Chapter 8

Nematode Parasites of Peanut

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The cultivated groundnut or peanut (*Arachis hypogaea* L.) is an annual, self-pollinating, herbaceous legume, native to South America (Hammons, 1982). It is geotropic, producing its pods (fruits) underground. Flowering begins four to six weeks after planting and extends over a period of several weeks. Within about one week after fertilization, a pointed needle-like structure, the carpophore, commonly called the "peg" develops, elongates and grows into the soil 2–7 cm deep. Upon entering the soil, the fertilized ovaries located behind the tip of the peg enlarge rapidly and pod growth begins. The length of time necessary for pod development to maturity may vary with cultivar and environmental conditions. Williams and Drexler (1981) determined that the cultivar Florunner required 63–70 days from the time the ovary began enlarging to maturity.

Peanut was listed as one of the twenty crop plants that stand between man and starvation (Wittwer, 1981). Peanut seeds are rich in calories and contain 25% protein. They may be boiled, broiled, roasted, fried, ground into peanut butter or crushed for oil. Peanut-containing foods such as peanut butter, salted peanuts, candies, and snack-type crackers and cookies are popular because of their unique roasted peanut flavour (McWatters & Cherry, 1982). On a worldwide scale, however, peanuts are grown primarily for cooking and salad oil. Oil extraction also produces a protein-rich byproduct which may be used for human consumption if processed from edible-grade peanuts, otherwise, it is used for animal feed.

The peanut today is cultivated on all six continents in about 80 countries. Eight countries, China, India, United States, Senegal, Sudan, Brazil, Argentina, and South Africa, produce about 77% of the world supply (United States Department of Agriculture, 1989). In 1988–89, approximately 21.98 million/t were produced on 18.95 million hectares. Production is distributed generally in the tropical, sub-tropical, and warm temperature zones. In addition, many of the major production regions are characterized by having loose friable sandy soils.

Nematodes of Peanut

Nematodes damage peanuts in all production regions of the world. Based on a worldwide survey of nematologists, annual losses caused by all nematodes to peanut were estimated at 12% and monetary losses were estimated at 1.03 billion U.S. dollars (Sasser & Freckman, 1987). The nematodes that are known to cause damage to peanut are *Meloidogyne* spp., *Pratylenchus brachyurus, Belonolaimus longicaudatus, Criconemella ornata, Aphelenchoides arachidis, Aphasmatylenchus straturatus, Scutel-*

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lonema cavenessi, Tylenchorhynchus brevilineatus, and *Ditylenchus destructor*, although many other species have been found in association with peanut (Sharma, 1985).

Meloidogyne

The three *Meloidoyne* species parasitizing peanut are *M. arenaria* (peanut root-knot nematode), *M. javanica* (Javanese root-knot nematode) and *M. hapla* (northern root-knot nematode). These three species are known to occur in North, Central and South America, Africa, India, Europe including the Mediterranean region, Japan, Australia and Fiji Islands (Sasser, 1977). Their distribution and economic importance are purported to be related to biological and environmental factors favourable to the nematodes. *Meloidogyne arenaria* and *M. javanica* are common in warm and hot regions of the world whereas *M. hapla* occurs only in cool regions.

According to a recent report, *M. arenaria* is the predominant *Meloidogyne* species parasitizing peanut in Alabama, Georgia, Texas and Arkansas, and *M. hapla* is the most damaging species in North Carolina, Virginia, and Oklahoma (Anon., 1987). Both *M. arenaria* and *M. hapla* were reported to cause damage in Georgia, North Carolina and Oklahoma. *Meloidogyne javanica* is present in some of the peanut growing regions of the United States, but it was reported parasitizing peanuts only in Georgia in one location (Minton *et al.*, 1969*b*).

In other regions of the world, *Meloidogyne arenaria* has been reported on peanut in Zimbabwe (Martin, 1958), Israel (Orion & Cohn, 1975), Egypt (Ibrahim & El-Saedy, 1976*a*), India (Sharma *et al.*, 1978; Dhruj & Vaishnav, 1981; Sakhuja & Sethi, 1985*c*), Taiwan (Cheng & Tu, 1980; Cheng *et al.*, 1981) and China (Zhang, 1985). In Senegal, Netscher (1975) reported that an isolate of *Meloidogyne* species resembling *M. arenaria* reproduced slightly on peanut, but that juveniles collected from peanut failed to reproduce on susceptible tomato. Even though *Meloidogyne* spp., including *M. arenaria* and *M. javanica*, occur in Senegal, they do not damage peanut.

Meloidogyne hapla was reported parasitizing peanut in Israel (Minz, 1956), South Africa (van der Linde, 1956), Australia (Colbran, 1958; Saint-Smith *et al.*, 1972), Zimbabwe (Martin, 1961), Japan (Mitsui *et al.*, 1976), Korea (Choi, 1981) and China (Yin & Feng, 1981; Yang, 1984; Zhang, 1985).

The first report of *M. javanica* parasitizing peanut was by Martin (1958) in Zimbabwe. A few years later Minton *et al.* (1969*b*) found this species parasitizing peanut in one location in Georgia, USA. In addition, *M. javanica* was reported on peanut in Egypt (Ibrahim & El-Saedy, 1976*b*), Brazil (Lordello & Gerin, 1981) and India (Sakhuja & Sethi, 1985*b*).

Symptoms of damage

Juveniles of *Meloidogyne* spp. enter and damage peanut roots, pegs and pods. Juveniles upon entering the root tips cause only slight mechanical injury, except when large numbers enter in a limited area.

Minton (1963) studied the infectivity and histopathology of *M. arenaria* on peanut. Roots inoculated with *M. arenaria* second stage juveniles were invaded by the second day. Large, multinucleate, densely stained giant cells developed by the eighth day. Hyperplasia occurred in tissue adjacent to the nematode and hyperplasia and hypertrophy resulted in disorganization of vascular tissue. Galls resulted as the parenchymatous cells associated with developing nematodes at the periphery of the stele multiplied and grew out into the cortex. Adjacent cortical cells were crushed by the expanding nematodes and parenchyma cells and necrosis was associated with the damage. Elongation of severely galled roots was slowed.

The anatomical changes induced by *M. javanica* in peanut root tissues include cell hyperplasia and hypertrophy that results in the formation of giant cells in the cortical and stelar tissues (Ibrahim & El-Saedy, 1976*b*). A major consequence of nematode development and giant cell formation in the stele was the malformation of the xylem elements and the inhibition of secondary growth of the xylem and phloem tissues.

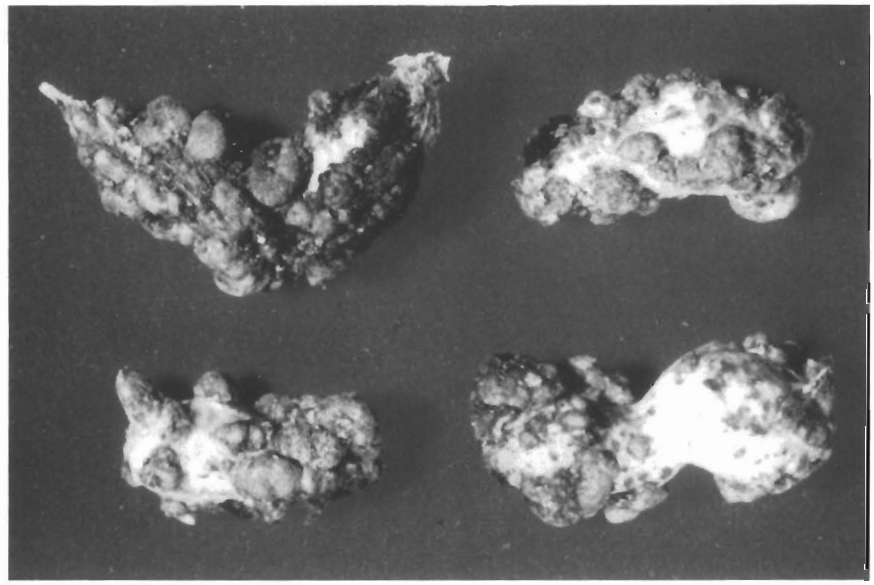

Fig. 1. *Meloidogyne arenaria* galls on peanut pods (Photo: D. W. Dickson).

Galls on peanut roots, pods and pegs caused by *M. arenaria* (Fig. 1, Plate 6A) and *M. javanica* are similar and are larger than those caused by *M. hapla* (Fig. 2, Plate 6B) (Sasser, 1954; Minton *et al.*, 1969*b*; Taylor & Sasser, 1978). Galls produced by *M. arenaria* and *M. javanica* may attain a diameter several times that of the adjacent root; thus the roots become much enlarged with fewer than normal small feeder roots. Sakhuji and Sethi (1985*b*) observed that roots infected with *M. javanica* proliferated with two to five lateral roots arising at the gall sites. *Rhizobium* nodules on peanut roots may be mistakenly diagnosed as root-knot galls. However, the appearance of root-knot nematode galls and *Rhizobium* nodules is distinctively different: galls result from an internal swelling of the root tissue and are of a woody consistency, whereas nodules are of a spongy consistency and are mostly appended laterally and can be rubbed off easily (Fig. 3). Damaged pods may become disfigured and fail to produce seeds (Fig. 1). *M. hapla* infected roots develop small galls and heavily infected roots develop extensive root proliferation above the galls resulting in a dense mat or bushy root system (Sasser, 1954).

Machmer (1951) described the field symptoms of *M. arenaria* on peanut as follows: "Galling occurs on all underground parts of peanut plants including the pods which appear warty. Pod stems are often heavily galled and are easily severed. Early infection of the peg (ovary) is detrimental to the seed embryo. Heavily galled plants frequently have a great many necrotic pegs but less than a dozen mature pods." Machmer further stated that the peanut vegetation usually does not exhibit conspicuous symptoms until near harvest. "At this time plants are becoming discoloured (Plate 6C) and are stunted so that they fail to cover the soil between rows (Fig. 4). The slowly dying and browning plants present a mottled effect among the greener plants and weeds. When the roots of such plants show conspicuous root-knot nematode galls the neighbouring plants in apparent vigour are usually well infected also." In China, Zhang (1985) reported that *M. arenaria* infected plants may become yellow and stunted as early as 40 days after planting. Above ground symptoms of *M. hapla* (Taylor & Sasser, 1978) and *M. javanica* (Minton *et al.*, 1969*b*) are similar to those of *M. arenaria*. Plants affected make poor top growth and yield poorly.

Fig. 2. Peanut roots galled by *Meloidogyne hapla*. Root proliferation near galls results in a matted root system (Photo: L. I. Miller).

Biological races

Taylor and Sasser (1978) suggested that the word "race" should be used only for populations of *Meloidogyne* that have been shown by numerous experiments to have host preferences significantly different from those established as "normal" for the species concerned, and also have wide geographical distribution. Sasser and Nusbaum (1955) observed that a population of *M. arenaria* in North Carolina did not infect peanut and suggested that this population differed from populations in Georgia and other states. Based on extensive differential host tests using populations of *M. arenaria, M. javanica* and *M. hapla* from widely separated regions of the world, Sasser (1972), Taylor and Sasser (1978) and Eisenback *et al.* (1981) separated *M. arenaria* into two races; race 1 reproduces on peanut but race 2 does not. Host races of *M. arenaria* are distributed throughout the world and are morphologically indistinguishable (Sasser, 1979*a*; Osman *et al.*, 1985). Reactions of the different populations of *M. javanica* and *M. hapla* to the differential hosts were relatively uniform, hence different biological races were not detected, although, some variability within populations of *M. javanica* was noted. Most *M. javanica* populations did not reproduce on peanut. *M. javanica* has been reported infecting peanut in Zimbabwe and Malawi (Martin, 1958), Georgia, USA (Minton *et al.*, 1969*b*) and Brazil (Lordello & Gerin, 1981).

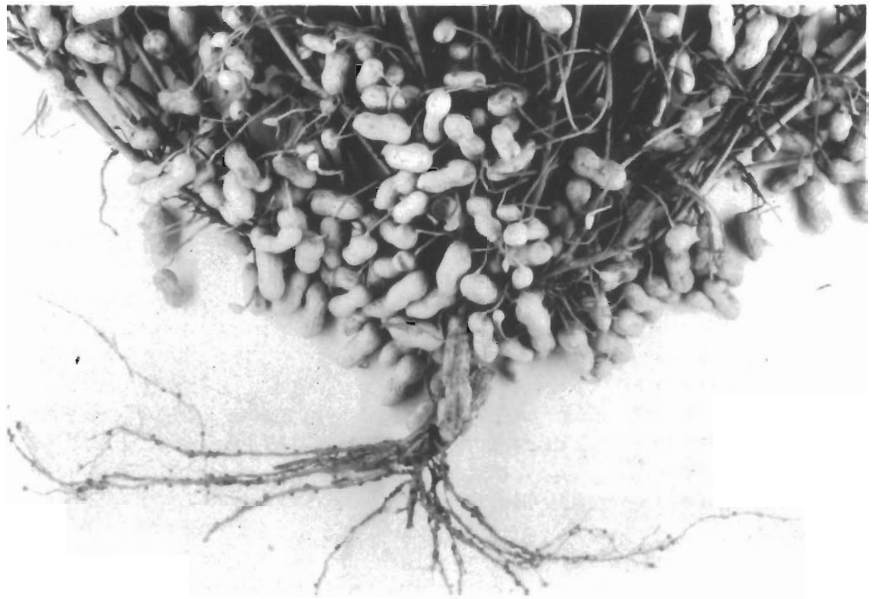

Fig. 3. Peanut plant with no nematode damage but with numerous nitrogen-fixing nodules attached to roots.

Fig. 4. A peanut field in Georgia with severe *Meloidogyne arenaria* damage in the foreground.

Survival and means of dissemination

Orr and Newton (1971) found *Meloidogyne* juveniles among 28 genera recovered from dust traps placed 2 m above the ground in Western Texas. Dispersal by surface run-off water and by irrigation also occurs (Faulkner & Bolander, 1966; Meagher, 1967; Sauer, 1968), but, for the most part, *Meloidogyne* species are disseminated widely by human activities. They may be spread in freshly dug peanut, including pods and roots, but generally they do not survive in well dried pods. Refuse from packing and processing plants that has not been thoroughly dried may harbour viable eggs and infective juveniles. Soil from infested fields transported on tillage equipment and the feet of farm animals and humans may also be a source of eggs and juveniles to infest fields.

Environmental factors affecting parasitism

Temperature is considered the most important environmental factor affecting *Meloidogyne* species survival and parasitism, and the lower and upper temperatures for survival are approximately 0° to 5°C and 35°C to 40°C, respectively (Taylor & Sasser, 1978). In general, the optimum temperature for survival of eggs and juveniles is 10°C–15°C (Bergeson, 1959; Thomason *et al.*, 1964). The optimum temperature for hatching of *M. hapla* and *M. javanica* is 25°C and 30°C, respectively (Bird & Wallace, 1965). *M. javanica* had a significantly higher hatch at 30° C than *M. hapla*. Milne and Du Plessis (1964) found that the life cycle time for *M. javanica* at 14.3°C was 56 days and 21 days at 26.1°C.

There is general agreement that *Meloidogyne* species damage is greater in sandy soils than in soils with a large percentage of clay. In Arizona, heaviest infestations of *M. incognita* occurred on coarse-textured soils (O'Bannon & Reynolds, 1961). Also in China the incidence and severity of *M. arenaria* on peanut was found to be related to soil texture (Zhang, 1985).

Soil moisture is necessary to sustain all activities of *Meloidogyne* spp. In moist soils, of 40–60% of field capacity, juveniles are active and move through the soil in water films. In dry soils they become inactive and die through desiccation (Van Gundy, 1985). In wet soils, hatching may be inhibited and juvenile movement slowed by lack of oxygen. Baxter and Blake (1969) found that all activities of *M. javanica* increased as oxygen concentrations increased from 0.2 to 21% and concluded that a favourable environment would be provided when moist soils drain rapidly and allow oxygen concentrations to increase above 10%. Zhang (1985) reported that *M. arenaria* is less serious in low fields that have a high water table than in well drained fields. Also, *M. arenaria* is less serious on peanut that follow a flooded crop than in fields that are not flooded. In controlled temperature studies, Vrain (1978) found that infectivity of *M. incognita* and *M. hapla* were lower after having been exposed to temperatures ranging from 20°C to –8°C in saturated soil than when exposed to these temperatures in soil at 51 cm moisture tension.

Meloidogyne species survive, hatch and reproduce over a wide pH range. If the soil pH is in the range favourable for plant growth the nematodes are active (Wallace, 1971).

The addition of organic amendments to the soil reduced the severity of *M. arenaria* on peanut (Zhang, 1985).

Disease complexes

Garcia and Mitchell (1975*a*) observed synergistic interactions in the incidence of pod rot of peanut when *Pythium myriotylum* was combined with *Fusarium solani* or *M. arenaria*, or a combination of both pathogens. Garcia and Mitchell (1975*b*) also reported that a combination of *P. myriotylum* and *M. arenaria* resulted in a significantly greater percentage of damping-off of peanut seedlings than the sum of the effects of the pathogens separately. Peanut plants inoculated with 2 g and 4 g of *F. solani* mycelia mat per pot plus 1000 to 2000 *M. arenaria* wilted sooner after inoculating than when *F. solani* was used alone (Patel *et al.*, 1985). Results of a two-year study in 25 cm clay pots indicated that the presence of *M. arenaria* had no effect on the incidence of *Aspergillus flavus* (Lk.) Fr. in peanut seeds (Minton & Jackson, 1967). However, one year the incidence of *A. flavus* was greater in shells of plants inoculated with both organisms than with only *A. flavus*. In a microplot study,

the incidence of *A. flavus* was greater in seeds of plants inoculated with *A. flavus* and *M. hapla* than in seeds of plants inoculated with only *A. flavus* (Minton *et al.*, 1969*a*). Aflatoxin was not detected in seeds of any treatment and was present in only one shell sample each of *A. flavus* or *A. flavus* plus *M. hapla* inoculated plants.

In greenhouse studies, Diomandé and Beute (1981*a*) demonstrated that cylindrocladium black rot (CBR) of peanut caused by *Cylindrocladium crotalariae* was increased in the presence of *M. hapla* on CBR-susceptible Florigiant and CBR-resistant NC 3033 cultivars. In field experiments, there was a significant positive correlation between final populations of *M. hapla* and *C. crotalariae* and CBR indicating that *M. hapla* affected CBR development (Diomandé & Beute, 1981*b*). Diomandé *et al.* (1981) also found that two populations of *M. arenaria* enhanced development of CBR on the CBR-resistant peanut, NC 3033 cultivar.

Economic importance and population damage threshold levels

Information on the economic importance of *Meloidogyne* species on peanut is unavailable in many areas of the world. Yield loss estimates for the individual species is difficult because damage is seldom confined to a single nematode species (Sasser *et al.*, 1970; 1975*a*).

In Georgia, Motsinger *et al.* (1976) found that 9.7% of the fields surveyed in seventeen counties were infested with *M. arenaria* or *M. hapla*. In eleven counties in Alabama, Ingram and Rodríguez-Kábana (1980) found 41.4% of the fields surveyed infested with *Meloidogyne* species. Twenty-six percent of the 127 fields surveyed or 15.5% of the 343 soil samples examined from five counties surveyed in Texas were infested with *Meloidogyne* species (Wheeler & Starr, 1987). At least 10% of the survey samples were estimated to have root-knot nematode population densities of 44–83 *M. arenaria*/500 cm^3 soil, exceeding that necessary for a 10% yield loss.

Losses in infested fields may exceed 50%, however, infestations are usually unevenly distributed and losses may average less than 50%. Recently, estimated production losses due to *M. arenaria* in major peanut-producing states of the USA ranged from 0.5% in Oklahoma to 5.4% in Alabama (Anon., 1987). Losses for *M. hapla* ranged from 0.3% in Georgia to 4.7% in North Carolina. Sasser (1979*b*) reported that the estimated peanut losses due to *Meloidogyne* species in West Africa and Southeast Asia was 15%.

In the Punjab State of India, *Meloidogyne* spp. juveniles were present in eleven peanut soil samples out of 28 examined from Ludhiana, seven out of twenty from Sangrur and eight out of twelve from Patiala districts (Sakhuja & Sethi, 1985*c*). *Meloidogyne* species were also found in Jalandhar and Kapurthala districts. Galling on peanut due to *Meloidogyne* spp. was noted in 22 locations of the 70 locations sampled. Ibrahim and El-Saedy (1976*a*) found that 65% of the 146 soil and root samples collected from declining peanut in Egypt contained *Meloidogyne* spp. *Meloidogyne javanica* was the dominant species with *M. arenaria* present in a few of the root samples. Singh (1972) reported that nine out of twelve soil samples collected around peanut plants in Guyana contained *Meloidogyne* spp.

Meloidogyne arenaria was reported to be a major disease of peanut in China (Zhang, 1985). *M. arenaria* occurs primarily in the southern area of the peanut production region and *M. hapla* in the northern area (Yang Baojun, pers. comm.). Investigations revealed that 61% of peanuts grown on 6200 ha in Leizhou Peninsula were infected with *M. arenaria* (Zhang, 1985).

In India, Sakhuja and Sethi (1985*b*) found that peanut plants grown in pots were stunted when inoculated with one *M. javanica* egg per cm^3 soil. A reduction of 27.3% in shoot length and 54.6% of dry shoot weight was obtained when plants were inoculated with eight eggs per cm^3 soil. The commonly used extraction procedures do not recover *Meloidogyne* eggs from the soil (Garcia, 1976; Rodríguez-Kábana *et al.*, 1986). Therefore, population evaluations for research and advisory purposes are usually based on numbers of juveniles in the soil. Population levels of *M. arenaria* juveniles in the soil in southeastern United States at planting time are usually relatively low and damaging populations may be near undetectable levels (Fig. 5). Hence, population levels for grower

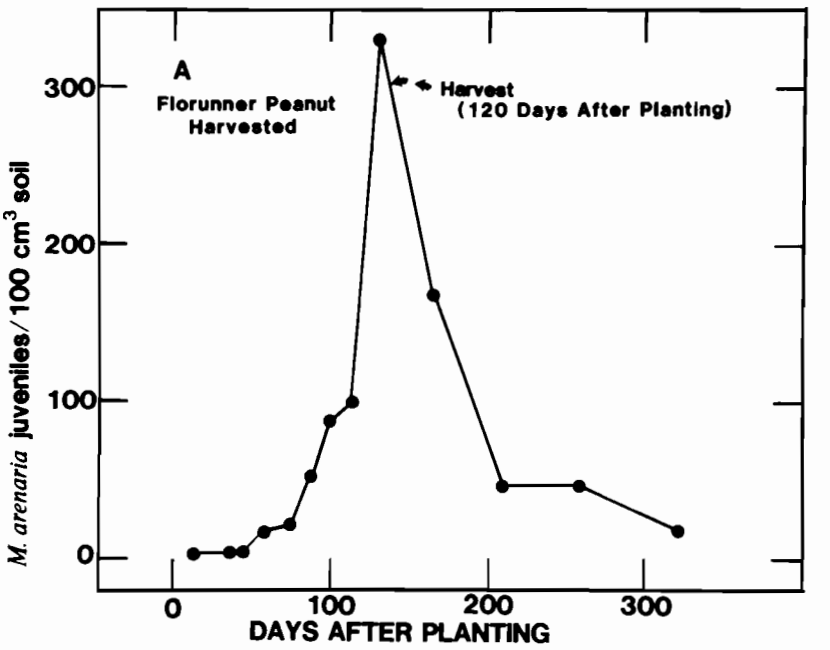

Fig. 5. Changes in the juvenile population of *Meloidogyne arenaria* during the peanut growing season and after harvest. (From Rodriguez-Kábana *et al.*, 1986.)

advisory purposes are usually determined as soon after harvest as practical rather than eight months later at planting.

In nematicide experiments, yields are usually negatively correlated with numbers of *Meloidogyne* juveniles in the soil. Regression analysis on data from 16 peanut experiments in Alabama indicated that yields were negatively related to numbers of *M. arenaria* juveniles in the soil determined near harvest (Rodríguez-Kábana *et al.*, 1982*b*).

On the basis of a linear regression model, Rickard *et al.* (1977) determined that peanut yield loss in microplots was 8.6% for each ten-fold increase in initial population of *M. hapla* juveniles in the soil.

Wheeler and Starr (1987) reported a significant negative relationship between initial populations of *M. arenaria* in microplot tests and peanut yields. A linear model estimated a 10% yield loss with initial populations of 44 to 83 eggs and juveniles per 500 cm^3 soil. Dhruj and Vaishnav (1981) found that 1000 *M. arenaria* juveniles per kg of soil caused a reduction of peanut plant shoot growth, shoot weight and root length of 23.9%, 33.1% and 31.9%, respectively.

Control measures

Control of *Meloidogyne* species may be necessary in some fields for profitable peanut production but not in others. Therefore, each field should be evaluated based on the history of nematode damage to peanut, the other crops growing in rotation with peanut, and the present nematode population level. Based on a survey, Motsinger *et al.* (1976) estimated that only 26.6% of the peanut fields in Georgia would respond to nematicides. However, this should not be interpreted to imply that other control measures such as rotations and cultural practices should not be considered in the remainder of the peanut production area. Preventing the development of a nematode problem may be more economical than managing the nematode once the problem develops.

Fig. 6. A peanut field in North Carolina infested with *Meloidogyne hapla*. Left, after cotton; right, after soybean. (Photo: J. N. Sasser).

Cultural practices

Rotations that include plants resistant to *M. arenaria, M. hapla* and *M. javanica* can be effective in reducing the damage to peanut caused by these nematodes (Fig. 6). When the cash value for peanut is low, this may be the only control method that can be used profitably.

Sasser (1954) published a susceptibility rating for a number of plants to *M. incognita, M. incognita* var. *acrita, M. javanica* and *M. arenaria*. In this publication he showed that peanut is susceptible to *M. arenaria* race 1 and *M. hapla*, but resistant to *M. incognita, M. javanica* and *M. incognita* var. *acrita*. However, subsequent research by Martin (1958) showed that some populations of *M. javanica* parasitize peanuts. Sasser listed a number of plants resistant to *M. arenaria* and *M. hapla* that were used effectively in rotation with peanut when one or more of these species is present. Since the pioneer research of Sasser (1954), many additional plants were found resistant to one or more *Meloidogyne* species.

A recently published check list (Sasser & Kirby, 1979) of crop plants listing over 450 cultivars in thirteen botanical families reported to carry resistance to at least one *Meloidogyne* species may serve as a useful guide for selecting cultivars to grow in rotation with peanut. In addition, Cheng *et al.* (1981), reported seven crop plants of thirty tested resistant (non-host) to *M. arenaria* in Taiwan. Care must be exercised in selecting cultivars to rotate with peanut because all cultivars of a crop do not respond the same. Maize (*Zea mays* L.) is an example of a crop that was for a long time considered an excellent rotational crop with peanut. But in recent years, some cultivars have been shown to support relatively high populations of *M. arenaria* and *M. javanica* (Baldwin & Barker, 1970; Norse, 1972). Conversely, most cultivars are resistant to *M. hapla* (Sasser, 1954; Baldwin & Barker, 1970).

Rodríguez-Kábana and Touchton (1984) in Alabama obtained a reduction of *M. arenaria* juveniles in sorghum (*Sorghum vulgare* Pers.) or maize to levels 10–20 times below those in peanut. Cotton effectively reduced *M. arenaria* population levels and yields of peanut planted after one year of cotton were significantly greater than yields in plots grown to peanut the previous year (Rodríguez-Kábana *et al.*, 1987). Rodríguez-Kábana and Morgan-Jones (1987) also found that sesame (*Sesamum indicum* L.), castor bean (*Ricinus communis* L.), joint vetch (*Aeschynomene indica* L.), partridge

peas (*Cassia fasiculata* Michx.), hairy indigo (*Indigofera hirsuta* L.) and bahiagrass (*Paspalum notatum* Flügge) are promising crops for managing *M. arenaria* in peanut.

Rotational crops recommended for *Meloidogyne* management on peanut in the United States varies with the nematode species present, cultivar of rotational crop, etc. Among the rotational crops that have been suggested by the various State Cooperative Extension Service specialists are cotton, maize, small grains and pasture grasses (Bailey, 1988; Dunn, 1988; Hagan, 1988). Maize is also a recommended rotational crop for managing *M. hapla* on peanut in Queensland Australia (Broadley, 1981; Vance, 1981).

Usually long rotations of three or more years out of peanut and other host crops are better than one- or two-year rotations. Rotations should not be expected to abruptly reduce root-knot nematode populations since 1) some nematodes of a population will survive the winter without a host, 2) the most "resistant" crop plant may support at least a low nematode population, and 3) most cultivated fields have at least a few weeds that are good hosts for nematode reproduction. Therefore, rotations should maintain population densities at low levels and reduce high population densities (Dunn, 1988).

Where practical, crop rotations in conjunction with flooding of the soil may effectively reduce damage due to *Meloidogyne* species (Thames & Stoner, 1953; Zhang, 1985).

Rotating a winter small grain crop with peanut can help prevent growth of weeds that are hosts of peanut nematodes, however, since some small grain cultivars may also support low population levels if grown during warm weather, planting should be delayed until cool weather when nematode development and reproduction is reduced (Dunn, 1988).

Destruction of roots of host crops that precede peanut in a rotation to interrupt reproduction will reduce the potential for damage to peanut; turning the soil several weeks before applying nematicides and planting peanut encourages the decay of live plant roots that protect nematodes from their enemies or from nematicides that are applied to the soil (Dunn, 1988). Drying of the soil after it has been turned may reduce the nematode population (Zhang, 1985). Clean fallowing for long periods of time may also be effective.

In China (Zhang, 1985), growers who fertilize well, especially with organic fertilizers, have less *M. arenaria* damage to peanut than growers who use less fertilizer. *M. arenaria* is less serious in China (Zhang, 1985) in low lying areas that have high water tables than in well drained soils. This nematode is also less serious in China in irrigated than in non-irrigated fields.

Resistance and tolerance

Peanut cultivars resistant to *M. arenaria* race 1, *M. hapla*, and populations of *M. javanica* that attack peanut have not been developed. Edwards (1956) reported the cultivars Natal Common and Kumawu Erect to be highly resistant to a root-knot nematode. The species was not reported, therefore it probably was not *M. arenaria* or *M. hapla*. Miller and Duke (1961) reported that a peanut of "a foreign introduction with a purple skin" resistant to *M. arenaria*, but Miller (1972*b*) later reported no resistance to the nematode in 2000 peanut introductions in field plots in Virginia. In greenhouse tests, Minton and Hammons (1975) and Holbrook *et al.* (1983) did not find a high level of resistance to *M. arenaria* among a total of 805 peanut entries. Recently Baltensperger *et al.* (1986) reported resistance to *M. arenaria* in another *Arachis* species, *A. glabrata* Benth., that may provide resistance to transfer to *A. hypogaea* should technology become available for making wide interspecific crosses.

Castillo *et al.* (1973*a*) reported resistance to *M. hapla* in four introductions of unidentified wild *Arachis* spp. and only moderate susceptibility in eight *A. hypogaea* entries. Also, Subrahmanyam *et al.* (1983) reported a wild *Arachis* sp. resistant to *M. hapla*. Sakhuja and Sethi (1985*a*) reported resistance to *M. javanica* in four cultivars.

Chemical control

Chemicals are one of the major means of controlling nematodes including *M. arenaria, M. hapla, Belonolaimus longicaudatus, Pratylenchus brachyurus* and other nematodes in peanut in the United

Fig. 7. A peanut field in Georgia infested with *M. arenaria*. Left, untreated; right, treated with phenamiphos at the rate of 2.8 kg a.i./h.a.

States (Fig. 7). The two general types of materials that have been effective are fumigants and non-fumigants that have systemic and/or contact properties.

Formulations of fumigants containing DD, I,3-D, EDB and DBCP were the first nematicides to be used to control nematodes of peanut (Miller, 1951; Good *et al.* 1958; Miller & Duke, 1961). DBCP was the principal nematicide for use in peanut in the United States during the 1960's and remained so until 1978 when it was suspended by the U. S. Environmental Protection Agency. There was increased use of EDB after the suspension of DBCP until 1983 when EDB was also suspended.

Manufacturers of DD withdrew this material from the market. The less hazardous 1,3-D, that contains only 1,3-dichloropropene as the active ingredient, is still available and is more effective than DD (Porter *et al.*, 1982). In recent years research has been conducted to determine more efficacious methods of applying 1,3-D. Rodríguez-Kábana *et al.* (1985) found that combination treatments of 1,3-D and aldicarb equalled or surpassed the performance of EDB treatments in increasing yields and controlling *M. arenaria* in peanut. There was some degree of phytoxicity in planting time application treatments of 1,3-D. Rodríguez-Kábana and Robertson (1987) later found that the efficacy of 1,3-D applied in the row preplant was dependent on the rates used and depth of application. Minton and Csinos (1986) obtained significant peanut yield increases when 1,3-D was applied in the mouldboard plow sole at rates as low as 41.7 kg ai/ha.

Several non-fumigant compounds having both nematicidal and insecticidal properties were introduced in the late 1950's (Dickson & Smart, 1971; Minton & Morgan, 1974; Sasser *et al.* 1975*b*). Materials such as ethoprop and fensulfothion are contact nematicides with no significant systemic properties. Other materials, such as aldicarb, carbofuran, oxamyl and phenamiphos kill by direct contact or are absorbed by plants and the parent compound or some metabolite in the plant are nematicidal. A number of additional compounds, some of which are used primarily as insecticides, have been found to have nematicidal properties but are usually less effective for nematode control on peanut than those listed above (Dickson & Smart, 1971; Minton & Morgan, 1974; Sasser *et al.*, 1975*b*).

The nonfumigant nematicides have been evaluated for the control of most major peanut nematodes under various cultural conditions. Much of the research with these materials has been done in the United States (Minton & Morgan, 1974; Dickson & Waites, 1978, 1982; Rodríguez-Kábana

Fig. 8. Liquid and granular applicators for applying non-fumigant nematicides mounted on tractor drawn rototiller for incorporating (Photo: A. W. Johnson).

et al., 1981, 1982*a*; Minton *et al.*, 1984; Rodríguez-Kábana & King, 1985); however, some research with these compounds has also been done in India (Singh & Sakhuja, 1984), Australia (Colbran, 1968; Broadley, 1981) and China (Zhang, 1985).

Generally, the non-fumigant nematicides are preferred over the fumigants because of their simplicity of application. Depending on the formulation used, they can be sprayed or applied in an 18–23 cm wide band over the row with a granular applicator mounted on a rototiller or on the planter equipment (Fig. 8). Incorporating these materials five to seven cm deep or less is preferred over deeper incorporation (Rodríguez-Kábana & King, 1979).

Methods of diagnosis

Sampling

Diagnosing *Meloidogyne* damage on peanut can best be done by periodic field observations and root and pod examination in conjunction with soil assays. Characteristic foliage symptoms and galling of underground plant parts may be detected. The type of galls on the roots and pods may be a useful indicator of the *Meloidogyne* species present (Sasser, 1954). Soil samples should be collected at or near harvest to determine the maximum population density. Root and pod samples for nematode extraction should also be collected late in the growing season.

Bioassays to establish the level of infestation (Ingram & Rodríguez-Kábana, 1980) may be useful if samples are collected during the winter or early spring when population levels are low.

Extraction

Meloidogyne juveniles and eggs may be extracted from soil and roots using standard laboratory procedures (Chapter 2). Adult females may be excised from root or pod tissues to be examined to assist with species identification.

Determining relationship of populations to crop loss

A measure of nematode involvement in peanut yield loss may be determined by correlating numbers of *Meloidogyne* juveniles per unit of soil or root-knot nematode indices with yield in nematicide treated and untreated soil. Negative relationships were found between yield and the initial soil population density of *M. hapla* (Rickard *et al.*, 1977) and *M. arenaria* (Dhruj & Vaishnav, 1981; Wheeler and Starr, 1987) as well as the final population denisty of *M. arenaria* in the soil (Rodríguez-Kábana *et al.*, 1982*b*). Root-knot nematode indices at harvest were correlated with yield for *M. arenaria* and *M. hapla* (Minton & Morgan, 1974). Models that will predict yield losses for a wide range of environmental conditions are not available.

Pratylenchus brachyurus

Pratylenchus brachyurus is the major lesion nematode parasitizing peanut. It is distributed chiefly in the warmer zones of the world (Loof, 1964). Steiner (1949) first reported *P. brachyurus* on peanut in Alabama, USA in 1942. *P. brachyurus* is now known to parasitize peanut in most of the peanut producing states in the USA. It has also been reported on peanut in several other countries of the world including Egypt (Oteifa, 1962), Australia (Colbran, 1968) and Zimbabwe (Anon., 1973). Also, *P. coffeae* was reported parasitizing peanut in India (Chabra & Mahajan, 1976).

Symptoms of damage

Lesion nematodes are migratory endoparasites that attack peanut roots, pegs and pods and feed within the parenchymatous tissues. Steiner (1945) and Boyle (1950) described conspicuous lesions on the pods of peanut infected with *P. brachyurus*. Good *et al.* (1958) later reported *P. brachyurus* in roots and pegs, as well as shells of mature pods, but indicated that nematodes were more numerous in the shells, where they colonize in dark-coloured necrotic lesions (Fig. 9, Plate 6D. Colbran (1968) indicated that *P. brachyurus* produces lesions on the underground portion of the stem, as well as on the roots, pegs and pods. Several hundred nematodes may colonize a single lesion and may

Fig. 9. Lesions on peanut caused by *Pratylenchus brachyurus* (Photo: T. E. Boswell).

include all developmental stages. Infected peanut roots develop lesions and with high population densities, these lesions coalesce and cause extensive discolouration and damage that result in slight stunting with unthrifty, yellow-green foliage and reduced root system and pod weight (Miller & Duke, 1961; Boswell, 1968).

In experiments conducted in sterilized soil inoculated with sterile *P. brachyurus*, Boswell (1968) found that unstained cells of peanut shells adjacent to the nematode or through which the nematode had passed had a slight tan to brownish granular appearance. Boswell (1968) characterized lesions on unstained shell tissue free of fungal mycelia by small black pin point to pin head size spots on the shell surface and usually near the center of the lesion with the remainder of the lesion having a somewhat lighter appearance as though the colour faded out into the surrounding tissue. Close examination of these lesions revealed that the colour was due primarily to necrotic parenchyma tissue. The margins of these lesions were not distinctly outlined as were lesions caused when *Rhizoctonia solani* was present. Lesions infected by both organisms were described as appearing rough and more like a scurf with brownish to black discolouration of the surface in the necrotic areas. These lesions have a definite margin even though the shape of the lesion may be irregular. An occasional lesion infected with both *P. brachyurus* and *R. solani* had a slightly raised dark centre and microscopic examinations revealed the presence of sclerotia. The symptoms of *P. brachyurus* damage on peanut shells grown in the field differ slightly from that described by Boswell (1968). Good *et al.* (1958) found that lesions on mature shells were "purplish-brown" and could be distinguished from lesions caused by soil microbial decomposition by their darker colour and distinct boundaries which did not fade gradually into the healthy surrounding tissue, as with microbial decomposition. Miller and Duke (1961) stated that severely infected pods grown in the field had small, brown lesions giving them a speckled appearance. They also stated that fungi and bacteria attack dead tissue of the peg and fruit and, under certain conditions, cause peg rot and seed decay. Reaction differences noted by the various researchers may be related to differences of infecting microorganisms, type of peanut or cultivar. Good *et al.* (1958) noted that lesions were less conspicuous on Virginia-type peanut than on Spanish and Runner types. Minton *et al.* (1970) found that lesions were not as conspicuous on pods of Virginia Bunch 67 and Georgia 186–26 (Virginia type) as on Florunner, Early Runner (Runner type), Argentine and Starr (Spanish type). All cultivars were equally infected by *P. brachyurus* as determined by the number of nematodes recovered from shells and pegs. *P. brachyurus* feeding within the pegs weakens them resulting in pod loss at harvest (Good *et al.*, 1958; Boswell, 1968; Jackson & Sturgeon, 1973). Good *et al.* (1958) reported that the microorganisms that colonize damaged pods may penetrate the shell and damage the seed, thus the yield, as well as quality and value of the crop may be reduced.

Survival and means of dissemination

P. brachyurus infects roots, pegs, and pods of peanut and, because it is able to withstand extremes in temperature and moisture, it may survive in the Southeastern USA in these dead tissues during the winter (Graham, 1951; Good *et al.*, 1958; Feldmesser & Rebois, 1965). In South Africa, Keon (1967) working with potato and maize, found that at the end of winter 66.1% of *P. brachyurus* were found in the soil organic matter although the organic matter constituted only 0.29% of the soil. Boswell (1968) recovered *P. brachyurus* from peanut shells that were stored at 24°C for three, six and 28 months. *P. brachyurus* is a polyphagous nematode and may survive and overwinter in live roots of many winter crops and weeds as well as in dead tissues.

P. brachyurus may be disseminated in many of the same ways as *Meloidogyne* species. Since this is a migratory parasite and attacks most underground plant structures, it can be transported in infected roots and other underground plant parts in the soil. Generally, the major method of spread is by human activity involving movement of plant material, soil and tillage equipment. Peanut shells used as mulch or ground and used as diluents in certain preparations may carry the live nematodes (Good *et al.*, 1958; Colbran, 1968). Also, water movement across the field as the result of either rainfall or irrigation may transport the nematode.

Environmental factors affecting parasitism

The distribution and parasitism of *P. brachyurus* is temperature related and it is restricted to the warmer zones of the world (Loof, 1964). Boswell (1968) found in controlled temperature studies that reproduction in root and shell tissue of peanut was greatest at 26°C. Soil types may also affect the parasitism of peanut by *P. brachyurus* (Endo, 1959; Boswell, 1968).

Soil moisture affected reproduction of *P. brachyurus* on peanut (Good & Stansell, 1965). Approximately ten times more nematodes were recovered from shell tissue of irrigated than from non-irrigated peanut.

Disease complexes

Good *et al.* (1958) suggested the possibility of a disease complex involving *P. brachyurus* and soil microorganisms that would produce a peg rot. They frequently found *P. brachyurus* and *Sclerotium rolfsii* occurring together as pathogens. Boswell (1968) found lesions of peanut pods to contain both *P. brachyurus* and mycelium of fungi, most notably *Rhizoctonia solani, Fusarium* spp. and *Penicillium* spp.

There is some indication that presence of *P. brachyurus* is related to an increase of *Aspergillus flavus* in peanut shells but not in seeds (Jackson & Minton, 1968). Jackson and Sturgeon (1973) reported that the lesion nematode feeds on the peanut root, pod, and peg, allowing fungi and bacteria to enter damaged cells, causing a peg and pot rot. They stated further that the peg is weakened or rots away and allows the mature pod to shed or to be lost during harvest.

Economic importance and population damage threshold levels

P. brachyurus damage often is overlooked. Consequently, damage estimates for this nematode may be low since it has been reported in a large percentage of the peanut production areas in the USA and in other countries.

Minton *et al.* (1963) reported *Pratylenchus* spp. in 37% of the peanut fields sampled in Alabama, USA. In a later survey Ingram and Rodríguez-Kábana (1980) found them in 83.9% of the Alabama fields they surveyed. Alexander (1963) found *P. brachyurus* spp. in two out of fourteen fields surveyed in South Carolina, and Motsinger *et al.* (1976) reported them in 16.9% of the 331 fields surveyed in Georgia. Wheeler and Starr (1987) found *P. brachyurus* in 15.7% of the samples collected from peanut fields in a five county survey in Texas. Severe damage to peanut by *Pratylenchus* spp. has also been reported in Florida (Dickson & Smart, 1971) and Arkansas (Jackson & Sturgeon, 1973). In Egypt, Oteifa (1962) found *P. brachyurus* in 81.2% of the peanut fields, but in a later survey, Ibrahim and El-Ṣaedy (1976*a*) found them in only 9.6% of their samples. *P. brachyurus* occurs in peanut fields in a variety of soils in South Burnett, Australia (Colbran, 1968); it is also widespread throughout Atherton Tablelands in North Queensland, Australia and was absent only in soils that had recently been brought into cultivation (Broadley, 1981). Singh (1972) found *Pratylenchus* spp. in 50% of the samples collected from peanut fields in Guyana.

Production losses due to *Pratylenchus* spp. were estimated for several states in the USA (Anon., 1987). The percentage losses for the various states was as follows: Alabama, 0.1%; North Carolina, 0.5% for 1984 and 0.25% for 1985; Texas, 2.0% and Virginia, trace.

Population damage thresholds for *P. brachyurus* have not been well defined. Numbers of *P. brachyurus* per g of shell have been correlated with yield (Good *et al.*, 1958; Boswell, 1968; Minton & Morgan, 1974). Boswell (1968) obtained significant yield increases in fumigant nematicide treated plots in which there were 242 or less *P. brachyurus* per g of shell compared to the untreated plots that had 2771 per g of shell. Minton and Morgan (1974) obtained a significant yield increase in fumigated plots in which there were 127 *P. brachyurus* per g of shell compared to 2280 per g of shell in untreated plots.

Control measures

Peanut yield losses due to *P. brachyurus* is relatively small in relation to the amount of infested area. Hence, control of this nematode in peanut has not been practiced extensively except in certain areas that have severe infestations and crop losses.

Cultural practices

Generally, crop rotations for control of *P. brachyurus* in peanut are not effective because of the wide host range of crops and weeds and because there are few alternative cash crops for use in rotations with peanut (Endo, 1959; Koen, 1967; Porter *et al.*, 1984).

Good *et al.* (1954) found that population levels of *P. brachyurus* were greater in maize than in peanut in a maize-peanut rotation. *P. brachyurus* were also present in the soil in rotations that included lupine (*Lupinus hirsutus* L.), oats (*Avena sativa* L.) and native grass cover but greater numbers were present in lupine than in oats and native grass (Good *et al.*, 1954). Brodie and Murphy (1975) in U.S.A. found that fallowing for six weeks (May-June) or nine months (May-March) reduced populations of *P. brachyurus* in the soil to zero or near zero.

Good *et al.* (1958) observed that timely harvesting removed more *P. brachyurus* infested pods from the field than late harvesting, hence fewer nematodes were left in the soil to infect subsequent crops. Boswell (1968) also found that timely harvesting increased yield and value of peanuts compared to late harvesting. Good and Stansell (1965) reported that the larger yields from *P. brachyurus* infested soil were from irrigated peanut grown in fumigated soil and harvested earlier than normal for non-irrigated peanut. Although, irrigated peanut yielded more, *P. brachyurus* were ten times more numerous in shell tissue in irrigated than in non-irrigated plots.

Resistance

No commercial peanut cultivar possesses useful levels of resistance to *P. brachyurus*. Minton *et al.* (1970) reported six cultivars of peanut to be equally infected with *P. brachyurus*, but lesion symptoms were not as conspicuous on two of them. Smith *et al.* (1978) reported resistance in two plant introductions, PI290606 and PI295233. Starr (1984) later confirmed their findings and reported an additional resistant plant introduction, PI365553.

Chemical

Severe infestations of *P. brachyurus* can reduce yields by as much as several hundred kilograms per hectare. Where severe infestations occur, chemical treatments may be justified. Nematicides that control *Meloidogyne* species also control *P. brachyurus* (Good & Stansell, 1965; Boswell, 1968; Jackson & Sturgeon, 1973; Minton & Morgan, 1974).

Methods of diagnosis

Sampling

Assays of both soil and subterranean plant parts should be made to assess population levels of *P. brachyurus*. Soil samples should be collected with a large sampling tube (5.0 cm ID) in order to also obtain roots. Alternatively, one may collect soil samples with a smaller sampling tube and collect peanut pods from several plants at harvest and assay shells. Shells usually yield more *P. brachyurus* per unit weight of tissue than roots. Soil samples should be collected shortly before or after harvest when soil populations are greatest. Bioassays to establish the level of infestation (Boswell, 1968) may be useful if samples are collected during the winter or early spring when population levels are low.

Extraction

P. brachyurus adults and juveniles may be extracted from roots by incubating roots in a mist chamber and from soil using standard laboratory procedures (Chapter 2).

Belonolaimus longicaudatus

The sting nematode, *Belonolaimus longicaudatus*, occurs in sandy soils along the Atlantic Coastal Plain from Connecticut and New Jersey to Florida and Westward to Texas, Oklahoma, Arkansas, and Kansas in the United States. Although *B. longicaudatus* has been associated with peanut in most of the peanut-producing states (Owens, 1951; Holderman, 1955; Rau, 1958; Wheeler & Starr, 1987), loss estimates were reported for only Virginia, North Carolina and Oklahoma (Anon., 1987). Cooper *et al.* (1959) reported that *B. longicaudatus* was known to be distributed in sixteen counties in North Carolina with eight of these being major peanut producing counties. In Virginia, *B. longicaudatus* is a serious problem in less than 5% of the peanut fields (P. M. Phipps, pers. comm.). *B. longicaudatus* has not been reported on peanut outside the United States.

Symptoms

B. longicaudatus feeds ectoparasitically at root tips and along the sides of succulent roots as well as on young pegs and pods. Small necrotic lesions may be observed on the roots, pegs and pods (Owens, 1951). Esser (1976) reported that, shortly after being fed up on, root tips swell slightly and some asymmetric recurving occurs. Heavy infestations may cause gnarled and stubby lateral roots

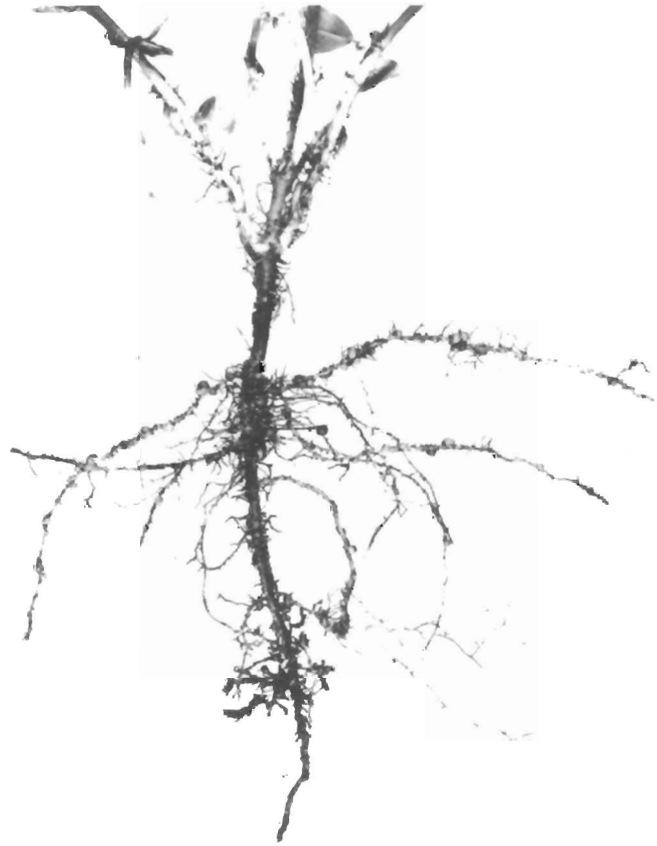

Fig. 10. Peanut plant with root system greatly reduced by *Belonolaimus longicaudatus* (Photo: J. N. Sasser).

and frequently only the taproot is left (Owens, 1951) (Fig. 10). Above ground symptoms include stunting and chlorosis. Peanut growth may be uneven in heavily infested fields and erratic stands may occur. Yield and quality of peanut may be severely reduced.

Biotypes

Rau (1958) described *B. longicaudatus* from Florida and suggested that it was probably the common sting nematode of the southeastern United States. Since Rau's publication, *B. longicaudatus* has been the species referred to most often on peanut. More recent investigations suggest that there are several pathotypes of physiological races or, perhaps, species. This nematode has been reported to be pathogenic on peanut in North Carolina and Virginia (Owens, 1951; Cooper *et al.*, 1959), but not in Georgia (Good, 1968). A population of *B. longicaudatus* from Gainesville, Florida did not cause damage or reproduce well on peanut, whereas a population from Sanford, Florida did (Perry & Norden, 1963).

Robbins and Hirschmann (1974) compared three populations of *B. longicaudatus* from each of Georgia and North Carolina and concluded that the Georgia and North Carolina nematodes were of different species and that neither were *B. longicaudatus* as described by Rau (1958). Their conclusions were based on differences in host range, morphology, and apparent infertility of inter-population offspring (Robbins, 1972; Robbins & Barker, 1973). However, the systematics have not yet been revised.

Survival and means of dissemination

The limited distribution of *B. longicaudatus* suggests that certain biological and environmental restraints affect its dispersal. Research results suggest that soil texture, soil temperature and soil moisture are critical to his reproduction (Perry, 1965; Robbins & Barker, 1974). *B. longicaudatus* can be readily established in new areas that have the required environmental conditions. It may move from one location to another by any means that will transport infested soil such as farm equipment, animal feet, water and transplants to which soil is attached.

Environmental factors affecting parasitism

The limited distribution of *B. longicaudatus* suggests that its ecological requirements may be very specific. Thames (1959) postulated that fine-textured soil inhibits its movement and reproduction. In Virginia, Miller (1972*a*) found *B. longicaudatus* only in the "A"-horizon of soils with a sand content of 84–94%.

Soil temperature and moisture also affect the reproduction and survival of *B. longicaudatus*. Reproduction was greatest at 25–30 °C (Robbins & Barker, 1974) which agreed with results obtained by Perry (1965). Perry observed that reproduction was greater for *B. longicaudatus* at 29.4°C than at 26.7°C and was greatly reduced at 35°C. Boyd and Perry (1969) concluded that this nematode either died or migrated downward when soil temperature at 2.5 cm below the bare soil surface reached 39.5°C or higher. Robbins and Barker (1974) found the optimum soil moisture for reproduction to be 7%.

Economic importance and population damage threshold levels

Economic losses for peanut in the USA due to *B. longicaudatus* are not great despite the extreme damage this nematode inflicts. Losses due to this nematode have been reported for only North Carolina (0.30%), Oklahoma (0.25%) and Virginia (0.50%) (Anon., 1987). Yields were increased as much as 400% in North Carolina (Cooper *et al.*, 1959) in nematicide treated plots compared to untreated soil in which the average population density of *B. longicaudatus* was approximately 50 per 473 cm^3 of soil from 9 June to 30 October. Sasser *et al.* (1960) also in North Carolina, obtained a yield increase of 109% with a nematicide in soil in which population levels ranged from 135 to 205 per 473 cm^3 soil in the untreated control during the growing season.

Fig. 11. A peanut field in North Carolina infested with *Belonolaimus longicaudatus*. Centre row was untreated; rows to right and left of centre were treated with different nematicides (Photo: A. W. Johnson).

Control measures

No commercial peanut cultivar is resistant to *B. longicaudatus*. The nematode has a wide host range and only a few crop plants such as small grain, tobacco (*Nicotiana tabacum* L.) and watermelon (*Citrullus vulgaris* Schrad.) have reduced population densities when grown in rotation with peanut (Holdeman & Graham, 1953; Bailey, 1988). The use of nematicides is the major means of control (Fig. 11). Both fumigant and non-fumigant nematicides have given excellent control and increased peanut yields (Cooper *et al.*, 1959; Sasser *et al.*, 1960; 1975*b*).

Methods of diagnosis

Sampling

Plant damage symptoms for *B. longicaudatus* may occur in the seedling stage of the peanut, especially if population levels are high. Examination of roots of seedlings for damage as well as assessment of population densities in the soil are recommended. Soil samples should be collected using procedures recommended for recovery of most ectoparasitic nematodes (Chapter 2).

Extraction

The extraction of *B. longicaudatus* from the soil may be done using one of a number of standard extraction procedures (Chapter 2).

Determining the relationship of populations to crop loss

The effects of *B. longicaudatus* on peanut is reflected in plant growth, yield and quality (Cooper *et al.*, 1959; Sasser *et al.*, 1975*a*). Significant negative correlations of number of nematodes in the soil with yield and growth may be obtained during most of the growing season.

Criconemella ornata

The ring nematode, *Criconemella ornata*, was first reported associated with peanut in Georgia (Boyle, 1950; Machmer, 1953). It is now known to occur in a large percentage of the peanut production regions of the United States (Minton *et. al.*, 1963; Alexander, 1963; Motsinger *et al.*, 1976; Ingram & Rodríguez-Kábana, 1980; Wheeler & Starr, 1987). *Criconemella* species have been reported in Burkina Faso (Germani & Dhéry, 1973), Egypt (Ibrahim & El-Saedy, 1976*a*) and Gambia (Merny *et al.*, 1974).

Symptoms

Machmer (1953) described a chlorotic condition of peanuts growing in Georgia in soil heavily infested with a species of *Criconemella* which he called "Peanut Yellows". Although he did not identify the species, it was probably *C. ornata*. Barker *et al.* (1982), reported that freshly extracted, greenhouse-grown inoculum caused the typical "Yellows disease" on peanut grown in microplots. As few as 178 freshly introduced *C. ornata*/500 cm^3 soil stunted peanuts. Roots, pods and pegs of peanut plants growing in microplots in soil heavily infested with *C. ornata* were severely discoloured with brown necrotic lesions (Fig. 12) (Minton & Bell, 1969). Small necrotic lesions were often superficial, but necrosis in large lesions usually extended deep into the tissues. Many lateral roots primordia and young roots were killed, resulting in reduced numbers of lateral roots. Pod yields from nematode-infected plants were reduced by about one-half.

Survival and means of dissemination

Information relative to factors affecting survival of *C. ornata* is limited. Little has been done to determine soil type preference, but survey results suggest it favours the lighter soils (Barker, 1974). Population levels decline rapidly in the presence of poor hosts. Since *C. ornata* is an ectoparasite,

Fig. 12. Lesions on peanut pods caused by *Criconemella ornata*. (From Minton and Bell., 1969.)

dispersal occurs primarily in soil transported on farm equipment, feet of animals, in water and in soil clinging to transplants.

Environmental factors affecting parasitism

The environmental factors affecting the parasitism of peanut by *C. ornata* has received little attention. Barker (1974) reported that the previous crop as well as geographic area in the state of North Carolina affected the occurrence and activity of *Criconemella* spp. The Coastal Plain, with warm, sandy soils, had a greater abundance of *Criconemella* spp. than the Piedmont and Mountain areas with soils that are cooler and contain more loam and clay. The frequencey of occurrence of *Criconemella* spp. on peanut (54%) was greater than for any other crop.

Disease complexes

Greenhouse studies in North Carolina revealed an interaction (enhancement of *Cylindrocladium* black rot, CBR) between *Cylindrocladium crotalariae* on CBR-susceptible Florunner but not on CBR-resistant NC 3033 peanut cultivars (Diomandé & Beute, 1981*a*). The severity of CBR on Florunner was increased when the density of *C. ornata* was 10^4 per 15-cm-diameter clay pot and *C. crotalariae* was 0.25 and 2.5 microsclerotia per cm^3 soil. Significant positive correlations between *C. ornata* and *C. crotalariae* and CBR indicated that this nematode can affect CBR development in the field (Diomandé & Beute, 1981*b*).

Economic importance and population damage threshold limits

Damage to peanut due to *C. ornata* in the field is subtle and low levels of damage may go undetected. Also, *C. ornata* is seldom present alone, but usually occurs in polyspecific communities. Therefore, losses due to *C. ornata* have not been well defined. Pod yield in a microplot experiment (Minton & Bell, 1969) was reduced by about one-half in heavily inoculated soil. In a field experiment in which the soil was infested with five genera of nematodes in addition to *C. ornata*, population densities of *C. ornata* were negatively correlated with peanut growth index and pod yield (Sasser *et al.*, 1975*a*).

Based on a linear regression model, Rickard *et al.* (1977) determined that peanut yield loss in microplots was 18.7% for each ten-fold increase in initial populations of *C. ornata* in the soil. Barker *et al.* (1982) found that as few as 178 *C. ornata*/500 cm^3 of soil in a microplot experiment caused a significant yield loss. In a second microplot experiment (Barker *et al.*, 1982), the *C. ornata* that reproduced the previous year on tobacco (a poor host) did not affect peanut yield. These researchers concluded that many of the nematodes present in the soil in the spring following tobacco may have been dead since tobacco is a poor host. Therefore, the previous host may affect the infectivity of the nematodes present in the soil resulting in an important problem for nematode advisory programmes.

Control

Since losses due to *C. ornata* have not been well defined, recommendations for control of this nematode when present as the primary pathogen are seldom made. Also, there is no known resistant commercial peanut cultivar. Certain crops such as cotton, soybean, corn and sorghum grown in rotation with peanut may reduce population levels (Good, 1968; Johnson *et al.*, 1974; Kinloch & Lutrick, 1975). Nematicides, both fumigant and nonfumigant, are effective against this nematode (Minton & Morgan, 1974).

Methods of diagnosis

Sampling

Evaluating soil population densities is the major means of diagnosing possible *C. ornata* damage to peanut. Soil samples should be collected using procedures recommended for recovery of ectoparasitic nematodes (Chapter 2).

Extraction

C. ornata may be extracted from the soil using one of several methods but the modified centrifuge-flotation method is, perhaps, the best for this nematode.

Determining the relationship of populations to crop loss

Even though *C. ornata* is a weakly, pathogenic nematode, negative correlations of population densities with yield and plant growth often suggest plant damage (Minton & Morgan, 1974; Sasser *et al.*, 1975*a*). Soil assays made early in the season (55–73 days after planting) may be more meaningful than assays made near harvest (Sasser *et al.*, 1975*a*).

Aphelenchoides arachidis

Aphelenchoides arachidis, the testa nematode, was described from northern Nigeria on peanut (Bos, 1977*a*; 1977*b*). It has been found at a significant level of infestation in only a limited area around Samaru. It was also found at a low level of infestation in peanut at Kadawa and in one peanut sample from Gwoza. It is not known to be a pest of peanut outside of Nigeria.

Symptoms

A. arachidis is a facultative endoparasite of peanut that parasitizes the tissues of the pods, testas, roots and hypocotyls, but not the cotyledons, embryos, or other parts of the plant (Bos, 1977*a*; Bridge *et al.*, 1977) (Fig. 13). Seed coats were discoloured when more than 2000 *A. arachidis*/testa were present (Bridge *et al.*, 1977) (Plate 6E). Heavily infested seeds, examined immediately after removal from fresh, mature pods, are a light brown, have translucent testas, and dark vascular strands within the testas. After infested seeds are dried, testas are often wrinkled and are darker brown than non-infested seeds (Plate 6E). Nematodes are found mainly in the sub-epidermal parenchymatous layer, and around the tracheids of the testa. Testas infested with *A. arachidis* are thicker and more uneven than normal testas. Nematodes are found in sub-epidermal parenchyma cells where walls are broken and cells enlarged. The epidermal layer of the seed coat is reduced in infested testas and the basal tissues, including the aleurone layer, is disorganized. Infested seeds of cultivar Spanish 205 weighed less than healthy seeds, but nematode damage had little effect on seed germination.

Biology and life cycle

A. arachidis is a facultative endoparasite of the seed, testa, pod shells, roots and hypocotyl of peanut (Bridge *et al.*, 1977). It has also been observed feeding ectoparasitically on roots and on two fungi, *Macrophomina phaseolina* and *Botrytis cinerea* associated with seeds on agar plates. *A. arachidis* were found in the parenchymatous tissues of the testa, root cortex and hypocotyl, but not in the central stele or vascular bundles (Bridge *et al.*, 1977). Pods had been invaded 10 days after the fruiting pegs had penetrated the soil, but numbers of nematodes in pods did not increase rapidly until after 30 days with largest numbers present at about day 60. All stages of the nematode, including eggs, were found throughout the testas, but at the end of the growing season, heavily infested testas of mature seeds contained mainly juvenile stages with few adults. Testas showing no external symptoms contained mostly adults and eggs, often arranged along the vascular elements of the seed coats.

Biotypes

Bos (1977*b*) suggested that there are two biotypes of *A. arachidis*, one occuring on cereals and one on both cereal and peanut.

Survival and means of dissemination

A. arachidis survived desiccation in stored peanut pods for 12 months (Bridge *et al.*, 1977). All juvenile stages were extracted from dried testas and shells with no particular stage predominating,

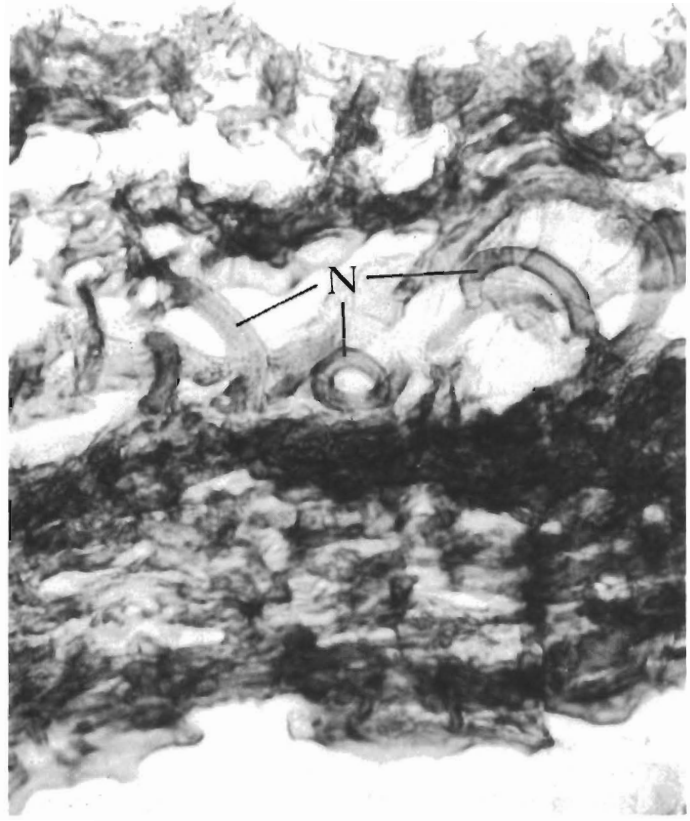

Fig. 13. Transverse section of peanut testa infected with *Aphelenchoides arachidis* (N = nematodes) (From Bridge *et al.*, 1977).

but adults were found alive only occasionally in either testas or shells of stored pods. No active nematodes were extracted from infested pods sun-dried in the field before storage. Volunteer plants in an infested field contained many adult nematodes suggesting that they continue to develop to maturity under natural conditions in pods left in the ground during the dry season in Nigeria. Unless appropriate precautions are taken, *A. arachidis* may become a serious pest world-wide since it can be disseminated in infested seeds (Bridge *et al.*, 1977).

Disease complexes

Aphelenchoides arachidis infestation of peanut seeds in field experiments predisposed seeds to invasion by fungi (McDonald *et al.*, 1979), Nematode infested seeds had higher levels of fungal infection (*Rhizoctonia solani, Sclerotium rolfsii, Macrophomina phaseolina* and *Fusarium* spp.) than the visually nematode-free seeds. Both rates of seedling emergence and total emergence were slightly lower for nematode-seeds than for clean seeds.

Economic importance and population damage threshold levels

A. arachidis devalues the confectionery peanut because it causes shriveled and discoloured seeds (Bridge *et al.*, 1977). Severe infestation of peanuts with *A. arachidis* not only has an adverse effect on the appearance and size of seed, but it may also predispose seeds to invasion by fungi which may lead to reduced seed emergence (McDonald *et al.*, 1979). It has not been shown to decrease yields.

Because of its limited distribution (Nigeria), *A. arachidis* has not caused major economic loss, but if it should become established in other peanut-producing regions of the world, it could possibly become a major economic pest.

Control

Only limited information is available on the control of *A. arachidis* on peanut. No field applied treatments have been reported, but a number of preventative measures are effective against further spread of the nematode. Immersing seeds in four times their volume of water heated to 60°C and allowing to cool for 5 minutes gave complete control of the nematodes without affecting germination (Bridge, 1975; McDonald & Misari, 1976; Bridge *et al.*, 1977). Sun drying the pods after harvest in very dry conditions as occurs in northern Nigeria, reduces the number of nematodes in the pods (Bridge *et al.*, 1977). In more humid areas, sun drying of pods may not be effective. Shelling peanut before planting will also eliminate the tissues in which most of the nematodes occur and in which they survive best (Bridge *et al.*, 1977).

Aphasmatylenchus straturatus

A. straturatus found around the roots of peanut in southwest Burkina Faso, West Africa near Niangoloko village was described in 1970 (Germani, 1970). It has not been reported to occur outside of Burkina Faso.

Symptoms

A. straturatus causes interveinal chlorosis, stunting, a poorly developed root system, reduction of *Rhizobium* nodules on the roots and peanut yield reduction (Germani & Dhéry, 1973; Germani & Luc, 1982*a*; 1982*b*).

Biology and life cycle

A. straturatus is a migratory endo/ectoparasitic nematode on peanut. Field observations indicate that it spends the dry season at a depth of 40 to 60 cm in the soil adjacent to roots of the karite (*Butyrospermum parkii* L.) tree or in the roots of this tree.

Peanuts are interplanted with the karite tree in many fields in Burkina Faso. Therefore, at the beginning of the rainy season, the nematodes move from the tree roots and enter peanut roots. The nematodes are most abundant in the peanut roots about 40 days after seeding the early maturing cultivars and 70 days after seeding the late maturing cultivars. Approximately 100–110 days after seeding, the nematode leaves the peanut roots and returns to roots of the karite tree. *A. straturatus* does not enter into anhydrobiosis.

Economic importance and population damage threshold levels

Yield reductions due to *A. straturatus* were estimated to range from 30 to 70%. In 1971, *A. straturatus* was estimated to infest approximately 4% of the peanut production area of Burkina Faso and in 1974 the estimate had risen to 25%. Since this nematode also parasitizes other economically important leguminous plants grown in Burkina Faso (Germani & Dhéry, 1973), its rapid spread poses a threat to peanut and other legumes.

Disease symptoms may occur in the field when as few as 600 nematodes per dm^3 of soil are present, but approximately 2000 nematodes per dm^3 are required in the greenhouse.

Control

Research to control *A. straturatus* on peanut has been limited, however DBCP applied at planting has given satisfactory control (Dhéry *et al.*, 1975).

Methods of diagnosis

Soil samples for nematode assays must be collected in the root zone of peanut or in the root zone of the karite tree during the dry season. If the samples are collected in the root zone of peanut they should be removed from the 0 to 20 cm depth, but if collected in the root zone of karite tree during the dry season, they should be removed from the 40 to 60 cm depth.

Scutellonema cavenessi

S. cavenessi was described from northern Nigeria (Sher, 1964) but has since been found associated with most cultivated plants in Senegal and Mali. In Senegal, *S. cavenessi* was associated with poor growth of peanut (Germani, 1979*b*; 1981*b*).

Symptoms

Foliage of peanut plants grown in soil infested with *S. cavenessi* were chlorotic (Germani, 1979*b*). *Scutellonema cavenessi* is associated with the reduction of number of lateral roots and *Rhizobium* nodules. Chlorosis was reduced in plots treated with DBCP which also reduced population densities of *S. cavenessi*. Chlorosis was associated with a reduced level of nitrogen fixation and less total nitrogen yield in pods and foliage (Germani, 1979*b*). Application of the fumigants, DBCP and EDB, to infested soil reduced the nematode population densities, increased vine and pod yield, the number and weight of *Rhizobium* nodules, the nitrogen and phosphorus content of foliage and seeds, and the level of endomycorrhyzae infestation (Germani, 1979*b*; 1981*b*; Germani *et al.*, 1981; 1982; 1985; Germani & Reversat, 1982; 1983).

Biology and survival

In Senegal, *S. cavenessi* showed seasonality in activity (Demeure, 1978*a*; Demeure *et al.*, 1980). This nematode is active during the rainy season, but as the dry season progresses and the humidity of the soil drops to approximately 0.2%, nematodes 0–25 cm deep in the soil enter into a state of anhydrobiosis, in which they remain until the next rainy season.

Economic importance and population damage thresholds

S. cavenessi is distributed throughout the peanut production area of Senegal, but the extent of the crop loss has not been fully evaluated. Nevertheless, in experimental plots, nematicides have increased yields of pods from 20% to 220% and vines 40% to 270% (Germani *et al.*, 1985).

Control

There are no known cultivars resistant to *S. cavenessi*. Also, all crops grow in rotation with peanut in the Sahelian zone of Senegal are susceptible to this nematode. Bare fallow between crops of peanut provided excellent control (Duncan, 1986) but because of the high cost, this practice is not practical in the Sahelian zone. Ethylene dibromide and DBCP are the only nematicides tested that have given practical control. These materials used at 20 kg per hectare of active ingredient have given excellent control and yield increases (Germani & Gautreau, 1976; Germani, 1979*a*; 1979*b*; 1981a; Duncan & Baujard, 1986; Baujard *et al.*, 1987). Growth and yield differences due to nematicides are shown in Plate 6F. There is also a residual effect of the nematicide on other crops grown in treated fields the following year. Ethylene dibromide and DBCP injected at an optimal depth of 15 cm at planting and up to 30 days after planting do not cause phytotoxicity. The fumigant nematicides are applied in or near the row with an animal drawn injector metered with a ground driven peristalic pump that applies a uniform rate as the apparatus is drawn across the field.

Methods of diagnosis

Soil samples for nematode assays should be collected in the peanut root zone to a depth of 25 cm using standard sampling and extraction techniques (Chapter 2). However, if samples are taken

during the dry season when the nematode are in the anhydrobiotic state, samples should be moistened before extraction by elutriation or Baermann techniques, or the centrifugation-flotation method should be used (Demeure, 1978*b*; Duncan, 1986; Duncan & Baujard, 1986).

Tylenchorhynchus brevilineatus

Tylenchorhynchus brevilineatus was first observed damaging peanut in 1976 in the Kalahasti area of Andhra Paradesh State, India (Reddy *et al.*, 1984). The disease caused by this nematode is known as "Kalahasti Malady". Since 1976, the disease has been widespread in the Kalahasti area and has also been observed in Nellore District in Andhra Pradesh (Reddy *et al.*, 1984). This nematode has not been reported as a pest of peanut in other parts of the world.

Symptoms of damage

The disease symptoms in farmers' fields (Reddy *et al.*, 1984) are characterized by small pods and a brownish-black discolouration of pod surface. Small, brownish-yellow lesions appear on the pegs and pod stalks and on young, developing pods. Margins of lesions are slightly elevated because of host cell proliferation around lesions. The length of pod stalks are greatly reduced, and in advanced stages of the disease, the pod surface becomes completely discoloured, but seeds from diseased pods are healthy. Discolouration is also observed on roots but is less severe than on pods.

Pathogenicity tests in the greenhouse corroborated field observations (Reddy *et al.*, 1984). Peanut plants inoculated with 500 *T. brevilineatus* per 12-cm-diameter pot were severely stunted and had reduced root systems. Lesions were present on the roots but were not extensive. Pods were severely discoloured and small, but seeds from the discoloured pods were healthy. Brownish-yellow lesions were observed on individually inoculated pods after 15 days. The number of lesions increased and extensive discolouration was observed by 30 days after inoculation.

Control

Aldicarb (10G) and carbofuran (3G) applied to peanut 20 days postplant controlled *T. brevilineatus* at 2.0, 4.0, 6.0 and 8.0 kg ai/ha. These treatments reduced soil population densities of *T. brevilineatus* and the percentage of diseased pods (Reddy *et al.*, 1984). These treatments increased plant height, pod yields and pod and kernel weights. Both materials were more effective at the higher than at the lower rates.

Ditylenchus destructor

D. destructor, the potato rot nematode, was first reported damaging peanut in the Transvaal Province of South Africa in 1987 (Jones & De Waele, 1988). A subsequent survey revealed the presence of this nematode in seven major peanut producing regions (De Waele *et al.*, 1988). Seventy-three percent of 877 seed samples that graded "damaged" were infested. An average of 160 nematodes per seed was recovered. This nematode has not been reported on peanuts in other parts of the world.

Symptoms of damage

D. destructor has been isolated from roots, pegs, shells and peanut seeds (De Waele *et al.*, 1988). Infected pods of cv Sellie were black resembling black hull caused by *Chalara elegans*. Approximately 40 to 60 percent of the pods and seeds were destroyed in heavily infested fields. *D. destructor* was present in both hulls and seeds.

In greenhouse pathogenicity tests (De Waele *et al.*, 1988), nematodes were present in the peg, exocarp, and endocarp, testa, embryo and on the cotyledons. The first symptom to develop was brown necrotic tissue at the pod base at the juncture of the peg and pod. The surface of infected tissue was dark brown and had a corky appearance. The most distinct symptom of advanced disease

was dark brown to black discolouration of veins which entended longitudinally in the exocarp just beneath the pod surface. Infected pods lacked the luster of healthy pods and appeared dead. Infected seeds were usually shrunken and the micropyles were dark brown to black. The testas were flacid, had dark vascular strands and were easily removed. The inner layer of the testa had a distinct yellow discolouration. Infected embryos were usually olive green to brown instead of having the normal colourless to yellow appearance. The extent of losses caused by this nematode and research results relative to its control have not been reported.

Other Nematodes

Sharma (1985) compiled a world list of nematode pathogens associated with peanut. The list is extensive and includes many genera and species that have not been proven to cause economic damage to peanut. Additional research may demonstrate that some of these species are, in fact, pathogenic and pose a serious threat to peanut production, while others may feed on peanut but cause no economic damage.

The possibility of the interaction of two nematodes, not considered serious pests of peanut, with a virus has been suggested. In Senegal, Merny and Mauboussin (1973) eliminated the clump disease of peanut caused by a virus by treating the soil with DD. They suggested that one or more nematodes was acting as a vector and pointed out that *Longidorus siddiqii* was present in soil samples. More recently, Singh and Sakhuja (1984) in India reduced the disease in field experiments by 97% and 84.1% with DBCP (45 l/ha) and aldicarb 3.0 kg a.i./ha, respectively. Soil samples collected from the rhizosphere of diseased plants always contained *Paralongidorus citri*. Occasionally, species of *Helicotylenchus, Tylenchorhynchus*, and *Hoplolaimus* were also present.

Conclusions and Future Prospects

Peanut yield losses due to parasitic nematodes occur in every major peanut production region of the world. If we can accept the estimated loss of 12% (Sasser & Freckman, 1987), it is apparent that losses are substantial and efforts to reduce these losses are needed.

Meloidogyne species are the major nematodes damaging peanut in most regions of the world, but in some regions, as in West Africa, other species may be more serious. In Senegal, for instance, *Meloidogyne* species do not damage peanut and peanut is often rotated with vegetables to suppress *M. arenaria* populations. A number of nematodes such as *A. arachidis, A. straturatus, S. cavenessi, T. brevilineatus* and *D. destructor* have been reported to cause serious damage to peanut in isolated areas of Africa and Asia but not in other areas of the world. *Belonolaimus longicaudatus* is a pathogen of peanut in only certain areas of the United States. Questions may be raised as to why these nematodes have been reported damaging peanuts only in these areas and what is the probability of their becoming pests in other regions of the world.

Nematode management in the past, particularly in industrialized countries has been based to a great extent on chemical control. In these countries, loss of the fumigants, DBCP, EDB and DD, for use on peanuts, the concern for environmental contaminants, and the increased cost of applying chemicals has increased the urgency to seek safer and more economical chemicals and to develop other means of control.

Ideally, nematode resistant cultivars would be the best and most economical means of control. Unfortunately, germplasm resistant to most nematode species that attack peanut has not been identified or has not been incorporated into commercial cultivars. Therefore, there is a need to accelerate research efforts in this area.

Expanded utilization of cultural practices such as crop rotations, cover crops, trap crops, fallow, flooding and organic amendments that reduce nematode damage may be necessary in the future to maintain economical peanut production. Efforts to prevent the spread of nematodes through sani-

tation and quarantine in extreme situations, may contribute to future containment of nematode problems.

Nematologists and advisors to growers in the future will be challenged to devise the most effective control measures that will yield quality peanuts and an economical return to the grower and protect the safety of the consumer and environment.

References

Alexander, P. M. (1963). Stylet-bearing nematodes associated with various plants in South Carolina, 1962–1963. *Plant Disease Reporter*, 47:978–982.

Anon. (1973). Plant nematology. In: *Secretary for Agriculture Report for the Period 1 October 1971 to 30 September 1972*. Salisbury, Rhodesia, 76 p.

Anon. (1987). Bibliography of estimated crop losses in the United States due to plant-parastic nematodes. *Annals of applied Nematology*, a supplement to the *Journal of Nematology*, 1:6–12.

Bailey, J. E. (1988). Peanut disease control. In: *North Carolina Agricultural Extension Service, North Carolina State University, Peanuts, 1988, AG–331*: 51–64.

Baldwin, J. G. & Barker, K. R. (1970). Host suitability of selected hybrids, varieties and inbreds of corn to populations of *Meloidogyne* spp. *Journal of Nematology*, 2:345–350.

Baltensperger, D. D., Prine, G. M. & Dunn, R. A. (1986). Root-knot nematode resistance in *Arachis glabrata*. *Peanut Science*, 13:78–80.

Barker, K. R. (1974). Influence of geographic area and previous crop on occurrence and density of plant-parasitic nematodes in North Carolina. *Plant Disease Reporter*, 11:991–995.

Barker, K. R., Schmitt, D. P. & Campos, V. P. (1982). Response of peanut, corn, tobacco, and soybean to *Criconemella ornata*. *Journal of Nematology*, 14:576–581.

Baujard, P., Duncan, L. W., Pariselle, A. & Sarr, E. (1987). Étude des effets de quatre nématicides fumigants sur les nématodes et l'arachide au Sénégal. *Revue de Nématologie*, 10:355–360.

Baxter, R. I. & Blake, C. D. (1969). Oxygen and the hatch of eggs and migration of larvae of *Meloidogyne javanica*. *Annals of applied Biology* 63:191–203.

Bergeson, G. B. (1959). The influence of temperature on the survival of some species of the genus *Meloidogyne* in the absence of a host. *Nematologica*, 4:344–354.

Bird, H. F. & Wallace, H. R. (1965). The influence of temperature on *Meloidogyne hapla* and *M. javanica*. *Nematologica*, 11:581–589.

Bos, W. S. (1977*a*). *Aphelenchoides arachidis* n. sp. (Nematoda: Aphelenchoidea) on endoparasite of the testa of groundnuts in Nigeria. *Zeitschrift für Pflanzenkrankheiten und Pflanzenschutz*, 84:95–99.

Bos, W. S. (1977*b*). A preliminary report on the distribution and host-range of the nematode *Aphelenchoides arachidis* Bos in the North of Nigeria. *Samaru agricultural Newsletter, 19:21–23.*

Boswell, T. E. (1968). *Pathogenicity* of Pratylenchus brachyurus *to Spanish peanut*. Ph.D. Dissertation. Texas A and M University College Station, 156 p.

Boyd, F. T. & Perry, V. G. (1969). The effect of sting nematodes on establishment, yield and growth of forage grasses on Florida sandy soils. *Proceedings of the Florida Soil Crop and Science Society*. 29:288–300.

Boyle, L. W. (1950). Several species of parasitic nematodes on peanuts in Georgia. *Plant Disease Reporter*, 34:61–62.

Bridge, J. (1975). Hotwater treatment to control plant parasitic nematodes of tropical crops. *Mededelingen van de Faculteit Landbouwwetendchappen Rijksuniversiteit Gent*, 40:249–259.

Bridge, J., Bos, W. S., Page, L. J. & McDonald, D. (1977). The biology and possible importance of *Aphelenchoides arachidis*, a seed-borne endoparasitic nematode of groundnuts from northern Nigeria. *Nematologica*, 23:253–259.

Broadley, R. A. (1981). Distribution and control of root-knot and lesion nematodes on peanuts in North Queensland. *Australian Journal of experimental Agriculture and Animal Husbandry. 21:223–226.*

Brodie, B. B. & Murphy, W. S. (1975). Population dynamics of plant nematodes as affected by combination of fallow and cropping sequences. *Journal of Nematology*, 7:91–92.

Castillo, M. B., Morrison, L. S., Russell, C. C. & Banks, D. J. (1973*a*). Resistance to *Meloidogyne hapla* in peanut. *Journal of Nematology*, 5:281–285.

Chabra, H. K. & Mahajan, R. (1976). *Pratylenchus coffeae*, the root-lesion nematode in groundnut and its control by granular nematicides. *Nematologia mediterranea*, 4:241–242.

Cheng, Y. H., Lo, W. C. & Tu, C. C. (1981). Studies on host range of *Meloidogyne arenaria* and its chemical control. *Research Bulletin of Tainan Dais*, 15:55–64.

Cheng, Y. H. & Tu, C. C. (1980). Identification of *Meloidogyne arenaria* of peanut on Taiwan. *Journal of agricultural Research of China*, 29:47–53.

Choi, Y. E. (1981). The root-knot nematodes, *Meloidogyne* spp., in Korea. In: *Proceedings of the 3rd Research Planning Conference on Root-knot Nematodes, Meloidogyne spp., Region VI, July 20–24, 1981*, Jakarta, Indonesia.

Colbran, R. C. (1958). Studies of plant and soil nematodes. 2. Queensland host records of root-knot nematodes (*Meloidogyne* spp.). *Queensland Journal of agricultural Science*, 15:101–136.

Colbran, R. C. (1968). Nematodes – important pests of peanuts. *Division of Plant Industry, Department of Primary Industries. Queensland, Australia, Advisory Leaflet No. 955*, 5 p.

Cooper, W. E., Wells, J. C., Sasser, J. N. & Bowery, T. G. (1959). The efficacy of preplant and postplant applications of 1,2-dibromo–3-chloropropane on control of the sting nematode, *Belonolaimus longicaudatus*. *Plant Disease Reporter*, 43:903–908.

De Waele, D., Jones, B. L., Bolton, C. & van den Berg, E. 1988. *Ditylenchus destructor* in hulls and seeds of peanut. *Journal of Nematology*, 21:10–15.

Demeure, Y. (1978*a*). Influence des températures élevées sur les états actifs et anhydrobiotiques du nématode *Scuetellonema cavenessi*. *Revue de Nématologie*, 1:13–19.

Demeure, Y. (1978*b*). Résistance à la sécheresse en zone sahélienne du nématode phytoparasite *Scutellonema cavenessi* Sher, 1963. *Cahiers de l'ORSTOM, Série Biologie*, 10:283–292.

Demeure, Y., Netscher, C. & Quénéhervé, P. (1980). Biology of the plant parasitic nematode *Scutellonema cavenessi* Sher, 1964: reproduction, development and life cycle. *Revue de Nématologie*, 3:213–225.

Dhéry, M., Germani, G. & Giard, A. (1975). Résultats de traitements nématicides contre la chlorose et le rabougrissement de l'arachide en Haute Volta. *Cahiers de L'ORSTOM, série Biologie*, 10:161–167.

Dhurj, I. & Vaishnav, M. U. (1981). Pathogenicity of root-knot nematode on groundnut. *Indian Journal of Nematology*, 11:217–218.

Dickson, D. W. & Smart, G. C., Jr. (1971). Control of *Meloidogyne arenaria* and *Pratylenchus penetrans* on peanut with foliar applications of a systemic nematicide. *Journal of Nematology*, 3:307–308.

Dickson, D. W. & Waites, R. E. (1978). Efficacy of at-plant and additional at-pegging applications of nematicides for control of *Meloidogyne arenaria* on peanut. *Proceedings of the American Peanut Research and Education Association*, 10:51 [Abstr.].

Dickson, D. W. & Waites, R. E. (1982). Evaluation of nematicides for managing the peanut root-knot nematode on peanut, 1980. *Fungicide and Nematicide Tests, American Phytopathological Society*, 37:195–196.

Diomandé, M. & Beute, M. K. (1981*a*). Effects of *Meloidogyne hapla* and *Macroposthonia ornata* on *Cylindrocladium* black rot on peanut. *Phytopathlogy*, 71:491–496.

Diomandé, M. & Beute, M. K. (1981*b*). Relations of *Meloidogyne hapla* and *Macroposthonia ornata* populations to *Cylindrocladium* black rot in peanuts. *Plant Disease*, 65:339–342.

Diomandé, M., Black, M. C., Beute, D. K. & Barker, K. R. (1981). Enhancement of *Cylindrocladium crotalaria* root rot by *Meloidogyne arenaria* (race 2) on a peanut cultivar resistant to both pathogens. *Journal of Nematology*, 13:321–327.

Duncan, L. W. (1986). Effect of bare fallow on plant parasitic nematodes in the Sahelian zone of Senegal. *Revue de Nématologie*, 9:75–81.

Duncan, L. W. & Baujard, P. (1986). Influence of nematicide placement depth and time of application on treatment efficacy on the Sahelian zone of Senegal. *Revue de Nématologie*, 9:135–139.

Dunn, R. A. (1988). Peanut nematode management. Florida Cooperative Extension Service, Institute of Food and Agricultural Sciences, University of Florida. *Nematode Plant Protection Pointer*, 3, 4 p.

Edwards, E. E. (1956). Studies on resistance to root-knot nematode *Meloidogyne* Goeldi, 1887. *Proceedings of the helminthological Society of Washington*, 23:112–118.

Eisenback, J. D., Hirschmann, H., Sasser, J. N. & Triantaphyllou, A. C. (1981). *A guide to the four most common species of root-knot nematodes (Meloidogyne species) with pictorial keys*. The Department of Plant Pathology and Genetics, North Carolina State University and USAID, Raleigh, NC, 47 p.

Endo, B. Y. (1959). Responses of root-lesion nematodes, *Pratylenchus brachyurus* and *P. zeae* to various plants and soil types. *Phytopathology*, 49:417–421.

Esser, R. P. (1976). Sting nematodes, devastating parasites of Florida crops. Division of Plant Industry, Florida Department of Agriculture and Consumer Service. *Nematology Circular*, 18:2 p.

Faulkner, L. R. & Bolander, W. J. (1966). Occurrence of large nematode populations in irrigation canals of South Central Washington. *Nematologica*, 12:591–600.

Feldmesser, J. & Rebois, R. V. (1965). Temperature and moisture effects on *Pratylenchus brachyurus*. *Nematologica*, 11:37–38.

Garcia, R. M. (1976). *Vertical distribution and survival stages of* Meloidogyne arenaria *in Florida with peanut as a host crop*. M. S. Thesis, University of Florida, Gainesville, 38 pp.

Garcia, R. & Mitchell, D. J.(1975*a*). Synergistic interactions of *Phythium myriotylum* with *Fusarium solani* and *Meloidogyne arenaria* in pod rot of peanut. *Phytopathlogy*, 65:832–833.

Garcia, R. & Mitchell, J. J. (1975*b*). Interactions of *Pythium myriotylum* with *Fusarium solani, Rhizoctonia solani*, and *Meloidogyne arenaria* in pre-emergence damping-off of peanut. *Plant Disease Reporter*, 59:665–669.

Germani, G. (1970). *Aphasmatylenchus straturatus* n. sp. (Nematode: Hoplolaimidae) from West Africa. *Proceedings of the helminthological Society of Washington*, 37:48–51.

Germani, G. (1979*a*). Nematicide as a tool to study the impact of nematodes on plant productivity. In: Mongi, H. O. & Huxley, P. A. (Eds) *Soil Research in Agroforestry: Proceedings of an Expert Consultation*. Nairobi, Kenya, 26–31 March:297–313.

Germani, G. (1979*b*). Action directe et rémanente d'un traitement nématicide du sol sur 2 cultivars d'arachide au Sénégal. *Oléagineux*, 34:399–404.

Germani, G. (1981*a*). Étude au champ de l'évolution des populations du nématode *Scutellonema cavenessi* et de la cinétique de la fixation de N2 sur 3 cultivars d'Arachide. *Oléagineux*, 36:247–249.

Germani, G. (1981*b*). Pathogenicity of the nematode *Scutellonema cavenessi* on peanut and soybean. *Revue de Nématologie*, 4:203–208.

Germani, G., Baujard, P. & Luc, M. (1985). *La lutte contre les nématodes dans le bassin arachidier sénégalais*. Dakar, ORSTOM, 1985, 8 p.

Germani, G., Cuany, A. & Merny, G. (1982). L'analyse factorielle des correspondances appliquée a l'influence de deux nématodes sur la croissance de l'arachide et la fixation symbiotique de l'azote. *Revue de Nématologie*, 5:161–168.

Germani, G. & Dhéry, M. (1973). Observations et expérimentations concernant le role des nématodes dans deux affections de l'arachide en Haute Volta: la "chlorose" et le "clump". *Oléagineux*, 28:235–242.

Germani, G. & Gautreau, J. (1976). Résultats agronomiques obtenus par des traitements nématicides sur arachide au Sénégal. *Cahiers de l'ORSTOM, série Biologie*, 11:193–202.

Germani, G. & Luc, M. (1982*a*). Études sur la "chlorose voltaïque" des légumineuses due au nématode *Aphasmatylenchus straturatus* Germani. I. *Revue de Nématologie*, 5:139–146.

Germani, G. & Luc, M. (1982*b*). Etudes sur la "chlorose voltaïque" des légumineuses due au nématode *Aphasmatylenchus straturatus* Germani. *Revue de Nématologie*, 5:195–199.

Germani, G., Ollivier, B. & Diem, H. G. (1981). Interaction of *Scutellonema cavenessi* and *Glomus mosseae* on growth and N2 fixation. *Revue de Nématologie*, 4:277–280.

Germani, G. & Reversat, G. (1982). Effet sur les rendements de l'arachide au Sénégal de deux produits nématicides, DBCP et EDB, et d'un amendement organique. *Oléagineux*, 37 ' 521–524.

Germani, G. & Reversat, G. (1983). Effect du dibromochloropropane sur quelques espèces de nématodes reviviscents, parasites de l'arachide au Sénégal. *Revue de Nématologie*, 6:73–78.

Good, J. M. (1968). Relation of plant parasitic nematodes to soil management practices. In: Smart, G. C. & Perry, V. G. (Eds) *Tropical Nematology*. Gainesville, University of Florida Press:113–118.

Good, J. M., Boyle, L. W. & Hammons, R. D. (1958). Studies of *Pratylenchus brachyurus* on peanuts. *Phytopathology*, 48:530–535.

Good, J. M., Robertson, W. K. & Thompson, L. C., Jr. (1954). Effect of crop rotation on populations of meadow nematode, *Pratylenchus leiocephalus*, in Norfolk loamy, fine sand. *Plant Disease Reporter*, 38:178–180.

Good, J. M. & Stansell, J. R. (1965). Effect of irrigation, soil fumigation, and date of harvest on *Pratylenchus brachyurus* (Godfrey). *Nematologica*, 11:38–39.

Graham, T. W. (1951). Nematode root rot of tobacco and other plants. *South Carolina Agricultural Experiment Station Technical Bulletin 390*, 25 p.

Hagan, A. (1988). Disease and nematode control. In: Alabama Cooperative Extension Service, Auburn University. *1988 Peanut Pest Mangement, Weed, Insect, Disease and Nematode Control Recommendations. Circular ANR–360*,:5–6.

Hammons, R. O. (1982). Origin and early history of the peanut. In: Patte, H. E. & Young, C. T. (Eds). *Peanut Science and Technology American Peanut Research and Education Society*. Yoakum, Texas: 1–20.

Holbrook, C. C., Knauft, D. A. & Dickson, D. W. (1983). A technique for screening peanut for resistance to *Meloidogyne arenaria. Plant Disease*, 67:957–958.

Holderman, Q. L. (1955). The present known distribution of the sting nematode, *Belonolaimus gracilis*, in the Coastal Plain of the Southeastern United States. *Plant Disease Reporter*, 39:5–8.

Holderman, Q. L. & Graham, T. W. (1953). The effect of different plant species on the population trends of the sting nematode. *Plant Disease Reporter*, 37:497–500.

Ibrahim, I. K. A. & El-Saedy, M. A. (1976*a*). Plant parasitic nematodes associated with peanuts in Egypt. *Egyptian Journal of Phytopathology*, 8:31–35.

Ibrahim, I. K. A. & El-Saedy, M. A. (1976*b*). Development and pathogenesis of *Meloidogyne javanica* in peanut roots. *Nematologica mediterranea*, 4:231–234.

Ingram, E. G. & Rodriguez-Kabana, R. (1980). Nematodes parasitic on peanuts in Alabama and evaluation of methods for detection and study of population dynamics. *Nematropica*, 10:21–30.

Jackson, C. R. & Minton, N. A. (1968). Pod invasion by fungi in the presence of lesion nematodes in Georgia. *Oléagineux*, 23:531–534.

Jackson, E. E. & Sturgeon, R. V., Jr. (1973). Effects of nematicides upon root lesion nematode populations. *Journal of the American Peanut Research and Education Association*, 5:178–181.

Johnson, A. W., Dowler, C. C. & Hauser, E. W. (1974). Seasonal population dynamics of selected plant parasitic nematodes on four monocultured crops. *Journal of Nematology*, 6:187–190.

Jones, B. L. & De Waele, D. (1988). First report of *Ditylenchus destructor* in pods and seeds of peanut. *Plant Disease*, 72:453.

Kinloch, R. A. & Lutrick, M. C. (1975). The relative abundance of nematodes in an established field crop rotation. *Proceedings of the Soil and Crop Science Society of Florida*, 34:192–194.

Koen, H. (1967). Notes on the host range, ecology and population dynamics of *Pratylenchus brachyurus*. *Nematologica*, 13:118–124.

Lordello, A. I. L. & Gerin, M. (1981). Nematóide javanes parasitando raízes e nódules de amendoim. *Revista de Agricultura Piracicaba*, 56:278.

Loof, P. A. A. (1964). Free-living and plant-parasitic nematodes from Venezuela. *Nematologica*, 10:201–300.

McDonald, D., Bos, W. S. & Gumel, M. H. (1979). Effects of infections of peanut (groundnut) seed by the testa nematode *Aphelenchoides arachidis*, on seed infection by fungi and on seeding emergence. *Plant Disease Reporter*, 63:464–467.

McDonald, D. & Misari, S. M. (1976). Disease and pests of groundnuts and their importance in crop exchange. Interafrican Symposium "The role of plant protection in crop improvement in Africa. Ibadan, Nigeria, 7–12 October, 1974". *African Journal of Plant Protection* 1:75–82.

McWatters, K. H. & Cherry, J. P. (1982). Potential food uses of peanut seed proteins. In: Pattee, H. E. & Young,

C. T. (Eds). *Peanut Science and Technology. American Peanut Research and Education Society, Inc.*, Yoakum, Texas:689–736.

Machmer, J. H. (1951). Root-knot of peanut. I. Distribution. *Plant Disease Reporter*, 35:364–366.

Machmer, J. H. (1953). *Criconemoides* sp., a ring nematode associated with peanut "yellows". *Plant Disease Reporter*, 37:156.

Martin, G. C. (1958). Root-knot nematodes (*Meloidogyne* spp.) in the Federation of Rhodesia and Nyasaland. *Nematologica*, 3:332–349.

Martin, G. C. (1961). Plant species attacked by root-knot nematodes (*Meloidogyne* spp.) in the Federation of Rhodesia and Nyasaland. *Nematologica*, 6:130–134.

Meagher, J. W. (1967). Observations on the transport of nematodes in subsoil drainage and irrigation water. *Australian Journal of experimental Agriculture and Animal Husbandry*, 7:577.

Merny, G., Fortuner, R., & Luc, M. (1974). Plant parasitic nematodes in Gambia. *L' Agronomie tropicale, Nogent*, 29:702–707.

Merny, G. & Mauboussin, J. C. (1973). Action possible des nématodes dans le rabougrissement ou "clump" de l'arachide. *Nematologica* 19:406–408.

Miller, L. I. (1951). A report on the effect of ethylene dibromide soil treatment on root-knot control, nodulation, and yield of peanuts. *Virginia Journal of Science 2, New Series (2)*:109–112.

Miller, L. I. (1972*a*). The influence of soil texture on the survival of *Belonolaimus longicaudatus*. *Phytopathology*, 62:670–671 (abst).

Miller, L. I. (1972*b*). Resistance of plant introductions of *Arachis hypogaea* to *Meloidogyne hapla, Meloidogyne arenaria*, and *Belonolaimus longicaudatus*. *Virginia Journal of Science*, 23:101 [Abstr.].

Miller, L. I. & Duke, G. B. (1961). Peanut nematode disease control. *Virginia Agricultural Experiment Station Bulletin 520*, 26 p.

Milne, D. L. & Du Plessis, D. P.(1964). Development of *Meloidogyne javanica* (Treub) Chitwood on tobacco under fluctuating soil temperatures. *South African Journal of Agricultural Science*, 7:673–680.

Minton, N. A. (1963). Effects of two populations of *Meloidogyne arenaria* on peanut roots. *Phytopathology*, 53:79–81.

Minton, N. A. & Bell, D. K. (1969). *Criconenoides ornatus* parasitic on peanuts. *Journal of Nematology*, 1:349–351.

Minton, N. A., Bell, D. K. & Doupnik, B., Jr. (1969*a*). Peanut pod invasion by *Aspergillus flavus* in the presence of *Meloidogyne hapla*. *Journal of Nematology*, 1:318–320.

Minton, N. A., Cairns, E. J., Minton, E. B. & Hopper, B. E. (1963). Occurrence of plant-parasitic nematodes in Alabama. *Plant Disease Reporter*, 47:743–745.

Minton, N. A. & Csinos, A. S. (1986). Peanut response to 1, 3-D in *Meloidogyne arenaria* and *Sclerotium rolfsii* infested soil. *Proceedings of the American Peanut Research and Education Society.*, 18:62 [Abstr.].

Minton, N. A., Csinos, A. S. & Bell, D. K. (1984). Effects of six nematicides and two fungicides, applied in various combinations on peanuts. *United States Department of Agriculture, Agricultural Research Service, ARS–3*, 14 p.

Minton, N. A. & Hammons, R. O. (1975). Evaluation of peanut for resistance to the peanut root-knot nematode, *Meloidogyne arenaria*. *Plant Disease Reporter*, 59:944–945.

Minton, N. A., Hammons, R. O. & Parham, S. A. (1970). Infection of shell and peg tissues of six peanut cultivars by *Pratylenchus brachyurus*. *Phytopathology*, 60:472–474.

Minton, N. A. & Jackson, C. R. (1967). Invasion of peanut pods by *Aspergillus flavus* and other fungi in the presence of root-knot nematodes. *Oléagineux*, 22:543–546.

Minton, N. A., McGill, J. F. & Golden, A. M. (1969*b*). *Meloidogyne javanica* attacks peanuts in Georgia. *Plant Disease Reporter*, 53:668.

Minton, N. A. & Morgan, L. W. (1974). Evaluation of systemic and nonsystemic pesticides for insect and nematode control in peanuts. *Plant Science*, 1:91–98.

Minz, G. (1956). The root-knot nematode, *Meloidogyne* spp., in Israel. *Plant Disease Reporter*, 40:798–801.

Mitsui, Y., Yoshida, T., Okamoto, K. & Ishii, R. (1976). Relationship between nematode-trapping fungi and *Meloidogyne hapla* in peanut field. *Japanese Journal of Nematology*, 6:47–55.

Motsinger, R. E., Crawford, J.L. & Thompson, S. S. (1976). Nematode survey of peanuts and cotton in Southwest Georgia. *Peanut Science*, 3:72–74.

Netscher, C. (1975). Studies on the resistance of groundnut to *Meloidogyne* spp. in Senegal. *Cahiers de l'ORSTOM, série Biologie*, 10:227–232.

Norse, D. (1972). Nematode populations in a maize-groundnut-tobacco rotation and the resistance of maize varieties to *Meloidogyne javanica*. *Tropical Agriculture, Trinidad*, 49:355–360.

O'Bannon, J. H. & Reynolds, H. W. (1961). Root-knot nematode damage and cotton yields in relation to certain soil properties. *Soil Science*, 92:384–386.

Orion, D. & Cohn, E. (1975). Crop rotation for control of certain nematodes. *Phytoparasitica*, 3:69 [Abstr.].

Orr, C. C. & Newton, O. H. (1971). Distribution of nematodes by wind. *Plant Disease Reporter*, 55:61–63.

Osman, H. A., Dickson, D. W., & Smart, G. C., Jr. (1985). Morphological comparisons of host races 1 and 2 of *Meloidogyne arenaria* from Florida. *Journal of Nematology*, 17:279–285.

Oteifa, B. A. (1962). Species of root-lesion nematodes commonly associated with economic crops in the Delta of the U.A.R. *Plant Disease Reporter*, 46:572–575.

Owens, J. V. (1951). The pathological effects of *Belonolaimus gracilis* on peanuts in Virginia. *Phytopathology*, 41:29 [Abstr.].

Patel, H. R., Vaishnav, M. U. & Dhruj, I. U. (1985). Interaction of *Meloidogyne arenaria* and *Fusarium solani* on *groundnut*. *Indian Journal of Nematology*, 15: 98–99.

Perry, V. G. (1965). Host-parasite relationships of certain nematodes and crop plants in Florida. *University of Florida Agricultural Experiment Stations Annual Report, Gainesville*: 115–116.

Perry, V. G. & Norden, A. J. (1963). Some effect of a cropping sequence on populations of certain plant nematodes. *Proceedings of the Soil and Crop Science Society of Florida*, 23:116–121.

Porter, D. M., Smith, D. H. & Rodríguez-Kábana, R. (1982). Peanut plant diseases. In: Patte, H. E. & Young, C. T. (Eds). *Peanut Science and Technology*. American Peanut Research and Education Society, Inc., Yoakum, Texas:326–410.

Porter, D. M., Smith, D. H. & Rodríguez-Kábana, R. (1984). Introduction. In: Porter, D. M., Smith, D. H. & Rodríguez-Kábana, R. (Eds). *Compendium of Peanut Diseases*. St. Paul, Minnesota, USA, American Phytopathological Society,:1–4.

Rau, G. J. (1958). A new species of sting nematode. *Proceedings of the helminthological Society of Washington*, 25:95–98.

Reddy, D. D. R., Subrahmanyam, P., Sankara Reddy, G. H., Raja Reddy, C. & Siva Ras, D. V. (1984). A nematode disease of peanut caused by *Tylenchorhynchus brevilineatus*. *Plant Disease*, 68:526–529.

Rickard, D. A., Barker, K. R. & Wells, J. C. (1977). Effects of *Macroposthonia ornata* and *Meloidogyne hapla* on yield and value of peanut. *Journal of Nematology*, 9:281. [Abstr.].

Robbins, R. T. (1972). *Morphology and ecology of the sting nematode*, Belonolaimus longicaudatus. Ph.D. Thesis, North Carolina State University, Raleigh:65 p.

Robbins, R. T. & Barker, K. R. (1973). Comparisons of host range and reproduction among populations of *Belonolaimus longicaudatus* from North Carolina and Georgia. *Plant Disease Reporter*, 57:750–754.

Robbins, R. T. & Barker, K. R. (1974). The effects of soil type, particle size, temperature, and moisture on reproduction of *Belonolaimus longicaudatus*. *Journal of Nematology*, 6:1–6.

Robbins, R. T. & Hirschmann, H. (1974). Variation among populations of *Belonolaimus longicautatus*. *Journal of Nematology*, 6:87–94.

Rodríguez-Kábana, R., Ivey, H. & Backman, P. A. (1987). Peanut-cotton rotations for the management of *Meloidogyne arenaria*. *Journal of Nematology*, 19:484–486.

Rodríguez-Kábana, R. & King, P. S. (1979). Relation between the method of incorporation of systemic nematicides into soil and their effectiveness against root-knot nematodes on peanuts. *Nematropica*, 9:167–172.

Rodríguez-Kábana, R. & King, P. S. (1985). Evaluation of selected nematicides for control of *Meloidogyne arenaria* in peanut: A multi-year study. *Nematropica*, 15:155–164.

Rodríguez-Kábana, R., King, P. S. & Pope, M. H. (1981). Comparison of in-furrow applications and banded treatments for control of *Meloidogyne arenaria* in peanuts. *Nematropica*, 11: 53–67.

Rodríguez-Kábana, R. & Morgan-Jones, G. (1987). Novel rotations and organic materials show promise for

management of nematodes. *Alabama Agricultural Experiment Station, Highlights of Agricultural Research*, 34:13.

Rodríguez-Kábana, R. & Robertson, D. G. (1987). Control of *Meloidogyne arenaria* in peanut with 1, 3-D: Relative efficacy and application depth. *Nematropica*, 17:17–29.

Rodríguez-Kábana, R., Shelby, R. A., King, P. S. & Pope, M. H. (1982*a*). Application time and effectiveness of four systemic nematicides against *Meloidogyne arenaria* on Florunner peanut. *Nematropica*, 12:85–96.

Rodríguez-Kábana, R. & Touchton, J. T. (1984). Corn and sorghum as rotational crops for management of *Meloidogyne arenaria* in peanut. *Nematropica*, 14:26–36.

Rodríguez-Kábana, R., Weaver, C. F. & King, P. S. (1985). Combinations of 1, 3-D and Aldicarb for management of *Meloidogyne arenaria* in peanuts. *Nematropica*, 15:93–106.

Rodríguez-Kábana, R., Weaver, C. F., Robertson, D. G. & Snoddy, E. L. (1986). Population dynamics of *Meloidogyne arenaria* juveniles in a field with Florunner peanut. *Nematropica*, 16:185–196.

Rodríguez-Kábana, R., Williams, J. C. & Shelby, R. A. (1982*b*). Assessment of peanut yield losses caused by *Meloidogyne arenaria*. *Nematropica*, 12:279–288.

Saint-Smith, J. H., McCarthy, G. J. P., Rawson, J. E., Langford, S. & Colbran, R. C. (1972). Peanut growing. *Queensland agricultural Journal*, 98:639–644.

Sakhuja, P. K. & Sethi, C. L.(1985*a*). Screening of groundnut germplasm for resistance to root-knot nematode, *Meloidogyne javanica*. *Indian Journal of Nematology*, 15:129–130.

Sakhuja, P. K. & Sethi, C. L. (1985*b*). Growth of groundnut as influenced by different inocula of *Meloidogyne javanica*. *Indian Journal of Nematology*, 15:135–137.

Sakhuja, P. K. & Sethi, C. L. (1985*c*). Frequency of occurrence of various plant parasitic nematodes and root-rot fungi on groundnut in Punjab. *Indian Journal of Nematology*, 15:191–194.

Sasser, J. N. (1954). Identification and host-parasite relationships of certain root-knot nematodes (*Meloidogyne* spp.). *University of Maryland, Agricultural Experiment Station, Technical Bulletin A–77*, 31 p.

Sasser, J. N. (1972). Physiological variations in the genus *Meloidogyne* as determined by differential hosts. *OEPP/EPPO Bulletin* 6:41–48.

Sasser, J. N. (1977). Worldwide dissimination and importance of the root-knot nematodes, *Meloidogyne* spp. *Journal of Nematology*, 9:26–29.

Sasser, J. N. (1979*a*). Pathogenicity, host ranges, and variability in *Meloidogyne* spp. In: Lamberti, F. & Taylor, C. E. (Eds.). *Root-knot nematodes (*Meloidogyne *species): Systematics, Biology, and Control.* London, New York, San Francisco, Academic Press:257–268.

Sasser, J. N. (1979*b*). Economic importance of *Meloidogyne* in tropical countries. In: Lamberti, F. & Taylor, C. E. (Eds). *Root-knot nematodes (*Meloidogyne *species): Systematics, Biology and Control.* London, New York, San Francisco Academic Press: 359–374.

Sasser, J. N., Barker, K. R. & Nelson, L. A. (1970). Relative effects of eight nematode species on growth, yield and value of peanut follwing chemical soil treatment. *Proceedings of the Second International Congress of Parasitology, 6–12 September 1970, Washington, DC, Journal of Parasitology 56, Number 4, Section II, part 1 of 3 parts*:300–301.

Sasser, J. N., Barker, K. R. & Nelson, L. A. (1975*a*). Correlations of field populations of nematodes with crop growth responses for determining relative involvement of species. *Journal of Nematology*, 7:193–198.

Sasser, J. N., Barker, K. R. & Nelson, L. A. (1975*b*). Chemical soil treatment for nematode control on peanut and soybean. *Plant Disease Reporter*, 59:154–155.

Sasser, J. N., Cooper, W. E. & Bowery, T. G. (1960). Recent developments in the control of sting nematode, *Belonolaimus longicaudatus*, on peanuts with 1,2-dibromo–3-chloropropane and EN 18133. *Plant Disease Reporter*, 44:733–737.

Sasser, J. N. & Freckman, D. (1987). A world perpspective on nematology: The role of the society. In: Veech, J. A. & Dickson, D. W. (Eds). *Vistas on Nematology*. Hyattsville, MD, USA, Society of Nematologists:7–14.

Sasser, J. N. & Kirby, M. F. (1979). Crop cultivars resistant to root-knot nematodes, *Meloidogyne* spp., with information on seed sources. *Cooperative Publication, Department of Plant Pathology, North Carolina State University and U. S. Agency of International Development*. Raleigh, NC. 24 p.

Sasser, J. N. & Nusbaum, C. J. (1955). Seasonal fluctuations and host specificity of root-knot nematode populations in two-year tobacco rotation plots. *Phytopathology*, 45:540–545.

Sauer, M. R. (1968). Nematodes in an irrigated vineyard. *Nematologica*, 14:457–458.

Sharma, S. B. (1985). A world list of nematode pathogens associated with chickpea, groundnut, pearl millet, pigeonpea and sorghum. *Pulse Pathology Progress Report 42. International Crops Research Institute for the Semi-Arid Tropics*:5–8.

Sharma, N. K., Sharma, S. K. & Midha, S. K. (1978). Root-knot disease of groundnut in Punjah. *Indian Farming*, 28:19–21.

Sher. S. A. (1964). Revision of the Hoplolaiminae (Nematoda). III. *Scutellonema*, Andrássy, 1958. *Nematologica* 9:421–443.

Singh, N. D. (1972). Plant-parasitic nematodes associated with some economic crops in Guyana. *Plant Disease Reporter*, 56:1059–1062.

Singh, I. & Sakhuja, P. K. (1984). Clump disease of groundnut and its control with nematicides. *Indian Journal of Nematology*, 14:54–55.

Smith, O. D., Boswell, T. E. & Thames, W. H. (1978). Lesion nematode resistance in peanuts. *Crop Science*, 18:1008–1011.

Starr, J. L. (1984). Expression of resistance in peanuts, *Arachis hypogaea*, to *Pratylenchus brachyurus*: Impact on screening for resistance. *Journal of Nematology*, 16:404–406.

Steiner, G. (1945). Meadow nematodes as the cause of root destruction. *Phytopathology*, 35:935–937.

Steiner, G. (1949). Plant nematodes the grower should know. *Proceedings of the Soil Science Society of Florida, 1942, 4-B*:72–117.

Subrahmanyam, P., Ghanekar, A. M., Nolt, B. L., Reedy, D. V. R., & McDonald, D. (1983). Resistance to groundnut diseases in wild *Arachis* species. *Proceedings of an International Workshop on Cytogenetics of* Arachis., *31 Oct. – 2 Nov. 1983*, ICRISAT Centre, Patancheru, India.

Taylor, A. L & Sasser, J. N. (1978). Biology, identification and control of root-knot nematodes (*Meloidogyne* species). *A cooperative publication of the Department of Plant Pathology, North Carolina Sate University and USAID*, North Carolina State University Graphics, 111 p.

Thames, W. H., Jr. (1959). Plant parasitic nematode populations of some Florida soils under cultivated and natural conditions. *Dissertation Abstract*, 20:1109–1110.

Thames, W. H., Jr. & Stoner, W. N. (1953). A preliminary trial of lowland culture rice in rotation with vegetable crops as a means of reducing root-knot nematode infestations in the Everglades. *Plant Disease Reporter*, 37:187–192.

Thomason, I. J., Van Gundy, S. D. & Kirkpatrick, J. D. (1964). Mobility and infectivity of *Meloidogyne javanica* as affected by storage time and temperature in water. *Phytopathology*, 54:192–195.

United States Department of Agriculture. *Foreign Agriculture Service. (1989). World OilseedSituation and Market Highlights*. Circular Series FOP 7–89 P9.

Van der Linde, J. W. (1956). The *Meloidogyne* problem in South Africa. *Nematologica*, 1:177–183.

Van Gundy, S. D. (1985). Ecology of *Meloidogyne* spp. – Emphasis on environmental factors affecting survival and pathogenicity. In: Sasser, J. N. & Carter. C. C. (Eds). *An Advanced Treatise on* Meloidogyne *Biology and Control* Volume I. Raleigh, NC, USA, University of North Carolina Press: 178–182.

Vance, P. N. (1981). Peanut growing in the South Burnett. *Queensland agricultural Journal*, 107:201–213.

Vrain, T. C. (1978). Influence of chilling and freezing temperatures on infestivity of *Meloidogyne incognita* and *M. hapla*. *Journal of Nematology*, 10:177–180.

Wallace, H. R. (1971). Abiotic influences in the soil environment. In: Zuckerman, B. M., Mai, W. F. & Rohde, R. A. (Eds). *Plant Parasitic Nematodes, Volume I.* New York & London Academic Press: 257–280.

Wheeler, T. A. & Starr, J. L. (1987). Incidence and economic importance of plant-parasitic nematodes on peanut in Texas. *Peanut Science*, 14:94–96.

Williams, E. J. & Drexler, J. S. (1981). A non-destructive method for determining peanut pod maturity. *Peanut Science*, 8:134–141.

Wilson, C. (1948). Root-knot nematodes on peanuts in Alabama. *Plant Disease Reporter*, 32:443.

Wittwer, S. H. (1981). The 20 crops that stand between man and starvation. *Farm Chemicals*, 144:17–28.

Yang, B. (1984). The identification of 15 root-knot nematode populations. *Acta Phytopathologica Sinica*, 11:107–112.

Yin, K. C. & Feng, Z. X. (1981). A preliminary survey on the parastic nematodes of Agricultural Crops. *Phytopathologica* 8:111–126.

Zhang, Y. (1985). Occurrence and control of peanut root-knot disease in non-irrigated sloping fields of Zhangjiang District. *Agricultural Science of Guamgdong Province*, 6:48–49.

Minton N.A., Baujard Pierre. (1990).

Nematode parasites of peanut.

In : Luc Michel (ed.), Sikora R.A. (ed.), Bridge J. (ed.). Plant parasitic nematodes in subtropical and tropical agriculture.

Wallingford : CAB International, p. 285-320. ISBN 0-85198-630-7.