery. However, the

October glyphosate treatment reversed the trend again. By 12 MAIT, rhizome biomass (280 g m^{-2}) was 64% lower compared with rhizome biomass before treatment initiation (784 g m^{-2}). Previous researchers obtained as high as 80% reduction in rhizome biomass 12 MAIT with a single glyphosate treatment, alone or in combination with disking (Enloe et al. 2012; Ramsey et al. 2003; Shilling et al. 1997; Willard et al. 1996). A second uptrend in rhizome biomass occurred at 18 MAIT i.e., in October when rhizome biomass almost doubled compared to that at 12 MAIT. However, the glyphosate treatment in October 2009 again reversed the trend. By 24 MAIT, 77% of the rhizome biomass (180 g m^{-2}) was eliminated compared with 784 g m^{-2} measured before treatment initiation. After the fourth glyphosate treatment, rhizome biomass followed a continuous downtrend and never recovered. By 30 MAIT, rhizome biomass (8 g m^{-2}) was 99% eliminated. Complete rhizome eradication in May + October glyphosate treated plots was achieved at 36 MAIT which was confirmed by all variables collected.

With the August imazapyr treatment, rhizome biomass was eliminated linearly from 3 MAIT to 15 MAIT. By 12 MAIT rhizome biomass was reduced significantly, by more than 74 % (to 235 g m^{-2}) compared with before treatment biomass of 896 g m^{-2} (Figure 4.3b). Similar results have been reported at 12 MAIT in previous studies (Enloe et al. 2012; Ramsey et al. 2003; Johnson et al.1999; Shilling et al. 1997). From 15 to 24 MAIT, more than 96% of rhizome biomass was eliminated and rhizomes were totally eliminated in three of the four blocks. Cogongrass eradication in all 4 blocks was achieved at 33 MAIT as confirmed by all variables collected.

With the October imazapyr treatment, rhizome biomass was eliminated almost linearly until 12 MAIT. By 12 MAIT, rhizome biomass was reduced by 90% (to 77 g m^{-2}) compared with 744 g m^{-2} before treatment initiation. From 12 to 24 MAIT, the rate of further elimination was

very low. By 24 MAIT, more than 99% of rhizome biomass was eliminated, with total elimination of rhizomes in three of the four blocks. At 30 MAIT, rhizome biomass was completely eliminated in all four blocks which was confirmed by all variables collected. These results indicate that the October imazapyr treatment resulted in eradication 3 months earlier than the August imazapyr treatment.

The May glyphosate + imazapyr treatment eliminated rhizome biomass almost linearly until 15 MAIT (Figure 4.3c). By 12 MAIT, rhizome biomass (307 g m^{-2}) was reduced by 61% and was significantly lower than rhizome biomass before treatment initiation (784 g m^{-2}) (Figure 4.3c). Minogue et al. (2012) obtained 60 to 65 % control of cogongrass two years after treatment. By 15 MAIT, rhizome biomass (86 g m^{-2}) was 89% eliminated and was significantly less than at 12 MAIT. By 24 MAIT, 99% of rhizome biomass was eliminated. At 27 MAIT, rhizome biomass was not detected within the sampling area (0.25 m^{-2}quadrat) in any of the four blocks. However, complete rhizome eradication as confirmed by all variables collected was not achieved until 30 MAIT. Similarly, with the August glyphosate + imazapyr treatment rhizome biomass was reduced by 65 % to 314 g m^{-2} by 12 MAIT, which was significantly less than the before treatment biomass (896 g m^{-2}). By 24 MAIT, rhizome biomass (77 g m^{-2}) was 95 % eliminated, which was a significant reduction from 12 MAIT levels. At 27 MAIT, no rhizomes were detected within the sampling area (0.25 m^{-2}quadrat) in any of the four blocks. However, as confirmed by all variables collected, complete eradication was not achieved until 33 MAIT which With regard to October glyphosate + imazapyr treatment, rhizome biomass (287g m^{-2}) was reduced by 61% and significantly lower by 12 MAIT than biomass before treatment initiation (744 g m^{-2}). Furthermore, by 15 MAIT, rhizome biomass was reduced by 93% to 49 g m^{-2} which was a significant drop from 12 MAIT. By 24 MAIT, 99% of the rhizome biomass was

eliminated and rhizomes were detected in only one of the four blocks. The rhizome biomass was completely eliminated, as confirmed by all variables collected, at 27 MAIT. Therefore, the October glyphosate + imazapyr treatment provided cogongrass eradication three and six months earlier than the similar May and August treatments, respectively. In contrast, Minogue et al. (2012) found a September glyphosate + imazapyr treatment more effective than an October treatment.

4.5.2. Bayou La Batre

The May + October glyphosate treatment was highly effective and indicated a steeper and more consistent pattern of rhizome biomass elimination compared with the same treatment at Tillman's Corner. The first glyphosate treatment resulted in only 43 % elimination of rhizome biomass. However, there was no upward trend in biomass at 6 MAIT as observed at the Tillman's Corner site. By 12 MAIT, rhizome biomass was reduced by 95% (to 34 g m^{-2}), compared with biomass of 756 g m^{-2} before treatment initiation (Figure 4.4a). In a previous study with the same treatments, Enloe et al. (2012) obtained 77% reduction in rhizome biomass 12 MAIT. After the third glyphosate application in May 2009, rhizomes were detected within the sampling area (0.25 m^{-2}quadrat) in only one of the four blocks. Complete rhizome eradication, as confirmed by all variables collected, occurred at 18 MAIT.

With the May imazapyr treatment, rhizome biomass was eliminated almost linearly through 15 MAIT. By 12 MAIT, rhizome biomass (161 g m^{-2}) was significantly reduced compared to 756 g m^{-2} measured before treatment initiation (Figure 4.4b). Similar results have been reported by previous researchers (Enloe et al. 2012; Ramsey et al. 2003; Shilling et al. 1997). By 15 MAIT, rhizome biomass was completely eliminated in three of the four blocks.

However, complete rhizome eradication, as confirmed by all variables collected, was not achieved until 27 MAIT. Similarly, with the August imazapyr treatment, rhizome biomass was eliminated linearly through 12 MAIT. By 12 MAIT, rhizome biomass was reduced 99%, from 922 g m^{-2} before treatment initiation to 4.7 g m^{-2}. By 18 MAIT, rhizome biomass was completely eliminated in three of the four blocks. However, complete rhizome eradication was not confirmed until 33 MAIT. With the October imazapyr treatment, most rhizome biomass reduction (88 %) occurred by 9 MAIT. By 12 MAIT, rhizome biomass was reduced by 95%, from 812 g m^{-2} before treatment initiation to 43 g m^{-2}. From 12 to 24 MAIT, rhizomes were detected within the sampling area (0.25 m^{-2}quadrat) in only two of the four blocks. By 24 MAIT, rhizomes were eliminated in three of the four blocks. However, complete rhizome eradication, as confirmed by all variables collected, was not achieved until 30 MAIT.

With the May glyphosate + imazapyr treatment, rhizome biomass was reduced significantly by 3 MAIT to 343 g m^{-2} compared with 756 g m^{-2} before treatment initiation. By 12 MAIT, 72% of rhizome biomass was eliminated (Figure 4.4c). Furthermore, by 15 MAIT, rhizome biomass was completely eliminated in two of the four blocks. Complete rhizome eradication was achieved at 27 MAIT as confirmed by all variables collected. Similarly with the August glyphosate + imazapyr treatment, rhizome biomass was eliminated linearly until 12 MAIT. Rhizome biomass was significantly reduced by 6 MAIT to117 g m^{-2} compared with 922 g m^{-2} before treatment initiation. By 12 MAIT, rhizome biomass was 97% eliminated. However, complete rhizome eradication, as confirmed by all variables collected, was not achieved until 27 MAIT. A similar trend of rhizome biomass elimination and eventual eradication was observed with the October glyphosate + imazapyr treatment.

Our research indicates significant differences in the response of cogongrass to May + October glyphosate treatment at two locations. Rhizome eradication occurred more quickly at Bayou La Batre (18 MAIT) than at Tillman's Corner (36 MAIT). Similar location related differences in response of cogongrass to foliar applications of glyphosate were reported from central Florida (Shilling et al.1997). The observed differences in glyphosate efficacy at two locations may be due to the morphological and ecotypic differences. Similarly, imazapyr and glyphosate + imazapyr resulted in higher initial elimination of rhizome biomass at Bayou La Batre than at Tillman's Corner. These differences may be attributed to the variation in the soil type (Shilling et al. 1997), cogongrass morphology (Bryson et al. 2010), and ecotypes (Capo-chichi et al. 2008) at two locations. Relatively fine textured soils at Tillman's Corner might have adsorbed more imazapyr and thereby reduced its efficacy when applied alone or in combination with the glyphosate. Nevertheless, it took 27 to 33 months for either imazapyr or glyphosate + imazapyr to eradicate cogongrass at each location. At both locations the October imazapyr or glyphosate + imazapyr treatments were quicker in terms of time to eradication than similar treatments in August. Willard et al. (1997) reported similar response of cogongrass to imazapyr and tank mixtures of glyphosate and imazapyr regardless of combination rate. However, they concluded that sequential applications of glyphosate + imazapyr provided the best control. Furthermore, fall-season applications (September to November) of glyphosate or imazapyr have been shown to be more effective due to better translocation of herbicides to the underground rhizomes as photosynthates are directed towards rhizomes in the fall (Faircloth et al. 2005; Johnson 1999 and 2000; Miller 2007; Minogue et al. 2012; Shilling et al. 1997).

Therefore, we conclude that cogongrass can be eradicated with a rigid sequential application program. Two glyphosate (May + October) treatments per year can eradicate

cogongrass; however, the time required for eradication may vary from location to location. Similarly, annual applications of either imazapyr or glyphosate + imazapyr, for at least three years, at any time of the year were more effective than glyphosate alone in eradicating cogongrass. Regardless, succession by suitable native vegetation is warranted to prevent reinvasion of the eradicated areas by windblown seeds from surrounding cogongrass infested areas.

4.6. Literature Cited

Akobundu, I. O. 1992. Integrated weed management techniques to reduce soil degradation. Proc. Intern. Weed Cont. Cong. Melbourne, Australia 1:278-288.

Akobundu, I. O. 1993. Chemical control of cogongrass (*Imperata cylindrica* (L.) Raeuschel) in arable fields. Proc. Weed Sci. Soc. Am. 33:6.

Akobundu, I.O., U.E. Udensi, and D. Chikoye. 2000. Velvetbean (*Mucuna spp*.) suppresses speargrass (*Imperata cylindrica* (L.) Raeuschel) and increases maize yield. Int. J. Pest Manage. 46:103-108.

Amy, A.C. 1932. Variations in the organic reserves in underground parts of five perennial weeds from late April to November. Minn. Agr. Exp. Sta. Tech. Bull. 84

Becker, R. L. and R. S. Fawcett. 1998. Seasonal carbohydrate fluctuations in hemp dogbane (*Apocynum cannabinum*) crown roots. Weed Sci. 46: 358–365.

Bomford, M. and P. O'Brien. 1995. Eradication or control for vertebrate pests? Wildland Soc. Bull. 23:249–255.

Bryson, C.T., and R. Carter. 1993. Cogongrass, *Imperata cylindrica*, in the United States. Weed Technol. 7:1005- 1009.

Bryson, C.T., L. J. Krutz, G. N. Ervin, K.N. Reddy, and J. D. Byrd, Jr. 2010. Ecotype variability and edaphic characteristics for cogongrass (*Imperata cylindrica*) populations in Mississippi. Invasive Plant Sci. Manag. 3:199–207.

Byrd, J. D., Jr. and C. T. Bryson. 1999. Biology, ecology, and control of cogongrass [*Imperata cylindrica* (L.) Beauv.]. Mississippi State, MS: Bureau of Plant Industry, Mississippi Department of Agriculture and Commerce Fact Sheet 1999-01. 2 p.

Capo-chichi, L. J. A., W. H. Faircloth, A. G. Williamson, M. G. Patterson, J. H. Miller, and E. van Santen. 2008. Invasion dynamics and genotypic diversity of cogongrass (*Imperata cylindrica*) at the point introduction in the southeastern United States. Invasive Plant Sci. Manag. 1:133–141.

Dewey, S.A., Jenkins, M.J., Tonioli, R.C., 1995. Wildfire suppression—a paradigm for noxious weed management. Weed Technol. 9: 621–627.

Dozier, H., J.F. Gaffney, S.K. McDonald, E.R.R.L. Johnson and D.G. Shilling. 1998. Cogongrass in the United States: history, ecology, impacts, and management. Weed Technol. 12:737-743.

Enloe, S. F., J. Belcher, N. Loewenstein, J. S. Aulakh and E. van Santen. 2012. Cogongrass control with aminocyclopyrachlor in pastures. Forage and Grazinglands. doi:10.1094/FG-2012-0828-02-RS.

Eplee, R.E. 2001. Coordination of witchweed eradication in the USA. In: Wittenberg R, Cock MJW (eds.) Invasive alien species: a toolkit of best prevention and management practices. CAB International, Wallingford, p 36.

Faircloth, W. H., M. G. Patterson, J. H. Miller, and D. H. Teem. 2005. Wanted dead or alive: cogongrass. Auburn, AL: Alabama Cooperative Extension Publication ANR-1241. 4 p.

Falvey, J. L. 1981. *Imperata cylindrica* and animal production in southeastern Asia: a review. Trop. Grassl. 15:52–56.

Flint, E. and C. Rehkemper. 2002. Control and eradication of the introduced grass, *Cenchrus echinatus*, at Laysan Island, Central Pacific Ocean. Pages 110–115 in C. R. Veitch and M. N. Clout, eds. Turning the Tide: The Eradication of Invasive Species. Auckland, New Zealand: Invasive Species Specialist Group of the World Conservation Union (IUCN).

Gaffney, J.F. 1996. Ecophysiological and technical factors influencing the management of cogongrass (*Imperata cylindrica*). Ph.D. dissertation, University of Florida, Gainesville, FL, USA. 111 p.

Greenfield, S.B. and D. Smith. 1974. Diurnal variations of nonstructural carbohydrates in the individual parts of switchgrass shoots at anthesis. J. Range Mgmt. 27:466-469.

Harding, V. J. and Downs, C. E. 1933. Notes on a Shaffer-Somogyi copper reagent. J. biol. Chem. IOI, 487.

Hodgson, J. M. 1968. The nature, ecology, and control of Canada thistle. United States Department of Agriculture, Technical Bulletin 1386, 32 pp.

Holm, L. G., D. L. Pucknett, J. B. Pancho, and J. P. Herberger. 1977. The World's Worst Weeds. Distribution and Biology. Univ. Press of Hawaii, Honolulu, HI. 609 p.

Johnson, E.R.R.L., J.F. Gaffney and D.G. Shilling. 1999. The influence of discing on the efficacy of imazapyr for cogongrass [*Imperata cylindrica* (L.) Beauv.] control. Proc. South. Weed Sci. Soc. 52:165.

Johnson, E.R.R.L., D.G. Shilling, G.E. MacDonald, J.F. Gaffney and B.J. Brecke. 2000. Time of year, rate of herbicide application, and revegetation: Factors that influence the control of cogongrass [*Imperata cylindrica* (L.) Beauv.] control. Proc. South. Weed Sci. Soc. 53:70.

Jose, S., J. Cox, D. L. Miller, D.G. Shilling and S. Merritt. 2002. Alien plant invasions: The story of cogongrass in southeastern forests. J. Forest. 100:41-44.

Klassen, W., Curtis, C. F.2005. History of the sterile insect technique. In Dyck V.A., Hendrichs J., Robinson A.S. (eds) Sterile insect technique. Principles and practice in area-wide integrated pest management. Springer, Dordrecht, The Netherlands. pp 3–36

Lippincott, C.L. 2000. Effects of *Imperata cylindrica* (L.) Beauv. (cogongrass) invasion on fire regime in Florida sandhill. Nat. Area. J. 20:140-149.

MacDonald, G.E., D.G. Shilling, J. Meeker, R. Charudattan, M. Minno, A. VanLoan, J. DeValerio, C. Yandoc and E.R.R.L. Johnson. 2001. Integrated management of non-native invasive plants in southeastern pine forest ecosystems - cogongrass as a model system. Final Report. USDA Forest Service, Forest Health Technologies. 50 pp.

MacDonald, G.E., E.R.R.L. Johnson, D.G. Shilling, D.L. Miller and B.J. Brecke. 2002. The use of imazapyr and imazapic for cogongrass [*Imperata cylindrica* (L.) Beauv.] control. Proc. South. Weed Sci. Soc. 55:110.

MacDonald, G.E. 2004. Cogongrass (*Imperata cylindrica*)— Biology, Ecology, and Management. Critical Reviews in Plant Sciences. 23:367-380.

Mack, R. N. and W. M. Lonsdale. 2002. Eradicating invasive plants: Hard-won lessons for islands. In: Veitch, C. R. and Clout, M. N. (eds.). Turning the tide: the eradication of

island invasives, pp. 164-172. IUCN SSC Invasive Species Specialist Group. IUCN, Gland, Switzerland and Cambridge, UK.

Mack, R.N. and S.K. Foster. 2004. Eradication or control? Combating plants through a lump sum payment or on the installment plan. In: Sindel BM, Johnson SB (eds) Proceedings of the fourteenth Australian weeds conference. Weed Science Society of New South Wales, Sydney, pp 55–61

Meinesz, A. 2001. Killer algae, 2nd edn. University of Chicago Press, Chicago.

Merkel & Associates, Inc. 2006. Final report on the eradication of the invasive seaweed *Caulerpa taxifolia* from Agua Hedionda Lagoon and Huntington Harbour, California.Southern California Caulerpa Action Team, San Diego.

Miller, J.H. 2000. Refining rates and treatment sequences for cogongrass (*Imperata cylindrica*) control with imazapyr and glyphosate. Proc. South. Weed Sci. Soc. 53:131-132.

Miller, J.H. 2007. What research has found about establishing loblolly pines in cogongrass infestations. In Loewenstein, N. J., and J. H. Miller, (eds) Proceedings of the regional cogongrass conference: a cogongrass management guide. Alabama Cooperative Extension System, Auburn University, Mobile, AL, pp 43–47.

Minogue, J. P., J. H. Miller, D. K. Lauer. 2012. Use of glyphosate and imazapyr for Cogongrass (*Imperata cylindrica*) management in southern pine forests. South. J. Appl. For. 36: 19-25.

Myers, J. H., D. Simberloff, A. M. Kuris, J. R. Carey. 2000. Eradication revisited: dealing with exotic species. Trends Ecol. Evol. 15:316–320.

Newsom, L. D. 1978. Eradication of plant pests-con. Bull. Entomol. Soc. Am. 24:35-40.

Omezine, A. and Fethia Skiri-Harzalla.(2009). Regenerative capacity of speargrass (*Imperata cylindrica* (L.) P. Beauv. Pak. J. Weed. Sci. Res. 15:53-69.

Patterson, D.T. and C.G. McWhorter. 1980. Distribution and control of cogongrass (*Imperata cylindrica*) in Mississippi. Proc. South. Weed Sci. Soc. 33:251.

Patterson, D.T., E.E. Terrell and R. Dickens. 1983. Cogongrass in Mississippi. Miss. Agric. For. Exp. Stn. Res. Rep. 46:1-3.

Potter, R. L., J. L. Petersen, and D.N. Ueckert. 1986. Seasonal trends of total nonstructural carbohydrates in Lindheimer pricklypear (*Opuntia lindheimeri*). Weed Sci. 34: 361-365.

Ramsey, C.L., S. Jose, D.L. Miller, J. Cox, K.M. Portier, D.G. Shilling and S. Merritt. 2003. Cogongrass [*Imperata cylindrica* (L.) Beauv.] response to herbicides and disking on a cutover site in a mid-rotation pine plantation in southern USA. Forest Ecol. Manage. 179:195-209.

Randall, R. 2001. Eradication of a deliberately introduced plant found to be invasive. In: Wittenberg R, Cock, M.J.W. (eds) 2001. Invasive alien species: a toolkit of best prevention and management practices. CAB International Wallingford, Oxon, p 174.

Reineke, L.H. 1933.Perfecting a stand-density index for even-aged forests. J. Agric. Res. 46:627-638.

Rejma´nek, M. and M. J. Pitcairn. 2002. When is eradication of exotic pest plants a realistic goal? Pages 249–253 in C. R. Veitch and M. N. Clout eds. Turning the Tide: The Eradication of Invasive Species. Auckland, New Zealand: Invasive Species Specialist Group of the World Conservation Union (IUCN).

Saldivar, A. J., W. R. Ocumpaugh, R. R. Gildersleeve, and G. M. Prine. 1992. Total nonstructural carbohydrates and nitrogen of 'Florigraze' rhizoma peanut. Agron. J. 84:439-44.

Saxena, K.G. and P.S. Ramakrishnan. 1983. Growth and allocation strategies of some perennial weeds of slash and burn agriculture (jhum) in northeastern India. Can. J. Bot. 61:1300-1306.

Schelhas, J. 2011. A socioeconomic assessment of Forest Service Recovery Act projects: the Alabama Cogongrass Control Center. General Technical Report - Pacific Northwest Research Station, USDA Forest Service 2011 No. PNW-GTR-831 pp. 5-22.

Shilling, D.G., T.A. Bewick, J.F. Gaffney, S.K. McDonald, C.A. Chase, and E.R.R.L. Johnson. 1997. Ecology, Physiology, and Management of Cogongrass (*Imperata cylindrica*). Final Report. Florida Institute of Phosphate Research. 128 p.

Simberloff, D. 2002a. Today Tiritiri Matangi, tomorrow the world! —are we aiming too low in invasives control? In: Veitch CR, Clout MN (eds.) Turning the tide: the eradication of invasive species. IUCN Species Survival Commission, Gland, pp 4–12.

Simberloff, D. 2002b. Why not eradication? In: Rapport D.J, Lasley W.L, Ralston D.E, Nielsen N.O, Qualset C.O, Damania A.B (eds) Managing for healthy ecosystems. CRC/ Lewis Press, Boca Raton, pp 541–548.

Simberloff, D. 2009. We can eliminate invasions or live with them. Successful management projects. Biol. Invas. 11: 149-157.

Soerjani, M. 1970. Alang-alang *Imperata cylindrica* (L.) Beav., pattern of growth as related to its problem of control. Biol. Trop. Bull. 1:88-96.

Soria, M., Gardener, M.R., and Tye, A. 2002. Eradication of potentially invasive plants with limited distributions in the Galapagos islands. In Veitch, C.R. and Clout, M.N. (eds.). Turning the tide: the eradication of invasive species, pp 287–292. IUCN SSC invasive species specialist group. IUCN, Gland, Switzerland and Cambridge, UK.

Stamm Katovich, E. J., R. L. Becker, and B. D. Kinkaid. 1996. Influence of nontarget neighbors and spray volume on retention and efficacy of triclopyr in purple loosestrife (*Lythrum salicaria*). Weed Sci. 44:143–147.

Suryatna, E.S. and J.L. McIntosh. 1980. Food crops production and control of *Imperata cylindrica* (L.) Beauv. on small farms, pp. 135-147. In: Proceedings of BIOTROP workshop on alang-alang in Bogor, 27-29 July 1976. pp. 15- 22. Biotropica Special Pub. No. 5 Bogor, Indonesia.

Tanner, G.W., J. M. Wood, and S.A. Jones. 1992. Cogongrass (*Imperata cylindrica*) control with glyphosate. Florida Scientist. 55:112-115.

Terry, P.J., G. Adjers, I.O. Akobundu, A.U. Anoka, M.E. Drilling, S. Tjitrosemito, and M. Utomo. 1997. Herbicides and mechanical control of *Imperata cylindrica* as a first step in grassland rehabilitation. Agroforest. Syst. 36: 151-179.

Udensi, U.E., I.O. Akobundu, A.O. Ayeni, and D. Chikoye. 1999. Management of cogongrass (*Imperata cylindrica*) with velvetbean (*Mucuna pruriens* var. utilis) and herbicides. Weed Technol. 13:201-208.

Van Loan, A.N., J.R. Meeker and M.C. Minno. 2002. Cogongrass, In: Biological control of invasive plants in the eastern United States, 413 p., Van Driesche, R., Ed., USDA Forest Service Publication FHTET-2002-04.

Wilcut, J. W., R. R. Dute, B. Truelove, and D. E. Davis. 1988a. Factors limiting distribution of cogongrass, *Imperata cylindrica*, and torpedograss, *Panicum repens*. Weed Sci. 36:577-582.

Wilcut, J. W., B. Truelove, D. E. Davis, and J. C. Williams. 1988b. Temperature factors limiting the spread of cogongrass (*Imperata cylindrica*) and torpedograss (*Panicum repens*). Weed Sci. 36:49-55.

Willard, T.R. 1988. Biology, Ecology and management of cogongrass [*Imperata cylindrica* (L.) Beauv.]. Ph.D. dissertation, University of Florida, Gainesville, FL, USA. 113 p.

Willard, T. R., D. W. Hall, D. G. Shilling, J. A. Lewis, and W. L. Currey. 1990. Cogongrass (*Imperata cylindrica*) distribution on Florida highway rights-of-way. Weed Technol. 4:658-660.

Willard, T.R., D.G. Shilling, J.F. Gaffney and W.L. Currey. 1996. Mechanical and chemical control of cogongrass (*Imperata cylindrica*). Weed Technol. 10:722-726.

Willard, T.R., J.F. Gaffney, and D.G. Shilling. 1997. Influence of herbicide combinations and application technology on cogongrass (*Imperata cylindrica*) control. Weed Technol. 11:76-80.

Wittenberg, R. and M. J. W. Cock, eds. 2001. Invasive Alien Species: A Toolkit of Best Prevention and Management Practices. Wallingford, UK: CAB International.

Yoda, K., T. Kira, H. Ogawa, and K.Hozumi. 1963. Self-thinning in crowded pure stands under cultivated and natural conditions. J. Biol. Osaka city Univ. 14:107-129.

Zamora, D. L., D. C. Thill, and R. E. Eplee. 1989. An eradication plan for plant invasions. Weed Technol. 3:2–12.

Table 4.1. Cogongrass parameters in the untreated control plots by location and treatment timing at 36 MAIT.

Location	Timing	----------Shoot----------		---------- Rhizome ----------		
		% Cover	Biomass (g m^{-2})	Biomass (g m^{-2})	Depth (cm)	TNC (%)
Tillman's Corner	May	51 b	156 b	624 a	15 a	27 b
	August	64 ab	244 ab	596 a	17 a	33 b
	October	73 a	356 a	668 a	17 a	47 a
	Mean	63	252	629	16	36
Bayou La Batre	May	58 b	300 b	460 b	18 a	42 a
	August	69 ab	296 b	564 b	14 a	32 b
	October	80 a	524 a	872 a	18 a	41 a
	Mean	69	373	632	17	38

[a] Abbreviations: MAIT; months after treatment initiation.
[b]Means within location and column followed by same letter are not significant different at $P = 0.05$.

Table 4.2. Cogongrass parameters in the treated plots by treatment and timing at 36 MAIT at Tillman's Corner, AL.

Treatment		Cogongrass response variables					
Herbicide	Application time	Green cover	Shoot biomass	Rhizome biomass	Visual control	Rhizome depth	Rhizome TNC
		----------% Reduction----------				cm	%
Glyphosate	May + October	100	100	100	100	0	0
	August	25	58	63	51	10	40
	October	73	83	88	81	8	41
	Mean	**75**	**80**	**83**	**77**	**6**	**27**
Imazapyr	May	96	100	100	99	0	0
	August	100	100	100	100	0	0
	October	100	100	100	100	0	0
	Mean	**100**	**100**	**100**	**99.7**	**0**	**0**
Glyphosate + Imazapyr	May	100	100	100	100	0	0
	August	100	100	100	100	0	0
	October	100	100	100	100	0	0
	Mean	**100**	**100**	**100**	**100**	**0**	**0**
	LSD (0.05)[c]	25	28	30	36	ns	ns

[a] Abbreviations: MAIT = months after initial treatment.
[b] Green cover, rhizome biomass, shoot biomass, and visual control data represent percent reduction compared to the untreated control.
[c] LSD (0.05); least significant difference for comparison between any two individual treatments within each column. Rhizome depth and TNC data at 36 MAIT were analyzed excluding 0 values.

Table 4.3. Cogongrass parameters in the treated plots by treatment and timing at 36 MAIT at Bayou La Batre, AL.

Treatment		Cogongrass response variables					
Herbicide	Application time	Green cover	Shoot biomass	Rhizome biomass	Visual control	Rhizome depth	Rhizome TNC
		%				cm	%
Glyphosate	May + October	100	100	100	100	0	0
	August	96	100	100	97	0	0
	October	98	100	100	98	0	0
	Mean	**98**	**100**	**100**	**98**	**0**	**0**
Imazapyr	May	100	100	100	100	0	0
	August	100	100	100	100	0	0
	October	100	100	100	100	0	0
	Mean	**100**	**100**	**100**	**100**	**0**	**0**
Glyphosate + Imazapyr	May	100	100	100	100	0	0
	August	100	100	100	100	0	0
	October	100	100	100	100	0	0
	Mean	**100**	**100**	**100**	**100**	**0**	**0**
	LSD (0.05)	ns	ns	ns	ns	ns	ns

[a] Abbreviations: MAIT; months after treatment initiation.
[b] Green cover, rhizome biomass, shoot biomass, and visual control data represent percent reduction compared to the untreated control.
[c] LSD (0.05); least significant difference for comparison between any two individual treatments within column.

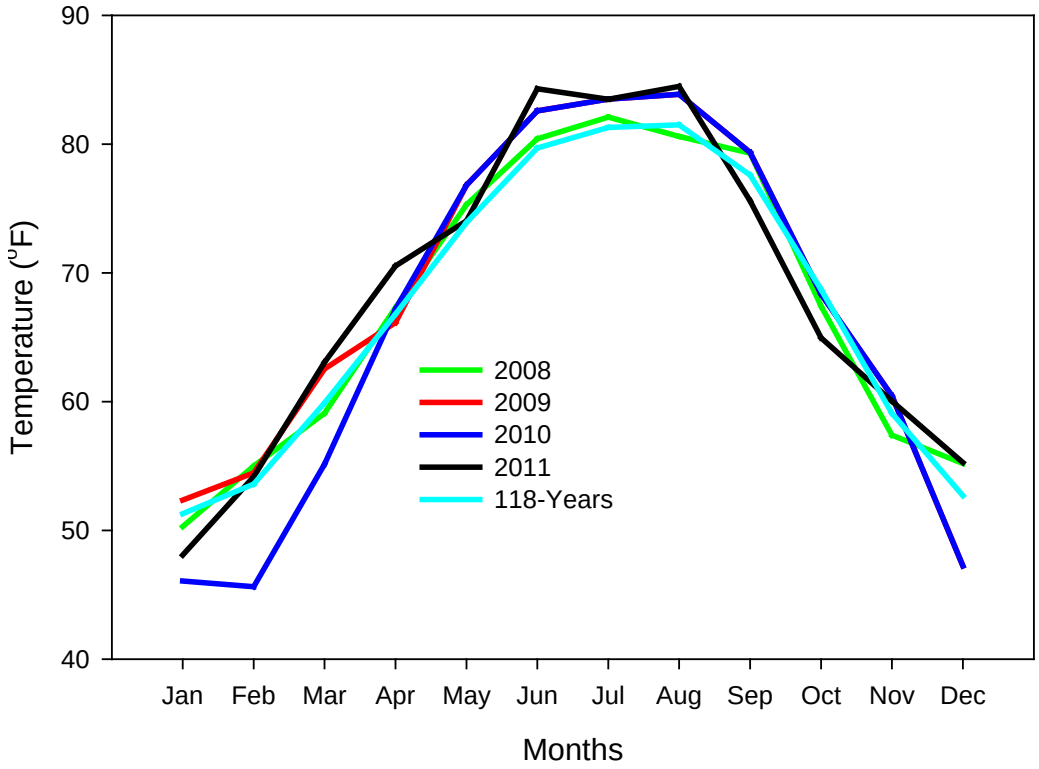


Figure 4.1. Mean monthly air temperature during the study period in relation to 118-years average.

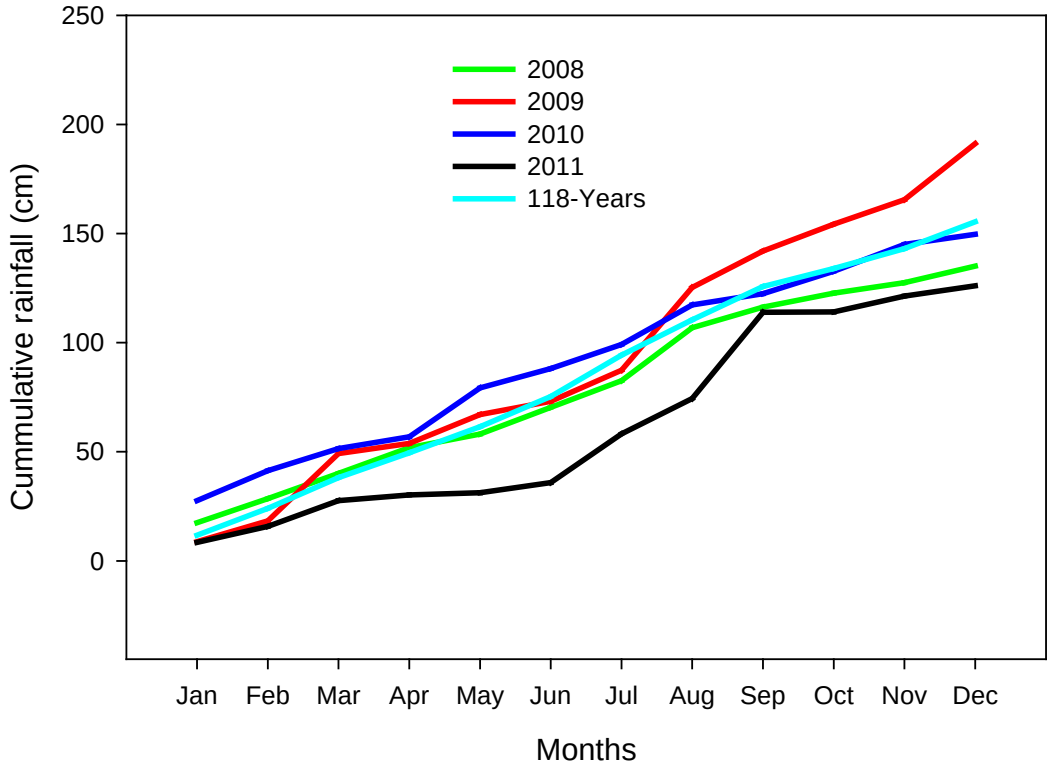
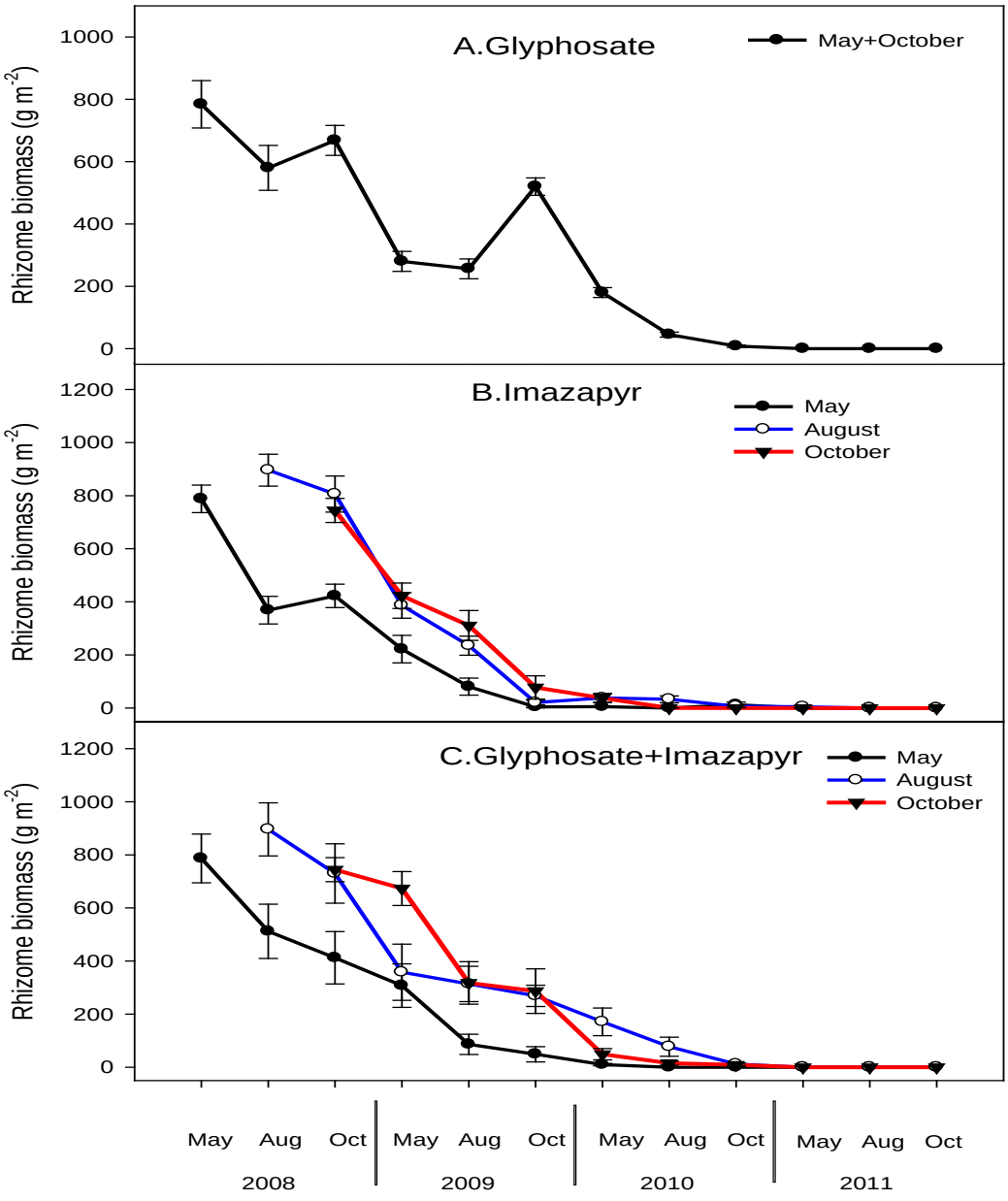


Figure 4.2. Cumulative precipitation during the study period in relation to 118-years average.



Figures 4.3. Pattern of rhizome biomass depletion at Tillman's Corner; A) Glyphosate; B) Imazapyr; C) Glyphosate + Imazapyr. biomass means with standard errors at different times of the year over four study years. Initial values for each treatment timing represent rhizome biomass mean and standard error in the nontreated control at the time of treatment initiation. For May or May + October timings, MAIT were: 0, 3, 6 in 2008; 12, 15, 18 in 2009; 24, 27, 30 in 2010; 36, 39, 42 in 2011. For August timing, MAIT were: 0, 3 in 2008; 9, 12, 15in 2009; 21, 24, 27 in 2010; 33, 36, 39 in 2011. For October timing, MAIT were: 0 in 2008; 6, 9, 12 in 2009; 18, 21, 24 in 2010; 30, 33, 36 in 2011.

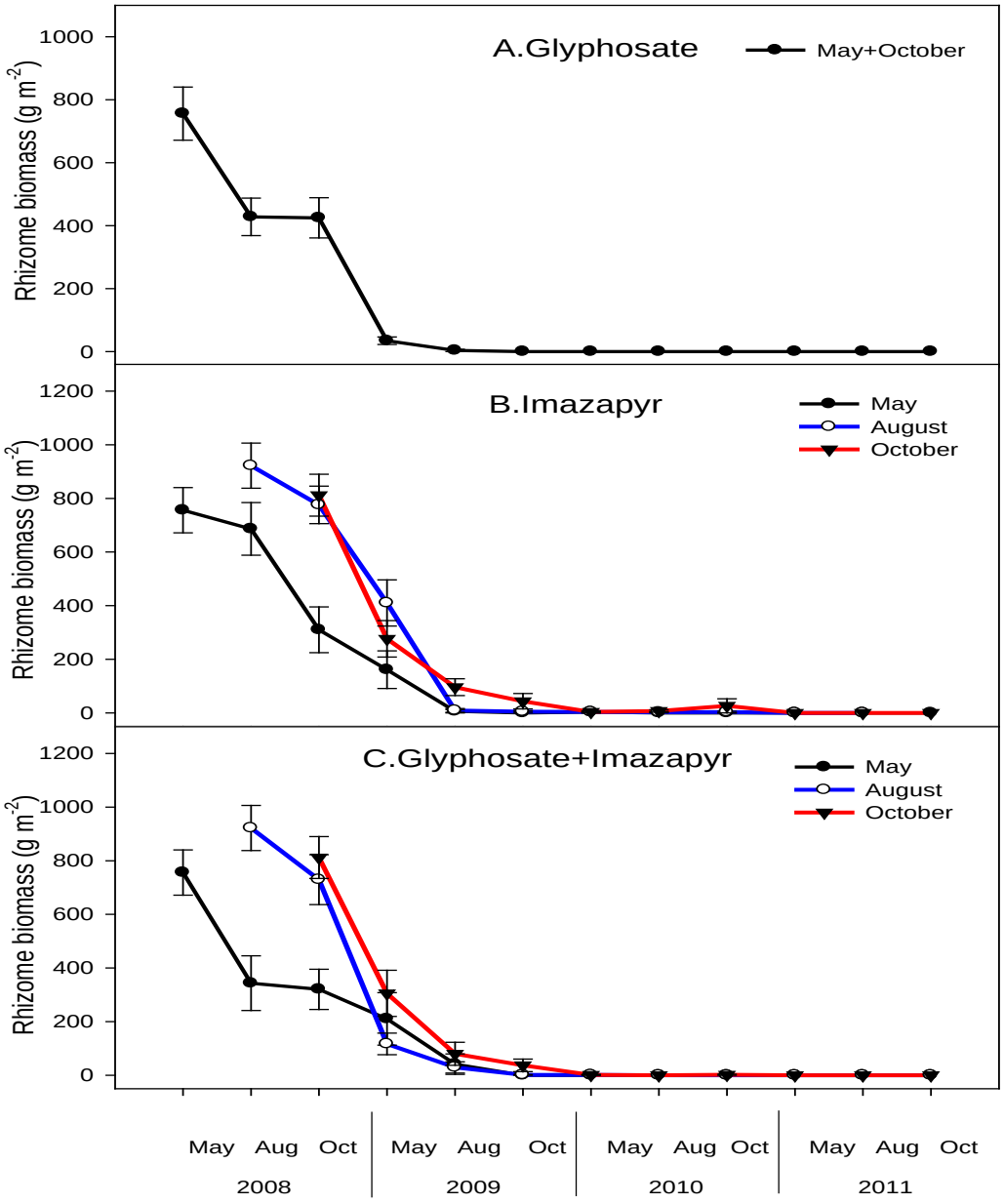


Figure 4.4. Pattern of rhizome biomass depletion at Bayou La Batre; A) Glyphosate; B) Imazapyr; C) Glyphosate + Imazapyr. Biomass means with standard errors at different times of the year over four study years. Initial values for each treatment timing represent rhizome biomass mean and standard error in the nontreated control at the time of treatment initiation. For May or May + October timings, MAIT were: 0, 3, 6 in 2008; 12, 15, 18 in 2009; 24, 27, 30 in 2010; 36, 39, 42 in 2011. For August timing, MAIT were: 0, 3 in 2008; 9, 12, 15in 2009; 21, 24, 27 in 2010; 33, 36, 39 in 2011. For October timing, MAIT were: 0 in 2008; 6, 9, 12 in 2009; 18, 21, 24 in 2010; 30, 33, 36 in 2011.

Chapter 5

Evaluation of Growth Dynamics and Glyphosate Sensitivity of Six Cogongrass Ecotypes

5.1. Abstract

Complementary field and laboratory studies were conducted to: 1) evaluate the growth dynamics of six cogongrass ecotypes ('Auburn', 'Mobile', 'Florida', 'Louisiana', 'Mississippi' and 'Red Baron') in response to a historic soil fertility gradient; and 2) their sensitivity to glyphosate herbicide. In a field study, ecotypes were grown over a wide range of soil fertility treatments (17 combinations of N, P, and K with or without lime and micronutrients). There was significant variation among ecotypes for tiller number, spread diameter, shoot and rhizome biomass, rhizome depth, and total nonstructural carbohydrates levels. All ecotypes except for 'Red Baron' were almost equally invasive. Glyphosate (3.36 kg ae ha^{-1}) completely controlled aboveground growth, but all ecotypes started to recover by 8 months after glyphosate treatment. By 12 months after glyphosate treatment up to 35% recovery was observed, with 'Red Baron' (1% cover) and 'Auburn' (35% cover) being the least and most resilient ecotypes, respectively. A complementary greenhouse dose-response study indicated variation in ecotype sensitivity to glyphosate. On the basis of LD_{50} and LD_{90} values for shoot biomass, three sensitivity groups were identified. 'Florida', 'Louisiana', and 'Mississippi' were the most sensitive; 'Auburn' and 'Mobile' were moderately sensitive; and 'Red Baron' was the least sensitive to glyphosate.

5.2. Introduction

Invasion of a habitat involves interplay between several biotic and abiotic factors and it is the interaction between invader, community and environment that determines the fate of an invasion (D'Antonio 1993; Robinson et al. 1995; Amsberry et al. 2000). Biotic factors include invader genotype, mechanism of seed dispersal, germination requirements, reproductive capability, competitive ability, plant community disturbance regime, and the type of native vegetation, among others. (Baker 1965; Rejmanek and Richardson 1996; Molofsky et al. 1999). Abiotic factors include availability of water, nutrients, and environmental conditions (Crawley 1987; Huenneke et al. 1990; Burke and Grime 1996; Lonsdale 1999; Bradley et al. 2010).

Cogongrass [*Imperata cylindrica* (L.) Beauv. var. *major* tribe *Andropogoneae*] is a highly invasive, alien, perennial C_4 grass which has encroached upon diverse habitat types in the southeastern United States (McDonald 2004). This rhizomatous grass is often found growing on dry sand dunes, wet swamps, river margins, rights of way, and forest ecosystems including commercial pine plantations (Daneshgar et al 2008; Dickens 1974; Chikoye et al. 1999; Garrity et al. 1997; Holm et al. 1977; Lippincot 1997; McDonald 2007; Yager et al. 2010). Currently, cogongrass occupies more than 500,000 hectares of land in the US and is a noxious weed in several southeastern states (Bryson and Carter 1993; Byrd and Bryson 1999, Faircloth et al. 2005; Van Loan et al. 2002). Cogongrass is perceived as serious threat to pineland ecosystems and native vegetation (Eussen and Wirjahardja, 1973; Seavoy, 1975; Eussen, 1980; Lippencott 2000; Yager et al. 2010).

Due to large scale pine plantation silviculture, the southern US is known as the wood basket of the United States (Schultz 1997). In 1952, pine plantations in the south occupied 1.8 million acres with the potential to produce 658 million ft^3 of timber (Fox et al. 2007). Currently,

pine plantations in the southern states occupy 32 million acres that contain 23.9 billion ft^3 of timber (Fox et al. 2007; Wear and Greis 2002). The practice of fertilization and control of competing hardwood vegetation were two of several factors that enhanced pine productivity. Fertilization became an operational silvicultural practice in the Southeast in 1962 when slash pine (*Pinus elliottii* Engelm.) showed larger growth response to phosphorus additions on poorly drained clay soils in Florida (Pritchett and Swinford 1961; Laird 1972). Consequently, fertilizer use in commercial plantations became a widespread practice; in 2004 over 1.2 million acres of pine plantations were fertilized (Fox et al. 2007). Phosphorus is typically the only nutrient applied prior to planting, as nitrogen and potassium fertilization are not recommended for trees less than five years of age (Dickens et al. 2003). However, 150 to 200 lbs of nitrogen, 25 to 50 lbs of phosphorus and 50 lbs of potassium are recommended during mid-rotation (5 to 10 years) to enhance pine productivity (Dickens et al. 2003). Over the past several decades, cogongrass invasion of the southern pine plantations, particularly longleaf pine (*Pinus palustris* Mill.), is rapidly increasing (Brewer, 2008; Daneshgar et al. 2008; Faircloth et al. 2005; King and Grace 2000; Lippincot 1997; Yager et al. 2010). Consequently, infested pine plantations may not produce optimally despite improved fertilization (Daneshgar and Jose 2009). In fact, the availability of nutrients may benefit cogongrass and further exacerbate the competition with pine trees and native vegetation (Daneshgar and Jose 2009).

Cogongrass is a very diverse species (Hubbard et al. 1944). Five varieties of cogongrass have been recognized worldwide: *I. cylindrica* var. *africana* (Andersson) C. E. Hubb., *I. cylindrica* var. *condensata* (Steud.) Hack. Ex Stuck., *I. cylindrica* var. *europaea* (Andersson) Asch. & Graebn., *I. cylindrica* var. *latifolia* (Hook f.) C. E. Hubb., and *I. cylindrica* var. *major* (Nees) C. E. Hubb. Additionally, substantial variation exists in morphological and physiological

characteristics of cogongrass due to genotypic differences (Capo-chichi et al. 2008; Vergara et al. 2008) and varying responses to edaphic factors (Bryson et al. 2010). In addition, some morphological and physiological differences also exist within a population among different ecotypes (Bryson et al. 2010). A population is classified into different ecotypes based on genetic variation within a species that is related to habitat or environment (Sosebee and Wester 1995).

Glyphosate is often used to control cogongrass in a variety of different situations including pine plantations (Dozier et al. 1998; Faircloth et al. 2005; Miller et al. 1999; Minogue et al. 2012; Ramsey et al. 2012; Udensi et al. 1999). Glyphosate inhibits the 5-enolpyruvylshikimate 3-phosphate enzyme thereby preventing the synthesis of the aromatic amino acids tryptophan, tyrosine and phenylalanine (Shaner 2005). However, cogongrass control with glyphosate is highly inconsistent. Previous research has documented locational differences in cogongrass response to glyphosate treatment (Shilling et al. 1997). Furthermore, despite the recognition of different cogongrass ecotypes, no research has explored the ability of cogongrass ecotypes to establish and spread. Nor is information available on how they would respond to enhanced soil fertility situations as may occur in pine plantations. In addition, the sensitivity of different ecotypes to glyphosate has not been quantified. An investigation of the response of different cogongrass ecotypes to diverse soil fertility situations can provide insight into cogongrass invasion dynamics. Similarly, knowledge about their sensitivity to glyphosate can provide critical information for explaining observed differential control of cogongrass by glyphosate, contributing to the development of efficacious cogongrass management strategies. Therefore, complementary field and greenhouse studies were conducted with following two objectives:

1) To evaluate growth dynamics of cogongrass ecotypes and their response to glyphosate under different soil fertility conditions in the field.
2) To quantify the level of sensitivity of cogongrass ecotypes to varying rates of glyphosate in the greenhouse.

5.3. Materials and Methods

5.3.1. Field Study-Response of Ecotypes to Fertility Treatments and Glyphosate

A field study was conducted from 2007 through 2010 at the Brewton Agricultural Research Unit, in Brewton, Alabama, US. Soil of the experimental site was a Benndale fine sandy loam (siliceous thermic Typic Paleudults) with < 1% organic matter. Soil pH was variable (4.9 to 6.2) because of previous long term fertilizer recommendation trials involving various liming and gypsum treatments conducted at the site. . The experiment was established in a split plot design with two replications across four blocks (A, B, C, and D). Whole plots (20.7 m by 6.3 m) consisted of 17 different fertility treatments (Table 5.1) adopted from previous long term fertilizer recommendation trials for cotton, corn, and soybean which were established in 1929 and continued through 2001. The subplots (3.4 m by 6.3 m) were six different cogongrass ecotypes: ‘Auburn’, ‘Mobile’, ‘Florida’, ‘Louisiana’, ‘Mississippi’, and ‘Red Baron’. These ecotypes were obtained from the cogongrass genotype research conducted at Auburn University by Capo-chichi et al. (2008). Before transplanting to the field, cogongrass clones from the six ecotypes were raised in greenhouse by vertically planting a three-node rhizome section in 2.5 cm diameter containers. A standard potting mixture was used as planting media. Pots were watered daily as required to prevent moisture stress and fertilized every week with 20-10-20 (N-P-K) Scotts® fertilizer mixture (Scotts Miracle-Gro, Com., Marysville, OH). Ambient temperature and light conditions were maintained inside the greenhouse. For the first experimental run,

cogongrass clones were transplanted into blocks A and B in August of 2007. For the second experimental run, clones were transplanted into blocks C and D in August of 2008. Data on cogongrass spread diameter and tiller number were recorded in blocks A and B in May 2008 and in C and D in May 2009. Tillers were counted from area (0.64 m^2) between small (radius = 17.1 cm) and big (radius = 48.3 cm) rings and converted to reflect 1 m^2 scale. Purpose of small ring was to exclude the original-clone tillers from those produced in field after transplantation. The spread diameter was recorded by measuring the horizontal distance between the two most distantly spaced tillers within in a subplot.

Percent live-cogongrass cover was estimated visually in each subplot immediately prior to glyphosate application in October using a 0 to 100% scale with zero being equivalent to no live aboveground shoot growth and 100 being vigorously growing and healthy cogongrass. Glyphosate (Accord® Concentrate, Dow AgroSciences LLC, Indianapolis, IN) was applied at 3.36 kg ae ha^{-1}, on October 31, 2008 on blocks A and B and on October 31, 2009 on blocks C and D as a strip treatment, on half of each subplot, across ecotypes. An ATV mounted boom sprayer, fitted with AI 11002 nozzles calibrated to deliver 187 liters ha^{-1} at 50 psi, was used. Cogongrass had been in the field for 15 months at the time of glyphosate application. Visual assessments of cover were made about eight and twelve months after each glyphosate treatment (MAT). Visual assessments were made on a 0 to 100% scale with zero being complete elimination of all cogongrass shoots in the treated plot and 100 being equivalent to the vegetative cover and vigor of each cogongrass ecotype in the corresponding control plot on the non-treated strip.

Data on shoot and rhizome biomass, maximum mean live-rhizome depth were recorded from a 0.25 m^2 quadrat randomly placed in each subplot in the non-treated strip. Green

cogongrass shoots were clipped off at the ground level inside the quadrat and oven dried at 60°C for 72 h for shoot dry weight. Rhizome biomass was quantified by excavating a 50 by 50 cm pit to a depth of 30 cm in each subplot on the non-treated side. The bottom of each pit was closely inspected to verify that no rhizomes were present below the excavation point. Maximum live-rhizome depth was recorded from all four sides/walls the pit, to determine mean live-rhizome depth. Excavated rhizomes were separated from the soil, washed and classified as alive or dead. Alive rhizomes within each quadrat were oven dried at 60°C for 72 h and weighed.

For determining total nonstructural carbohydrate content (TNC), 15 grams of healthy white rhizomes were collected, placed inside a plastic freezer bag and stored on dry ice inside a cooler to prevent respiration losses during transportation. The rhizome samples were kept in a freezer at -20^{o}C until analysis. TNC content was determined by using a tailored version of the Shaffer-Somogyi method (Harding and Downs, 1933). The method consisted of digesting 0.20 to 0.25 g of finely ground (to less than 1 mm) rhizome samples with 50 ml of 0.05 N H_2SO4 and boiling for 15 minutes. Subsequently, the samples were cooled in a shallow ice-water bath, followed by adding 2.5 to 3.0 ml 1.0 N NaOH solution and stirring. The pH of the samples was maintained at 4.5 ± 0.1 while stirring using 1.0 N and 0.1 N H_2SO_4 and 1.0 N and 0.1 N NaOH. One ml of glucoamaylase enzyme solution was added and stirring was continued. The samples were then incubated at 60^{o}Cfor one hour and filtered in 250 ml volumetric flasks using 541 Whatman filter paper or glass wool. Ten ml of diluted sample was transferred to a test tube, followed by the addition of 10 ml of Shaffer-Somogyi solution. Samples were again boiled for 15 minutes and cooled immediately in an ice-bath. The cooled samples were treated with 2 ml of potassium iodide-oxalate, 10 ml of 1 N H_2SO4, 0.2 ml of ‘Fastbreak’ defoamer (1:100) solution, and 1 ml of 1% starch solution. Finally, the samples were titrated with 0.02 N sodium thiosulphate

solution until a clear light blue end-point. The percentage of TNC in the rhizomes was calculated on a dry weight basis using following formula.

$$\text{Percent TNC } = \frac{\text{Sample TNC(mg)} - \text{Enzyme TNC (mg)} * 100}{\text{Dry sample weight (mg)}}$$

The sample TNC was determined by subtracting the sample titer value from the blank. Enzyme TNC signifies the milligrams of glucoamaylase enzyme used.

5.3.2. Statistical Analysis

Statistical analysis was performed using generalized linear mixed models or linear mixed models methodology as implemented in SAS® PROC GLIMMIX (version 9.2, SAS Institute, Inc., Cary, NC). Data on tiller number, spread diameter, shoot and rhizome biomass, rhizome depth, and TNC were analyzed in a split plot design. Fertility, ecotypes, year and all their interactions were treated as fixed effects. Block nested within year, block nested within year × treatment factors were treated as random effects. The split plot nature of the experiment required four different residual terms: 1) block nested within year × fertility as the appropriate error term for fertility; (2) block nested within year × fertility × ecotype as the appropriate error term for ecotype and its interaction with fertility; 3) block nested within year as the appropriate error term for year; and (4) the residual error term as the appropriate error term for all interactions effects of year with fertility and ecotype.

Since the MAT factor was considered as split plot in time, therefore, the percent cover data were analyzed in a split-split plot design. Fertility, ecotypes, MAT, year and all their interactions were treated as fixed effects. Block nested within year, block nested within year × treatment factors were treated as random effects. The split-split plot nature of the experiment required five different residual terms: 1) block nested within year × fertility as the appropriate

error term for fertility; (2) block nested within year × fertility × ecotype as the appropriate error term for ecotype and its interaction with fertility; 3) block nested within year × fertility × ecotype × MAT as the appropriate error term for MAT and its interaction with fertility and ecotype; 4) block nested with in year as the appropriate error term for year; and (5) the residual error term as the appropriate error term for all interactions effects of year with fertility, ecotype, and MAT.

The factor MAT is of a repeated measures nature that induces a covariance relationship because of the lack of re-randomization. Therefore, an autoregressive covariance structure AR (1) was most suitable to model the correlation between observations taken from the same plot at different MATs. Rhizome and shoot biomass data were square root transformed. Percent cover and TNC data were arcsine square root-transformed. Therefore, back transformed means along with 95 % confidence intervals are reported. No transformation was needed for spread diameter, tiller number and rhizome depth data. Multiple means' comparisons of significant effects were made using the 'Adj=simulate' option in statistical analysis system at the 5% significance level.

5.3.3. Greenhouse Study-Response of Ecotypes to a Range of Glyphosate Rates

Two runs of experiments were conducted at the Plant Science Research Center greenhouse at Auburn University, Auburn, AL during 2009 –2010 to evaluate the sensitivity of six cogongrass ecotypes described above to seven different rates of glyphosate. The first run was conducted from February 7, 2009 through September 29, 2009. The second run was conducted from October 23, 2009 through August 23, 2010. The experiments were established in a completely randomized design with five replications. The seven glyphosate rates used were: 0, 0.56, 1.12, 2.24, 3.36, 4.48, 8.96 kg ae ha^{-1} (0, 0.5, 1.0, 2.0, 3.0, 4.0 and 8.0 lbs ae a^{-1}, respectively).

Two sets were propagated one in 2009 and the other in 2010 from 15 cm long, mature, white rhizome sections with five to six intact internodes. Potting mixture consisted of a typical field soil with sandy loam texture. Two rhizomes were planted horizontally, about 2.5 cm deep, in 22.5 cm diameter pots and eventually thinned to one plant per pot a week after emergence. Pots were watered daily as required to prevent moisture stress and fertilized every week with 20-10-20 (N-P-K) Scotts® fertilizer mixture (Scotts® Miracle-Gro, Com., Marysville, OH). Ambient temperature and light conditions were maintained inside the greenhouse. Ecotypes were allowed to grow until a well-developed rhizome system was attained. For run 1 in 2009, cogongrass plants were 136 days old at the time of glyphosate (Accord® Concentrate, Dow AgroSciences LLC, Indianapolis, IN) treatment (June 23). For run 2 in 2010, plants were 210 days old at the time of treatment (May 21). Glyphosate treatments were applied inside a spray chamber using 8002 flat fan nozzle calibrated to deliver 187 liters ha^{-1} at 207 kPa. A non-ionic surfactant (Timberland® 90, UAP, Loveland Products INC, Loveland, CO) was added on 0.5% v/v basis. Shoot growth was used to evaluate sensitivity to glyphosate. Therefore, two weeks after the glyphosate treatment, all shoots were clipped off about 2.5 cm above the soil level to allow measurement of regrowth. Five weeks after glyphosate treatment (5WAT) shoot regrowth from each pot was clipped off, oven dried at 60^{o}C for 72 h, and shoot dry weight was recorded.

5.3.4. Statistical Analysis

Before statistical analysis, shoot biomass data were converted to a relative biomass compared to the non-treated control of each biotype. Data were subjected to ANOVA using Proc GLIMMIX in SAS® (version 9.2, SAS® Institute, Inc., Cary, NC). Experimental run, ecotype, glyphosate rate, and their interactions were treated as fixed effects. The error term was defined as

the residual in the random effects statement. Subsequently a non-linear regression equation was calculated for each ecotype, using the two parameters exponential decay model:

$$Y = A \exp(-b.\ x)$$

Where Y is the shoot biomass of the treated plant, A is the shoot biomass of the non-treated control (intercept), b is constant for the reduction in shoot biomass (slope), and x is the glyphosate rate. Data were combined across experimental runs based on a non-significant effect of experimental run and its interactions with other factors ($P > 0.05$).

5.4. Results and Discussion

5.4.1. Response of Ecotypes to Fertility Treatments

5.4.1.1. Tiller Number

Fertility effect on tiller number was not significant. However, there was significant variation among different ecotypes for tiller number ($P < 0.0001$). 'Auburn' produced the maximum number of tillers (24 tillers m^{-2}) that were significantly more than 'Louisiana' and 'Red Baron' (Figure 5.1). 'Mobile', 'Florida', and 'Mississippi' had similar tiller counts (18 to 22 tillers m^{-2}). However, 'Louisiana' (12 tillers m^{-2}) produced significantly fewer tillers than 'Mobile' and 'Florida'. Of all ecotypes, 'Red Baron' had the lowest tiller number (1 tiller m^{-2}).

5.4.1.2. Spread Diameter

A year by ecotype interaction was highly significant ($P < 0.05$). In 2008, 'Mississippi' was the fastest spreading ecotype, at 147 cm (Figure 5.2). Similarly, 'Mobile', 'Florida' and 'Louisiana' spread 122 to 140 cm in a year, spreading significantly more than 'Auburn' and 'Red Baron' which spread only 82 and 28 cm, respectively. However, 'Auburn' was significantly more spreading than 'Red Baron'. In 2009, 'Louisiana' ecotype spread 214 cm, which was

significantly more than all the other ecotypes except ‘Florida’ (195 cm). Spread diameters of ‘Mobile’, ‘Florida’ and ‘Mississippi’ ranged between 180 and 195 cm. As in 2008, ‘Auburn’ and ‘Red Baron’ spread significantly less than the other ecotypes, although ‘Auburn’ was significantly more expansive than ‘Red Baron’.

5.4.1.3. Shoot and Rhizome Biomass

Data analysis revealed a significant effect of fertility and ecotype on shoot and rhizome biomass. Significantly less shoot and rhizome biomass occurred in the nontreated control (0-0-0) and the no-lime (90-60-60) treatment when compared with all other fertility treatments (Table 5.2). However, different levels of N, P and K were not significantly different for rhizome and shoot biomass.

A decrease in shoot and rhizome biomass with the addition of N and P may have resulted from accompanied decreases in soil pH, cation exchange capacity, calcium, and/or magnesium levels (Table 5.1). In contrast, as the K levels increased soil pH, cation exchange capacity, calcium, and magnesium levels tended to increase. Furthermore, with regard to an N effect, no baseline soil nitrogen test was done to determine differences in nitrogen levels among different historic N treatments. Since nitrogen is not adsorbed on the exchange complex like phosphorus and is subject to losses such as volatilization, leaching and microbial uptake by the time the study was initiated, there may not have been any significant differences in N between different N treatments. Furthermore, the minimum rhizome and shoot biomass in both the nontreated control (0-0-0) and the no-lime (90-60-60) treatments might have resulted from low pH, cation exchange capacity and limited availability of calcium and magnesium associated with these treatments (Table 5.1). However, the treatment with lime only had two times more rhizome and three times more shoot biomass than the historic nontreated or no-lime treatments. Interestingly, the plots

treated with lime only had soil pH, cation exchange capacity, calcium, and magnesium levels that were similar to those observed in the N, P, and K treatments but higher than historic nontreated or no-lime treated plots.

With regard to an ecotype main effect, the highest shoot and rhizome biomass was produced by ‘Florida’ although values were not significantly different from ‘Mobile’, ‘Louisiana’ and ‘Mississippi’ (Table 5.3). Both ‘Auburn’ and ‘Red Baron’ produced significantly less shoot biomass than the other ecotypes. However, rhizome biomass of ‘Auburn’ ecotype was not different from that of ‘Mobile’, ‘Florida’, ‘Louisiana’, and ‘Mississippi’. Furthermore, the shoot and rhizome biomass of ‘Auburn’ was significantly more than that of ‘Red Baron’.

5.4.1.4. Rhizome Depth

For rhizome depth, only the fertility and ecotype main effects were significant ($P < 0.005$). Different rates of N, P, and K did not affect rhizome depth. However, rhizome depth was significantly less in the historic nontreated control plots (12 cm) than in the plots that received any of the fertility treatments (16 cm). Furthermore, the rhizome depth in plots treated with lime only (15 cm) was similar to the plots that received any of the fertility treatments. With regard to ecotypes, the rhizome depth of ‘Mobile’, ‘Florida’ and ‘Mississippi’ was similar (≥18 cm) and significantly more than the other ecotypes. ‘Red Baron’ produced significantly shallower rhizomes (7 cm) than any of the other ecotypes. Furthermore, the rhizome depth of ‘Louisiana’ (15 cm) was similar to that of ‘Auburn’ (13 cm).

5.4.1.5. Total Nonstructural Carbohydrates

A year by ecotype interaction was highly significant (P = 0.003) for TNC. In 2009, the TNC levels varied from 31 to 33% across ecotypes with no significant differences between ecotypes. In 2010, 'Auburn' had significantly higher TNC levels (40 %) than 'Louisiana' (33 %) and 'Red Baron' (28 %) which did not differ significantly. TNC levels in 'Mobile', 'Florida', and 'Mississippi' were similar TNC (~39 %) and were significantly greater than observed for 'Louisiana' and 'Red Baron'.

5.4.2. Response of Ecotypes to Glyphosate

5.4.2.1. Field Study

Cogongrass cover before and after glyphosate treatment was significantly affected by fertility and ecotype. Before glyphosate treatment (0 MAT), the nontreated control and no-lime plots had significantly less cover compared with other fertility treatments (Table 5.4). Glyphosate treatment completely controlled the aboveground growth through mid-summer. At 8 MAT, cogongrass regrowth varied from 1 to 10% across fertility treatments. By 12 MAT, cogongrass growth had recovered to 35 to 50% cover in all fertility treatments except the nontreated control and no-lime plots in which cover was still ≤ 3 % (Table 5.4).

With regard to ecotypes, cover was significantly less in 'Red Baron' when compared with other ecotypes at 0, 8 and 12 MAT (Table 5.5). Regrowth was not observed in any of the ecotypes until mid-summer. However, by 8 MAT, cogongrass started growing back. The 'Auburn' ecotype recovered faster than the other ecotypes. By 12 MAT, cogongrass cover in all ecotypes except 'Red Baron' had recovered to 16 to 35%. However, only the difference between 'Auburn' with maximum cover (35%) and 'Red Baron' (1%) was significant.

5.4.2.2. Greenhouse Study

Anova indicated significant ecotype by glyphosate interaction with respect to shoot biomass ($P < 0.006$). Further analysis revealed a non-linear response of ecotypes to increasing glyphosate rates. Thus a non-linear regression equation was calculated for shoot biomass for each ecotype, using the exponential decay model (Figures 5.3A and 5.3B).

'Mississippi' had the lowest LD_{50} (0.64 kg ae ha^{-1}) and LD_{90} (2.17 kg ae ha^{-1}) values, indicating its high sensitivity to glyphosate (Table 5.6). Although the LD_{50} values of 'Florida' (0.73 kg ae ha^{-1}) and 'Louisiana' (0.77 kg ae ha^{-1}) were similar only 'Louisiana' values were significantly different than those of 'Mississippi'. Nevertheless, 'Florida', 'Mississippi', and 'Louisiana' did not differ in LD_{90} values. 'Auburn' and 'Mobile' had similar LD_{50} and LD_{90} values and were moderate in sensitivity to glyphosate. The minimum reduction in shoot biomass was recorded in 'Red Baron' indicating higher tolerance to glyphosate than other ecotypes. The LD_{50} and LD_{90} values for 'Red Baron' were 1.85 and 6.73 kg ae ha^{-1}, respectively.

Significant differences were reported with respect to the rate of decline/slope (-b) in shoot biomass among cogongrass ecotypes with varying glyphosate concentration. The rate of decline was the maximum for 'Mississippi' ($-b = 1.17 \pm 0.10$) which was similar to that of 'Florida' ($-b = 1.04 \pm 0.08$) and 'Louisiana' ($-b = 1.00 \pm 0.09$) indicating steeper reduction in the biomass of these ecotypes with increasing glyphosate. Similarly, 'Auburn' and the 'Mobile' had similar rate of decline in shoot biomass which was different from that of other ecotypes. Of all the ecotypes evaluated, the 'Red Baron' had the minimum rate of decline ($-b = 0.37 \pm 0.05$) indicating higher glyphosate tolerance compared to the other ecotypes.

5.5. Conclusions

The growth dynamics of the six ecotypes of cogongrass did not vary significantly across most fertility treatments except for the nontreated control and no-lime treatments. These results indicate an adaptability of cogongrass to a broad range of soil fertility conditions. However, contrary to the previous findings by Wilcut et al. (1988), all cogongrass ecotypes performed poorly at soil pH < 5.0 compared to soil at pH ≥ 5.7. Minimum shoot and rhizome biomass was recorded from both the nontreated control and no-lime treated plots that had soil pH = 4.9. It may be concluded that under managed production systems such as pine plantations, cogongrass can grow stably over a wide range of soil fertility. However, it appears that cogongrass growth may be more sensitive to soil pH which can modify the availability of various macro- and micro-nutrients.

On the basis of growth dynamics, ecotypes could be categorized as; 1) highly aggressive which includes 'Auburn', 'Mobile', 'Florida', 'Louisiana', and Mississippi' 2) non-aggressive which includes 'Red Baron' only. Under field conditions, all ecotypes expressed similar sensitivity to glyphosate (3.36 kg ae ha^{-1}). However, on the basis of greenhouse dose-response study, the ecotypes could be distinguished into three sensitivity categories. 'Red Baron' was the least sensitive, 'Auburn' and the 'Mobile' ecotypes were moderately sensitive, and 'Florida', 'Louisiana', and 'Mississippi' were the most sensitive.

Although 'Red Baron' is not as aggressive as the others, there are concerns that it could transfer cold resistance into other ecotypes and it may revert to green invasive from and can produce viable seeds (Cseke and Talley 2012). Additionally, more research is needed to determine the causes of low sensitivity of 'Red Baron' to glyphosate as it might raise concerns of transfer of glyphosate tolerance trait as well.

5.6. Literature Cited

Amsberry, L. M., A. Baker, P. J. Ewanchuk, M. D. Bertness. 2000. Clonal integration and the expansion of *Phragmites australis*. Ecol. Appl. 10: 1110–1118.

Baker, H. G. 1965. Characteristics and modes of origin of weeds. In: Baker, H. G., Stebbins, G. L.(eds) The genetics of colonizing species. Academic Press, New York, pp 147–169.

Bradley, B. A., D.S. Wilcove, M. Oppenheimer. 2010. Climate change increases risk of plant invasion in the Eastern United States. Biol. Invasions. 12:1855–1872.

Brewer, S. 2008. Declines in plant species richness and endemic plant species in longleaf pine savannas invaded by *Imperata cylindrica*. Biol. Invasions. 10: 1257-1264.

Bryson, C. T., and R. Carter. 1993. Cogongrass, *Imperata cylindrica*, in the United States. Weed Technol. 7:1005- 1009.

Bryson, C. T., L. J. Krutz, G. N. Ervin, K.N. Reddy, and J. D. Byrd, Jr. 2010. Ecotype Variability and Edaphic Characteristics for Cogongrass (*Imperata cylindrica*) Populations in Mississippi. Invasive Plant Sci. Manag. 3:199–207.

Burke, M. J. W., P. J. Grime. 1996. An experimental study of plant community invasibility. Ecology. 77: 776–790.

Byrd, J. D., Jr. and C. T. Bryson. 1999. Biology, ecology, and control of cogongrass [*Imperata cylindrica* (L.) Beauv.]. Mississippi State, MS: Bureau of Plant Industry, Mississippi Department of Agriculture and Commerce Fact Sheet 1999-01. 2 p.

Capo-chichi, L. J. A., W. H. Faircloth, A. G. Williamson, M. G. Patterson, J. H. Miller, and E. van Santen. 2008. Invasion dynamics and genotypic diversity of cogongrass (*Imperata cylindrica*) at the point introduction in the southeastern United States. Invasive Plant Sci. Manag. 1:133–141.

Chikoye, D., F. Ekeleme, and J. T. Ambe. 1999. Survey of distribution and farmers' perceptions of speargrass [*Imperata cylindrica* (L.) Raeuschel] in cassava-based systems in West Africa. Int. J. Pest Manag. 45:305–311.

Crawley, M. J. 1987. What makes a community invasible? In: Gray, J, Crawley, M. J, Edwards, P. J (eds) Colonization, succession, and stability. Blackwell, London, pp 429–454.

Cseke, L. J., S. M. Talley. 2012. A PCR-based genotyping method to distinguish Between wild-type and ornamental varieties of *Imperata cylindrica*. J. Vis. Exp. 60:e3265, DOI : 10.3791/3265.

D'Antonio, C. M. 1993.Mechanisms controlling invasion of coastal plant communities by the alien succulent *Carpobrotus edulis*. Ecology. 74: 83–95.

Daneshgar, P., S. Jose, A. R. Collins, and C. Ramsey. 2008. Cogongrass (*Imperata cylindrica*), an alien invasive grass, reduces survival and productivity of an establishing pine forest. For Sci. 54:579–587.

Daneshgar, P., S. Jose. 2009. *Imperata cylindrica*, an alien invasive grass, maintains control over nitrogen availability in an establishing pine forest. Plant Soil. 320: 209-218.

Dickens, E. D., D. J. Moorehead, and B. McElvany. 2003. Pine plantation fertilization. Better crops 87: 12-15.

Dickens, R. 1974. Cogongrass in Alabama after sixty years. Weed Sci. 22:177–179.

Dozier, H., J. F. Gaffney, S. K. McDonald, E. R. R. L. Johnson and D. G. Shilling. 1998. Cogongrass in the United States: history, ecology, impacts, and management. Weed Technol. 12:737-743.

Eussen, J. H. H., and S. Wirjahardja. 1973. Studies of an alang-alang (*Imperata cylindrica* (L.) Beauv.) vegetation. Biotrop. Bull. 6:1-24.

Eussen, J. H. H. 1980. Biological and ecological aspects of alang–alang [*Imperata cylindrica* (L.) Beauv.]. In: Proceedings of BIOTROP workshop on alang–alang in Bogor, 27–29 July 1976. Biotropica Special Pub. No. 5 Bogor, Indonesia, pp 15–22.

Faircloth, W. H., M. G. Patterson, J. H. Miller, and D. H. Teem. 2005. Wanted dead or alive: cogongrass. Auburn, AL: Alabama Cooperative Extension Publication ANR-1241. 4 p.

Falvey, J. L. 1981. *Imperata cylindrica* and animal production in southeastern Asia: a review. Trop. Grassl. 15:52–56.

Fox, T. R., E. J. Jokela, and H. L. Allen. 2007. The development of pine plantation silviculture in the southern United States. J. Forest. 105: 337-347.

Garrity, D. P., M. Soekardi, M. Van Noordwijk, R. De La Cruz, P. S. Pathak, H. P. M. Gunasena, N. Van So, G. Huijun, and N. M. Majid. 1996. The Imperata grasslands of tropical Asia: area, distribution, and typology. Agrofor. Syst. 36:3–29.

Harding, V. J. and C. E. Downs. 1933. Notes on a Shaffer-Somogyi copper reagent. J. biol. Chem. IOI, 487.

Holm, L. G., D. L. Pucknett, J. B. Pancho, and J. P. Herberger. 1977. The world's worst weeds. Distribution and biology. Univ. Press of Hawaii, Honolulu, HI. 609 p.

Hubbard, C. E., R. O. Whyte, D. Brown, and A. P. Gray. 1944. *Imperata cylindrica*- taxonomy, distribution, economic significance, and control. Oxfordshire, UK: Imperial Agricultural Bureaux Joint Publication. 7:1–63.

Huenneke, L. F., S. P. Hamburg, R. Koide, H. A. Mooney, P. M. Vitousek. 1990. Effects of soil resources on plant invasion and community structure in California serpentine grassland. Ecology. 7: 478–491.

Johnson, E. R. R. L., J. F. Gaffney, and D. G. Shilling. 1999. The influence of discing on the efficacy of imazapyr for cogongrass [*Imperata cylindrica* (L.) Beauv.] control. Proc. South. Weed Sci. Soc. 52:165.

King S. M., J. B. Grace. 2000. The effects of gap size and disturbance type on invasion of wet pine savanna by cogongrass, *Imperata cylindrica* (Poaceae). Am. J. Bot. 87:1279–1286.

Laird, C.R. 1972. Fertilization of slash pine on poorly drained soils in northwest Florida. Circ. 378, Florida Cooperative Extensive Serv., Univ. of Florida, Gainesville, FL. 12 p

Lippincott, C. L. 1997. Ecological consequences of *Imperata cylindrica* (cogongrass) invasion in Florida sandhill. Ph.D. dissertation, University of Florida, Gainesville, Florida, USA.

Lippincott, C. L. 2000. Effects of *Imperata cylindrica* (L.) Beauv. (cogongrass) invasion on fire regime in Florida sandhill. Nat. Areas J. 20:140–149.

Lonsdale, W. M. 1999. Global patterns on plant invasions and the concept of invasibility. Ecology. 80:1522–1536.

MacDonald, G. E. 2004. Cogongrass (*Imperata cylindrica*) — biology, ecology, and management. Critical Rev. Plant Sci. 23: 367-380.

MacDonald, G. E. 2007. Cogongrass: the plant's biology, distribution, and impacts in the Southeastern US. In: Loewenstein, N. J., Miller, J. H. (eds) Proceedings of the regional cogongrass conference: a cogongrass management guide. Alabama Cooperative Extension System, Auburn University, Mobile, AL, pp 10–23.

Miller, J. H. 1999. Refining rates and treatment sequences for cogongrass (*Imperata cylindrica*) control with imazapyr and glyphosate. Proc. South. Weed Sci. Soc. 53:181

Minogue, P. J., J. H. Miller, and D. K. Laur. 2012. Use of glyphosate and imazapyr for cogongrass (*Imperata cylindrica*) management in southern pine forests. Southern J. Appl. Forest. 36: 19-25.

Molofsky, J., S. L. Morrison, C. J. Goodnight.1999. Genetic and environmental controls of the establishment of invasive grass, *Phalaris arundinacea*. Biol. Invasions. 1: 181–188.\

Pritchett, W. L., and K. R. Swinford. 1961. Response of slash pine to colloidal phosphate fertilization. S.S.S.A.P. 25: 397-400.

Ramsey, C.L., S. Jose, D. Zamora. 2012. Cogongrass (*Imperata cylindrica*) control with imazapyr and glyphosate combined with and without four adjuvants. Southern. J. Appl. Forest. 36: 204-210.

Rejmanek, M., D. M. Richardson.1996. What attributes make some plant species more invasive? Ecology 77:1651–1661.

Robinson, G. R., J. F. Quinn, M. L. Stanton. 1995. Invasibility of experimental habitat islands in a California winter annual grassland. Ecology. 76: 786–794.

Schultz, R. P. 1997. Loblolly pine: the ecology and culture of loblolly pine (*Pinus taeda* L.). Agriculture Handbook (Washington) 713.

Seavoy, R. E. 1975. The origin of tropical grasslands in Kalimantan, Indonesia. J. Trop. Geograph. 40: 48-52.

Shaner, D. L., T. Nadler-Hassar, W. B. Henry, and C. H. Koger. 2005. A rapid in vivo shikimate accumulation assay with excised leaf discs. Weed Sci. 53:769–774.

Shilling, D. G., T. A. Bewick, J. F. Gaffney, S. K. McDonald, C .A. Chase, and E. R. R. L. Johnson. 1997. Ecology, physiology, and management of cogongrass (*Imperata cylindrica*). Final report. Florida institute of phosphate research. pp 128.

Sosebee, R. E., and D. B. Wester. 1995. Genetic variation in plants: environmental and biotic effects. p. 1–21. In D. J. Bedunah and R.E. Sosebee, (ed.) Wildland plants: Physiological ecology and developmental morphology. Society for Range Management, Denver, CO.

Udensi, U. E., I. O. Akobundu, A. O. Ayeni, and D. Chikoye. 1999. Management of cogongrass (*Imperata cylindrica*) with velvetbean (*Mucuna pruriens* var. utilis) and herbicides. Weed Technol. 13:201-8.Van Loan, A. N., J. R. Meeker and M. C. Minno. 2002. Cogongrass, In: Biological control of invasive plants in the eastern United States, 413 p., Van Driesche, R., Ed., USDA Forest Service Publication FHTET-2002-04.

Vergara, R., M. C. Minno, M. Minno, S. E. Soltis, and P. S. Soltis. 2008. Preliminary study using ISSRs to differentiate Imperata taxa (Poaceae: Andropogoneae) growing in the US. Southeast. Nat. 7: 267–276.

Wear, D. N., and J. G. Greis. 2002. Southern forest resource assessment: summary of findings. J. Forest. 100: 6-14.

Wilcut, J. W., B. Truelove, D. E. Davis, J. C. Williams. 1988. Temperature factors limiting the spread of cogongrass (*Imperata cylindrica*) and torpedograss (*Panicum repens*). Weed Sci 36:49–55.

Yager, L. Y., D. L. Miller, J. Jones. 2010. Susceptibility of longleaf pine forest associations in south Mississippi to invasion by cogongrass [*Imperata cylindrica* (L.) Beauv.]. Nat. Areas J. 30: 226-232.

Table 5.1. Soil pH, cation exchange capacity, phosphorus, potassium, calcium and magnesium levels in different historic fertility treatments at the time of start of study as per soil test report.

N-P-K	Treatment Description	pH	Cation exchange capacity cmol kg^{-1}	Phosphorus kg ha^{-1}	Potassium kg ha^{-1}	Calcium kg ha^{-1}	Magnesium kg ha^{-1}
0-60-60	No nitrogen	6.2	3.3	28	66	590	59
30-60-60	Low nitrogen	6.1	3	18	66	542	60
60-60-60	Intermediate nitrogen	6.1	2.8	20	60	489	53
120-60-60	High nitrogen	6	3.2	18	62	447	53
150-60-60	Very high nitrogen	5.7	2.3	21	53	333	45
90-0-60	No phosphorus	6.2	2.5	9	65	447	51
90-30-60	Intermediate phosphorus	5.7	2.6	30	61	354	44
90-60-0	No potassium	5.8	2.7	41	43	383	47
90-60-30	Low potassium	5.9	3	39	58	564	67
90-60-120	High potassium	6	3	35	70	521	57
0-0-0	Only dolomitic lime	6.1	2.7	10	42	458	52
90-60-60	Std. NPK and no-lime	4.9	2	49	34	101	14
90-60-60	Std. NPK and low magnesium	5.7	2.6	39	52	394	25
90-60-60	Std. NPK and micronutrients	6	2.5	16	53	438	52
90-60-60	Std. NPK 1	6	3	60	72	539	58
90-60-60	Std. NPK 2	6	3	18	60	492	59
0-0-0	Nontreated control	4.9	2.2	17	29	79	13

[a] Low magnesium treatment received calcitic lime only, all other treatments, excepting no-lime treatment, received dolomitic lime; Std. NPK 1, 90-60-60 annually until 2001; Std. NPK 2, nontreated until 1979 and 90-60-60 thereafter.

Table 5.2. Fertility means with 95% confidence limits for shoot and rhizome biomass.

N-P-K	Fertility treatment	Shoot biomass g m^{-2}	95% CL	Rhizome biomass g m^{-2}	95% CL
0-60-60	No nitrogen	248	180, 328	452	332, 588
30-60-60	Low nitrogen	228	162, 304	360	252, 484
60-60-60	Intermediate nitrogen	158	104, 222	352	248, 476
120-60-60	High nitrogen	153	100, 216	280	188, 392
150-60-60	Very high nitrogen	160	106, 225	312	212, 428
90-0-60	No phosphorus	150	98, 213	336	236, 456
90-30-60	Intermediate phosphorus	123	77, 181	288	192, 400
90-60-0	No potassium	139	89, 200	268	190, 376
90-60-30	Low potassium	189	130, 259	352	248, 476
90-60-120	High potassium	212	150, 286	428	312, 564
0-0-0	Only dolomitic lime	102*	76, 155	220*	140, 320
90-60-60	Std. NPK and no-lime	33*	12, 66	108*	56, 180
90-60-60	Std. NPK and low magnesium	154	101, 217	340	240, 464
90-60-60	Std. NPK and micronutrients	178	121, 246	336	232, 456
90-60-60	Std. NPK 1	225	161, 301	444	328, 584
90-60-60	Std. NPK 2	226	162, 303	400	288, 532
0-0-0	Nontreated control	33*	12, 66	112*	56, 188

[a] Shoot and rhizome biomass means represent back transformed data. 95% CL represents lower and upper confidence limits at $P = 0.05$.

[b] Fertility means averaged over ecotypes and years.

Table 5.3. Ecotype means and 95% confidence limits for shoot and rhizome biomass and rhizome depth.

Ecotype	Shoot biomass ($g\ m^{-2}$)	95% CL	Rhizome biomass ($g\ m^{-2}$)	95% CL	Rhizome depth (cm)
Red Baron	21	11, 34	69	30, 125	7 c
Auburn	90	68, 115	292	202, 398	13 b
Louisiana	218	175, 246	361	261, 478	15 ab
Mississippi	209	183, 256	382	278, 502	18 a
Mobile	227	191, 266	396	290, 517	18 a
Florida	242	205, 282	461	347, 593	19 a

[a] Shoot and rhizome biomass means represent back transformed data.95% CL represents lower and upper confidence limits at $P = 0.05$.
[b] Ecotype means averaged over fertility treatments and years.

Table 5.4. Fertility means with 95% confidence limits for cogongrass cover before and after glyphosate treatment.

N-P-K	Fertility treatment	Percent cover					
		0 MAT	95% CL	8 MAT	95% CL	12 MAT	95% CL
0-60-60	No nitrogen	70	56,82	7	3, 14	31	22, 40
30-60-60	Low nitrogen	53	39,67	8	3, 14	31	23, 41
60-60-60	Intermediate nitrogen	57	43,70	7	2, 13	29	19, 37
120-60-60	High nitrogen	50	36,64	4	2, 10	25	17, 34
150-60-60	Very high nitrogen	54	40,68	7	2, 13	28	19, 37
90-0-60	No phosphorus	56	41,69	8	3, 15	26	18, 35
90-30-60	Intermediate phosphorus	52	38,66	6	2, 12	20	12, 28
90-60-0	No potassium	45	31,59	10	5, 18	28	20, 37
90-60-30	Low potassium	56	42,70	9	3, 16	30	21, 39
90-60-120	High potassium	71	58,83	8	3, 15	36	27, 46
0-0-0	Only lime	50	36,64	8	3, 16	22	14, 31
90-60-60	Std. NPK and no-lime	17*	8,29	1*	0, 2	1*	0, 4
90-60-60	Std. NPK and low magnesium	53	39,67	10	4, 17	29	21, 38
90-60-60	Std. NPK and micronutrients	60	45,73	8	3, 15	28	20, 37
90-60-60	Std. NPK 1	71	57,83	5	2, 11	30	22, 40
90-60-60	Std. NPK 2	61	47,74	4	2, 6	21	13, 29
0-0-0	Nontreated control	16*	7,27	1*	0, 2	3*	0, 7

[a] Percent cover means at 0, 8, and 12 MAIT represent back transformed data. 95% CL represents lower and upper confidence limits for percent cover means at $P = 0.05$.

[b] Fertility means averaged over ecotypes and years.

Table 5.5. Ecotype means and 95% confidence limits for cogongrass cover before and after glyphosate treatment.

Ecotype	Percent cover					
	0 MAT	95% CL	8 MAT	95% CL	12 MAT	95% CL
Red Baron	5	2,10	0	0,1	1	0,2
Auburn	52	46,58	11	6,18	35	29,42
Louisiana	49	43,56	3	1,6	16	11,22
Mississippi	52	46,58	6	2,11	23	17,29
Mobile	52	46,58	5	2,9	22	16,28
Florida	55	49,62	6	2,10	21	15,27

[a] Percent cover means at 0, 8 and 12 months after glyphosate treatment (MAIT) represent back transformed data. 95% CL represents lower and upper confidence limits for percent cover means at $P = 0.05$.
[b] Ecotype means averaged over fertility treatments and years.

Table 5.6. Best fit regression parameters and their standard errors for each cogongrass ecotype describing relative shoot biomass response to glyphosate 5 WAT.

Biotype	Parameters		LD_{50}*	LD_{90}*	Pseudo R^2
	A	-b*			
			kg ae ha^{-1}	kg ae ha^{-1}	
Auburn	1.0 ± 0.03	0.75 ± 0.05	1.03 ± 0.06	3.42 ± 0.21	0.99
Mississippi	1.0 ± 0.04	1.17 ± 0.10	0.64 ± 0.04	2.17 ± 0.18	0.96
Florida	1.0 ± 0.04	1.04 ± 0.08	0.73 ± 0.04	2.46 ± 0.18	0.97
Louisiana	1.0 ± 0.04	1.00 ± 0.09	0.77 ± 0.07	2.56 ± 0.22	0.98
Mobile	1.0 ± 0.04	0.66 ± 0.06	1.18 ± 0.09	3.91 ± 0.32	0.95
Red Baron	1.0 ± 0.05	0.37 ± 0.05	1.85 ± 0.19	6.73 ± 0.78	0.91

[a] Abbreviations: WAT, weeks after glyphosate treatment; A, intercept; -b, slope; LD_{50} and LD_{90}, glyphosate concentrations resulting in 50 and 90 %, respectively, reduction in biomass.
[b] Significantly different among ecotypes at $P = 0.05$.

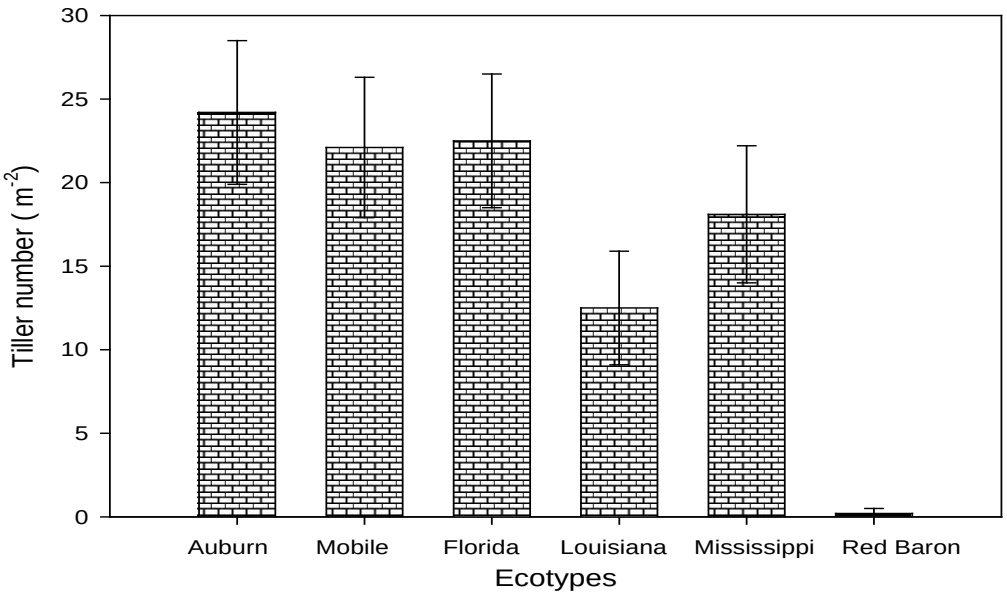


Figure 5.1. Mean tiller number with standard error for different cogongrass ecotypes.

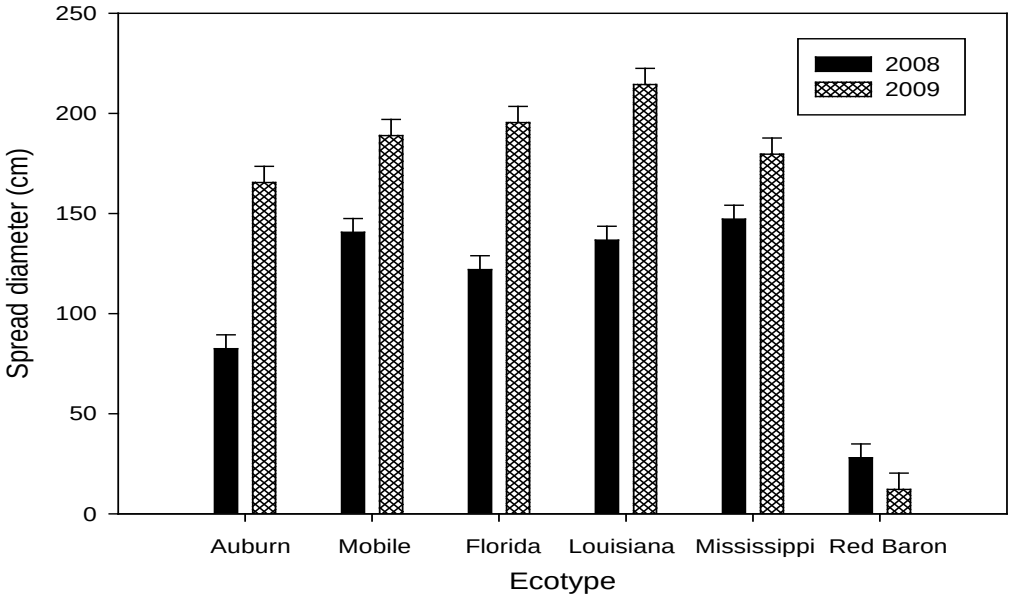


Figure 5.2. Mean spread diameter with standard error for different cogongrass ecotypes.

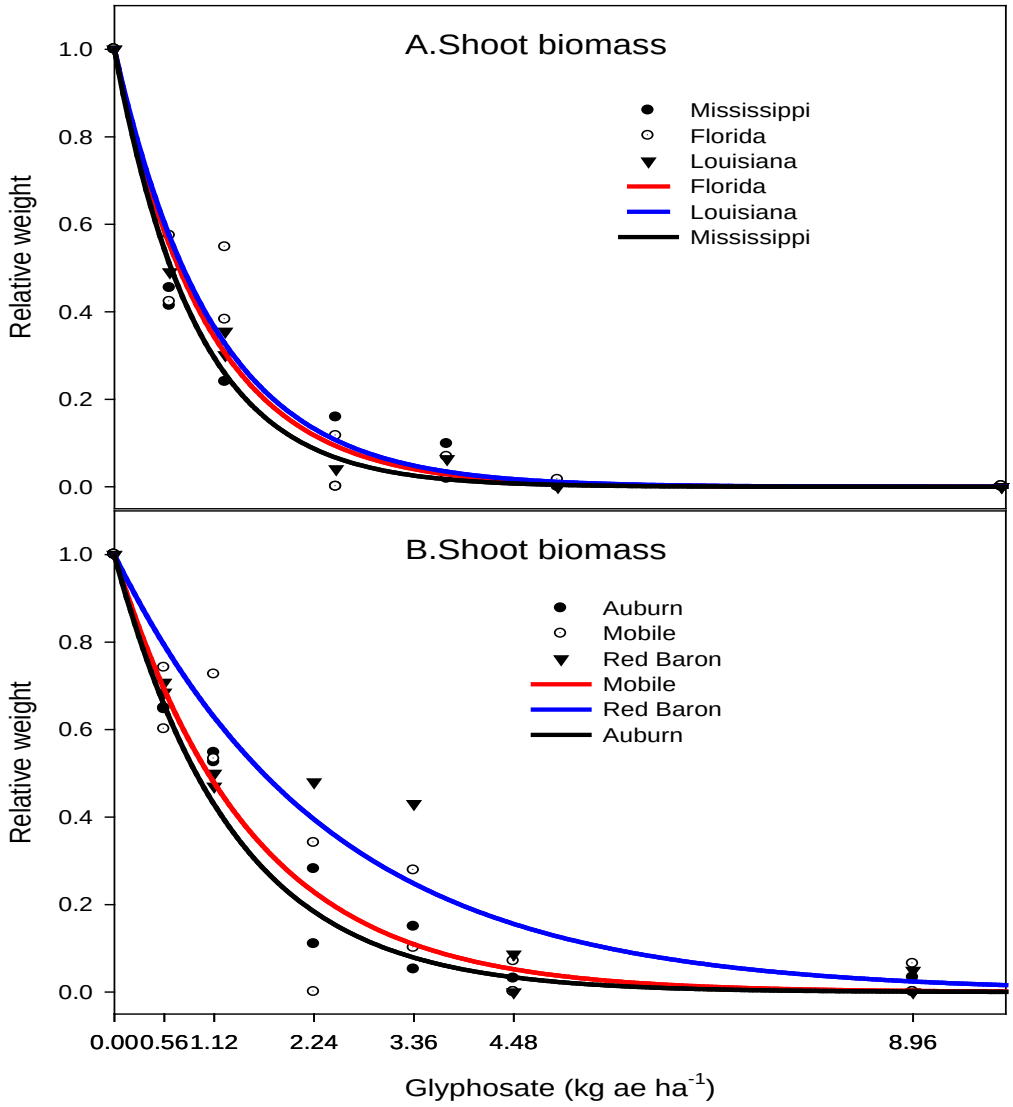


Figure 5.3. Shoot biomass response; A) Glyphosate sensitive ecotypes; B) Glyphosate tolerant ecotypes. Nonlinear regression parameters, calculated using two parameter exponential decay model: Y = A exp (-b.x), where Y is the relative shoot biomass, x is the glyphosate rate A is the shoot biomass of the nontreated, b is a constant for decline in biomass.