Chapter 3

Chemical Control of Peanut Diseases: Targeting Leaves, Stems, Roots, and Pods with Foliar-Applied Fungicides

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Additional information is available at the end of the chapter

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1. Introduction

Peanut (*Arachis hypogaea* L.) is an important food crop with high levels of proteins, carbohydrates, vitamins and minerals contained within seeds (Moss and Rao, 1995). While the cultivation of peanut may occur over a wide range of climatic conditions, close attention should be paid to soil type, temperature range, and rainfall amount and distribution. Well-drained, sandy soils are best suited for peanut production (Beasley et al., 1997). Williams and Boote (1995), found the optimal temperature range for peanut production to be between 27 and 33°C. Furthermore, peanut plants require large amounts of rainfall, 50-75 cm, during production to optimize growth, yield and seed maturity (Beasley et al., 1997). If ample water and optimum temperatures are available after planting, peanut plants will emerge within 2 weeks of planting. These plants form self-pollinating flowers approximately 30-40 days after emergence and may continue to produce new flowers throughout the growing season until harvest. Fertilized flowers will form pointed needle-like carpophores (commonly referred to as "pegs"), that grow geotropically. The tissue at the tip of the peg becomes lignified, thus protecting the fertilized ovaries located behind the tip. Pegs grow into the soil to a depth of 2-7 cm (Porter, 1997). Peanut pod growth is initiated as the tip of the peg becomes horizontally oriented. The mature pods are oblong and may contain as many as five seeds. There are four market types of peanuts: runner, spanish, valencia and virginia. Runner and virginia types are most commonly grown throughout production regions in the United States; however, spanish and valencia market types are grown in the Southwest. The aforementioned market types differ in growth habit, days to maturity, yield potential, as well as susceptibility to diseases.

The worlds leading peanut producing countries include India, China and the United States. In 2011, approximately 444,500 hectares of peanuts were harvested in the United States

(NASS, 2011); with largest production region being the southeastern states of Alabama, Florida, Georgia, and South Carolina. Production is concentrated in this region due to the semi-tropical temperate climate conditions. Unfortunately, these environmental conditions are conducive for many pests, including weeds, insects and diseases. Other production regions in the United States include the southwestern region (New Mexico, Oklahoma, and Texas), as well as the Virginia-Carolina region (North Carolina and Virginia), each of which has their own disease issues.

1.1. Peanut leaf spot

Several fungal diseases are known to affect peanut leaves. Most notibly are early and late leaf spot, caused by *Cercospora arachidicola* (Hori), (Teleomorph: *Mycosphaerella arachidis* Deighton), and *Cercosporidium personatum* (Berk. & Curt.) Deighton, (Teleomorph: *Mycosphaerella berkeleyi* Jenk.), respectively. Either disease may be present within a given area or year. While both pathogens are destructive on leaves, they are also capable of causing lesions on petioles, pegs, main stems and lateral branches (Shokes and Culbreath, 1997). Leaf spot symptoms are initially seen as small necrotic flecks that appear approximately 10 days after spore deposition. Over several weeks, the lesions will enlarge from 1-10 mm in diameter and sporulate. The physical appearance of the two diseases is similar (**Fig. 1**); however, early leaf spot can be distinguished from late leaf spot based on lesion characteristics; the most noteworthy is the color of the lesion on the adaxial surface. Light to dark brown lesions are characteristic of *C. arachidicola*; while *C. personatum* lesions have more of a black appearance (Smith and Littrell, 1980; Sholar et al., 1993; Shokes and Culbreath, 1997). The orientation of sporulation may also be used in distinguishing between the two diseases. *Cercospora arachidicola* sporulates on the adaxial leaf surface; whereas *C. personatum* sporulates on the abaxial surface of the leaf. Microscopic examination of conidia may be required to further differentiate the two pathogens. Conidiophores of *C. arachidicola* are dark at the base, unbranched, and septate; giving rise to curved, subhyaline, septate conidia (15-45 × 3-6 μm). Conidia (20-70 × 4-9 μm) of *C. personatum* are typically straight, rounded at the apex and not constricted, and are produced on smooth, brown conidiophores (Shokes and Culbreath, 1997).

Optimal environmental conditions for infection and reproduction for the two pathogens are quite similar; 16-24 °C and 20-26 °C for *C. arachidicola* and *C. personatum*, respectively, and both require long periods of relative humidity greater than 90% (Shokes and Culbreath, 1997). Primary inoculum for either pathogen originates from infected residue in the soil from previous peanut crops (Shokes and Culbreath, 1997). Both *C. arachidicola* and *C. personatum* overwinter as dormant stromata on infected residue until environmental conditions are conducive for sporulation and dispersal. Initial inoculum is responsible for the onset of leaf spot epidemics, and subsequent sporulation increases the disease. If left unmanaged, yield reductions as great as 70% may be incurred (Nutter and Shokes, 1995; Shokes and Culbreath, 1997).

Several other diseases including pepper spot, caused by *Leptosphaerulina crassiasca* (Sechet) C.R. Jackson & D.K. Bell, web blotch, caused by and peanut rust, caused by *Puccinia arachidis*

Speg., are also capable of infecting peanut foliage with the latter causing substantial losses throughout many production areas around the world. Peanut rust occurs sporadically in the southeastern United States and is generally considered a late season disease. Widespread use of chlorothalonil for managment of leaf spot is believed to have kept problems with rust to a minimum (Hagan, 1998).

Figure 1. Early and late leafspot of peanut.

1.2. Diseases of peanut stems, roots, pegs and pods

Numerous other fungal diseases are known to affect peanut stems, roots, pegs and pods. Diseases such as stem rot (Fig. 2), Rhizoctonia limb and pod rot (Fig. 3), Pythium pod rot (Fig. 4), Cylindrocladium black (Fig. 5) rot and Sclerotinia blight (Fig. 6) are among the most difficult to manage.

Stem rot, caused by the soilborne fungus *Sclerotium rolfsii* Sacc., is a very destrucive disease. *Sclerotium rolfsii* has a worldwide distribution and is capable of infecting a wide variety of row crops including crucifers, grasses and legumes (Aycock, 1966; Punja, 1985). Although the sexual stage of *S. rolfsii*, the basidiomycete *Athelia rolfsii* (Cruz) Tu & Kimbrough, has been identified, it is rarely seen under field conditions (Backman and Brenneman, 1997). *Sclerotium rolfsii* does not produce conidia and is classified as a Deuteromycete in the group

'Mycelia Sterilia' (Alexopolous et al., 1992). The fungus overwinters in the soil as hard, round, brown sclerotia (Backman and Brenneman, 1997). Mature sclerotia have a melanized outer layer, the rind, which allows the fungus to survive periods of adverse environmental conditions and remain viable for up to 3 years (Punja, 1985).

Upon germination of sclerotia, *S. rolfsii* may survive saprophytically as mycelium in organic matter in the soil or directly infect a susceptible host plant (Aycock, 1966). After an infection site is established, the fungus becomes necrotrophic, meaning an external energy source is needed to breach host defenses (Punja, 1985). Initial symptoms of infection include chlorosis and/or wilting of a lateral branch; however, if main stems become infected, the entire plant may appear wilted or chlorotic (Backman and Brenneman, 1997). Infected leaves typically have a water-soaked or necrotic appearance. Symptoms may appear rather quickly if temperatures are favorable. In very young pods the rot is clear and watery. As the pods mature, the damage on the pod is white or brownish in color and white mycelia is present. Sclerotia may be found in the vicinity of the plant stem or pods as well. The incubation period typically ranges from 2 to 4 days; however, wounding of plants may decrease the time required (Aycock, 1966).

Figure 2. Stem rot of peanut, caused by *Sclerotium rolfsii*.

Rhizoctonia solani (Kühn) anastomosis group 4 (AG-4) is capable of causing seed decay, pre- and post-emergence damping-off, as well as hypocotyl and root rot; however, it is most

devastating on mature plants causing a rot of pegs, pods, and stems. Although variable from year to year, Rhizoctonia limb rot is considered a major disease of peanut in the southeast (Brenneman, 1997; Thompson, 1982) and Texas (*personal observation)*. Substantial losses due to limb and pod rot can be experienced (Kemerait, 2003). Limb rot is more severe during cool wet periods and may be exacerbated by excessive nitrogen fertility (*Brenneman, unpublished*). Generally limb rot symptoms are first observed on lower branches that are in contact with the soil surface. Circular lesions, yellow to dark brown in color, occur at infection sites and have distinct target spot appearance. As lesion development progresses, infected limbs become girdled and die (Franke, 1999). The fungus may produce irregularly shaped sclerotia within host tissue as nutrient sources become depleted (Brenneman, 1997). Hypae of *R. solani* are typically white to brown in color, 4-15 μm thick, septate and branched at right angles (Taber and Pettit, 1970). During infection, hyphae quickly invade the epidermis and advance intracellularly (Christou, 1962). Studies conducted by Bateman (1970) suggest that *R. solani* produces various phytotoxins and degradative enzymes to kill host tissue, resulting in the release of nutrients that promote fungal growth. The disease is somewhat sporadic in nature (Thompson, 1982; Barnes et al., 1990), and cannot easily be assessed until after digging. *Rhizoctonia* spp. are comonly found associated with peanut pods that are left in the soil, and may also actively colonize and rot developing pods. Symptoms of Rizoctonia pod rot consist of a dry-rot, where the reticulations of the pods are exposed, having a skeletonized appearance. Cream to brown colored mycelia may be observed on diseased kerenls.

In addition, Pythium pod rot can also be responsible for considerable losses and frequently occurs throughout Texas, Oklahoma and North Carolina. A severe pod rot in Nicaragua was also recently determined to be caused by *Pythium myriotylum (*Augusto et al., 2010, II) Several *Pythium* spp., including *P. myriotylum*, *P. ultimum*, *P. irregulare*, *P. vexam*, and *P. dimorphum*, have been found to incite pod rot (Wheeler et al., 2005). Frank, 1972 reported that pod rot in Israel results from synergistic interactions between *P. myriotylum* and *Fusarium solani*. Pythium pod rot can be characterized by the appearance of wet, greasy pods which often exhibit a very unpleasant odor. White mycelial growth may be observed on decaying pods. Depending on the stage of infection and species involved, the examination of infected tissue may reveal the presence of spherical oospores. Due to the nature of peanut pod rot and similarity of symptoms laboratory diagnosis is often required to differentiate the causal agents

Cylindrocladium black root rot, caused by the fungus *Cylindrocladium parasiticum*, is a disease of economic importance in Georgia, Florida, Alabama, North and South Carolina, and Virginia (Phipps, 1990). The disease is of particular concern in Virginia where 20% of peanut fields are thought to be infested. Under favorable conditions, overwintering microsclerotia of the pathogen germinate and infect roots, causing decay (Fig. 5). Dark, red perithecia of the fungus are produced on the stems of infected plants; however, the sexual stage of the fungus does not appear to play a role in the disease cycle. Various studies have proven that *C. parasiticum* can be seedborne (Glenn et al., 2003). This mechanism is thought to have played a role in the spread of the pathogen from Georgia where it was first reported

in the 1960's (Bell and Sobers, 1965). Recent studies suggest that populations in Georgia are mainly clonal and genetically homogeneous (Wright, et al., 2010)

Figure 3. Peanut pod rot, caused by *Rhizoctonia solani*.

Sclerotinia blight, caused primarily by *Sclerotinia minor* Jagger and to a lesser extent *S. sclerotiorum*, is a destructive and economically important disease throughout areas of North Carolina and Virginia (Porter and Beute, 1974), Oklahoma (Sturgeon, 1982) and Texas (Wadsworth, 1979). Under favorable conditions, sclerotia of the pathogen eruptively germinate at the soil surface and initiate direct infections, with the resulting yield loss ranging from 10 to 50% (Comp). Symptoms consist of wilting and yellowing of the lateral branches. Dense mats of white mycelia develop on diseased areas, and small water-soaked lesions may be apparent near the soil line. Lesions become bleached due to the production of oxalic acid and have a distinct shredded appearance (Woodward et al., 2006). Small, black, angular sclerotia are produced on and within infected tissues. Infected peanut seed and crop debris may serve as initial inoculum (Woodward et al., 2006). Porter et al. (1989) found that disease incidence was correlated to discoloration, indicating that infestations of seed lots were restricted to mycelial infections on the seed testa. The sclerotia are easily capable of surviving 3-4 year crop rotations with non-hosts, and are spread primarily by soil movement through equipment or farming operations.

Figure 4. Peanut pod rot, caused by *Pythium myriotylium*.

Figure 5. Peanut plants with perithecia of *Cylindrocladium parasiticum*.

Figure 6. Sclerotinia blight of peanut, caused by *Sclerotinia minor*.

2. Chemical management of peanut diseases

Numerous chemical fungicides are available for control of the aforementioned diseases of peanut (Table 1). Applications of these products are made for the management of both foliar and soilborne diseases. These products have traditionally been the second largest variable expense in peanut production, behind seed cost. In the United States, management tactics vary among production regions; however, multiple applications of fungicides are typically required to minimize disease-associated losses within a given growing season (Melouk and Backman, 1995; Shokes and Culbreath, 1997). In the southeastern United States, applications of fungicides are typically made on calendar-based schedule; with initial applications beginning approximately 30 days after planting (DAP) and subsequent applications made on 14-day intervals. Due to the long growing season and high disease pressure in this region, a total of six to eight applications may be warranted. Whereas, two-to-three applications may be made in more arid production regions, such as west Texas. In the Virginia/Carolina region, several weather-based spray advisories have been developed and are currently being used to properly time applications (Phipps et al., 1997).

2.1. Management of peanut leaf spot with fungicides

Copper and sulfur dusts, were among the first fungicides used in peanut production for management of foliar diseases (Smith and Littrell, 1980). Most inorganic copper and sulfur

compounds are relatively insoluble, thus, preventative applications create a protectant barrier on leaf surfaces. Small quantities are absorbed by fungal spores, and accumulations result in their lethal effect. Dust formulations are no longer being utilized due to high usage rates, poor plant coverage, and the potential contamination of non-target locations (Backman et al., 1975; Backman, 1978). Some of the early liquid fungicides, such as benomyl and chlorothalonil were or are used to manage *C. arachidicola* and *C. personatum*. Benomyl was very effective at controlling leaf spot (Porter, 1970); however, widespread resistance to benomyl occurred in both *C. arachidicola* and *C. personatum* shortly after use began (Smith and Litrell, 1980).

Chlorothalonil, a broad-spectrum fungicide, is among the most effective fungicides registered for leaf spot control and has been the standard fungicide for leaf spot management since the 1970s (Smith and Litrell, 1980; Culbreath et al., 1992). Unfortunately, chlorothalonil is not active against *S. rolfsii* or *R. solani*, thus other fungicide chemistries are required. The registrations of tebuconazole and azoxystrobin in 1994 and 1997, respectively, greatly expanded fungicide options for peanut since they have excellent efficacy on both foliar and soilborne diseases. Other fungicides, primarily triazoles, strobilurins and carboximides, have been subsequently registered which provide peanut growers numerous options for broad spectrum disease management (Table 1). Although these new fungicides are generally quite active against both foliar and soilborne diseases, they have site specific modes of action, and therefore pose a significant risk for resistance development (Bertrand and Padgett, 1997). Therefore these products have been used as spray blocks or as tank mixes in combination with other chemistries in accordance with FRAC guidelines (www.frac.info). Field trials to evaluate the effects of ergosterol biosynthesis inhibiting fungicides in combination with chlorothalonil demonstrated that using reduced rates of chlorothalonil tank mixed with either propiconazole or cyproconazole improved the control of leaf spot over that of a full rate of chlorothalonil alone (Culbreath et al., 1992; Culbreath et al., 1995). However, tank-mix combinations of fungicides may result in added cost. Culbreath et al. (2001) evaluated the efficacy of various alternations and combinations of chlorothalonil and benomyl for managing benomyl-resistant *C. arachidicola* and *C. personatum* populations. Results of that study showed that full-season tank mixes of the compounds provided leaf spot control comparable to the standard chlorothalonil program, suggesting that tank-mixing is a valid resistance management tool where fungicide resistance is already a problem.

Brenneman and Culbreath (1994) studied various application schedules of chlorothalonil and tebuconazole for leaf spot and stem rot.. They evaluated different application schedules and found that a block of four applications of tebuconazole beginning at the third spray, reduced the severity of both foliar and soilborne diseases, and increased pod yields and kernel quality when compared to the full-season chlorothalonil program. Similar trends were observed when less than four tebuconazole applications were made (Brenneman and Culbreath, 1994). Recommendations in eastern production regions call for chlorothalonil to be added to tebuconazole due to the development of tebuconazole insensitive populations of *C. arachidicola* and *C. personatum* (Stevenson and Culbreath, 2006). It is interesting to note that later generation triazoles such as prothiconazole still maintain field control of leaf spot populations resistant to tebuconazole (Culbreath et al 2008).

Mode of action	Target site and FRAC codes[1]	Group name	Common name	Trade name(s)	Mobility
Nucleic acid synthesis	A1 (4)	Phenylamide	mefenoxam or metalaxyl	Ridomil Gold EC, Ridomil Gold GR, Ridomil Gold SL	locally systemic
Mitosis and cell division	B1 (1)	Benzimidazole	thiophanate-methyl	Topsin M	locally systemic
Respiration	C2 (7)	Carboxamide	penthiopyrad	Fontelis	locally systemic
			boscalid	Endura	systemic
			flutolanil	Artisan (+ propiconazole), Convoy, Moncut	systemic
	C3 (11)	Strobilurin - Quinone outside inhibitor (QoI)	azoxystrobin	Abound	locally systemic
			fluoxastrobin	Evito	locally systemic
			pyraclostrobin	Headline	locally systemic
			trifloxystrobin	Absolute (+tebuconazole), Stratego (+propiconazole)	locally systemic
	C5 (29)	Dinitroanaline	fluazinam	Omega	protectant
Lipids and membranes	F1 (2)	Dicarboximide	iprodione	Rovral	locally systemic
	F3 (14)	Aromatic hydrocarbon	dichloran	Botran	protectant
			PCNB	PCNB	protectant
Sterol synthesis	G1 (3)	Demethylation inhibitor - DMI	cyproconazole	Alto	systemic
			metconazole	Quash	locally systemic
			propiconazole	Tilt, Propiconazole, Propimax, Artisan (+ flutolanil), Stratego (+ trifloxystrobin)	locally systemic
			prothioconazole	Proline, Provost (+ tebuconazole)	systemic
			tebuconazole	Folicur, Muscle, Orius, Tebuzole, Trisum, Absolute (+ trifloxystrobin)	locally systemic
Multi-site activity	M1 (M1)	Inorganic	copper salts	Kocide, Copper-Count-N	protectant
	M2 (M2)		sulfur	*numerous*[2]	protectant
	M3 (M3)	Dithiocarbamate	mancozeb	Mancozeb	protectant

Mode of action	Target site and FRAC codes[1]	Group name	Common name	Trade name(s)	Mobility
			maneb	Maneb	protectant
	M4 (M4)	Phthalimide	captan	Captan	protectant
	M5 (M5)	Chloronitrile	chlorothalonil	Bravo, Equus, Echo	protectant
	M7 (M7)	Guanadine	dodine	Elast	protectant
Unknown	unknown (33)	Phosphonate	phosphorous acid	Phostrol, AgriFos	systemic
			potassium phosphite	Fosphite, Prophyt	systemic
	n/a	n/a	Chlorpyrifos[3]	Lorsban	n/a

Table 1. Peanut fungicides registered in the United States grouped by mode of action

2.2. Management of diseases caused by soilborne pathogens with fungicides

Peanut producers have more options now than ever when it comes to fungicides. While many of the products currently on the market have activity against diseases caused by both foliar and soilborne pathogens, flutolanil was registered in 1995 and is only active against *S. rolfsii* and *R. solani*. Therefore it must be used in combination with products with leaf spot activity (Hagan et al., 2004). However, to effectively use any fungicide for management of soilborne pathogens, the technical difficulties of getting the fungicide to the lower stem and around the pegs and pods must be considered. The most active fungicides will fail to control soilborne diseases if they cannot be placed appropriately. Pentachloronitrobenzene (PCNB), an organochlorine fungicide, was the first fungicide used extensively against stem rot; however, high costs and inconsistent field results limited producer usage (Csinos, 1989). This fungicide was applied as a granule, the logic being that granules were needed to filter down through the canopy to the soil surface for control of soilborne diseases (Csinos, 1989).

This same strategy was applied to newer fungicides, such as the ergosterol biosynthesis inhibitors as they were evaluated in peanut. Granular formulations of diniconazole and tebuconazole were examined, but results were inconsistent (Csinos, 1987). Suppression of diseases caused by soilborne pathogens was observed when liquid formulations of these compounds were applied to foliage in leaf spot studies (Backman and Crawford, 1985; Csinos et al., 1987; Brenneman and Culbreath, 1994; Besler et al., 2003). By mixing dyes with the foliar-applied fungicides and applying irrigation, Csinos (1988) documented how these materials were delivered to the soil. He demonstrated that the architecture of the peanut plant served to funnel rain or irrigation water along the stems and increase deposition of fungicides at the plant crown and pegs. This redistribution is important since these structures serve as primary infection courts for several pathogens (Melouk and Backman, 1995).

Various factors are known to affect fungicide deposition and efficacy. Differences in the leaf cuticle can influence the retention of fungicides (Neely, 1970; Neely, 1971), and changes in the composition of the cuticle have been attributed to different environmental factors (Skoss,

1955). Pesticide deposition is also greatly affected by canopy density. Researchers have found that higher levels of chlorothalonil are deposited on the upper plant canopy, compared to the lower canopy (Brenneman et al., 1990; Hamm and Clough, 1999). Zhu et al. (2004) demonstrated that spray deposits in the upper and lower peanut canopy differed significantly, and deposits in the lower canopy decreased as plants aged. The deposition and retention of chlorothalonil may differ within the peanut canopy layer and volume of water used for application (Brenneman et al., 1990). O'leary et al. (1997) found that both formulation and application method of flutolanil resulted in significant increases in chemical residues on subterranean plant parts and the lower canopy, respectively, characteristics that impacted managment of stem rot.

2.3. Improving fungicide deposition and efficacy via application method

Thorough coverage of foliage or the ability of fungicides to reach target organisms is essential in maximizing disease control. Environmental conditions such as relative humidity, wind speed, temperature and rainfall can greatly affect fungicide deposition. Changes in nozzle type, carrier volumes or pressure may also improve deposition. Application method is known to affect the deposition of fungicide by influencing penetration within the the plant canopy (Brenneman et al., 1990). Fungicides can be applied to peanut through various ground sprayers, fixed wing aerial applicators, or injected through irrigation systems (chemigation). Brenneman and Sumner (1990) reported that cholorthalonil applied via chemigation provided a similar level of leaf spot control as ground applications under low to moderate levels of disease; however, control was not sufficient with severe epidemics. Chemigation with propiconazole (Brenneman et al., 1994) or tebuconazole (Brenneman and Sumner, 1989) in place of foliar applications of chlorothalonil resulted in increased leaf spot incidence. Chemigation wets the entire leaf surface and residues may be displaced from the tissues due to the cuticle (Neely, 1970; Neely, 1971; Skoss, 1955). Johnson et al. (1986) found that only 10% of chlorothalonil applied was retained on the foliage after chemigation. Backman (1982) speculated that the displacement of PCNB and carboxin due to chemigation led to improved efficacy of stem rot in Alabama. A subsequent report evalauting tebuconazole found that Rhizoctonia limb rot was less severe where the fungicide was applied via chemigation (Brenneman and Sumner, 1989). Chemigation is permitted on several fungicide labels including azoxystrobin, metalaxyl and mefenoxam which are used predominantly for pod rot in in Texas where the majority of peanut acres are irrigated (Woodward and Black, 2007). In greenhouse studies simulating chemigation with mefenoxam, Wheeler et al. (2007) found that the chemical should be applied in an appropriate volume of water that places the fungicide at a depth where pods are developing. Higher irrigation rates led to increased concentrations at depths of 10 and 20 cm; however, excesive irrigation can leach the fungicide from the zone completely and compromise efficacy.

Fungicide penetration and deposition may also be affected by canopy density and architecture. Older peanut plants tend to have a more dense canopy, thus reducing deopsits to the lower canopy (Zhu et al., 2003). Much research has been conducted to evaluate

methods of improving fungicide penetration into the lower canopy for control of soilborne diseases. The application of benomyl in conjuntion with the pruning of peanut vines increased stem rot control (Backman et al., 1975). Likewise, the application of iprodione following pruning has improved control of Sclerotinia blight (Bailey and Brune, 1997; Butzler et al., 1998). Implements designed to open the canopy have been used to concentrate fungicides near the crown area. Grichar (1995) found that use of an A-sweep boom attachment improved the efficacy of several fungicides towards stem rot. Targeting applications of fluazinam using a canopy opener allowed for reduced rates to be used in the control of Sclerotinia blight in Oklahoma (Damicone and Jackson, 2001).

More recently, Augusto et al. (2010a) found that fungicide applications made at night (when peanut leaves are folded) rather than the day (when peanut leaves are unfolded) were more effective for the control of stem rot and increased yields. While stem rot control was enhanced, incidence of early leaf spot was not affected by application timing with systemic fungicides, but protectants such as chlorothalonil were less effective for leaf spot when sprayed at night. Additional studies found that early morning applications (applied betwen 3:00 and 5:00 A.M.) of pyraclostrobin and prothioconazole plus tebuconazole decreased stem rot compared to day-time or evening (between 9:00 and 10:00 PM) applications (Augusto et al., 2010b). In that study, applications of systemic fungicides applied prior to sunrise increased yields compared to day applications. This resulted from increased spray coverage, density and droplet size in the lower canopy, as well as improved redistribution downward with movement in dew that was present in the morning applications.

2.4. Redistribution of fungicides via irrigation

Historically, suppresion of soilborne pathogens was achieved through applications of granular fungicides banded over the center of the row (Csinos, 1987). These formulations were thought to sift through the canopy ultimately arriving at the soil; however, control using these materials was costly and inconsistent. The registration of the flutolanil has provided producers with a more effective means of managing soilborne diseases (Hagan et al., 2004). Furthermore, the registration of tebuconazole and azoxystrobin, has greatly improved both stem rot and leaf spot management over the past decade (Brenneman and Culbreath, 1994; Brenneman and Murphy, 1991; Grichar et al., 2000). In contrast to granular fungicides, broadcast-spray applications of these compounds are made to peanut foliage. Fungicide deposition within the canopy contributes to efficacy for leaf spot, but the management of stem rot is more difficult since the target of spray deposition for stem rot control is at the base of the plant or even below ground (Punja, 1985). The mechanism by which foliar-applied fungicides affect stem rot is not fully understood. It is believed that initial deposits of fungicides within the upper canopy are washed on to stems and pegs at the base of the plant via dew, rainfall, or irrigation (Taylor, 1996). This hypothesis was tested by Csinos and Kvien (1988), by using methyl-blue dye to demonstrate fungicide redistribution with irrigation. As a result of these studies and observations of sporadic reductions in efficacy of foliar-applied fungicides in non-irrigated fields, producers in

Georgia are advised to administer irrigation following fungicide applications in order to maximize stem rot control (Kemerait et al., 2006). It is recognized that administering irrigation too quickly may compromise leaf spot control, but the timings needed to optimize control of diseases caused by foliar and soilborne pathogens are not well documented.

There is currently limited information available regarding the redistribution of fungicides from rainfall or irrigation. Most of what has been reported pertains to the influence of rainfall and the rainfastness of protectant compounds in vegetables or fruit crops (Smith and MacHardy, 1984; Neely, 1971; Kudsk et al., 1991). Information regarding mechanisms of suppressing soilborne pathogens with foliar applied fungicides is even more limited. Csinos and Kvien (1988) suggested that initial fungicide deposits applied to peanut foliage are washed to the base of the plant, thus improving contact with soilborne pathogens. Presumably, fungicides were redistributed from the foliage to crowns and pegs.

Using *S. rolfsii* to bioassay peanut tissues, Woodward (2006) was able to quantify the redistribution of azoxystrobin, flutolanil and tebuconazole applied to foliage using irrigation, and to examine the effects of different irrigation timings (0-96 hours after application). In that study, irrigation timing was found to affect the efficacy towards both foliar and soilborne pathogens. Leaf spot was more severe when irrigation was administered immediately after fungicides were applied, whereas, a significant reduction was was observed following a 6 to 12 hour delay in applying irrigation. Maximum leaf spot control was obtained when fungicides were allowed to dry for 24 hours. Inversely, pod colonization (indicating potential for pod rot) increased significantly as irrigation was delayed. Overall, pod colonization was similar for all the fungicides evaluated; however, supression was greatest for tebuconazole at earlier timings. Smaller differences between timings were observed for azoxystrobin. Differences in physiochemical properties of these fungicides, such as affinity to the leaf surface, permeability, and the rate of uptake could have attributed to these differences.

Flutolanil (Araki, 1980) and tebuconazole (Taylor, 1996) are rapidly absorbed by the leaf, whereas, azoxystrobin remains on the leaf surface for a longer period (Bartlett et al., 1995). The persistence of azoxystrobin on the leaf surface may help explain the differences in the pod colonization for the non-irrigated controls. Earlier irrigation timings led to maximum stem rot control, while longer drying times were required to maximize leaf spot control. In the study conducted by Woodward (2006), a period of 18 hours drying time was required between the application of select fungicides and admistering an irrigation event. More recently, Augusto and Brenneman (2011) evaluated the interactive effects of fungicide timing and subsequent irrigation. Leaf spot control was not effected by irrigation, which was applied approximately 24 hours after fungicide applications. Overall, the application of irrigation was less effective at reducing stem rot incidence compared to nighttime applications of fungicides; however, effects of neither fungicide timing or subsequent irrigation were the same for all fungicides evaluated. This could be attributed to differences in retention, absorbtion or systemicity of the fungicides. Systemic fungicides used to manage leaf spot and stem rot move acropetally within the plant; however, applications of

prothioconazole, or prothioconazole plus tebuconazole have been shown to reduce disease in the lower non-treated areas of the plant (Augusto and Brenneman, 2012). A better understanding of fungicide systemicity is needed to maximize foliar and soilborne disease control in peanut. Furthermore, the increased residual activity of newer peanut fungicides has led to changes of commercial fungicide regimes under reduced disease pressure.

2.5. Use of extended interval fungicide programs and forecasting models

While fungicides are typically applied on a 14-day schedule to manage fungal diseases, the use of extended spray intervals could certainly be beneficial to producers by reducing production costs if they could maintain similar yields. In a study conducted by Brenneman and Culbreath (1994), fungicides applied on a 14-day schedule and 21-day schedule provided similar levels of leaf spot and stem rot suppression. Disease suppression decreased in plots treated on a 28-day interval; however, leaf spot and stem rot suppression was lower than what was observed in the non-treated control. A similar trend was observed for yield, where 3-year averages for the non-treated control, 14-day and 21-day intervals were 2914, 5153, and 4704 kg per hectare, respectively. Additional studies have shown that fungicides applied on 21- or 28-day intervals are capable of providing sufficient control of diseases and provide yields comparable to those achieved by the standard 14-day applications interval (Brenneman et al., 2001; Culbreath, 1993; Culbreath et al., 1992; Monfort, 2002; Phatak et al., 2002). Results of one study in particular showed that plots receiving as few as four chlorothalonil applications applied on a 28-day interval had yields as high as plots treated with seven applications made on a 14-day interval (Culbreath et al., 1992). Chandra et al. (1998), found that one properly timed application provided adequate control of leaf spot; however, timings differed within years. More recently Culbreath et al. 2006 also demonstrated excellent leaf spot control with pyraclostrobin applied at more extended intervals, and even when the initial sprays were greatly delayed. Delayed initial applications with this fungicide are now widely used by growers in the southeastern United States with good results.

By better defining the environmental conditions that favor disease development, peanut producers can improve disease control by timely application of fungicides. Forecasting models use environmental data such as temperature, rainfall and relative humidity, to predict when conditions are favorable for pathogen and disease development (Campbell and Madden, 1990). Over the past 40 years, various forecasting models have been developed and successfully implemented for peanut diseases. Jenson and Boyle (1966) and Phipps and Powell (1984) are credited with developing some of the first forecasting models to manage peanut leaf spot. More recently, an early leaf spot spray advisory, developed in Virginia, was effective in reducing number of sprays required for satisfactory disease control and has been highly accepted by growers (Cu and Phipps, 1993; Phipps, 1993). Spray advisories for late leaf spot have been implemented in other peanut producing states, such as Georgia, Alabama, North Carolina and Oklahoma (Nutter and Brenneman, 1989; Davis et al., 1993; Bailey et al., 1994; Damicone 1994).

In Georgia, AU-Pnut is the predominant leaf spot advisory used in research; however, it is not widely used by producers. This model was developed in the late 1980s, and is based solely on precipitation (the number of precipitation events and the five-day forecasted probability of precipitation) (Davis et al., 1993). Studies to evaluate the AU-Pnut advisory for timing applications of fungicides aimed at soilborne fungi have shown suppression of stem rot, but the results have been inconsistent (Brenneman and Culbreath, 1994; Rideout, 2003).

Several spray advisories based on the environmental conditions that incite Sclerotinia blight have been developed in Virginia and North Carolina (Phipps, 1995, Langston, 1998, Langston et al., 2002). Such advisories have been shown to improve disease control when compared to calendar applications. These advisories are based on air and soil temperatures, precipitation, relative humidity, vine growth, and canopy closure. Adaptations of these models have been evaluated for the control of stem rot. Rideout (2003) demonstrated that fungicide application timing has a significant effect on stem rot control and yield in Georgia. Furthermore, he concluded that the application of fungicides according to advisories based on soil temperature, precipitation and host growth provided similar or better disease control than the typical calendar-based programs.

3. Conclusions

Peanut is susceptible to various foliar and soilborne pathogens. Currently there is a wide range of fungicides labeled for management of peanut diseases (Table 1). Standard fungicides, such as chlorothalonil or tebuconazole, commonly comprise fungicide regimes designed to control leaf spot and stem rot, respectively. Other diseases, such as pod rot and Sclerotinia blight are managed with fungicides such as azoxystrobin and fluazinam, respectively. Several other fungicides with different modes of action are avaliable for use in peanut. While some fungicides, such as pyraclostrobin have post-infection activity, efficacy is typically greatest when applications are made in a preventative manner. Utilization of integrated disease managment strategies that incorporate factors such as field history, cultural practices and partially resistant cultivars may be used to reduce disease pressure and increase profitability. Resistance to several classes of fungicides used in peanut have been identified in populations of leaf spot pathogens. Most recently, resistance to triazole fungicides, such as tebuconazole, have been reported in eastern production regions of the United States. Furthermore, the potential exists for resistance to develop in other fungicide classes, primarily the strobilurin; therefore, it is imperative that producers rotate chemistries to ensure the sustainability and longterm use of these fungicides. Future research evaluating aspects of peanut fungicides, such as initial application timing, systemic and residual activity and interactive effects of tank-mixtures are warranted. For diseases caused by soilborne pathogens, a better understanding of spatial and temporial aspects of the pathogen could allow for more precise applications of fungicides.

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