MECHANISMS OF COGONGRASS [*Imperata cylindrica* (L.) Beauv.] COMPETITION, LOW LIGHT SURVIVAL, AND RHIZOME DORMANCY.

By

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To my parents.
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MECHANISMS OF COGONGRASS \textit{[Imperata cylindrica (L.) Beauv.]} COMPETITION, LOW LIGHT SURVIVAL, AND RHIZOME DORMANCY

By

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Cogongrass \textit{[Imperata cylindrica (L.) Beauv.]} has been reported as a serious perennial pest throughout the tropical and subtropical areas of the world and has been ranked the 7th worst weed worldwide. It is an invasive C4 grass weed, and possesses a very strong and aggressive rhizome system, which is the major reason for its survivability and competitiveness. The low shoot to rhizome ratio greatly contributes to energy conservation and rapid re-growth after chemical and mechanical control methods such as burning or cutting. Cogongrass thrives better in soils with low pH (pH about 4.7), low fertility, low organic matter, although it can grow in a wide range of soils. The goal of this research is to provide biological information concerning cogongrass growth and competitiveness to facilitate future research on integrated management of cogongrass.

The first experiment evaluated the influence of soil pH on the relative competitiveness of cogongrass and bahiagrass. Based on previous research on different pH requirements for optimal growth of cogongrass and bahiagrass (cogongrass about 4.7, bahiagrass about 5.5 to 6.5), we hypothesized soil pH would influence the competition between these 2 species. Our results showed that when soil pH increased, bahiagrass competitiveness was increased and cogongrass
competitiveness was decreased. Therefore, cogongrass invasion into bahiagrass pastures appears to be strongly related to soil pH.

The second experiment evaluated light intensity effects on cogongrass growth. Mature and young cogongrass plants were grown under different light intensity levels (75, 25, 15, 2, and 1% of full sunlight) to measure growth patterns and changes in rhizome to shoot ratio. Mature plants had a completely developed rhizome system, and under low light intensities, the plants maintained a constant rhizome to shoot ratio. At light intensity below the light compensation point, rhizome to shoot ratio increased dramatically indicating possible rhizome dormancy. Young plants sacrificed rhizome growth for shoot production with rhizome to shoot ratio decreasing as light intensity decreased.

The third experiment evaluated abscisic acid (ABA) concentration in cogongrass rhizomes and rhizome scale leaves as a function of apical position. The relationship between ABA concentration and rhizome dormancy was evaluated. Our results showed that ABA likely plays a role in cogongrass rhizome dormancy. The rhizome position that includes rhizome tips, the ABA concentration was significantly higher than in axillary nodes, including shoots and scale leaf tissue. Further research is still needed to confirm the actual function of ABA in cogongrass rhizome dormancy and the complex interaction between different plant growth hormones, such as auxin, also needs to be addressed in the future.
CHAPTER 1
INTRODUCTION

Invasive plant species are currently disrupting natural areas by displacing native flora and fauna through several mechanisms. Non-native species are often more proliferate and less sensitive to environmental changes (Marion, 1986) and able to adapt to a wide range of environmental conditions. These species are also capable of exploiting a variety of niches minimizing the effects of external pressure from the ecosystem (Reichard, 1996; Grace, 1999).

Cogongrass Problem Statement

Cogongrass (*Imperata cylindrica* (L.) Beauv.), named as japgrass, bladygrass, speargrass, alang-alang and lalang-lalang (Dozier et al., 1998), is a cosmopolitan and disruptive grass species found throughout the tropical and subtropical areas of the world. It has been found on virtually every continent and is considered a troublesome weed in over 73 countries (MacDonald, 2004). Cogongrass is also regarded as the 7th most hard-to-control weed worldwide (Holm et al., 1977).

Pendleton (1948) first stated that the complete eradication of this noxious weed should be undertaken, as the hazard of this weedy species far outweighs its benefits.

Cogongrass has infested more than 500 million hectares of land worldwide (Holm et al., 1977; Dozier et al., 1998) including areas in Europe, northern Africa, the Middle East, Australia and New Zealand, and more than 35 million hectares in Asia (Garrity et al., 1996; Holm et al., 1977). In the United States, cogongrass is found throughout much of the southern Gulf Region (Dickens, 1974; Elmore, 1986) including southern Alabama, Georgia, Louisiana, Mississippi and Florida (Shilling et al., 1995). A survey of cogongrass distribution on Florida highway rights-of-way from 1984 to 1985 showed cogongrass widely distributed from the north central region southward through the central Florida ridge north of Lake Okeechobee (Willard et al., 1990).
The highest distribution frequencies were in counties where cogongrass was used for forage and soil stabilization during the 1950s (Willard et al., 1990).

There were two mechanisms of cogongrass introduction into the United States. Cogongrass was inadvertently introduced from Japan to Alabama as a packing material in 1912 (Dickens, 1974; Tabor, 1949, 1952) and intentionally introduced as a potential forage from the Philippines to Mississippi in 1921 (Hubbard et al., 1944; Dickens and Buchanan, 1975; Patterson et al., 1983; Tabor, 1949, 1952). Forage trials conducted in Texas, Mississippi, Alabama and Florida proved that cogongrass was not a desirable forage species, but by this time cogongrass had spread from the original points of introduction. Points of introduction into Florida were at The University of Florida Experiment Station in Gainesville, USDA Plant Introduction Station in Brooksville and the Soil Conservation Service Reclamation Area in Withlacoochee (Hall, 1983; Willard, 1988). The quick spread of cogongrass was aided by cattlemen who took the grass from the Florida Experiment Station and established it on ranches throughout the state. By the 1950s, it had already covered more than 1000 acres of land in central and northwest Florida (Dickens, 1974).

Cogongrass is generally considered to have little utility. It is undesirable to grazing animals because the leaves have serrated margins that accumulate silicon, becoming sharp and abrasive (Coile and Shilling, 1993). However, sometimes it can be used for thatch, short-term forage production, soil stabilization and even paper-making (Watson and Dallwitz, 1992). An Imperata cylindrica, var. ‘rubra’ ornamental variety of this grass was developed in the United States called “Rubra”, “Red Baron” or “Japanese Blood Grass”, which is not aggressive (Greenlee, 1992). Several natural compounds with medicinal value have also been discovered in cogongrass such as imperanene and cylindrene (Matsunaga et al., 1994).
**Taxonomy**

Imperata is a genus of the tribe Andropogoneae, subtribe Saccharine. This genus has nine species including *I. brasiliensis, I. brevifolia, I. cheesemanii, I. condensate, I. congerta, I. contracta, I. cylindrical, I. minutiflora* and *I. teniis* (Gabel, 1982). Cogongrass is the most variable and important species in this genus and can be identified from other species as having two anthers instead of one (Gabel, 1982; Hitchcock, 1951). Cogongrass was grouped by Hubbard et al., (1944) and Santiago (1980) into five varieties. Varieties Major and Africana are the most widespread, damaging and variable (Brook, 1989). However, recent studies show all 5 varieties to be integrated; making true distinctions difficult (Clayton and Renvoize, 1982).

**Morphology and Biology**

Cogongrass is a stemless, tufted grass species. The above-ground portion of the plant is slender, with flat leaf blades arising from rhizomes. Leaf blades are 1 to 2 cm wide and 15 to 120 cm long with prominent, white and slightly off-center mid-veins and very narrow and sharp tips (Hubbard et al., 1944). Stomata can be found on both surfaces of the leaf blades (Bryson and Carter, 1993).

Cogongrass inflorescence is a 10 to 20 cm long panicle containing an average of 460 individual spikelets (Bryson and Carter, 1993; Holm et al., 1977; Shilling et al., 1997). A single plant can produce as many as 3000 seeds (Sajise, 1972). Seeds are small and attached to a plume of silky white hairs allowing seeds to travel as long as 15 miles over open country (Hubbard et al., 1944). Larger spikelet clumps and greater wind speed favor larger dispersal distances (McDonald et al., 1996). Flowering is promoted through external stresses such as cold, mowing, tillage, or burning (Sajise, 1972), although some studies suggested flowers produced under such conditions cannot produce viable seeds (Eussen, 1980). In the Philippines, cogongrass flowers year-round (Holm et al., 1977) while flowering usually occurs in the late winter and early spring.
in the United States (Shilling et al., 1997; Willard, 1988). Cogongrass has been observed flowering throughout the year (Gaffney, 1996) supporting the argument by Dickens and Moore (1974) and Shilling et al. (1997) that cogongrass flowering, at least in southern US, does not depend on photoperiod.

Cogongrass seed germination requires light and appears to be photochrome mediated (Sajise, 1972; Soemarwoto, 1959, Dickens and Moore, 1974). The pH requirement of cogongrass seed germination is usually less than 5.0 (Sajise, 1972) and the optimum temperature for germination was reported to be 30 C (Dickens and Moore, 1974). Most seeds will remain viable for at least three months if stored under cold and dry conditions. Germination rates are as high as 95 to 98% immediately after harvest, providing adequate spikelet fill occurs (Santiago, 1965; Shilling et al., 1997). Seed viability has been shown to drop to 50% by 7 months and 0% by 11 months (Shilling et al., 1997). Cogongrass seedlings generally emerge in groups (Shilling et al., 1997) and are surprisingly weak competitors with other grass seedlings (Dozier et al., 1998; Shilling et al., 1997; Willard and Shilling, 1990). Cogongrass seedlings were shown to be less competitive than bahiagrass seedlings and were not able to establish in areas with > 75% coverage of bahiagrass sod. However, cogongrass ramets arising from rhizomes were shown to be more competitive than bahiagrass seedlings (Shilling et al., 1997; Willard and Shilling, 1990). Cogongrass seedling growth has been shown to be enhanced primarily by decreasing interspecific competition (King and Grace, 2000a, 2000b).

Cogongrass is a prolific seed producer; however, only cross-pollination from geographically isolated and heterogenous populations can produce viable seeds. McDonald et al. (1996), showed that cogongrass populations in Florida only produce self-incompatible seeds (McDonald et al., 1996). However, viable seed production may be occurring in Florida recently.
due to co-mingling of distinct populations. Seed would explain cogongrass invasion into new sites over large geographic areas and infestations has been found in isolated areas of Alabama and Mississippi, however, rhizome spread through movement of contaminated soil during construction or intentional forage planting has been acknowledged as the major mechanism of spread in the U.S. (Hubbard et al., 1944; Wilcut et al., 1988; Patterson et al., 1980, Patterson and McWhorter, 1980).

Cogongrass rhizomes are defined as C-strategist that can thrive in established population of other plants (Tominaga, 2003). Rhizomes are long, white and tough with prominent nodes and short internodes. The cataphylls (scale leaves) are brownish red and serve as a protective sheath around the rhizome (Ayeni, 1985; English, 1998). The sharp tip of the rhizome can grow into other plants roots, bulbs and tubers and cause physical damage as well as infection (Boonitee and Ritchit, 1984; Eussen and Soerjani, 1975). The sclerenchymous fibers under the epidermis and the sclerotic tissue surrounding the vascular bundles help rhizomes to conserve water and resist breakage, disruption, desiccation and heat from fire (Holm et al., 1977). Axillary bud dormancy is maintained through auxin-imposed apical dominance (Gaffney, 1996).

Rhizomes can produce over 40 tons per hectare fresh weight with density as high as 89 m linear length within 1 m² of soil surface area (Lee, 1977). Rhizomes form a dense mat underground and comprise over 60% of the total plant biomass. These structures facilitate rapid re-growth after defoliation (Sajise, 1976) and can give rise to 350 shoots in 6 weeks and cover a 4 m² area in 11 weeks (Eussen, 1980). Rhizomes are resistant to heat but susceptible to cold temperatures (Wilcut et al., 1988, Hubbard et al., 1944). Rhizomes are usually found within the top 8 to 40 cm of soil depending on soil type, but have the ability to grow as deep as 120 cm in the soil (Gaffney, 1996).
**Physiology**

Cogongrass is a perennial, rhizomatous, C4 plant that is able to tolerate drought, fire, cultivation and short-term shade (Terry et al., 1997). The light compensation point for cogongrass is 32 to 35 µmol m$^{-2}$ s$^{-1}$ (Gaffney, 1996; Ramsey et al., 2003), which is approximately 2% of ambient light intensity in North Central Florida (Gaffney, 1996). Some research suggested shading is beneficial for cogongrass control because it reduces carbohydrate storage, rhizome production and shoot weight. This results in decreased regeneration vigor increasing susceptibility to herbicides (MacDicken et al., 1997). Patterson (1980) showed that shading changed the total biomass, biomass distribution and plant morphology of cogongrass. Compared to plants in exposed habitats, cogongrass in shaded habitats possessed decreased total plant biomass, biomass distribution in rhizomes and an increase in biomass distribution in leaves. Cogongrass adaptations to light intensity level changes were realized by specific leaf area and leaf area ratio changes, tolerating a 50% reduction in sunlight. Moosavi-Nia and Dore (1979) observed the changes of herbicide movement under increasing shade. They found that glyphosate symptoms appearance was delayed in the shaded plants with increased efficacy. Light is a requirement for cogongrass rhizome propagation with light stimulating rhizome bud growth (Soerjani and Soemarwoto, 1969).

Vegetative reproduction by rhizomes is responsible for the local spread of cogongrass and new population establishment. A single rhizome system contains huge numbers of viable buds each with the potential to give rise to new shoots. Rhizome regeneration capacity increases with increasing age, weight, length, thickness and number of these visible buds. Younger buds have reduced regeneration capacity because of the lack of roots (Terry et al., 1997).

Ayeni (1985) developed a model of cogongrass rhizome growth and development. Early rhizome growth starting at the third or fourth leaf stage is vertical. When rhizomes start to
develop scale leaves at the fifth leaf stage, rhizome growth becomes horizontal. Rhizome tips then begin to grow upward and form secondary shoots and rhizomes. Buds on the convex side of the rhizome usually form new rhizomes but buds on the concave side will either develop shoots or remain dormant. Rhizomes and shoots develop at the same time on more mature plants but shoots develop first in stressed plants.

**Rhizome Physiology**

Correlative inhibition is a term defining the growth inhibiting effects of one part of the plant on another (Goebel, 1900; Smith and Rogan, 1980). In the case of apical dominance, apex buds exert control on the growth of axillary buds. There are four stages of apical dominance development: 1) axillary bud formation, 2) imposition of inhibition, 3) the release of apical dominance and 4) subsequent growth of axillary buds. Correlative inhibition is primarily present in the first two stages (Cline, 1997). In these stages the growing apex is a stronger competitor for water and nutrients, with a higher sink activity than axillary buds. This activity also influences herbicide efficacy because systemic herbicides move primarily to actively growing plant parts, and dormant buds with lower sink activities will accumulate a sub-lethal herbicide dose. The apex will be killed by the herbicides, releasing axillary buds which will start to grow. Therefore, the whole plant will escape from herbicide treatment.

There are several proposed mechanisms of apical dominance. The most widely accepted is the auxin inhibition hypothesis. Auxin produced at apical buds will move basipetally along the rhizome and directly inhibit lateral bud growth. This theory was confirmed by reproducing apical dominance through the application of exogenous auxin after removal of the apex (Leakey and Chancellor, 1975). Another theory is nutritional differences among plant parts. The apex is a stronger sink than basal parts so nitrogen and carbohydrates accumulate in the apex thus reducing growth of non-sink regions (McIntyre, 1990). McIntyre (1971, 1987) also suggested
that the rhizome apex could be a stronger competitor for water than axillary buds. The ratio of plant hormone concentration also appears to have effects in apical dominance. For example, auxin may cause inhibition of axillary buds in development stage II, but has promotional effects on stage IV. Cytokinin promotes the development of stage I, which is bud formation and will cause release of apical dominance (stage III) when exogenously applied (Cline, 1997; Tamas, 1987). Collectively these data infer apical dominance is the comprehensive effect of several factors including hormones, nutrients, and growth factors.

In terms of rhizome activity, generally lower light intensity will increase apical dominance because of the reduced amount of carbohydrates available and the competition between nodes for these carbohydrates (McIntyre, 1990). However, research on quackgrass showed that under lower light intensity, shoots will be produced in all rhizome nodes instead of being restricted to the nodes close to the apical end due to apical dominance (McIntyre, 1970). Some research showed that the percentage of buds producing shoots was not influenced by light in quackgrass (Chancellor, 1968; Jonson and Buchholtz, 1963), whereas Leakey et al. (1978) found that light would delay or prevent shoot formation, which is called light-induced inhibition.

It was suspected to due to the low rate of transpiration resulting to low water potential in the rhizomes under low light intensity.

Parental plants also have effects on axillary bud growth. Research on quackgrass rhizomes showed that the viability of buds increased with increasing distance from the parent plant (Dekker and Chandler, 1985; Tardif and Leroux, 1990) and it was suggested that the smaller internodes providing less nutrition might be the reason for the low viability of the basal buds (Tardif and Leroux, 1990). Leakey et al. (1975) found that in quackgrass rhizomes the removal of the apex would not fully release axillary buds from dormancy unless the whole rhizome was
removed from the parent plant. It was suggested that cytokinins from the roots on the parental plants, as well as auxins, nutrients and other hormonal factors are the reasons for parent plant factors.

In cogongrass, removal of the apex will release the axillary buds from apical dominance, with buds closest to the actively growing apex being less dormant and more likely to sprout (Gaffney, 1996). Scale leaves may also contribute to bud dormancy. One scale leaf is produced per rhizome node and its major function is as a protective sheath. Robertson et al. (1989) found that removal of scale leaves from quackgrass rhizomes was effective in promoting sprouting of previously dormant buds. English (1998) found similar results with cogongrass. Roberston et al. (1989) suggested that the reason might not be due to the factor of scale leaves but the exposure of buds to light and the secondary effects of light. This may also be the case in cogongrass as sprouting of cogongrass rhizomes is two to three times greater in light than under dark conditions (Holm et al., 1977). Research also suggests that the ABA produced in scale leaves might contribute to bud dormancy, but low levels of ABA were found in the scale leaves (Taylor et al., 1995).

**Habitat**

Cogongrass usually thrives in tropical and subtropical areas where annual rainfall is 75-500 cm (Bryson and Carter, 1993). This species does not infest cultivated areas but is found in disturbed areas such as roadways, pastures, mine sites, forest land, park and other recreational areas. It grows under a wide range of soil conditions (Bryson and Carter 1993, Gaffney, 1996) and can tolerate soils with low pH (4.7), poor fertility, and low organic matter (Sajise, 1980). Ramakrishnan and Saxena (1983) reported its high efficiency in nutrient uptake, associations with mycorrhiza (Brook, 1989), and high utility of phosphorus (Brewer and Cralle, 2003). The competitive nature of cogongrass contributes to its ability to exclude other species and establish
monotypic stands. In addition to direct competition, cogongrass interferes other plant growth through allelopathy (Eussen, 1979; Casini et al., 1998; Koger and Bryson, 2003).

Management

Herbicide trials have been conducted extensively on cogongrass with hundreds of materials tested, but imazapyr and glyphosate have been shown to be the only consistently effective herbicides. The most effective application dosages are 4.5 kg/ha and 0.8 kg/ha, for glyphosate and imazapyr respectively (Willard et al., 1996). Compared to glyphosate, imazapyr provides a longer period of control due to soil activity, but off-target effects are a concern with its usage (MacDonald et al., 2002). It was shown that imazapyr applied in late summer or early fall can provide cogongrass control as long as 18 month (Dozier et al., 1998).

Herbicide efficacy can be influenced by many factors. Developmental stage at time of application influences efficacy. Under greenhouse conditions, glyphosate applied to 8-week-old cogongrass provided better control than at 12 weeks after planting. It was hypothesized that the delay in application timing allowed the rhizome system to become more established (Willard, 1988). Seasonal application timing is another important consideration for control. Application in the fall has consistently shown better herbicide activity relative to spring applications due to basipetal movement of photosynthates in the fall of the year (Johnson et al., 1999; Gaffney, 1996; Tanner et al., 1992). Essentially, fall applications result in greater herbicide loading into the rhizome complex. However, low rainfall during that time of the year may cause low available soil moisture thus reducing herbicide movement and activity (Dozier et al., 1998).

Cogongrass responds differently to mechanical treatment. Shallow tillage only provides short-term reductions in shoot growth, while deep tillage provides better control by cutting rhizomes into fragments and by bringing material to the soil surface to desiccate. Multiple and deep tillage more than 15 cm over a period of months to 2 years may achieve good results
(Shilling et al., 1995). Disking has been shown to provide a 27% decrease in rhizome biomass one time and a 66% decrease when performed twice (Willard et al., 1997). Mowing alone does not control cogongrass but can reduce rhizome and foliage biomass (Willard and Shilling, 1990; Willard et al., 1996). It has been suggested that one of the most effective cogongrass management practices is integrating disking with herbicide applications. Deep disking 20 to 30 cm to break apical dominance and promote new leaf growth, followed by herbicide application to regrown shoots, followed by postapplication discing will enhance herbicide incorporation in the soil (Dozier et al., 1998).

Integrated management is necessary for cogongrass management since single management methods have often failed to provide effective management. Incorporating different methods including burning, tilling, mowing, cultural and chemical control will provide more effective long term control (Gaffney, 1996). After control, the niche released by cogongrass must be rapidly replaced by desirable plant species to exclude cogongrass reinvasion. The choice of a desirable species is dependent on species tolerance to the previous cogongrass management techniques and soil type. Some have suggested bahiagrass (*Paspalum notatum*) and hairy indigo (*Indigofera hirsuta* Harv.) might be alternative choices.

**Rationale**

Emphasis has been made on the importance of an integrated management system to suppress cogongrass. Although field trials integrate different weed control methods, physiological and biological information concerning cogongrass will be helpful in refining control strategies. The biggest issues with cogongrass management include competitiveness, low light tolerance, and rhizome dormancy. These factors aid in the ability of cogongrass to compete and exclude other species. These studies will focus on the biological characteristics of cogongrass competitiveness, rhizome system development under different environmental
conditions, and the potential for rhizome dormancy. The specific objectives of these experiments are:

- Evaluate the competitiveness of cogongrass and bahiagrass under different soil pH conditions
- Evaluate the growth patterns of cogongrass as a function of maturity level under different light intensities
- Evaluate the level of abscisic acid in cogongrass rhizomes as a function of node position

The hypotheses addressed follow:

-Cogongrass competitiveness with bahiagrass changes as a function of soil pH
- Shoot to rhizome ratios in cogongrass will change as a function of light intensity and maturity level
- Abscisic acid is present in cogongrass rhizomes at levels and locations that indicate a role in bud dormancy
CHAPTER 2
COGONGRASS-BAHIAGRASS COMPETITION AS A FUNCTION OF SOIL PH

Introduction

Cogongrass [Imperata cylindrica (L.) Beauv.] is a warm-season, rhizomatous, perennial, C4 grass that has become a serious pest throughout the tropical and subtropical areas of the world (Favley, 1981; Holm et al., 1977). It is listed on the Federal Noxious Weed List (USDA Animal and Plant Health Inspection Service, 2006) and several states’ Noxious Weed Lists including Florida, Alabama, Mississippi, North Carolina, Vermont and Hawaii (USDA Natural Resources Conservation Service Plants Profile, 2008).

Florida has large tracts of grazing land ranging from improved pastures to rangeland. Rangelands here means long-term pasture settings that generally receive lower levels of input and thus experience a greater degree of species invasion that further reduces forage performance. Cogongrass is currently increasing in importance in Florida’s rangeland grazing areas.

Bahiagrass (Paspalum notatum) is a pasture species at risk of infestation by cogongrass (Shilling et al., 1997; Willard and Shilling, 1990). Bahiagrass is the most widely utilized forage in Florida, covering an estimated 2.5 million acres (Chambliss, 1996). Bahiagrass is a warm season perennial species with a deep fibrous root system, and can thrive in dry, infertile soils (Beard, 1980; Watson and Burson, 1985). It requires little to no irrigation, minimal fertilization and is susceptible to relatively few insect or disease pests (Chambliss, 1996). The optimal condition for bahiagrass growth is a soil pH of about 5.5 to 6.5. Seeds are commonly used to establish bahiagrass, but seedlings are weak competitors (Beard, 1980; Watson and Burson, 1985). This slow establishment rate from seeds and poor growth under moderate or heavy shade makes bahiagrass susceptible to competition from aggressive grass species (Busey and Myers, 1979; Watson and Burson, 1985).
Cogongrass generally infests areas with low soil pH (pH≈4.7), poor fertility and low organic matter (Sajise, 1980; Wilcut et al., 1988). Cogongrass does not tolerate tillage, and is therefore, commonly found on roadways, pastures, reclaimed mining areas, forest land, parks and other recreational areas (Gaffney, 1996). Cogongrass is highly competitive, often forming monotypic stands. Mechanisms of interference can include competition, allelopathy and physical injury (Eussen and Soerjani, 1975; Eussen, 1979). These mechanisms retard growth, cause yellowing and die-back and otherwise reduce yield and/or growth of other crops and desirable plants (Hubbard et al., 1944; Soerjani, 1970). Once cogongrass has invaded an area, establishment of other desirable grass species is difficult due to the dense rhizome system. This extensive rhizome network physically excludes other vegetation and quickly extracts soil moisture and nutrients from shallow soil layers (Boonitee and Ritdhit, 1984; Terry et al., 1997; Casini et al., 1998; Koger and Bryson, 2003).

Cogongrass is capable of reinvading an unoccupied ecological niche after effective control methods are implemented. Therefore, the reestablishment of desirable forages is critical to prevent re-invasion by cogongrass (Shilling et al., 1997; Yandoc et al., 2004). According to Gaffney (1996), the success of this strategy relies on the tolerance of the desirable species to the herbicide used for cogongrass and agronomic conditions favorable for growth. He suggested that the use of herbicides followed by the establishment of either bermudagrass (*Cynodon dactylon*) will effectively suppress cogongrass and prevent reinfestation. Barron et al., (2003) reported similar results using bahiagrass and bermudagrass under field conditions.

According to Shilling (1997), the degree of invasiveness of a given species at a given time can be defined as the relative ability of that species to displace other species. Competitiveness can be regarded as the determinant of displacement ability. Therefore, the relative
competitiveness of bahiagrass and cogongrass can be a crucial factor in cogongrass invasion into bahiagrass pastures. Bahiagrass is usually established from seeds with a relatively slow growth rate (Beard, 1980), while cogongrass most commonly spreads from rhizomes. Willard and Shilling (1990) showed that the rate of cogongrass establishment is more rapid than that of seedling bahiagrass. Additional greenhouse studies by Willard et al., (1990) demonstrated the influence of growth stage on competition between bahiagrass and cogongrass. Results showed bahiagrass seedlings to be less competitive than cogongrass emerging from rhizomes, while established bahiagrass, i.e. in this case bahiagrass planted 8 weeks before cogongrass, showed more competitiveness than cogongrass. Furthermore, bahiagrass establishment combined with one mowing maximized competitiveness. Under conditions of no nutrient or water stress, cogongrass effectively competed with seedling bahiagrass but not with established bahiagrass. This information provides us with the basic theory of evaluating the relative competitiveness between these two species. Under optimal environmental conditions and at certain growth stages, bahiagrass can actually be more competitive than cogongrass (Shilling 1997). Therefore, if we improve growth conditions for bahiagrass, it will likely prevent the establishment of cogongrass.

As stated previously, cogongrass has the ability to reinvade into an unoccupied ecological niche. Therefore, if conditions to improve bahiagrass seedlings’ competitiveness could be achieved, long-term cogongrass control may be realized.

The classic replacement-series experimental design (de Wit, 1960) has been widely used in experimental studies of interspecies competition. For a two-species design, the experiment consists of a pure stand of each species alone and combinations of mixtures of different ratios while maintaining constant overall density. The biomass each species contributes to the total biomass at each different planting ratio determines competitiveness at a given ratio. Performance
of the species in mixture compared with that in a pure stand is used to assess relative competitiveness and aggression. Environmental conditions such as fertilizer, water status, soil pH etc. can also be manipulated to study the influence of different parameters on competitiveness.

In summary, it is noted that optimum soil pH for bahiagrass is 5.5 to 6.5 while cogongrass is often found in acidic soils as low as pH≈4.7. While both species can survive on low fertility soils, low soil pH appears to have a greater negative impact on bahiagrass than on cogongrass (Shilling, 1997). Soil pH could play a major role in the management of competitiveness between these two species.

**Research Objectives and hypothesis**

**Objective**

The objective of this experiment was to study the competition between rhizomatous emerging cogongrass established from rhizomes and seedling bahiagrass as affected by two different levels of soil pH (pH=6.8, pH=4.5).

**Hypothesis**

Our hypothesis is that the relative competitiveness of cogongrass and bahiagrass will be changed by different soil pH levels.

**Materials and Methods**

Preliminary density and fertility studies performed by Shilling et al., (1997) showed that cogongrass density of 144 plants/m² and bahiagrass density of 720 plants/m² are optimal plant densities for both species. To minimize intraspecific competition and maximize interspecific competition, soil fertility level was achieved based on previous research by Shilling et al. (1997)
with 45.4 kg ha\textsuperscript{1} N by using 14-14-14 fertilizer\textsuperscript{1}. These methods will be used in the following experiments.

Cogongrass plants used in this experiment were propagated from 10 cm long rhizome segments collected from a population in Gainesville, FL. Rhizomes were planted in plastic flats and covered with commercial potting mix\textsuperscript{2} and were placed in direct sunlight area and watered daily. After two weeks, plants with 2 leaves attached to a single node were selected and transplanted. Bahiagrass seeds were planted in plastic flats similar to cogongrass and approximately 3 weeks after planting, 2-leaf-stage seedlings of equal size were selected and transplanted. Both species were transplanted at the same time into 4L volume pots at desired density levels. Plants were then placed in a greenhouse under the following environmental conditions: 16-h day: 8-h night photoperiod and 30 °C day: 20 °C night temperatures.

In this study, two pH treatment levels were evaluated, pH=6.8 and pH=4.5. Soil was a Chandler fine sand and was gathered from the University of Florida Plant Science Research and Education Center in Citra, FL. For each soil pH level, a replacement-series model for studying competition between the two species was established. Cogongrass densities were 0, 1, 2, 4, and 8 shoots/pot and corresponding bahiagrass densities were: 40, 20, 10, 1, and 0 plants/pot, respectively. The resulting proportions were: (40:0), (20:1), (10:2), (1:4) and (0:8) for a single pot; these proportions were based on previous research (Shilling et al., 1997). The experimental unit was one pot and it was considered as one replication. The experiment was established as a 2 (pH levels) by 5 (densities) factorial in a completely randomized design with 4 replications. The study was conducted in August 2007 and repeated in October 2007. At 8 wks after transplanting,

\textsuperscript{1} Scotts Osmocote 14-14-14
\textsuperscript{2} Fafrad super-fine germination mix. Conrad Fafrad. Inc. P.O.Box 790. Agawam, MA 01001-0790
species were harvested and separated. Plant tissues were placed in a 75 °C oven for 3 days and dry weights were determined.

Competitive indices were based upon shoot biomass and calculated as follows (de Wit, 1960; de Wit and van den Bergh, 1965; McGilchrist and Trenbath, 1971; Rejmanek et al., 1989; Radosevich, 1988):

**Relative yield (RY):** the yield of each species in mixture as a percent of its monoculture yield under the same growing conditions.

\[ RY \text{ of species A} = \frac{\text{biomass production of species A at a particular proportion}}{\text{biomass production of species A in monoculture}} \]

**Relative yield total (RYT):** the absolute yield of each species within a given proportion.

\[ RYT = RY \text{ of species A} + RY \text{ of species B} \]

**Relative crowding coefficient (RCC):** a measure of the relative competitiveness of one species over another.

\[ \text{RCC of species A with respect to species B} = \frac{\text{dry weight/plant of species A at a particular proportion}}{\text{dry weight/plant of species B at a particular proportion}} \times \frac{\text{dry weight/plant of species A at monoculture}}{\text{dry weight/plant of species B at monoculture}} \]

RCC is an index. An RCC of 1.00 indicates equal competitiveness between the two species and RCC increases as a certain species’ competitiveness increases.

**Aggressivity (A):** another way to measure the relative competitive ability of each species

\[ A \text{ of species A} = \frac{\text{dry weight/plant of species A at a particular proportion}}{\text{dry weight/plant of species A at monoculture}} - \frac{\text{dry weight/plant of species B at a particular proportion}}{\text{dry weight/plant of species B at monoculture}}. \]

A value of 0 denotes equal competitiveness while species with positive values are considered to be more competitive.

All data were subjected to ANOVA. A replacement series curve model under different pH levels was created based on RY and RTY of both cogongrass and bahiagrass.
Results

Plant Biomass

ANOVA results indicated no run by density interaction; therefore, data from both experiments were pooled.

As expected, total biomass for cogongrass increased as the proportion of cogongrass plants per density level increased. However, there was no significant difference (P>0.05) for cogongrass biomass between the two pH levels within each level of plant density (Figure 2-1A). This trend was also observed for bahiagrass (Figure 2-1 B). There was also no significant difference (P>0.05) between bahiagrass shoot biomass within each level of plant density between the two different soil pH levels.

On a per-plant basis, cogongrass biomass declined slightly at the higher densities (4:01 and 8:0). There was no significant difference between biomass at each pH level within a given density (Figure 2-2 A). For bahiagrass (Figure 2-2 B), single plant shoot biomass also decreased as the proportion of bahiagrass in the mixture increased. There was no difference in individual plant biomass as a function of soil pH at each density level, except at the lowest bahiagrass: cogongrass proportion (4:01). At this level, bahiagrasss shoot biomass was two times greater under high soil pH condition.

In terms of total plant biomass per pot and single plant biomass, cogongrass performed better under low soil pH, while, bahiagrass performed better under high soil pH. Our results indicated that cogongrass growth is similar under widely different soil pH values, while low soil pH level negatively impacted bahiagrass biomass production. For single plant biomass, there was no statistical difference between cogongrass at different density levels. The situation for bahiagrass was different. Single plant biomass increased as plant density decreased. However, there was an obvious trend that under higher levels of competition (1:20, 2:10), cogongrass
performed better under low pH conditions. Under higher levels of competition (2:10, 4:1), bahiagrass performs better under high soil pH conditions.

**Replacement Series**

Under low soil pH (Figure 2-3 A), relative yield curves of cogongrass and bahiagrass crossed at the density level of cogon: bahia=2:10, indicating similar relative yield and thus similar competitiveness at this density. Under high soil pH conditions (Figure 2-3 B), relative yield curves of cogongrass and bahigrass crossed at the density level between cogon:bahia=2:10 and cogon:bahia=4:1. At pH 6.8, a lower density of bahiagrass (i.e. greater density of cogongrass) achieved the same level of competitiveness as the pH 4.7 soils. Therefore, high soil pH favors bahiagrass competitiveness. A similar conclusion can also be drawn by the species contribution to total yield. The area below relative yield (RY) line of a certain species shows yield contribution of that species to total yield and the area below relative yield total (RYT) line shows total yield. Under low soil pH, cogongrass and bahiagrass contributed similarly to the total yield, while bahiagrass contributed more to the total yield under high soil pH. Moreover, the area difference of relative yield of cogongrass/bahiagrass between different soil pH levels indicated the competitiveness changes for both species. Comparing with low soil pH, cogongrass area, i.e., its relative yield was slightly decreased and bahiagrass area, i.e., its relative yield was increased under high soil pH.

Numeric values for relative yield (RY) and relative yield total (RYT) are presented in table 2-1. ANOVA results indicated that for cogongrass RY was only significantly higher at low soil pH at 2:10 densities, but not significantly different between two pH levels at 1:20 and 4:01 density level. However, the overall trend is that cogongrass RY at all densities is higher at low soil pH. Bahiagrass RY at high pH was significantly higher than that at low pH at 4:01, however, there is also a obvious trend that bahiagrass RY are higher under high soil pH at all densities.
The biggest RY difference for both species between high and low soil pH occurred at the cogon: bahia density of 2: 10. For RYT, there was no significant difference between pH levels (P=0.79). RYT of mixed species in each density level was always below 1, indicating antagonistic effects between cogongrass and bahiagrass.

**Relative Crowding Coefficient (RCC) and Aggressivity (A)**

RCC is an index indicating the relative competitiveness of one species to another. The relative competitiveness of a species increases as the RCC increases. A RCC of 1 indicates both species are equally competitive (Harper, 1977). ANOVA results showed that cogongrass RCC at low pH was not significantly higher than at high pH, however, the overall trend was cogongrass RCC was higher at low soil pH. Conversely bahiagrass RCC at low pH was significantly lower than at high pH except for cogongra : bahiagrass=2:10. At low soil pH, bahiagrass was more competitive than cogongrass at cogongrass: bahiagrass=1:20 and 4:1 but less competitive than cogongrass at cogongrass: bahiagrass=2:10 (Table 2-2). However, at high soil pH, bahiagrass RCC was significantly higher than cogongrass at almost all density levels except for cogongrass: bahiagrass=1:20.

For aggressivity (A), the species with a positive A value is considered to be the more competitive, negative value the less competitive, and a A value of 0 means equal competitiveness between species. For cogongrass aggressivity (A) value, ANOVA indicated a pH by density interaction (P=0.01). The aggressivity of cogongrass at low soil pH had positive A values at cogon: bahia =1:20 and 2: 10, but negative values at cogon: bahia =4:1 while interacting with bahiagrass. At low densities and low pH, cogongrass showed more aggressivity than bahiagrass. Under high soil pH, cogongrass A values were all negative at all density levels, indicating bahiagrass was more aggressive than cogongrass.
RCC and A values did not reflect the exact same trend compared to relative competitiveness of cogongrass and bahiagrass as a function of density and soil pH. However, it is clear by both indices that bahiagrass competitiveness was greatly enhanced at high soil pH. The largest difference between both RCC and A of bahiagrass and cogongrass occurred at the ratio of cogon: bahia= 2:10. Once again this appears to be the density at which bahiagrass competitiveness against cogongrass is most profoundly affected by soil pH.

Therefore, there is a significant difference between high and low soil pH on the relative competitiveness of cogongrass and bahiagrass. High soil pH increased bahiagrass competitiveness. Both cogongrass and bahiagrass competitiveness were significantly different at two different soil pH levels and cogongrass competitiveness was not greater than bahiagrass at low soil pH.

**Discussion**

A deWit replacement model was used to determine the relative competitiveness of the two plant species. Willard and Shilling (1990) mentioned in their research on competition between cogongrass and bahiagrass that since comparisons were made between two species at different stages of development, prudence should be exercised when drawing conclusions based on the replacement model. However, the data provide essential background information for further research on competition between cogongrass and bahiagrass.

Although high and low soil pH treatments did not result in significant plant biomass differences for either species, relative competitiveness was shown to be significantly different as a function of soil pH. Cogongrass RY, RCC and A were all lower under low soil pH, conversely, bahiagrass RY, RCC and A were all significantly greater under high soil pH conditions.

Our results differ somewhat from previous research which focused on the influence of propagule type for cogongrass and bahiagrass. Shilling et al. (1997) compared the relative
competitiveness between cogongrass seedlings and bahiagrass seedlings and cogongrass ramets and bahiagrass seedlings. These data indicated that bahiagrass seedlings were not as competitive as cogongrass ramets (i.e., cogongrass established from rhizomes, but were more competitive than cogongrass seedlings. Therefore, propagule type is an important factor in the relative competitiveness between the two species. Research conducted by Willard et al. (1990) reflected similar results as Shilling et al. (1997) where cogongrass maintained advantage over seedling bahiagrass regardless of the species mixture, but was less competitive than established bahiagrass. However, under field conditions, it is more common that bahiagrass pastures are established from seeds and cogongrass is introduced or invades by rhizomes. For established bahiagrass pastures, human or natural disturbance may play an important role in cogongrass invasion. In many areas cogongrass does not become dominant until disturbance releases it from competition, however, after invasion, cogongrass can successfully displace the original species and establish a monoculture (Willard et al., 1990; Eussen and Soerjani, 1975).

In previous research (Willard et al., 1990 and Shilling et al., 1997), neither study noted the pH of the soil used in the experiments. Since previous research is somewhat contradictory as to the competition between cogongrass and bahiagrass, it is important to discern whether differing soil conditions would change competitiveness.

Our results indicated that at pH value < 5, the competitiveness between cogongrass and bahiagrass was similar. At pH> 5, bahiagrass showed greater competitiveness over cogongrass, supporting the environmental conditions which favor cogongrass and bahiagrass. Optimum soil pH for bahiagrass is 5.5 to 6.5 (Chambliss, 1996), while cogongrass has the ability to thrive in habitats with acidic soils as low as pH≈4.7 (Sajise, 1980; Wilcut et al., 1988). The significant differences between cogongrass and bahiagrass performance at different soil pH indicated soil
pH plays an important role in the competitiveness between the two species. Cogongrass ramets have been shown to effectively compete with bahiagrass seedlings, however, when soil pH is favorable for bahiagrass, the competitive edge of cogongrass is lost. Moreover, our studies demonstrated that cogongrass ramets did not show overwhelmingly greater competitiveness over bahiagrass even at a soil pH that is considered to favor cogongrass growth. Bahiagrass seedlings are generally regarded as weak competitors because of slow growth rate; however, our research showed that under certain conditions, bahiagrass competitiveness can be elevated. Therefore, proper soil conditions could defer cogongrass invasion into bahiagrass.
Figure 2-1 Average shoot biomass under different plant density levels and different soil pH levels. Data represented as the mean of 8 replications with standard error. A) Cogongrass shoot biomass per pot/rep B) Bahiagrass shoot biomass per pot/rep
Figure 2-2 Shoot biomass per plant under different plant density levels and different soil pH levels. Data represented as the mean of 8 replications with standard error. A) Cogongrass shoot biomass per plant B) Bahiagrass shoot biomass per plant
Figure 2-3 Relative yield (RY) and relative yield total (RYT) of cogongrass and bahiagrass shoot biomass. Data represented as the mean of 8 replications with standard error. A) Low soil pH 4.7 B) High soil pH 6.8
Table 2-1 Cogongrass and bahiagrass under different soil pH levels (low and high) and density levels as measured by relative yield (RY) and relative yield total (RYT).

<table>
<thead>
<tr>
<th>Density (cogon: bahia)</th>
<th>RY Cogon</th>
<th>RY Bahia</th>
<th>RYT</th>
</tr>
</thead>
<tbody>
<tr>
<td></td>
<td>low</td>
<td>high</td>
<td>low</td>
</tr>
<tr>
<td>0:40</td>
<td>-</td>
<td>-</td>
<td>1</td>
</tr>
<tr>
<td>1:20</td>
<td>0.19 ± 0.05A</td>
<td>0.12 ± 0.02A</td>
<td>0.66 ± 0.35A</td>
</tr>
<tr>
<td>2:10</td>
<td>0.42 ± 0.06A</td>
<td>0.24 ± 0.03B</td>
<td>0.39 ± 0.05A</td>
</tr>
<tr>
<td>4:01</td>
<td>0.66 ± 0.05A</td>
<td>0.59 ± 0.05A</td>
<td>0.04 ± 0.01B</td>
</tr>
<tr>
<td>8:00</td>
<td>1</td>
<td>1</td>
<td>-</td>
</tr>
</tbody>
</table>

*Data based on aboveground shoot biomass and are represented as the mean of 8 replications with standard error.

*Mean separation is only shown between different soil pH (low and high) within each density level for RY cogongrass, RY bahiagrass and RYT respectively. The two data within the same row between low and high soil pH with the same letter means they are not significantly different according to Fisher’s Protected LSD test at P≤ 0.05.*
Table 2-2 Cogongrass and bahiagrass as measured by relative crowding coefficient (RCC) and aggressivity (A).

<table>
<thead>
<tr>
<th>Density (cogon: bahia)</th>
<th>RCC Cogon low</th>
<th>RCC Cogon high</th>
<th>RCC Bahia low</th>
<th>RCC Bahia high</th>
<th>A cogon(^b) low</th>
<th>A cogon(^b) high</th>
</tr>
</thead>
<tbody>
<tr>
<td>0:40</td>
<td>0 ± 0(^a)</td>
<td>0 ± 0</td>
<td>N/A</td>
<td>N/A</td>
<td>-1 ± 0</td>
<td>-1 ± 0</td>
</tr>
<tr>
<td>1:20</td>
<td>1.23 ± 0.44A</td>
<td>0.64 ± 0.12A</td>
<td>1.25 ± 0.22A</td>
<td>2.55 ± 0.73A</td>
<td>0.06 ± 0.04A</td>
<td>-0.52 ± 0.17B</td>
</tr>
<tr>
<td>2:10</td>
<td>1.49 ± 0.49A</td>
<td>0.63 ± 0.20A</td>
<td>1.10 ± 0.22B</td>
<td>3.26 ± 0.65A</td>
<td>0.11 ± 0.03A</td>
<td>-1.31 ± 0.32B</td>
</tr>
<tr>
<td>4:01</td>
<td>0.91 ± 0.17A</td>
<td>0.53 ± 0.19A</td>
<td>1.44 ± 0.25B</td>
<td>4.71 ± 0.65A</td>
<td>-0.47 ± 0.19A</td>
<td>-3.52 ± 1.34B</td>
</tr>
<tr>
<td>8:00</td>
<td>N/A</td>
<td>N/A</td>
<td>0 ± 0</td>
<td>0 ± 0</td>
<td>1 ± 0</td>
<td>1 ± 0</td>
</tr>
</tbody>
</table>

\(^a\) Data based on aboveground shoot biomass and are represented as the mean of 8 replications with standard error.

\(^b\) Bahiagrass has the same aggressivity value as cogongrass, but the sign is opposite.

\(^c\) Mean separation is only shown between different soil pH (low and high) within each density level for RCC cogongrass, RCC bahiagrass and A cogongrass respectively. The two data within the same row between low and high soil pH with the same letter means they are not significantly different according to Fisher’s Protected LSD test at P≤ 0.05.
CHAPTER 3
GROWTH AND PHOTOPRODUCTIVE PARTITIONING OF COGONGRASS UNDER DIFFERENT LIGHT INTENSITIES

Introduction

Cogongrass [Imperata cylindrica (L.) Beauv.] is a tropical, rhizomatous grass native to southeast Asia (Holm et al., 1977). It is a serious weed throughout the warmer regions of the world and mainly infests non-cultivated areas (Dickens, 1974; Holm et al., 1977).

Cogongrass grows vigorously in a wide variety of environmental conditions (Holm et al., 1977) and competitive against other plant species for nutrients and light (Boonitee and Ritdhit, 1984). According to Eussen and Wirjahardja (1973), plants which can survive competition with cogongrass usually have a deeper root system and taller canopy than cogongrass.

Previous studies have investigated the impact of shading on cogongrass (Patterson et al., 1980; Patterson, 1980). These studies focused on the presence of sun and shade cogongrass ecotypes. They evaluated cogongrass growth under shade levels which simulate conditions under agronomic crop canopies. Flint and Patterson (1980) collected mature cogongrass plants from shaded and exposed habitats and found that plants from both areas exhibited adaptation to shade through an increase in specific leaf area, leaf weight ratio and leaf area ratio. Short term (90 days) shading decreased dry matter, leaf area, growth rate and net assimilation rate in plants, although partitioning of plant biomass increased in leaves. Plants from shaded and full sun habitats responded similarly to shading, indicating little evidence to support the existence of sun and shaded ecotypes of cogongrass.

However, considering the low (32-35 µmol m\(^{-2}\) s\(^{-1}\)) light compensation point of cogongrass (Ramsey et al., 2003), the light intensity levels of this previous study may not have imposed significant stress for cogongrass. Additionally, Flint and Patterson (1980) only collected aboveground biomass data, while changes in underground biomass were not mentioned.
Furthermore, only mature plants were examined and they did not address the impacted of low light on cogongrass at establishment. However, MacDickens et al., (1997) stated that Imperata spp. is shade-intolerant, with shading quickly reducing carbohydrate storage, rhizome production, and shoot dry weight, with increased susceptibility to competition and herbicides. Their research results indicated that herbaceous cover crops and tree fallows with fast-growing species such as Egyptian riverhemp (Sesbania sesban), thorn mimosa (Acacia nilotica) or lead tree (Leucaena leucocephala) could provide shade-based control of cogongrass. Menz and Grist (1996) also suggested increasing rubber (Ficus elastica) planting density to shade Imperata would be a bioeconomic approach to control Imperata spp.

The success of cogongrass is largely due to rhizome regeneration capacity. Rhizomes can reproduce and spread at a rate of 350 shoots in 6 weeks and can cover 4 m² in 11 weeks (Eussen, 1980). Additionally, the presence of apical dominance prevents axillary bud formation and maintains large rhizome base and carbohydrate storage. Decreasing light intensity will likely change the growth pattern and carbon-assimilate partitioning of the whole plant, especially the rhizome system. It is plausible that cogongrass will reduce rhizome biomass to support greater leaf biomass, thus changing the plant shoot to rhizome ratio under low light intensities.

Low light conditions are often associated with a shift in biomass allocation patterns between shoots and roots. Light quantity will influence the photosynthesis rate, which will determine the amount of substrate available for growth. A model Thornley (1972) developed can be applied directly to light influences on the shoot to rhizome ratio. This model states that the growth of shoots and rhizomes both depend on the concentration of labile carbon (C) and nitrogen (N) and the transport of C and N between shoots and rhizomes. Translocation depends on the difference in C and N concentration between shoot and rhizome. Rhizome growth is
determined by the residue of assimilate available after shoot growth. Therefore, in this model, greater light levels will result in greater available carbon for rhizome growth. Conversely, low light levels will shift carbon allocation to shoots, since the shoot is the dominant sink, regardless of light regime.

**Objectives and Hypothesis**

The objective of this experiment was to examine the effect of different light intensities on the growth and carbon allocation of newly established and mature cogongrass.

Our hypothesis is that lower light intensities will enhance carbon assimilate partitioning to the shoot at the expense of the rhizome, changing shoot to rhizome ratio. It is also assumed that changes in root: shoot partitioning will differ between newly established and mature cogongrass plants.

**Materials and Methods**

**Plant Material Preparation:** Cogongrass rhizome materials were collected from a single population in Gainesville, FL. For mature plants, rhizomes were transplanted in 4 L pots filled with commercial potting soil. Plants were allowed to grow under natural sunlight conditions for 12 weeks to ensure full rhizome development. All leaves were removed prior to shading exposure. For newly established plants, 10 cm long rhizome segments that were sprouted in plastic trays. Trays were filled with commercial potting mix and placed outside under natural conditions in August 2007. After 3 weeks, uniform shoots were selected and the shoots with their connected rhizome nodes were detached from the original rhizome segments and were transplanted to pots, similar to mature plants, and remained outside for an additional 2 weeks.

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1 Fafrad super-fine germination mix. Conrad Fafrad. Inc. P.O.Box 790. Agawam, MA 01001-0790
before treatment. All plants were fertilized and watered as needed for the duration of the study. Initial density was 4 plants per pot for both mature and newly established plants.

**Light Intensity Treatment:** Both mature cogongrass plants and newly established plants were placed in a greenhouse (25 °C day/20 °C night) under different light intensities. Respective light treatments were 75%, 25%, 15%, 2% and 1% of full sunlight (2200 µmol m$^{-2}$ s$^{-1}$). The 75% of full sunlight was not achieved by artificial treatment; instead, it is due to the sunlight reduction from the top of greenhouse. The 2% sunlight treatment reflects the light compensation point (near 35 µmol m$^{-2}$ s$^{-1}$) for cogongrass (Ramsey et al., 2003). 1% was lower than cogongrass light compensation point and was considered as stress. Layers of shade cloth placed 1 meter above the plants were used to achieve the desired light levels and actual light intensity was measured by quantum sensor$^2$. The experimental design was a 2 (plant age) by 5 (light intensity) factorial blocked by light regime with 6 replications. The study was initiated on Sept. 8th, 2007 and repeated on Jan. 20th, 2008.

**Data Collection and Mathematical Analysis of Growth:** At the time of study initiation, 5 pots were harvested from each maturity group and separated into aboveground (shoot) and belowground (rhizome) biomass. This served as a time zero from calculating growth rate and other parameters from the time course of the studies. Plants were harvested after 12 weeks of shade exposure to evaluate shoot to rhizome ratios. Plant height was measured just prior to harvesting and plants were separated into shoots and underground biomass. Leaf area was measured using a automatic leaf area meter$^3$. All plant materials were dried at 70 °C oven for 3 days and shoot dry weight (DW), rhizome dry weight (DW) and total plant dry weight (DM)

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$^2$ Li-Cor LI-170  
$^3$ Li-Cor Model 3100 Area Meter, Li-Cor Bioscience 4647 Superior Street Lincoln Nebraska 68504
were measured. From these data, parameters relating to growth analysis were calculated.

Parameters included leaf weight ratio (LWR), rhizome weight ratio (RWR), rhizome leaf ratio (RLR), specific leaf area (SLA), leaf area ratio (LAR), leaf area duration (LAD), net assimilating rate (NAR), relative growth rate (RGR, Kw), relative leaf area expansion rate (Ka), relative leaf growth rate (Ki), leaf area partition coefficient (LAP), leaf weight partition coefficient (LWP) and dry matter production over the period (ΔW). All calculations were based on Potter and Jones (1977) and Patterson (1980) as follows:

\[
\begin{align*}
LWR &= \text{shoot DW/total DW, g g}^{-1} \\
RWR &= \text{rhizome DW/total DW, g g}^{-1} \\
RLR &= \text{rhizome DW/shoot DW, g g}^{-1} \\
SLA &= \text{leaf area/shoot DW, dm}^2 \text{g}^{-1} \\
LAR &= \text{leaf area/total DW, dm}^2 \text{g}^{-1} \\
LAD &= (\text{leaf area-1}/\ln(\text{leaf area})) \times \text{time duration, total amount of leaf area present during the interval, dm}^2 \text{days} \\
Ka &= \ln(\text{leaf area})/\text{time duration, relative leaf area expansion rate/day} \\
Ki &= \ln(\text{shoot DW})/\text{time duration, relative leaf growth rate/day} \\
Kw (RGR) &= \ln(\text{total DW})/\text{time duration, relative growth rate/day} \\
LAP &= (Ka \times \text{leaf area})/(Kw \times \text{total DW}) \\
LWP &= (Ki \times \text{shoot DW})/(Kw \times \text{total DW}) \\
NAR &= (Kw \times \text{total DW})/\text{leaf area, average DW production per unit leaf area, g dm}^2 \text{day}^{-1} \\
\Delta W &= NAR \times LAD
\end{align*}
\]

Data were subjected to analysis of variance to test for treatment differences. Means are presented with standard errors and separated using Fisher’s protected least significant difference (LSD).

**Results and Discussion**

Statistical analysis detected a significant treatment by run interaction; therefore, data for each experiment will be presented separately but will be discussed together.

**Results from Experiment 1 and 2**

In both experiment 1 and 2, for mature cogongrass, plant height, shoot dry matter (DW), rhizome DW, total DW and leaf area were all significantly reduced as light intensity decreased
(Table 3-1 and Table 3-3). Leaf weight ratio (LWR), was significantly lower at the 1% light intensity compared to the higher light regimes. Rhizome weight ratio (RWR) and rhizome to leaf ratio (RLR) were significantly higher at the 1% light intensity compared to the other light intensities.

Newly established cogongrass plant height, leaf area, shoot DW, rhizome DW and total DW significantly decreased in both experiments as light intensity decreased to ≤ 15% (Table 3-2 and Table 3-4). In experiment 1, the LWR of newly established plants was significantly lower at 75% full sunlight as compared to the lower light intensities. RWR and RLR were conversely significantly higher at 75% full sunlight than other light intensity levels. For all 3 ratios, there were no significant differences between light intensity levels ≤ 25%.

RLR was the principle difference between mature and young cogongrass growth and dry matter production, relative to light intensity. This is likely because mature plants possessed significant rhizome biomass before shade treatments while young plants did not. Mature plants had similar RLR even at 2% of full sunlight, which is close to cogongrass light compensation point. This indicates mature cogongrass was likely able to maintain an adequate RLR through sacrificing rhizome reserve to maintain shoot growth. At the 1% light intensity, the higher RLR suggests cogongrass sacrificed leaf production to maintain a viable rhizome system. This further suggests that mature cogongrass could become dormant when adverse conditions such as intense shading arise. Therefore, mature cogongrass showed the ability to adjust to different light intensities to either maintain its low rate growth by balancing the ratio between rhizome and shoot production, or tend to become dormant.

Young plants did not withstand shading as well as mature plants. When the light intensity declined to 25% of full sunlight, the decreased RLR suggests the plants sacrificed rhizome
production in favor of shoot production. Additionally, young plants, with immature rhizome systems, do not appear to have the ability to sacrifice leaf biomass as a means of conserving rhizome biomass for future growth.

LWR represents the investment of plant biomass into photosynthetic tissue and LAR, the product of SLA and LWR, represents the level of distribution of this tissue as light-harvesting structure in the form of leaf area (Patterson, 1980; Patterson et al, 1980; Patterson et al., 1979). As previously stated, LWR was decreased significantly under shading for mature plants and increased significantly for young plants (P<0.05). As light intensity decreased, mature cogongrass did not devote more carbon assimilation products into photosynthetic tissue, but rather kept a balance between photosynthetic and rhizome tissue. Young cogongrass did not have the same level of reserved rhizome biomass to support the whole plant; therefore, the increase in LWR indicates adaptation to low irradiance at the whole-plant level. For mature cogongrass, LAR was constant regardless of light intensity variation, indicating the ratio of leaves to total plant weight remained constant. Young cogongrass LAR increased significantly as light intensity decreased; therefore, the increase of leaf area compared to the plant total weight also implied adaption to low irradiance.

NAR did not vary with light intensity for mature cogongrass plants (Table 3-1 and Table3-3), but sharply declined in young cogongrass plants (Table 3-2 and Table 3-4). Mature plants were able to maintain the same rate of dry matter production per unit leaf area regardless of light intensity. This is probably due to the mature rhizome system that allows a larger proportion of photosynthate to be invested into shoot growth instead of further rhizome development. Conversely, young plants were attempting to develop shoots and rhizomes simultaneously.
Kw, SLA and LAD did not differ between young and mature plants (Table 3-1, Table 3-2, Table 3-3 and Table 3-4). Additionally, their trend with respect to light intensity was similar. Kw and LAD decreases significantly as light intensity decreased while SLA increased significantly as light intensity increased. Based on previous research by Patterson et al., (1979), our results indicated that cogongrass leaves were thinner under shading, but the distribution of leaf biomass as leaf area was significantly increased under low light intensity.

The parameters most related to leaf partitioning are Ka, Kl, LAP and LWP. LAP indicates the partitioning of total daily biomass accumulation into new leaf area and similarly, LWP indicates the partitioning of total daily biomass accumulation into new leaf weight. Ka is the actual leaf area expansion rate and Kl is the actual leaf weight growth rate. The partitioning of daily biomass accumulation into leaf area/weight will determine the area/weight growth of the next day, which will depend on the partitioning rate to a larger extent (Potter and Jones, 1977). The Ka and Kl of mature cogongrass both decreased significantly as light intensity decreased but LAP and LWP did not. As mentioned before, mature cogongrass assimilation rate remained constant throughout all light intensities, therefore, the proportion of carbon assimilates partitioning to leaf area expansion and leaf weight growth were constant. However, actual leaf weight growth and leaf expansion rate declined. Once again, this indicates that mature cogongrass has the ability to maintain regular growth patterns or self-impose dormancy under adverse conditions.

Young cogongrass Ka and Kl also decreased significantly as light intensity decreased (P<0.05) but LAP and LWP increased significantly from 75 to 2% full sunlight and only LWP decreased significantly from 2% to 1% full sunlight (P<0.05). Young cogongrass assimilation
rate decreased significantly throughout decreasing light intensities, therefore, although the partitioning of assimilate to leaves increased, the actual growth was still decreased.

The correlation of parameters \( K_a, K_l, LAP \) and \( LWP \) with relative growth rate (\( Kw \)) is shown in Figure 3.1 and Figure 3.2. In both experiments, young plants showed correlation between \( K_l \) and \( Kw \) as well as \( K_a \) and \( Kw \). It indicates for young plants, increases in growth were associated with increases in both relative leaf area and leaf biomass, but poorly with the partitioning coefficients, i.e. the actual leaf area and leaf weight increase instead of the partitioning proportion determines plant growth. Mature cogongrass growth correlation with leaf area expansion was less than young plants, but it was still well correlated with the increase in relative leaf biomass. It suggests for mature plants, leaf weight increase is more crucial for their growth instead of leaf area expansion.

**Overall Discussion**

Previous research on parameters related with growth rate and leaf partitioning rate showed a decrease with shading (Patterson, 1980; Patterson et al., 1979; Holly and Ervin, 2007). Our experiments with young cogongrass plants showed similar results to previous research. Decreases in growth, leaf partitioning and RLR indicates how young cogongrass adapts to decreasing light intensity. However, mature cogongrass, with a fully established rhizome system, has the ability to maintain balance between rhizomes and shoot growth. These data suggest that mature plants have the ability to conserve rhizome biomass when placed under intense shade.

These data do not support our hypothesis that lower light intensity will enhance more carbon assimilation partitioning to the shoot, increasing the shoot to rhizome ratio. Rather, the influence of shading to cogongrass growth and carbon partitioning is a function of maturity level. Our results indicated that cogongrass without an established rhizome system, shading from neighboring plants might provide a powerful competitive function and could help to explain why
cogongrass does not invade and displace heavily shaded ecosystems. Utilization and cultivation of competitive neighboring crops could produce a shading canopy, reducing plant vigor and consequently reducing cogongrass spread. Moreover, as shading promotes high relative aboveground biomass allocation, aboveground control practices might be more effective.
Table 3-1 Experiment 1: Growth of mature cogongrass plants as affected by light intensity for 12 weeks

<table>
<thead>
<tr>
<th>Parameter</th>
<th>Light Intensity % of full sunlight</th>
</tr>
</thead>
<tbody>
<tr>
<td></td>
<td>75%</td>
</tr>
<tr>
<td>Plant Height (cm)</td>
<td>86±3ab</td>
</tr>
<tr>
<td>Leaf Area (cm²)</td>
<td>1214±122a</td>
</tr>
<tr>
<td>Shoot DW (g)</td>
<td>9.87±1.09a</td>
</tr>
<tr>
<td>Rhizome DW (g)</td>
<td>41.54±4.13a</td>
</tr>
<tr>
<td>Total DW (g)</td>
<td>51.4±5.15a</td>
</tr>
<tr>
<td>Leaf Weight Ratio (LWR)</td>
<td>0.19±0.01a</td>
</tr>
<tr>
<td>Rhizome Weight Ratio (RWR)</td>
<td>0.81±0.01b</td>
</tr>
<tr>
<td>Rhizome: Leaf Ratio (RLR)</td>
<td>4.27±0.23b</td>
</tr>
<tr>
<td>Specific Leaf Area (dm²/g) (SLA)</td>
<td>1.24±0.04c</td>
</tr>
<tr>
<td>Leaf Area Ratio (dm²/g) (LAR)</td>
<td>0.24±0.01a</td>
</tr>
<tr>
<td>Leaf Area Duration (cm² days) (LAD)</td>
<td>15334±1332a</td>
</tr>
<tr>
<td>Relative Leaf Area Expansion Rate/day (Ka)</td>
<td>0.078±0.001a</td>
</tr>
<tr>
<td>Relative Growth Rate/day (Kw)</td>
<td>0.095±0.001a</td>
</tr>
<tr>
<td>Relative Leaf Growth Rate/day (Kl)</td>
<td>0.076±0.001a</td>
</tr>
<tr>
<td>Leaf Area Partition Coefficient (LAP)</td>
<td>0.197±0.009a</td>
</tr>
<tr>
<td>Leaf Weight Partition Coefficient (LWP)</td>
<td>0.154±0.007a</td>
</tr>
<tr>
<td>Net Assimilation Rate (g/dm² day) (NAR)</td>
<td>0.40±0.01a</td>
</tr>
</tbody>
</table>

*aAll data represents the mean of 6 replications followed by standard error. *bFor each parameter, values sharing the same letter within a row are not significantly different according to Fisher’s Protected LSD test at P≤ 0.05.
Table 3-2 Experiment 1: Growth of newly established cogongrass plants as affected by light intensity for 12 weeks

<table>
<thead>
<tr>
<th>Parameter</th>
<th>75%</th>
<th>25%</th>
<th>15%</th>
<th>2%</th>
<th>1%</th>
</tr>
</thead>
<tbody>
<tr>
<td>Plant Height (cm)</td>
<td>92±1a</td>
<td>93±3a</td>
<td>43±5b</td>
<td>52±3c</td>
<td>51±1c</td>
</tr>
<tr>
<td>Leaf Area (cm²)</td>
<td>2598±158a</td>
<td>1124±91b</td>
<td>365±70c</td>
<td>241±42cd</td>
<td>45±8d</td>
</tr>
<tr>
<td>Shoot DW (g)</td>
<td>19.20±1.47a</td>
<td>6.68±0.53b</td>
<td>1.71±0.34c</td>
<td>1.08±0.17c</td>
<td>0.31±0.07c</td>
</tr>
<tr>
<td>Rhizome DW (g)</td>
<td>35.49±4.59a</td>
<td>5.14±0.74b</td>
<td>0.93±0.28c</td>
<td>0.71±0.33c</td>
<td>0.27±0.12c</td>
</tr>
<tr>
<td>Total DW (g)</td>
<td>54.68±5.90a</td>
<td>11.81±1.21b</td>
<td>2.65±0.61c</td>
<td>1.80±0.47c</td>
<td>0.58±0.18c</td>
</tr>
<tr>
<td>Leaf Weight Ratio (LWR)</td>
<td>0.36±0.02b</td>
<td>0.58±0.03a</td>
<td>0.66±0.03a</td>
<td>0.67±0.06a</td>
<td>0.58±0.04a</td>
</tr>
<tr>
<td>Rhizome Weight Ratio (RWR)</td>
<td>0.64±0.02a</td>
<td>0.42±0.03b</td>
<td>0.34±0.03b</td>
<td>0.33±0.14b</td>
<td>0.42±0.04b</td>
</tr>
<tr>
<td>Rhizome: Leaf Ratio (RLR)</td>
<td>1.83±0.15a</td>
<td>0.75±0.08b</td>
<td>0.52±0.07b</td>
<td>0.57±0.17b</td>
<td>0.79±0.17b</td>
</tr>
<tr>
<td>Specific Leaf Area (dm²/g)(SLA)</td>
<td>1.37±0.07b</td>
<td>1.69±0.07b</td>
<td>2.13±0.18a</td>
<td>2.17±0.11a</td>
<td>1.57±0.14b</td>
</tr>
<tr>
<td>Leaf Area Ratio (dm²/g)(LAR)</td>
<td>0.50±0.05c</td>
<td>0.97±0.04b</td>
<td>1.43±0.18a</td>
<td>1.44±0.11a</td>
<td>0.12±0.09a</td>
</tr>
<tr>
<td>Leaf Area Duration (cm² days)(LAD)</td>
<td>29706±158a</td>
<td>14368±1021b</td>
<td>5490±897c</td>
<td>3907±582c</td>
<td>1037±156d</td>
</tr>
<tr>
<td>Relative Leaf Area Expansion Rate/day (K_a)</td>
<td>0.087±0.001a</td>
<td>0.078±0.001b</td>
<td>0.064±0.002c</td>
<td>0.060±0.003c</td>
<td>0.041±0.002d</td>
</tr>
<tr>
<td>Relative Growth Rate/day (Kw)</td>
<td>0.095±0.001a</td>
<td>0.078±0.001b</td>
<td>0.068±0.002c</td>
<td>0.056±0.002c</td>
<td>0.043±0.003d</td>
</tr>
<tr>
<td>Relative Leaf Growth Rate/day (K_l)</td>
<td>0.084±0.001a</td>
<td>0.072±0.001b</td>
<td>0.056±0.002c</td>
<td>0.051±0.002c</td>
<td>0.036±0.003d</td>
</tr>
<tr>
<td>Leaf Area Partition Coefficient (LAP)</td>
<td>0.458±0.050c</td>
<td>0.963±0.043b</td>
<td>1.534±0.236a</td>
<td>1.549±0.16a</td>
<td>0.896±0.110b</td>
</tr>
<tr>
<td>Leaf Weight Partition Coefficient (LWP)</td>
<td>0.316±0.019c</td>
<td>0.532±0.027ab</td>
<td>0.614±0.034a</td>
<td>0.619±0.059a</td>
<td>0.498±0.044b</td>
</tr>
<tr>
<td>Net Assimilation Rate (g/dm² day) (NAR)</td>
<td>0.203±0.023a</td>
<td>0.082±0.005b</td>
<td>0.046±0.005c</td>
<td>0.041±0.005c</td>
<td>0.052±0.011bc</td>
</tr>
</tbody>
</table>

"All data represents the mean of 6 replications followed by standard error. b) For each parameter, values sharing the same letter within a row are not significantly different according to Fisher’s Protected LSD test at P≤ 0.05."
Figure 3.1 Experiment 1: Relative growth rate (Kw) compared with: A) relative leaf expansion rate (Ka) B) relative leaf weight growth rate (Kl) C) leaf area partitioning coefficient (LAP) D) leaf weight partitioning coefficient (LWP). All data represents means of 6 replications.
Table 3-3 Experiment 2: Growth of mature cogongrass plants as affected by light intensity for 12 weeks

<table>
<thead>
<tr>
<th>Parameter</th>
<th>75%</th>
<th>25%</th>
<th>15%</th>
<th>2%</th>
<th>1%</th>
</tr>
</thead>
<tbody>
<tr>
<td>Plant Height (cm)</td>
<td>119±8ab</td>
<td>132±5a</td>
<td>119±7ab</td>
<td>104±5b</td>
<td>106±2b</td>
</tr>
<tr>
<td>Leaf Area (cm²)</td>
<td>7113±588a</td>
<td>4385±123b</td>
<td>2757±170bc</td>
<td>1522±239c</td>
<td>1437±239c</td>
</tr>
<tr>
<td>Shoot DW (g)</td>
<td>75.98±13.91a</td>
<td>38.33±10.92b</td>
<td>22.62±1.32b</td>
<td>13.06±1.87b</td>
<td>9.58±1.25b</td>
</tr>
<tr>
<td>Rhizome DW (g)</td>
<td>83.49±18.54a</td>
<td>53.32±7.43ab</td>
<td>54.35±1.26ab</td>
<td>38.24±3.60b</td>
<td>33.26±4.51b</td>
</tr>
<tr>
<td>Total DW (g)</td>
<td>159.47±31.99a</td>
<td>91.66±18.19ab</td>
<td>76.97±0.06ab</td>
<td>51.3±1.83b</td>
<td>42.8±5.10b</td>
</tr>
<tr>
<td>Leaf Weight Ratio (LWR)</td>
<td>0.48±0.03a</td>
<td>0.39±0.04ab</td>
<td>0.29±0.02ab</td>
<td>0.26±0.04c</td>
<td>0.23±0.03c</td>
</tr>
<tr>
<td>Rhizome Weight Ratio (RWR)</td>
<td>0.52±0.03c</td>
<td>0.60±0.04bc</td>
<td>0.71±0.02ab</td>
<td>0.74±0.05a</td>
<td>0.77±0.03a</td>
</tr>
<tr>
<td>Rhizome: Leaf Ratio (RLR)</td>
<td>1.09±0.11c</td>
<td>1.61±0.26bc</td>
<td>2.41±0.19ab</td>
<td>3.10±0.65a</td>
<td>3.55±0.52a</td>
</tr>
<tr>
<td>Specific Leaf Area (dm²/g) (SLA)</td>
<td>1.01±0.16b</td>
<td>1.43±0.02b</td>
<td>1.22±0.01ab</td>
<td>1.16±0.02b</td>
<td>1.48±0.07a</td>
</tr>
<tr>
<td>Leaf Area Ratio (dm²/g) (LAR)</td>
<td>0.48±0.07a</td>
<td>0.45±0.04ab</td>
<td>0.36±0.02ab</td>
<td>0.31±0.06b</td>
<td>0.34±0.04ab</td>
</tr>
<tr>
<td>Leaf Area Duration (cm² days) (LAD)</td>
<td>72104±5302a</td>
<td>46517±11744b</td>
<td>31309±1692bc</td>
<td>18640±2520c</td>
<td>17715±2531c</td>
</tr>
<tr>
<td>Relative Leaf Area Expansion Rate/day (Ka)</td>
<td>0.098±0.001ab</td>
<td>0.092±0.003ab</td>
<td>0.088±0.001bc</td>
<td>0.081±0.001cd</td>
<td>0.080±0.002d</td>
</tr>
<tr>
<td>Relative Growth Rate/day (Kw)</td>
<td>0.107±0.002a</td>
<td>0.101±0.002b</td>
<td>0.099±0.001b</td>
<td>0.095±0.001bc</td>
<td>0.093±0.001c</td>
</tr>
<tr>
<td>Relative Leaf Growth Rate/day (Kl)</td>
<td>0.099±0.002a</td>
<td>0.090±0.003b</td>
<td>0.086±0.001bc</td>
<td>0.079±0.001cd</td>
<td>0.076±0.001d</td>
</tr>
<tr>
<td>Leaf Area Partition Coefficient (LAP)</td>
<td>0.45±0.07a</td>
<td>0.41±0.05ab</td>
<td>0.32±0.02ab</td>
<td>0.26±0.06ab</td>
<td>0.29±0.004b</td>
</tr>
<tr>
<td>Leaf Weight Partition Coefficient (LWP)</td>
<td>0.45±0.02a</td>
<td>0.36±0.04ab</td>
<td>0.25±0.04bc</td>
<td>0.22±0.04c</td>
<td>0.19±0.02c</td>
</tr>
<tr>
<td>Net Assimilation Rate (g/dm² day) (NAR)</td>
<td>0.24±0.04a</td>
<td>0.23±0.02a</td>
<td>0.28±0.02a</td>
<td>0.34±0.06a</td>
<td>0.28±0.03a</td>
</tr>
</tbody>
</table>

All data represents the mean of 6 replications followed by standard error. For each parameter, values sharing the same letter in a row are not significantly different according to Fisher’s Protected LSD test at P ≤ 0.05.
Table 3-4 Experiment 2: Growth of newly established cogongrass plants as affected by light intensity for 12 weeks

<table>
<thead>
<tr>
<th>Parameter</th>
<th>Light Intensity % of full sunlight</th>
</tr>
</thead>
<tbody>
<tr>
<td></td>
<td>75%</td>
</tr>
<tr>
<td>Plant Height (cm)</td>
<td>124±5a</td>
</tr>
<tr>
<td>Leaf Area (cm²)</td>
<td>1447±39a</td>
</tr>
<tr>
<td>Shoot DW (g)</td>
<td>24.19±1.98a</td>
</tr>
<tr>
<td>Rhizome DW (g)</td>
<td>32.33±2.82a</td>
</tr>
<tr>
<td>Total DW (g)</td>
<td>56.52±4.63a</td>
</tr>
<tr>
<td>Leaf Weight Ratio (LWR)</td>
<td>0.43±0.01c</td>
</tr>
<tr>
<td>Rhizome Weight Ratio (RWR)</td>
<td>0.57±0.01a</td>
</tr>
<tr>
<td>Rhizome: Leaf Ratio (RLR)</td>
<td>1.34±0.06a</td>
</tr>
<tr>
<td>Specific Leaf Area (dm²/g)(SLA)</td>
<td>0.59±0.04d</td>
</tr>
<tr>
<td>Leaf Area Ratio (dm²/g)(LAR)</td>
<td>0.26±0.02d</td>
</tr>
<tr>
<td>Leaf Area Duration (cm² days)(LAD)</td>
<td>17824±1904a</td>
</tr>
<tr>
<td>Relative Leaf Area Expansion Rate/day (Ka)</td>
<td>0.081±0.001a</td>
</tr>
<tr>
<td>Relative Growth Rate/day (Kw)</td>
<td>0.096±0.001a</td>
</tr>
<tr>
<td>Relative Leaf Growth Rate/day (KI)</td>
<td>0.086±0.001a</td>
</tr>
<tr>
<td>Leaf Area Partition Coefficient (LAP)</td>
<td>0.22±0.02c</td>
</tr>
<tr>
<td>Leaf Weight Partition Coefficient (LWP)</td>
<td>0.38±0.01c</td>
</tr>
<tr>
<td>Net Assimilation Rate (g/dm² day)(NAR)</td>
<td>0.39±0.04a</td>
</tr>
</tbody>
</table>

*aAll data represents the mean of 6 replications followed by standard error. *bFor each parameter, values sharing the same letter in a row are not significantly different according to Fisher’s Protected LSD test at P ≤ 0.05.*
Figure 3-2 Relative growth rate (Kw) compared with: a) relative leaf expansion rate (Ka) b) relative leaf weight growth rate (Kl) c) leaf area partitioning coefficient (LAP) d) leaf weight partitioning coefficient (LWP). All data represents means of 6 replications from Experiment 2.
CHAPTER 4
ABSCISIC ACID CONTENT IN DORMANT COGONGRASS RHIZOMES

Introduction

Cogongrass (*Imperata cylindrica* (L.) Beauv.) is one of the most troublesome weedy species in the world. As a persistent invasive species, it possesses several survival strategies including an extensive rhizome system, adaptation to poor soils, drought tolerance, prolific wind-disseminated seed, fire adaptation, and high genetic plasticity (Hubbard et al., 1944; Holm et al., 1977; Brook, 1989; Dozier et al., 1998).

Among these strategies, the most important one is the persistent and aggressive rhizome system. Cogongrass can colonize new habitats by seed dispersal, but spread is mostly from vegetative rhizome growth (Tominaga, 2003). Growth from a single rhizome can extend 1 m per year and total rhizome growth from one plant can be more than 12 m per year (Tominaga, 2003). Research has shown this extensive rhizome network can exceed 40 tons of fresh weight per hectare (Terry et al., 1970). Rhizomes are mainly found in the top 15 cm of fine soils and top 40 cm of coarse soils (Holm et al., 1977; Gaffney, 1996). Cogongrass has a low shoot-to-rhizome ratio with rhizomes comprising over 60% of the plant total biomass (Holm et al., 1977).

This extensive rhizome system allows cogongrass to survive control procedures. The regeneration capacity of rhizomes has been shown to be positively correlated with increased age, weight, length and thickness (Ayeni, 1985). Rhizomes are highly resistant to breakage and disruption, and drought (Ayeni, 1985; English, 1998).

Apical dominance plays an important role in cogongrass rhizome growth. Apical dominance means the apical bud (or tip) of the rhizome produces auxin, which not only promotes cell division on the apical bud, but also diffuses basipetally and inhibits the development of lateral bud growth (Cline, 1994). These buds would otherwise compete with the apical tip for
light and nutrients. The persistent and large rhizome mass with numerous dormant buds provides a means of energy conservation and regeneration capacity. Apical dominance contributes to the ability of cogongrass to survive control methods, therefore, repeated treatments are needed for long term control.

Axillary buds are abundant in cogongrass, but often dormant. Therefore, better understanding of bud dormancy could lead to better manipulation of rhizome growth and the development of more effective control measures. Research done by Gaffney and Shilling (1995) confirmed apical dominance in cogongrass through auxin (indole-3-acetic acid (IAA))-imposed dormancy. They also showed that Napthalam, a chemical that inhibits polar auxin transport, can activate axillary buds. Extensive research performed on quackgrass (*Agropyron repens* (L.)), which is a rhizomatous, perennial grass, also reported IAA-induced apical dominance (Chancellor and Leakey, 1972).

English (1998) performed research on cogongrass rhizomes, and her results indicated that different parts of the rhizome performed differently in the same growing stage. Sprouting of apical and central buds was significantly higher than the basal buds. Therefore, further investigation into the viability and level of the dormancy of basal buds may provide useful information. English (1998) also mentioned that apex, scale leaves and root removal will influence axillary bud development.

Releasing rhizome dormancy could improve cogongrass control in different ways. Dormant buds are not susceptible to translocated herbicides because they are not active metabolic sinks. As the top growth of plants is killed by herbicides, axillary buds are often viable and able to sprout. Releasing rhizome dormancy will enhance bud sprouting at the expense of carbohydrates reserved in rhizomes. This will deplete the potential for regeneration and increase
the concentration of herbicide into axillary bud tissue. Moreover, it will increase the shoot to rhizome ratio and increase the target for foliar-applied herbicides.

As bud dormancy is enforced by apical dominance, it can be regarded as a “secondary dormancy”. Research has shown secondary dormancy is closely related to abscisic acid (ABA) concentration, especially in the case of seed dormancy and germination (Bewley, 1997). ABA is also thought to play a role in apical dominance. It has been postulated as a possible auxin-induced second messenger that directly represses axillary bud outgrowth (Tucker, 1978).

Research on quackgrass showed that ABA plays a principle role in bud dormancy where exogenously applied ABA inhibited sprouting in quackgrass buds (Taylor et al., 1995. Pearce et al., 1995). ABA applied to quackgrass rhizomes with the apex intact stimulated sprouting in axillary buds, but had no effect with the apex removed. Research by Cline and Oh (2006) showed that basally applied ABA could restore apical dominance in Ipomoea and Solanum suggesting the interaction among ABA and auxin. Research on purple nutsedge (Cyperus rotundus L.) and yellow nutsedge (Cyperus esculentus L.) showed that exogenous ABA inhibited nutsedge tuber sprouting and this might be a natural dormancy mechanism in nutsedge (Jangaard et al., 1971). In contrast, Kojima (1993) found asparagus (Asparagus officinalis) spears lateral buds initiated growth, ABA concentration in lateral buds plus scale leaves associated with the tip region was the highest among all tested tissue. In asparagus rhizomes, ABA concentration was also higher in younger regions where buds would readily grow.

Therefore, we suspect that the ABA concentration will be different between dormant and non-dormant buds of cogongrass, determine if this relationship between ABA level is positively or negatively correlated to dormancy.
In summary, the objective of this experiment is to determine the levels of abscisic acid in cogongrass rhizomes as a function of nodal position. We hypothesize that the secondary dormancy at rhizome nodes enforced by apical dominance is related to ABA content and that ABA content differs along the length of the rhizome.

**Materials and Methods**

Rhizomes from mature cogongrass plants were collected from a single population in Gainesville, FL. Rhizomes were transplanted in 4 L pots containing commercial potting mix\(^1\). Plants were placed under greenhouse conditions with temperatures of 25 °C day /20 °C night. Plants were watered and fertilized as needed.

After 6 months of growth, rhizomes extending from the bottom of four pots were randomly selected. These rhizomes were covered with scale leaves that had accumulated large amount of anthocyanins. Although extended from the pot and exposed to light conditions, these rhizomes remained dormant and did not form shoots. Two rhizomes > 20 cm length were detached from each pot and quick-frozen in liquid nitrogen. Rhizomes were then segmented into 4 sections each containing 7 nodes, from the tip basipetally. The actual separation between sections was made between two buds. For each section, nodal tissue and scale leaves were separated.

All tissue was placed into a freeze dryer\(^2\) for 3 days to achieve complete dryness and ground with a mortar and pestle using liquid nitrogen. After grinding, ABA was extracted from the tissues with 10 ml 100% methanol for 12 hours on a rotary shaker\(^3\) (150 rpm) at 4 C.

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\(^1\) Fafrad super-fine germination mix. Conrad Fafrad. Inc. P.O.Box 790. Agawam, MA 01001-0790

\(^2\) Microprocessor control corrosion resistant freeze-dryer, Kinetics Dura-Dry™ MP, FTS system

\(^3\) Gyrotiry® rotary shaker, Model No. G-2, New Brunswick Scientific Inc. Edison New Jersey
Methanol was evaporated at 0 °C and the residue resuspended in 1.0 ml tris-buffered saline⁴. All above stated steps were based on MacDonald (1994).

Analysis for ABA was performed using an enzyme-linked immunoassay specific for ABA while using (±) cis-trans ABA as a standard. Detailed steps and solution preparation, based on the immunoassay instruction manual, can be found in the Appendix.

The experiment was repeated twice and ANOVA was performed to test differences between experiments. Means were separated by using Fisher’s protected least significant difference (LSD).

**Results and Discussion**

Statistical analysis detected a significant interaction (P>0.05) between the two experimental runs, therefore, data from the two runs are presented separately.

![ABA concentration (pmol/g DW) in cogongrass rhizome and scale leaves at different sections. All data represents means of 8 replications followed by standard error. Section 1-4 represents sections from farther away from to being closer to parental plants.](image)

⁴ Turbo Vap® LV Evaporator, Zymark
Figure 4-2 Experiment 2: ABA concentration (pmol/g DW) in cogongrass rhizome and scale leaves at different sections. All data represents means of 8 replications followed by standard error. Section 1-4 represents sections from farther away from to being closer to parental plants.

Abscisic acid content (pmol/g DW) in cogongrass rhizome tissue and scale leaves at different nodal positions are shown in Figures 4-1 and 4-2, for Experiment 1 and 2 respectively. Section 1 refers to those nodes nearest to and including the tip, while position 2, 3, and 4 represent segments closest to farthest away from the tip, respectively. Although the actual ABA concentration from the two sets of experiments was significantly different (P<0.05), the relative trend of ABA concentration in the two experiments was very similar. Rhizome segments and scale leaves closest to the rhizome tip had the highest concentration of ABA and the level was significantly higher (P<0.05) than all the other parts of the same rhizome. For nodes and scale leaves in segments 2, 3 and 4, the ABA concentration levels were similar and not significantly different (P>0.05).

These results indicate a possible relationship between the level of ABA concentration and cogongrass rhizome dormancy. Buds closest to the apex are usually less dormant than basal buds.
(Taylor et al., 1995). Previous research on quackgrass (*Elytrigia repens*) also showed similar results (Taylor et al., 1995; Pearce et al., 1995). They concluded that rhizome tips and axillary buds closest to the tip had the highest level of ABA concentration. The actual ABA level present in rhizomes harvested at different experiment periods varied differently, which is similar to our experiment. However, the actual value of ABA content in cogongrass rhizomes and quackgrass rhizomes are quite different. Leaky (1975) suggested that the small amount of ABA (≤ 200 pg/mg DW) was not sufficient to account for bud inhibition, while Taylor et al. (1995) suggested it might be the balance of several growth hormones interacting together. Therefore, further research on cogongrass relative to this point is also necessary. The ABA concentration in scale leaves for cogongrass and quackgrass were also very different. Quackgrass scale leaves have about 20 times less ABA concentration in scale leaves compared to rhizome tissues (Taylor et al., 1995). Our results showed in each section of cogongrass rhizome, the ABA concentration in rhizome tissue and scale leaves were comparable. Previous research by English (1998) showed that cogongrass scale leaves were an inhibitory factor to bud growth and removal of scale leaves around the axillary buds of the rhizome lead to increased sprouting of the axillary buds. The comparable level of ABA concentration in scale leaves suggests the possibility of ABA functioning as a dormancy-enhancing role of scale leaves.
CHAPTER 5
CONCLUSIONS

These three projects were focused on studying cogongrass growth responses. As a strong competitor, cogongrass competitiveness against bahiagrass was evaluated at two different soil pH conditions. At soil pH 4.5, cogongrass ramets were more competitive than bahiagrass seedlings. Cogongrass was able to thrive on its own at low pH conditions while bahiagrass was slightly affected by cogongrass. At a soil pH of 6.8, bahiagrass seedlings showed significantly greater competitiveness against cogongrass ramets. Therefore, as soil pH shifted from low to high, bahiagrass seedling competitivenss was increased while cogongrass competitiveness was decreased. At optimal soil pH for forages, bahiagrass seedlings are strong competitors against cogongrass. As the soil pH decreases, bahiagrass weakens, while cogongrass has a strong adaptation ability to thrive in low pH conditions. Therefore, cogongrass becomes the more competitive species when the soil pH decreased. Our results indicate that decreases in soil pH, often associated with poor soil fertility might be the reason for cogongrass invasion into bahiagrass pastures.

Under low light intensities, mature cogongrass plants with a fully developed rhizome system and young cogongrass without strong rhizome system responded differently. As light intensity decreased to below the cogongrass light compensation point, mature plants went from full growth to maintenance growth to dormancy. It indicates that at low light intensity conditions, mature cogongrass plants sacrificed rhizome tissue for shoot production to maintain a low level of growth. As light intensity decreases below the light compensation point, cogongrass stopped shoot production, but maintained rhizome biomass. In mature plants, the rhizome to shoot ratio increased as light intensity decreased. Young plants sacrificed rhizome production to produce
more shoots to maintain growth rate, with a marked decline in rhizome to shoot ratio as light intensity decreased.

Based on previously suggested auxin imposed apical dominance theory, our results showed that abscisic acid might also play a role in cogongrass rhizome dormancy. Nodes nearest to the rhizome tips have significantly higher amount of abscisic acid content than all other positions along rhizomes. Nodal tissue and associated scale leaves showed comparable amounts of abscisic acid content. The content level decreased in the same trend as compared to positions without rhizome tips. Further research is still needed to confirm the actual function of abscisic acid in cogongrass rhizome dormancy.
APPENDIX
PHYTODETEK ABSCISIC ACID TEST KIT EXPERIMENT PROTOCOL

Catalog number: PDK 09347/0096
Competitive ELISA, for the quantitative determination of Abscisic Acid

**Procedures**

Prepare tracer solution:

Add 1 mL distilled water to each vial of lyophilized ABA-tracer. Wait 5 minutes to allow for complete reconstitution.
Add 4 mL of tracer diluent to each ABA-tracer vial and mix well to insure proper ABA-tracer dilution.

Prepare standards:

Weigh 26.43 mg of 2-cis-(S)-ABA and dissolve in 10.0 mL of absolute methanol. If an enantiomeric ABA compound is used, weigh 52.86 mg of the compound. Add 100 μL of this solution to 9.90 mL of Absolute methanol. This makes a stock solution (SS) with a concentration of 0.1 μmole ABA/mL. Store this stock solution in an amber bottle, in the dark at -20° C or lower.
Following the chart below, prepare standards by diluting the stock solution in TBS buffer (buffer formulation on page 7). New standards should be prepared each time the test is run.
Note: SS = 0.1 μmole ABA/ml stock solution = 100,000 picomoles/ml.

NSB=Nonspecific Binding, Bo=100% Binding

<table>
<thead>
<tr>
<th>Cup</th>
<th>ABA Solution</th>
<th>TBS Buffer</th>
<th>Picomoles (ABA/mL)</th>
<th>Dilution</th>
</tr>
</thead>
<tbody>
<tr>
<td>A1= NSB</td>
<td>50μL of SS</td>
<td>4.95 mL</td>
<td>1000</td>
<td>1:100</td>
</tr>
<tr>
<td>B1</td>
<td>200 μL of A1</td>
<td>1.80 mL</td>
<td>100</td>
<td>1:10</td>
</tr>
<tr>
<td>C1</td>
<td>500 μL of B1</td>
<td>2.00 mL</td>
<td>20</td>
<td>1:5</td>
</tr>
<tr>
<td>D1</td>
<td>500 μL of C1</td>
<td>2.00 mL</td>
<td>4</td>
<td>1:5</td>
</tr>
<tr>
<td>E1</td>
<td>500 μL of D1</td>
<td>2.00 mL</td>
<td>0.8</td>
<td>1:5</td>
</tr>
<tr>
<td>F1</td>
<td>500 μL of E1</td>
<td>2.00 mL</td>
<td>0.16</td>
<td>1:5</td>
</tr>
<tr>
<td>G1</td>
<td>500 μL of F1</td>
<td>2.00 mL</td>
<td>0.032</td>
<td>1:5</td>
</tr>
<tr>
<td>H1=Bo</td>
<td>100 μL of TBS</td>
<td></td>
<td></td>
<td></td>
</tr>
</tbody>
</table>

The sensitivity is optimum between 0.0064 and 0.16 picomoles ABA/mL.
Remove desired number of test wells from the pouch and place in test well holder. Seal pouch and return to freezer.

Add 100 μL of standard or sample extract to each well. Standards and samples should be run in duplicate.

Add 100 μL diluted Tracer prepared in Step 1 to each well using a multichannel pipette.

Mix by gently tapping plate. Cover test wells with Plate sealer or place in a humid box (airtight plastic box lined with damp paper towel).

Incubate test wells in refrigerator at 4° C for 3 hours.

Prior to the end of the incubation period prepare Substrate solution: Dissolve 1 Substrate tablet in 5 mL Substrate diluent. One tablet is sufficient to perform 16 test wells.

After the 3 hour incubation, remove the test wells from the refrigerator and dump contents of the test wells into the sink.

Wash test wells by adding 200 μL of the Wash solution to each well with a multichannel pipette. Dump contents of the test wells into the sink. Repeat this step 2 more times. Then, grasping the test well holder upside down, firmly tap on paper towel to shake out remaining drops of Wash solution.

Add 200 μL of substrate solution to each well using a multichannel pipette.

Cover test wells with Plate sealer or place in a humid box.

Incubate at 37° C for 60 minutes. Test is not valid unless Bo reads greater than 0.750 O.D. If the value is below this, increase the Substrate incubation time until the desired O.D. is obtained (not to exceed 30 additional minutes).

Remove test well from incubator and add 50 μL (1 drop) Stop reagent to each well. Wait 5 minutes.

Read color absorbance at 405 nm. Record optical densities.

Calculations

Calculate the means of the optical densities of duplicate standards or samples.

Calculate the % Binding of each standard point or sample by the following:

% Binding = (Standard or Sample O.D. - NSB O.D.) / (Bo O.D. - NSB O.D.) x 100

Note: B = STD or Sample O.D.

NSB = 100 μL of A1 (100 picomoles ABA/mL) + 100 μL Tracer = 0% Binding.

Bo= 100 μL of H1 (TBS buffer) + 100 μL Tracer = 100% Binding.
Plot the % Binding (B/Bo) versus the ABA concentration (picomoles/mL) and draw the best fit curve on 4-cycle semi-log paper (sigmoid curve).

Determine ABA concentration by interpolation of the sample % Binding from the standard curve.

Linear standard curves can also be drawn using a Log-Logit transformation of the data as follows:

\[ \text{Logit (B/Bo)} = \ln \left( \frac{B/Bo}{100 - (B/Bo)} \right) \]
Buffer formulations

Stop Reagent

Dissolve in 800 mL distilled water:
Sodium hydroxide 40.0 g
Adjust volume to 1 L. Store at room temperature.

Substrate Diluent

Dissolve in 800 mL distilled water:
Magnesium chloride 0.1 g
Sodium azide 0.2 g
Diethanolamine 97.0 mL
Adjust pH to 9.8 with hydrochloric acid. Adjust final volume to 1 L with distilled water. Store at 4° C.

TBS Buffer / Tracer Diluent

Dissolve in 800 mL distilled water:
Trizma base 3.03 g
Sodium chloride 5.84 g
Magnesium chloride hexahydrate 0.20 g
Sodium azide 0.20 g
Adjust pH to 7.5. Adjust volume to 1 L. Store at 4° C.

Wash Solution

Dissolve in 1000 mL distilled water:
Sodium chloride 8.00 g
Sodium phosphate, dibasic (anhydrous) 1.15 g
Potassium phosphate, monobasic (anhydrous) 0.20 g
Potassium chloride 0.20 g
Tween-20 0.50 g
Sodium azide 0.20 g
Adjust pH to 7.4
LIST OF REFERENCES


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BIOGRAPHICAL SKETCH

Jingjing was born in 1984 in a small city called Suzhou in China. She grew up and went to school there until she went to college in Nanjing, China. She received her Bachelor of Science in plant biotechnology from Nanjing Agriculture University. Afterwards, she decided to travel to the US to pursue further study in agriculture. She came to University of Florida in August 2006 starting her master’s research in the Department of Agronomy specializing in weed science. Her research focused on biological characteristics of the invasive weed cogongrass. She is expected to receive a Master of Science in agronomy in December 2008. Jingjing plans to attend business school at the University of Florida in pursuit of another Master of Science degree in management. Her future plan is to use what she learnt from both degrees and find a position in agricultural business in China.