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RECENT ADVANCES IN CASSAVA PEST MANAGEMENT

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ABSTRACT

Cassava (*Manihot esculenta*) occupies a uniquely important position as a food security crop for smallholder farmers in areas of the tropics where climate, soils, or societal stresses constrain production. Given its reliability and productivity, cassava is the most important locally produced food in a third of the world's low-income, food-deficit countries. It is the fourth most important source of carbohydrates for human consumption in the tropics, after rice, sugar, and maize. World production of cassava from 1994–1996 averaged 166 million tons/year grown on 16.6 million hectares (ha), for an average yield of 9.9 tons/ha. Approximately 57% is used for human consumption, 32% for animal feed and industrial purposes, and 11% is waste. Africa accounts for 51.3% of the production; Asia, 29.4%; and Latin America, 19.3%. The area planted to cassava in Africa, Asia, and Latin America is 10.3, 3.7, and 2.6 million ha, respectively.

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INTRODUCTION

Cassava originated and was domesticated in the neotropics (146). Today cassava is a major crop across an extensive area of western and central Africa, referred to as the "cassava belt," where it was introduced by returning slave ships in the 1500s. In the seventeenth century, cassava was introduced to Asia, where it is grown for both human consumption and animal feed, often for the European export market (93, 113).

The cassava germplasm bank maintained at the Centro Internacional de Agricultura Tropical (CIAT) in Cali, Colombia, consists of $\approx$ 6000 accessions and locally selected cultivars (landraces) collected primarily in South America. These traditional cultivars represent centuries of cassava cultivation in diverse habitats (113), having been selected by farmers over long periods in the presence of a high diversity of herbivores. These landraces often possess traits that confer low levels of resistance to multiple pests. Farmers also rely on intercropping, rotations, burning, and other cultural practices to control pests (113).

Large-scale production and product diversification for industrial applications are increasing in some areas of South America (e.g. southern Brazil, northern Colombia, Venezuela) and Asia (e.g. China, Indonesia, Vietnam), but most cassava is produced for immediate consumption and provides basic sustenance to large populations in marginal areas (111). Cassava is cultivated primarily on small plots with minimum inputs. The plant is well adapted to seasonally dry environments, where rainfall is scarce and unpredictable. There is, however, a large gap between potential yield [21.3 tons/ha (110)] and that realized by producers [a mean of 11.2 tons/ha in Latin America; 10.0 in northeast (NE) Brazil, where half of Brazil's cassava is produced; 9.0 in Africa (110)]. Contributors to yield reduction include low-fertility soils, harsh environmental conditions, arthropod pests, and diseases, as well as the limited use of inputs (e.g. fertilizers and pesticides). Stem cuttings rather than seeds are planted, and vegetative propagation contributes to the buildup of pathogens and pests, resulting in yield reduction. Few improved cultivars or stem cuttings free of pathogens or pests are available to farmers, in part owing to poor articulation of research and extension agencies with marginalized populations.

The technical options for cassava pest control in many areas of Latin America and Africa are limited by farmers' inability to invest in inputs and by weak national agricultural research and extension agencies. Although classical biological control succeeded in controlling introduced pests in Africa (112), farmer education and participation in the decision-making processes are essential for controlling most pest problems and achieving sustainable increases in productivity. Participatory research and development with farmers is showing promise in overcoming these obstacles in South America (41, 63; B Ospina, unpublished data).

Cassava has the ability to generate hydrogen cyanide (HCN) (70, 156). Many farmers and indigenous peoples in the neotropics prefer to grow "bitter" cultivars with high (fresh root parenchymal tissue concentration of >100 mg kg^{-1} HCN) cyanogenic potential (81, 178), based on the belief that the concentration of cyanogenic glucosides in the leaves and roots is a defense mechanism against pathogens, as well as arthropod and mammalian pests. The evidence for this hypothesis in cassava is speculative (28). Specialist pests such as the hornworm (*Erinnyis ello*) and the green mite (*Mononychellus tanajoa*), which have presumably coevolved with cassava, show no preference with respect to concentration of cyanogenic compounds (12). On the other hand, generalist feeders such as the African grasshopper (*Zonocerus variegatus*) and the root-feeding burrower bug (*Cyrtomenus bergi*) display a preference for low HCN (16, 28, 36, 147), indicating that cyanogen levels may be an important factor in pest resistance. The key factor appears to be coevolution, in that cyanogens in the leaves and roots may deter damage by generalist feeders, which have not coevolved with cassava.

An extensive review of cassava pests was undertaken two decades ago (30) when cassava was an under-studied crop. During the past 20 years, however, considerable research has been conducted at international agricultural research centers such as CIAT and the International Institute of Tropical Agriculture (IITA, Nigeria) and by numerous national research programs in Latin America (e.g. Brazil, Colombia, Cuba), Africa (e.g. Cameroon, Nigeria, Uganda), and Asia (e.g. India, Indonesia, Philippines, Thailand). Thus we now have a more complete understanding of the cassava arthropod complex, crop losses, and possible control techniques (19, 20, 42, 67, 102, 112, 132, 161, 179). This review focuses on pests that can significantly reduce cassava production, with emphasis on the neotropics.

YIELD-PEST RELATIONSHIPS

Cassava is considered more tolerant to pests than most crops because it does not have critical periods that affect yield-forming organs (66). Nevertheless, recent research has shown that several pests can reduce yields significantly when pest populations are high and/or environmental conditions are unfavorable. An estimated 200 species of arthropods feed on cassava in the Americas alone (30). Many of these species are specific to cassava and have adapted in varying degrees to an array of natural biochemical defenses that include laticifers and HCN content (28). In Africa, there are some native generalist pests of cassava (*Bemisia tabaci* and the variegated grasshopper, *Z. variegatus*), but there are few serious specialist pests. Two major pests were accidentally introduced from South America: the cassava mealybug *Phenacoccus manihoti* and the cassava green mite *M. tanajoa*. *P. manihoti* was the target of a highly successful classical biological control effort (112, 134). Advances in the use of phytoseiid predator mites for controlling *M. tanajoa* are reviewed below. In Asia, none of the major neotropical cassava pests has become established. Although several native arthropod pests feed on cassava, none is reported causing serious yield losses (82, 127, 151). Lack of solid data on yield reductions in cassava stimulated research to determine the effect of arthropod pest damage on root yield and quality.

Indirect Feeding Damage

The damage to cassava is often indirect because most arthropod pests are foliage- or stem-feeders, reducing leaf area, leaf life, or photosynthetic rate. Recent studies show that pests that attack crops over prolonged periods (3–6 months)—such as mites, mealybugs, thrips, whiteflies, and lacebugs—can cause severe root yield reductions (19, 32) as a result of their feeding on leaf cell fluids and the subsequent decrease in photosynthesis. Severe attacks can induce premature leaf drop and death of the apical meristem. The potential for yield reduction by these pests is much greater than that of cyclical pests such as hornworms and leaf-cutting ants, which cause sporadic defoliation; nevertheless, these highly visible pests often cause farmers to apply insecticides (41).

In experimental trials, mites reduced fresh root yield in susceptible cultivars by 21, 25, and 53% during a 3-, 4-, and 6-month attack, respectively, and by 15% in resistant cultivars (32, 43). Under field conditions with higher mite populations, there was ≤73% yield loss in susceptible cultivars and 67% of the stem-cutting planting material was damaged (42, 43). In the case of the whitefly *Aleurotrachelus socialis*, there is a correlation between duration of attack and yield loss: Infestations of 1, 6, and 11 months resulted in a 5, 42, and 79% yield reduction, respectively (168). Losses due to mealybug (*Phenacoccus herreni*) infestations in farmers' fields in Brazil are estimated at 80% (AC Bellotti, personal observation); on experimental plots, reduction ranged from 68–88%, depending on the cultivar (168). In Africa, mealybug attacks have caused yield losses of ≈80% (112, 155). Lacebugs (*Amblystiria machalana* and *Vatiga manihotae*) reduced yield by 39% in experimental plots at CIAT (57).

In the neotropics, a complex of pests attacks the crop over its 8- to 24-month growing cycle. In seasonally dry regions, it is common to observe populations of mites, mealybugs, lacebugs, whiteflies, thrips, and other minor pests infesting cassava plants (20, 57, 126). Preliminary research indicates that controlling

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only one of these pests may not improve yields. In an experiment evaluating a predominantly mite and whitefly complex in NE Brazil, yields improved only when both pest populations were controlled; controlling only one pest had no positive impact on yields (ARN Farias, unpublished data).

Direct Feeding Damage

The burrower bug (*C. bergi*; Hemiptera: Cydnidae) is one of the few pests that damage the cassava root directly. As a result of punctures during feeding, the root is exposed to invasive fungal pathogens, reducing both yield and root quality. Brown to black lesions on the white root parenchyma in 30–40% of the roots has resulted in total loss of commercial roots (31).

Seasonal Yield Losses

Arthropod pests do not appear to cause significant damage to cassava in areas where there is considerable and consistent rainfall (130); however, the crop is often grown under conditions of irregular, limited rainfall. The cassava plant is well adapted to long periods of limited water, responding to water shortage by reducing its evaporative (leaf) surface rapidly and efficiently and by partially closing the stomata, thereby increasing water-use efficiency (65, 69, 82, 83). Negative effects of excess solar radiation are minimized when the leaves change their orientation in order to intercept less light. The combination of these stress-avoidance mechanisms is highly effective. In water-deprived plants, both the accelerated shedding of old leaves and the pronounced decrease in their photosynthetic activity means that the younger leaves play a key role in the plant's carbon nutrition. Given that pests prefer the younger canopy leaves, dry-season feeding tends to cause the greatest yield losses in cassava.

Once the crop enters into a wet cycle (rain or irrigation), it has the potential to recuperate and compensate for yield losses from severe drought as well as from pest attacks because of the formation of new leaf canopy and the higher photosynthetic rate in newly formed leaves (82). When yield losses due to mite attack were evaluated in the absence of water stress (through irrigation), for example, yield reduction was <25% (B Ospina, unpublished data).

Secondary Pests

Several arthropod pests attack cassava only occasionally, or they may be present continually but at such low populations that yield is not affected (Table 1). If their populations increase or outbreaks occur, several of these pests can cause severe damage. In recent years, for example, the lepidopteran stemborer *Chilomima clarkei* has caused considerable crop damage in several areas of Colombia, with >7000 ha under attack (172). Borers not only reduce yields by 45–62% (122); they also reduce both the quality and quantity of stem cuttings for planting material. Severe attacks of whitegrubs (Scarabaeidae) and

Table 1 Occasional and incidental pests of cassava

Common name	Important species	Region	Type of damage	Reported yield loss	Control strategy	References
Scales	*Aonidomytilus albus* *Saissetia miranda*	Americas, Africa, Asia	Attack stems causing leaf fall; use of infested stems reduces germination	<20% fresh root yields; 50–60% loss in stem cutting	Destroy infested stems; use scale-free planting material	29, 30, 54, 95, 124, 167
Fruitflies	*Anastrepha pickeli* *Anastrepha manihoti*	Americas, Costa Rica, Panama, Venezuela, Colombia, Brazil, Peru	Bore fruit (seed) and stems, causing rotting of pith area	0–30% when infested stems used as planting material	Use of damage-free planting material	26, 29, 30, 52, 118, 124, 125
Shootflies	*Neosilba perezi*	Americas	Larvae kill apical buds, retarding plant growth, inducing lateral branching	Not reported; reduced quality of planting material	None required	29, 30, 52, 66, 124, 125, 143
Gallmidges	*Jatrophobia (Eudiplosis) brasiliensis*	Americas	Yellowish green to red galls on upper leaf surface	Not reported	None required	29, 30, 124, 150
Whitegrubs	*Leucopholis rurida* *Phyllophaga* spp. Several others	All regions	Feed on planting material, roots	95% loss in germination	Soil pesticide treatment at planting	29, 30, 120, 124, 125, 139, 143
Termites	*Coptotermes voltkevi* *Coptotermes paradoxis*	All regions	Tunnel in planting material, roots, stems, swollen roots	46–100% loss of planting material	Dusting of planting material with pesticide	11, 29, 30, 51, 54, 57, 120, 124

Stemborers	*Chilomima clarkei*	Americas, Colombia, Venezuela, Paraguay	Tunnel stems, stem breakage	45–62%	Cultural practices to maintain clean fields; destroy infested stems	29, 30, 52, 53, 95, 106, 122, 125, 150, 172, 173
	Coelosternus spp.	Americas, Brazil	Tunnel stems, stem breakage	Unknown		
	Lagochirus sp.	All regions	Tunnel stems, stem breakage	Unknown		
Leaf-cutting ants	*Atta* spp. *Acromyrmex* spp.	Americas	Foliage removed	Unknown	Toxic baits	29, 30, 76, 77, 150; B Ospina, unpublished data
Root mealybugs	*Pseudococcus mandioca*	Brazil, Paraguay, Bolivia	Scarring of root parenchyma, quality reduced, some defoliation	17%	Crop rotation	58, 142, 176
	Stictococcus vayssierei	Cameroon	Leaf fall, wilting; root quality reduced	Unknown	Unknown	136
Grasshoppers	*Zonocerus elegans* *Zonocerus variegatus*	Mainly Africa but also Americas	Defoliation, stripping of bark	Unknown	Entomopathogens being investigated	28, 29, 30, 79, 123, 129, 152
Thrips	*Frankliniella williamsi*	Americas, Africa	Leaf distortion, bud reduction	17–25%	Host plant resistance	25, 29, 30, 95, 124

termites can eliminate stake germination completely, destroying whole plantations (29, 30). Scale populations, normally under natural biological control, can occasionally increase rapidly, causing yield losses and reduced planting material. Leaf-cutting ants can defoliate part or all of small plantations, probably reducing yields. Thrips, which are often present in seasonally dry areas, can cause yield losses. They can, however, be controlled easily through the use of resistant pubescent cultivars that are commonly grown in these areas (25, 166); consequently, this pest is not discussed in detail in this review.

MANAGEMENT OF MAJOR PESTS

Cassava Green Mite

The cassava green mite *M. tanajoa* (syn = *Mononychellus progresivus*; 108) is probably native to NE Brazil, where it was first reported in 1938. The natives had long known the damage symptoms, which provided the name tanajoa (a sick or diseased plant). It attacks young leaves and meristems of cassava in the neotropics (29, 30), but it is normally a serious problem only in dry regions (42, 171, 183). The mite first appeared in Africa (Uganda) in 1971; by 1985 it had spread across most of the African cassava belt, occurring in 27 countries (182). Today it is one of the principal pests of cassava in Africa, causing estimated root yield losses of 13–80% (112, 157, 184).

CONTROL *Cultural* Application of acaricides is not an economic option for low-income farmers in view of cassava's long growing period. Cultural methods are not known, and even if they were available, it would be a formidable task to disseminate this knowledge across such large regions characterized by political instability, lack of infrastructure, weak institutions, and low-input farming.

Host plant resistance Cultivars from either Africa or Latin America have not shown promise for obtaining high levels of resistance; nevertheless, substantial efforts were mounted to develop resistant cultivars (e.g. 21, 22, 78, 84, 109). Cultivars with low to moderate levels of resistance are grown by farmers in Colombia and Ecuador (148).

Biological The most promising approach is to find a practical, long-lasting solution in the form of biological control (33). The International Institute of Biological Control (IIBC, formerly the Commonwealth Institute of Biological Control) began exploring for natural enemies of the mite in the neotropics in 1974 (188). A staphylinid predator *Oligota minuta* was released in Kenya in 1977 but failed to become established. In the early 1980s, IITA began working on biological control and built a laboratory in Benin to receive and mass-rear imported natural enemies. Meanwhile, CIAT began intensive exploration and evaluation of phytoseiids and other predators in Latin America. Ten species of phytoseiids in >60 shipments (>14,000 individuals) were sent from CIAT to Benin via quarantine at IIBC in England. Despite massive releases (5.5 million individuals at 348 sites in 10 African countries) from 1984 to 1988, none of these predators became established (186). In 1988, IITA began receiving phytoseiid shipments from the Brazilian national research center on cassava CNPMF/EMBRAPA (Centro Nacional de Pesquisa de Mandioca e Fruiticultura/Empresa Brasileira de Pesquisa Agropecuaria) in Cruz das Almas, Bahia (186). Three Brazilian species of phytoseiids are now established in Africa:

1. *Typhlodromalus* (*Amblyseius*) *manihoti*, introduced in 1993, is now in four countries and spreading slowly (JS Yaninek, personal communication).

2. *Typhlodromalus* (*Amblyseius*) *aripo*, as of 1993, spread 12 km the first season and ≤200 km the second, and is now in 14 countries, covering ≈400,000 km^2 (114). On-farm field trials indicate that *T. aripo* reduces cassava green mite populations by 35–60% and increases fresh root yields by 30–37%. It is estimated that farmers' profits increased $70/ha/season (JS Yaninek, personal communication).

3. *Neoseiulus idaeus*, introduced in 1990, is spreading extremely slowly in two countries. It does not appear to affect green mite populations (185; JS Yaninek, personal communication).

Dry conditions favor the mite's survival while hindering that of phytoseiid predators (13). There appear to be fewer species of natural enemies in NE Brazil than in other parts of South America with more rainfall (74; L Smith, unpublished data). Predator exclusion trials in northern Colombia indicated that mite populations decreased fresh root yield by 33% in the absence of predators. Application of acaricides did not increase yield, indicating good natural biological control (39, 40). CIAT and CNPMF/EMBRAPA are importing phytoseiid predators from coastal Ecuador, which has a similar dry climate. The two species released thus far—*Neoseiulus californicus* and *Galendromus annectens*—have been recovered only occasionally and in small numbers.

An Entomophthorales fungal pathogen, *Neozygites* cf *floridana*, causes dramatic natural epizootics in the cassava green mite in NE Brazil (73). Some strains appear to be specific to the genus *Mononychellus* (75; L Smith, unpublished data). Although this pathogen has also been found in cassava green mites in Africa, epizootics have not been observed (187), suggesting that the Brazilian strains may be more virulent than the African strains. A Colombian strain (3) is currently being evaluated as a potential biological control agent for Africa.

Cassava Mealybug

Although *P. herreni* is distributed throughout northern South America, it has—from the 1970s onward—caused serious losses only in NE Brazil (19). Surveys found few parasitoids in the region, suggesting that *P. herreni* may be an exotic pest, probably coming from northern South America, which is geographically separated from NE Brazil by the Amazon basin (177). Farmers estimated their losses to be >80% and implemented cultural practices such as pest-free cutting selection and destruction (burning) of infested growing points where the initial mealybug attack is concentrated. These practices met with limited success, however, and cassava production decreased in the region. Although numerous natural enemies of mealybugs were observed in other regions of South America, little was done to introduce appropriate biological control agents into NE Brazil at that time (27).

In Africa, an unidentified mealybug first appeared on cassava in the Congo and Zaire in 1973. By 1986 it had spread across 70% of the African cassava belt, causing losses of up to 84%. The African mealybug was identified as a new species, *P. manihoti*, presumed to have come from the neotropics, where cassava originated.

CONTROL Management of the cassava mealybug has become a well-known example of classical biological control (see 112, 133). Explorations for natural enemies were conducted by IIBC and IITA in Central America and northern South America. Then in 1980 it was learned that the mealybug from northern South America has males, whereas the African mealybug does not. The neotropical population was subsequently described as a separate species, *P. herreni*. The target species, *P. manihoti*, was finally located in Paraguay by Bellotti in 1980 (112). IIBC subsequently collected natural enemies of *P. manihoti* and sent them via quarantine in London to IITA in Benin for multiplication and release in Africa. The encyrtid parasitoid *Apoanagyrus* (*Epidinocarsis*) *lopezi* and the coccinellid predators *Hyperaspis notata*, *Hyperaspis raynevali*, and *Diomus* sp. became established in Africa. It appears that the parasitoid is the principal agent reducing the mealybug population (64) despite some interference by native ants (72) and hyperparasitoids (115). Stunting of cassava tips by mealybugs decreased from 88 to 3%, and yield increased by 2500 kg/ha in savanna regions. The parasitoid was established in all ecological zones occupied by the mealybug, and by 1990 *A. lopezi* was established in 25 countries, covering an area of 2.7 million km^2. The cost-benefit ratio of the project has been conservatively estimated to be 1:149 (137). Poor soil fertility conditions can produce smaller hosts, resulting in low production of *A. lopezi* female progeny and higher mealybug populations. Predators were therefore released during the late 1980s and early 1990s to control the pest where soil fertility is poor (134). In some regions, such as the People's Republic of the Congo, mealybug outbreaks at the onset of the rainy season continued to occur. The Entomophthorales fungus *Neozygites fumosa* was primarily responsible for reductions in pest density (121).

In 1993, UNDP (United Nations Development Program) funded a project for ecological and sustainable cassava crop protection [PROFISMA/ESCaPP (Proteção Fitossanitária Sustentâvel de Mandioca/Ecologically Sustainable Cassava Plant Protection)], involving CIAT, CNPMF/EMBRAPA, and IITA. CIAT had already conducted field surveys and basic biological studies on several natural enemies of *P. herreni* in support of biological control of *P. manihoti* in Africa. Three encyrtid parasitoids [*Apoanagyrus* (*Epidinocarsis*) *diversicornis, Aenasius vexans*, and *Acerophagus coccois*] were identified as effective parasitoids of *P. herreni* (164, 165). All three parasitoids were attracted to cassava mealybug infestations (37). Surveys were conducted in nine states of NE Brazil to measure damage and collect natural enemies (61). The three parasitoids were then imported from CIAT and released in NE Brazil in 1994–1995 (158; JMS Bento, unpublished data). Within 6 months, *A. diversicornis* had migrated 130 km from its release site. *A. coccois* also became established and was recovered in high numbers at ≤180 km from its release site 9 months later. *A. vexans*, despite being consistently recaptured at its release site, did not migrate at all (158).

Whiteflies

As direct-feeding pests and vectors of plant viruses, whiteflies constitute a major problem in cassava production in Africa, the neotropics, and, to a lesser degree, Asia. The largest complex is in the neotropics, where 11 species are reported: *A. socialis, Trialeurodes variabilis, Bemisia tuberculata, Aleurothrixus aepim, B. tabaci* (19, 29, 30), *Bemisia argentifolii* (94), *Trialeurodes abutiloneus* (170), *Aleurodicus dispersus, Paraleyrodes* sp., *Aleuronudus* sp., and *Tetraleurodes* sp. (49). *A. socialis* is the predominant species in northern South America (19, 29, 30, 99) but is also found in Brazil and other areas (89). *A. aepim* is the predominant species in Brazil (89); *B. tuberculata* and *T. variabilis* are reported in low populations from Brazil, Colombia, and several other countries (87). The spiraling whitefly *A. dispersus* is reported to feed on cassava in Nigeria (2, 135), as does *Bemisia afer* in Kenya (131) and the Ivory Coast (92). *B. tabaci* has a pantropical distribution, feeding on cassava throughout Africa and several countries in Asia, including India (120) and Malaysia (140). *Aleurodicus* sp. is also reported feeding on cassava in India (138).

DAMAGE Whitefly feeding affects cassava in three ways. Direct damage is caused by feeding on the phloem of the leaves, inducing chlorosis and leaf fall, which results in considerable reduction in root yield if prolonged feeding occurs. Yield losses of this type are common in the neotropics owing to *A. socialis* and *A. aepim* activity (168; ARN Farias, personal communication, unpublished data). They also produce a honeydew, which provides a medium for sooty mold growth that can reduce yields (52). Most importantly, whiteflies are major vectors of cassava viruses (4, 80).

Two cassava viruses are known to be transmitted by whiteflies. African cassava mosaic disease (ACMD) is caused by several geminiviruses transmitted by *B. tabaci*. ACMD is reported causing crop losses of 28–40% from all African cassava-producing countries (159, 160); it has not been found in the neotropics. Until recently, the *B. tabaci* biotypes found in the Americas did not feed on cassava (71, 180, 181). It has been speculated that the absence of ACMD was related to the inability of its vector, *B. tabaci*, to colonize cassava. Since the early 1990s, however, a new biotype (B) of *B. tabaci*, considered by some to be a separate species (*B. argentifolii*; 145), has been reported feeding on cassava in the neotropics (United States, Dominican Republic, Ecuador, Brazil, Puerto Rico; 57, 94). Now that the B biotype has been found feeding on cassava in the Americas, ACMD poses a serious threat to cassava production, as most traditional varieties are highly susceptible to the disease. *B. tuberculata* is the reported vector of cassava frog-skin disease in the neotropics (4). Thus a continued strong research emphasis on whiteflies in cassava is justified.

ECOLOGY/BIOLOGY Most research efforts in the neotropics have concentrated on *A. socialis* and, to a lesser degree, *A. aepim*. Populations of both species are highest during the rainy season but are present throughout the crop cycle (91, 100, 107, 168). Egg to adult development time of *A. socialis* in growth chamber studies was 32 days (5). Field studies on the biology of *A. socialis* show an egg stage of 10–12 days and immature stages of 24–33 days (53).

CONTROL Research on whitefly control in the neotropics has emphasized host plant resistance (HPR) and biological control.

Cultural Intercropping cassava with cowpeas reduced egg populations of two whitefly species (*A. socialis* and *T. variabilis*) compared with those in monoculture. These effects were residual, persisting up to six months after harvest (106). Intercropping with maize did not reduce egg populations. Yield losses in cassava/maize, cassava monoculture, and mixed variety systems were ≈60%; whereas in cassava/cowpea intercrops, yield losses were only 12% (101, 104). Intercropping may offer the small-scale farmer an effective means of reducing pest populations (100).

Host plant resistance HPR offers a low-cost, sustainable solution to cassava losses from whitefly damage. HPR studies at CIAT with *A. socialis*, *T. variabilis*, and *B. tuberculata* were initiated some 15 years ago, and >3000 clones have been evaluated. Several sources of resistance have been identified, and the clone M Ecu 72 has consistently expressed the highest level of resistance. This and other selected clones were used in a crossing program to provide high-yielding, whitefly-resistant clones, which showed no significant differences in yield between insecticide-treated and untreated plots (59). Greenhouse and field studies showed that *A. socialis* feeding on resistant clones had less oviposition, longer development periods, reduced size, and higher mortality than those feeding on susceptible ones. *A. socialis* nymphal instars feeding on M Ecu 72 suffered 72.5% mortality (5, 60). Given that HPR to whiteflies is rare in cultivated crops, these results offer an encouraging control option. Whitefly-resistant cassava clones are being evaluated by CORPOICA, the Colombian agricultural research corporation, for subsequent release to farmers.

Biological Numerous natural enemies of the whitefly complex have been recorded (49, 103, 104). Recent surveys in Colombia show that the most representative group is the microhymenopteran parasitoid complex, which includes the genera *Encarsia, Eretmocerus*, and *Amitus* associated with *A. socialis, B. tuberculata*, and *T. variabilis* (49). The predominant species were *Encarsia hispida, Amitus* sp., and *Eretmocerus* sp. (undescribed). *A. socialis* was the most frequently parasitized whitefly species, and parasitism was higher in the Andean zone than in the coastal and eastern plains region of Colombia. Parasitism of *A. socialis* by *Eretmocerus aleurodiphagus* and *A. aleurodinus* ranged from 49–54% in certain regions (56). The predominant predator species was *Delphastus pusillus*, although predators appear to play a minor role (103). The fungal entomopathogen *cladosporium* sp. has been observed causing high mortality (82%) of *A. aepim* in cassava fields in Brazil (90).

Cassava Hornworm

E. ello (Lepidoptera: Sphingidae) is a serious pest of cassava in the neotropics (17, 29, 30), with a broad geographic range extending from southern Brazil, Argentina, and Paraguay to the Caribbean basin and the southern United States.

DAMAGE Although several species of *Erinnyis* feed on cassava, *E. ello* causes the worst damage. Severe attacks can cause complete plant defoliation, resulting in bulk root loss and poor root quality. Losses in root production are influenced by plant age, soil fertility, environmental factors (especially rainfall), and frequency of attack. In simulated damage studies, yield losses in fertile soils ranged from 0–25% after one attack, and up to 47% after two consecutive attacks. On infertile soil, losses varied from 15–45% after one attack, and up to ≤ 64% after two attacks (6). Losses in farmers' fields have been measured at 18% after one attack. After plants have reached six months of age, losses are less severe, although root quality may be adversely affected (88). Repeated attacks, which are more common in large cassava plantations, cause severe yield losses but do not kill the plant (17). The carbohydrates stored in the roots enable the plant to recover, especially during the more favorable rainy season.

Sporadic attacks usually coincide with the rainy season. The mass migratory flight capacity of *E. ello* (116, 117) accounts for the sudden "invasions" and subsequent increase in oviposition in cassava fields (6, 32). Although a large complex of natural enemies exists, their effectiveness is greatly reduced because of the migratory behavior of the hornworm adults (17, 174).

BIOLOGY Hornworm adults are nocturnal grey moths that oviposit round, light green to yellow eggs individually on the upper surface of cassava leaves. Eggs hatch in 3–5 days. In field cage studies, females oviposited an average of 450 eggs, although as many as 1850 eggs per female were observed. This fecundity partially explains the rapid buildup of hornworm populations. At 15, 20, 25, and 30°C, the average length of the larval stage is 105, 52, 29, and 23 days, respectively, indicating that peak hornworm activity may occur at lower altitudes (<1200 m) (15).

CONTROL *Chemical* Pesticides give adequate control if hornworm populations are detected and treated during the first three instars. Larval populations in the fourth and fifth instars are not only difficult to control but also uneconomical because considerable defoliation has already occurred. Moreover, pesticide use disrupts natural enemy populations and can lead to more frequent attacks (163).

Biological The large complex of natural enemies associated with *E. ello* has been extensively reviewed (15, 17, 18, 29, 30, 88, 153). More than 30 species of parasites, predators, and pathogens of the egg, larval, and pupal stages have been identified. Eight microhymenopteran species of the families Trichogrammatidae, Scelionidae, and Encyrtidae are egg parasites. *Trichogramma* and *Telenomus* spp. are the most important and have been studied in detail (15, 29, 30, 32, 105, 162). Natural parasitism by *Trichogramma* spp. ranges from 53–57%, which cannot reduce high hornworm populations below economic injury levels (15, 32). Releasing parasites to augment natural parasitism will increase levels of egg parasitism; however, it is difficult to synchronize releases to coincide with *E. ello* oviposition.

Tachinid flies are the most important dipteran parasite of *E. ello*. The Braconidae, particularly *Cotesia* spp., are the most important hymenopteran larval parasites (17, 88, 153). Several predator species feed on hornworm eggs, larvae, and pupae, the most important being *Polistes* spp. (Hymenoptera: Vespidae), *Podisus* spp. (Hemiptera: Pentatomidae), and various spider species. *Polistes* predation levels are determined primarily by the number of wasp larvae contained in the nest. Each *Polistes* larva consumes ≈0.5 hornworm larvae daily (15, 128).

Releasing predators and parasites to augment field populations is difficult and not economically feasible for resource-limited cassava farmers, as predator and parasitoid effectiveness are limited by poor functional response during hornworm eruptions, which last ≈15 days.

A granulosis virus (Baculoviridae) was found attacking *E. ello* in cassava fields at CIAT in the early 1970s. Subsequent studies have determined the value and potential use of this virus (8, 15, 17, 18, 60, 154). Infested larvae are collected from the field, macerated in a blender, filtered through cheesecloth, mixed with water, and applied to hornworm-infested fields. Mortality as high as 99.8% has been measured in field evaluations (18). The effect of virus concentration and mortality of larval instars showed a sigmoidal relationship for the first, second, and fourth instars. LC_{50} studies indicate that progressively higher concentrations are needed for adequate control of each succeeding larval instar. Most fifth instar larvae reached the prepupal stage, but few female adults emerged. They had wing deformities and died without producing progeny. The virus can be kept refrigerated (17).

The hornworm virus provides an attractive management option because of its ease of manipulation and storage at low cost. This technique was first implemented in southern Brazil, where light traps were used to detect adult movement and invasions. Virus applications were made when populations were in their early instars, resulting in almost complete control (154). Pesticide applications in certain regions were reduced by 60% (17, 41). In Venezuela the virus has replaced the use of pesticides on large cassava plantations (7,000 ha), where the hornworm is endemic, reaching levels of 60–390,000 adults on 1,000 ha of cassava during a semester. Populations are highest during the rainy season (50). Applications (70 ml/ha) can be made through overhead sprinkler irrigation systems when the larvae are in the first and second instars, resulting in 100% hornworm control and savings in pesticide costs. The direct cost of gathering, processing, storing, and applying the hornworm virus is just $4/ha (61, 119).

Cassava Burrower Bug

C. bergi is a polyphagous feeder that attacks a wide range of crop plants other than cassava. It has been reported only from the neotropics, with damage to cassava reported in Colombia (98), Panama (1), and Costa Rica (47). Its geographic distribution has yet to be determined, but an attempt is being made to map potential risk zones of infestation by means of the Geographic Information System (GIS; 147).

C. bergi was first recorded as a pest on cassava in Valle, Colombia in 1980 (98). Other hosts include onion, forage peanuts (*Arachis pintoi*), maize, sorghum, sugarcane, coffee, coriander, pasture grasses, potato, asparagus, and numerous weed species (28, 47, 48, 141, 147). Some host plants are strongly preferred over others (147). Optimal development and survival of *C. bergi* occurred on forage peanuts (*A. pintoi*) and maize; low-HCN cassava, sorghum, and welsh onion were less favorable hosts. *C. bergi* was unable to complete its development cycle on "bitter" cassava (fresh root parenchyma tissue concentration of cyanide >100 mg kg^{-1} HCN) (28, 147).

DAMAGE Nymphs and adults penetrate the root peel and parenchyma by means of a strong, thin stylet, feeding on the starch. Soil-borne pathogens (e.g. *Aspergillus, Diplodia, Fusarium, Genicularia, Phytophora*, and *Pythium* spp.) can penetrate the root parenchyma during wounding or feeding (31). Brown to black lesions develop on the root, rendering it commercially unacceptable (7).

BIOLOGY All five nymphal stages and the adult stage of *C. bergi* live in the soil, and populations are present throughout the cassava crop cycle. Root damage increases with plant age, reaching 70–80% of total roots and >50% reduction in starch content (7). *C. bergi* is strongly attracted to moist soil; it will migrate when soil water content is below 22% and is most persistent when it is >31% (147). The rainy season greatly favors adult and nymphal survival, behavior, and migration, whereas low soil water content during the dry season restricts adult burrowing and migration and increased nymphal mortality (147). Laboratory studies show that the lower temperature threshold for development is 14.7°C, and population growth is optimal at ≈26°C. Soil temperatures >31°C appear detrimental to egg eclosion and molting from the fifth instar to adult stage (147).

Both field trials and laboratory experiments indicate that cassava feeding preferences in *C. bergi* may be related to cyanogenic glucoside levels. Adults and nymphs that fed on a high-HCN clone had longer nymphal development, reduced egg production, and increased mortality (28, 55). An exponential decline in oviposition is observed with increasing levels of cyanogenic potential (CNP), beginning 12 days after exposure, especially at levels >150 ppm fresh weight. Oviposition on clones <45 ppm (CNP) was significantly higher, while oviposition varied on clones with a CNP between 45 and 150 ppm (147). Cyanogenesis did not, however, have a negative effect on adult survival.

Field trials have shown that low-HCN clones suffer more damage than high-HCN ones (28, 48). More recent research has found a negative relation between CNP and damage caused by *C. bergi*, indicating that CNP in cassava may act as a deterrent. Further research is required, however, as results are not conclusive (147).

CONTROL Controlling *C. bergi* is difficult because of its polyphagous nature and its adaptation to the soil environment. Pesticide applications are costly, environmentally hazardous, and not always effective (7, 48, 169). Intercropping cassava with *Crotalaria* sp. (sunne hemp) reduced root damage to 4%, compared with 61% in cassava monoculture (48). Yields were reduced by 22% when intercropped, so farmers have been reluctant to adopt this technology.

Recent studies indicate that entomopathogenic nematodes and fungi may offer a more acceptable solution for controlling *C. bergi*. The nematode *Steinernema carpocapsae* successfully parasitized *C. bergi* in the laboratory (44). A native species (*Heterorhabditis bacteriophora*), found parasitizing *C. bergi* in the field, has resulted in 84% average parasitism of the instars (14, 45, 61). The entomopathogenic fungus *Metarhizium anisopliae* has been observed parasitizing *C. bergi* in the field. In laboratory studies, mortality was highest (61%) during the fifth instar, while overall average mortality was 33% (60). The effectiveness of these biological control agents in the field remains to be evaluated.

Cassava Lacebugs

Several species of lacebugs (Hemiptera: Tingidae) have been reported feeding on cassava in the neotropics. In a recent review, Froeschner (96) identified five species of the genus *Vatiga* that show a decided preference for cassava: *V. illudens, V. manihotae, V. pauxilla, V. varianta*, and *V. cassiae*. The first two are the most widely distributed. *V. illudens* predominates in Brazil but is found throughout the Caribbean area. *V. manihotae* predominates in Colombia and Venezuela but is also reported from Cuba, Trinidad, Peru, Ecuador, Paraguay, Argentina, and Brazil. *V. varianta* is reported from Brazil and Colombia, *V. cassiae* from Brazil, and *V. pauxilla* from Argentina. In addition, the species *A. machalana*, referred to as the black lacebug, causes damage to cassava in Colombia, Venezuela, and Ecuador (57).

DAMAGE A prolonged dry period is favorable for increasing lacebug populations (149). Adults and nymphs feed on the undersurface of lower leaves. Initially, white feeding spots appear on the leaves, increasing in number and area until the leaf centers turn white and eventually tan. High lacebug populations will cause leaves to curl and die. Younger plants (4–5 months) attract higher populations, which tend to decline on older plants (149).

The relationship between damage, population density, and duration is unknown. A field trial at CIAT with a natural infestation by *A. machalana* resulted in 39% yield reduction compared with pesticide-treated plots (57). Field observations in Colombia and Ecuador indicate that populations of *A. machalana* are higher than other lacebug species (AC Bellotti, personal observation).

BIOLOGY The egg stage of *V. manihotae* is 8–15 days, followed by five nymphal stages averaging 16–17 days. Adult longevity under field conditions averages 40 days (38). Laboratory studies with *V. illudens* in Brazil show a nymphal duration of 13.5 days and average adult longevity of 27 days (86). In laboratory studies with *A. machalana*, the egg stage averaged 8.2 days, the five nymphal instars 14 days, and adult longevity 18 and 22 days for females and males, respectively (57).

CONTROL Lacebug control appears to be difficult, as few natural enemies have been observed (38, 85, 149). Preliminary screening of cassava germplasm indicates that HPR may be available (24, 46, 57), but considerable research is still required before implementation is possible.

TRENDS AND FUTURE CONSIDERATIONS

To develop the cassava crop to its full potential as a carbohydrate source, a concentrated research effort is required. Arthropod pest management research should concentrate on a more comprehensive understanding of the biodiversity in cassava cropping systems. Another important issue is that of quarantine measures to prevent the movement of pests, especially into Asia, in light of what occurred in Africa (95). There are several pests found in the neotropics that could potentially cause severe crop losses in these areas. These include the cassava hornworm, several mite species, lacebugs, and whiteflies. Moreover, what is considered a secondary pest in the neotropics at present could become a major pest outside its center of origin, where native natural enemies or tolerant germplasm are not available.

A successful integrated pest management (IPM) program in cassava will depend on having effective, environmentally sound, low-cost pest management technologies available to cassava farmers in developing countries. Biotechnology tools presently available offer the potential to develop improved pest-resistant varieties and to enhance the effectiveness of natural control organisms, including parasitoids and entomopathogens. The new generation of genetic pest management technologies presently being integrated with traditional IPM offers alternative technologies for controlling stemborers, leaf-cutting ants, grasshoppers, whitegrubs, and other difficult-to-control pests. Research activities in these areas are already underway and should be available to farmers in the near future.

IPM requires decision-making guides and strategies for appropriate implementation of control tactics. In traditional production systems, few options are available to resource-limited farmers. Choice of cultivar is often limited to local landraces, despite the diversity in germplasm banks, because of poor communication between research and extension agencies and farmers, the neglected status of cassava as a crop for marginal populations, and farmers' and consumers' strong local preferences for particular cultivars. Availability of clean planting material is also limiting, especially during periods of drought. Good stake sanitation greatly increases yield potential, but pathogen-free planting material is often scarce. Pesticides are seldom used in traditional systems given their prohibitive cost; nevertheless, pesticide use in Latin America is increasing, especially when cassava is grown on a plantation scale (23). The use of a baculovirus to control the cassava hornworm has been successful but requires refrigeration facilities (17). Cultural methods and planting systems are also highly influenced by local preferences. Because of these restrictions, cassava IPM systems are still rudimentary.

A promising approach to overcome slow technological diffusion and to increase productivity in small-scale production systems combines grass-roots community development with a holistic approach to crop management and diversification of markets. Pest control is often a low priority for poor farmers who depend on cassava as an "insurance crop" that requires minimal input. Moreover, the highly perishable nature of fresh cassava roots makes transport and marketing riskier (68), resulting in peaks in availability and low prices at harvest time. In some traditional cassava-producing areas, increased productivity has been tied to product diversification in local and regional markets to provide an acceptable return on farmers' investments (59, 175). This has been done most successfully by small-scale drying and chipping of cassava for animal feed (144). By opening new markets, profitability of cassava production increases, and pest control becomes economically viable through proper agronomic and phytosanitary practices, use of resistant cultivars, and biological and cultural control methods.

Area-wide classical biological control has had spectacular success in Africa; however, management of most cassava pests in Latin America will require farmer involvement in the identification and implementation of solutions. Participatory research methods have been developed and tested in Brazil, Colombia, and Ecuador (9, 10; B Ospina, unpublished data).

Classical biological control is largely independent of farmer input and, in the case of cassava, requires little or no modification of traditional practices. For the majority of pests not amenable to control through biological methods alone, farmer practices offer the best opportunity for intervention. IPM models for developing countries have emphasized integration of farmers' knowledge and their participation in experimentation (9, 10, 34, 35, 41). For example, improved cultivars must meet highly localized preferences for characters such as root color, texture, and growth habit. Attempts to incorporate pest resistance or higher yield must consider local preferences as selection criteria through farmer participation in germplasm evaluation (97).

Recent projects have emphasized direct involvement of international and national research agencies with growers to promote improved technologies and to inform the research and development process (60–63). Key to the success of such participatory research are identification of needs by farmers, inclusion of farmers in field research and validation, training of research and extension personnel in participatory methods, and integration of production with expanded markets. In Colombia and NE Brazil, these requirements have been accomplished through the organization of farmer research committees. By working with trained extension agents and researchers, farmers select problems to address, participate in the design of experiments, and conduct trials on their own land. Although it is still too early to judge the impact of this approach on cassava productivity, these efforts have been marked by enthusiastic response from growers and extension agencies. The concepts of community-based research and participatory methods have been adopted by state agencies and financed by regional banks. In some cases, communities have spontaneously adopted the model after visiting neighboring community research projects (62). The network of cassava farmer research organizations appears to be fostering communication and technology transfer and should contribute to adoption of IPM strategies and development of new technologies appropriate to the region's sociobiological environments. The infrastructure created by farmer organizations and research committees will be necessary to implement pest control tactics as they become available for incorporation into cassava IPM systems.

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Literature Cited

1. Aguilar JA, González A, López J, Concepción J, Gordon R, et al. 1991. *Evaluación y Control del Daño Ocasionado por el Chinche Subterraneo* Cyrtomenus bergi *Froeschner (Hemiptera: Cydnidae) en el Cultivo de la Yuca Var. Brasileña.* El Dorado, Panama: Inst. Invest. Agropec. Panamá (In Spanish)
2. Akinbosotu TA, Jackai LEN, Ntonifor NN, Hassan AT, Agyakwa CW. 1993. Spiralling whitefly *Aleurodicus dispersus* in Nigeria. *FAO Plant Prot. Bull.* 41(2):127–29
3. Alvarez JM, Acosta A, Bellotti AC, Braun AR. 1993. Estudios de patogenicidad de un hongo asociado a *Mononychellus tanajoa* (Bondar), ácaro plaga de la yuca (*Manihot esculenta* Crantz). *Rev. Colomb.*

Entomol. 19(1):10–20 (In Spanish)

4. Angel JC, Pineda BL, Nolt B, Velasco AC. 1990. Mosca blanca (Homoptera: Aleyrodidae) asociadas a transmisión de virus en yuca. *Fitopatol. Colomb.* 13:65–71(In Spanish)
5. Arias B. 1995. *Estudio sobre el comportamiento de la "mosca blanca"* Aleurotrachellus socialis *Bondar (Homoptera: Aleyrodidae) en diferentes genotipos de yuca, Manihot esculenta Crantz.* MS thesis. Univ. Nac. Colomb., Palmira. 181 pp. (In Spanish)
6. Arias B, Bellotti C. 1984. Pérdidas en rendimiento (daño simulado) causadas por *Erinnyis ello* (L.) y niveles críticos de población en diferentes etapas de desarrollo en tres clones de yuca. *Rev. Colomb. Entomol.* 10(3–4):28–35 (In Spanish)
7. Arias B, Bellotti AC. 1985. Aspectos biológicos y de manejo de *Cyrtomenus bergi* Froeschner, chinche de la viruela en el cultivo de yuca (*Manihot esculenta* Crantz). *Rev. Colomb. Entomol.* 11(2):42–46 (In Spanish)
8. Arias B, Bellotti AC. 1987. Control de *Erinnyis ello* (L) (Lep: Sphingidae) gusano cachón de la yuca *Manihot esculenta* (Crantz) con *Baculovirus erinnyis* NGV. *Rev. Colomb. Entomol.* 13(2):29–35 (In Spanish)
9. Ashby JA. 1990. Small farmers' participation in the design of technologies. In *Agroecology and Small Farm Development*, ed. MA Altieri, SB Heath, 23:245–55. Boca Raton, FL: CRC Press
10. Ashby JA, Gracia T, Guerrero MDP, Quirós CA, Roa JI, et al. 1995. *Institutionalising Farmer Participation in Adaptive Technology Testing with the 'CIAL.'* London: Overseas Dev. Inst. Agric. Res. Ext. Netw., Pap. 57. 43 pp.
11. Atu UG. 1993. Cultural practices for the control of termite (Isoptera) damage to yams and cassava in south-eastern Nigeria. *Int. J. Pest Manage.* 39(4):462–66
12. Ayanru DKG, Sharma VC. 1984. Changes in total cyanide content of tissues from cassava plants infested by mites (*Mononychellus tanajoa*) and mealybugs (*Phenacoccus manihoti*). *Agric. Ecosyst. Environ.* 12:35–46
13. Bakker FM, Klein ME, Mesa NC, Braun AR. 1993. Saturation deficit tolerance spectra of phytophagous mites and their phytoseiid predators on cassava. *Exp. Appl. Acarol.* 17(1–2):97–113
14. Barbarena MA. 1996. *Capacidad parasítica de dos razas del nematodo* Heterohabditis bacteriophora *Poinar (Rhabditida: Heterohabditidae) sobre la chinche de la viruela de la yuca (Cyrtomenus bergi Froeschner) (Hemiptera: Cydnidae) en condiciones de laboratorio.* BS thesis. Univ. Valle, Cali, Colomb. 89 pp. (In Spanish)
15. Bellotti AC, Arias B. 1988. *Manejo integrado de* Erinnyis ello (*L.*). Cali, Colomb.: Cent. Int. Agric. Trop.
16. Bellotti AC, Arias B. 1993. The possible role of HCN in the biology and feeding behavior of the cassava burrowing bug (*Cyrtomenus bergi* Froeschner: Cydnidae: Hemiptera). *Proc. Int. Sci. Meet. Cassava Biotechnol. Netw., 1st, Cartagena de Indias, Aug. 25–28, 1992,* pp. 406–9. Cali, Colomb.: Cent. Int. Agric. Trop.
17. Bellotti AC, Arias B, Guzmán OL. 1992. Biological control of the cassava hornworm *Erinnyis ello* (Lepidoptera: Sphingidae). *Fla. Entomol.* 75:506–15
18. Bellotti AC, Arias B, Reyes JA. 1990. Biological control of the cassava hornworm, *Erinnyis ello* (Lepidoptera: Sphingidae), with emphasis on the hornworm virus. *Proc. Symp. Int. Soc. Trop. Root Tuber Crops, 8th, Bangkok, 1988,* pp. 354–62. Bangkok: Dept. Agric. Thai., Centr. Int. Agric. Trop., Centr. Int. Papa
19. Bellotti AC, Braun AR, Arias B, Castillo JA, Guerrero JM. 1994. Origin and management of neotropical cassava arthropod pests. *Afr. Crop Sci. J.* 2(4):407–17
20. Bellotti AC, Braun AR, Yaninek JS, Neuenschwander P, Herren HR. 1987. Cassava agro-ecosystems and evaluation of pest complexes. *Proc. Int. Congr. Plant Prot., 11th, Manila, Oct. 5–9,* 2:81–89. Manila: Pest Control Counc. Philipp., Dept. Agric. Philipp., Univ. Philipp. Baños, Philipp. Counc. Agric. For. Nat. Res. Dev., Int. Rice Res. Inst.
21. Bellotti AC, Byrne D. 1979. Host plant resistance to mite pests of cassava. *Rec. Adv. Acarol.* 1:13–21
22. Bellotti AC, Byrne DH, Hershey CH, Vargas O, Varela AM. 1985. The potential of host-plant resistance in cassava for mites and mealybugs. In *Cassava: Research, Production and Utilization*, ed. JH Cock, JA Reyes, pp. 417–39. Cali, Colomb.: Cent. Int. Agric. Trop./United Nations Dev. Prog.
23. Bellotti AC, Cardona C, Lapointe SL. 1990. Trends in pesticide use in Colombia and Brazil. *J. Agric. Entomol.* 7:191–201
24. Bellotti AC, Hershey CH, Vargas O. 1987. Recent advances in resistance to insect and mite pests of cassava. *Proc. Workshop Cassava Breeding: Multidiscip. Rev., Manila, March 4–7, 1985,* pp. 117–46.

Cali, Colomb.: Cent. Int. Agric. Trop.

25. Bellotti AC, Kawano K. 1980. Breeding approaches in cassava. In *Breeding Plants Resistant to Insects*, ed. FG Maxwell, PR Jennings, pp. 314–35. New York: Wiley
26. Bellotti AC, Peña JE. 1978. Studies on the cassava fruit fly *Anastrepha* spp. *Proc. Cassava Prot. Workshop, Cali, Nov. 7–12, 1977*, pp. 203–8. Cali, Colomb.: Cent. Int. Agric. Trop.
27. Bellotti AC, Reyes JA, Varela AM. 1983. Observaciones de los piojos harinosos de la yuca en las Américas; su biología, ecología y enemigos naturales. In *Yuca: Control Integrado de Plagas*, ed. JA Reyes, pp. 313–39. Cali, Colomb.: Cent. Int. Agric. Trop. (In Spanish)
28. Bellotti AC, Riss L. 1994. Cassava cyanogenic potential and resistance to pests and diseases. *Acta Hortic.* 375:141–51
29. Bellotti AC, van Schoonhoven A. 1978. *Cassava Pests and Their Control.* Cali., Colomb.: Cent. Int. Agric. Trop. 71 pp.
30. Bellotti AC, van Schoonhoven A. 1978. Mite and insect pests of cassava. *Annu. Rev. Entomol.* 23(1):39–67
31. Bellotti AC, Vargas O, Arias B, Castaño O, Garcia C. 1988. *Cyrtomenus bergi* Froeschner, a new pest of cassava: biology, ecology and control. In *Symp. Int. Soc. Trop. Root Crops, Gosier (Guadeloupe), 7th, July 1–6, 1985*, ed. LM Degras, pp. 551–61. Paris: Inst. Nat. Rech. Agron.
32. Bellotti AC, Vargas O, Peña JE, Arias B. 1983. Pérdidas en rendimiento en yuca causadas por insectos y ácaros. In *Yuca: Investigation, Production y Utilization*, ed. C Domínguez, pp. 393–407. Cali, Colomb.: Cent. Int. Agric. Trop. (In Spanish)
33. Bennett FD. 1986. Biological control of *Phenacoccus manihoti* and *Mononychellus tanajoa*: why and how. *Proc. Int. Workshop Biol. Control HPR Control Cassava Mealybug Green Mite Afr., Ibadan, 1982*, pp. 6–14. Ibadan, Niger.: Int. Inst. Trop. Agric.
34. Bentley JW. 1990. Conocimiento y experimentos espontáneos de campesinos hondureños sobre el maíz muerto. *Manejo Integr. Plagas* 17:16–26 (In Spanish)
35. Bentley JW, Andrews KL. 1991. Peasants, pests and publications: anthropological and entomological views of an integrated pest management program for small-scale Honduran farmers. *Human Organ.* 50:113–22
36. Bernays EA, Chapman RF, Leather EM, McCaffery AR, Modder WWD. 1977. The relationship of *Zonocerus variegatus* (L.) (Acridoidea: Pyrgomorphidae) with cassava (*Manihot esculenta*). *Bull. Entomol. Res.* 67:391–401
37. Bertschy C, Turlings TCL, Bellotti A, Dorn S. 1997. Chemically-mediated attraction of three parasitoid species to mealybug-infested cassava leaves. *Fla. Entomol.* 80(3):383–95
38. Borrero HM, Bellotti AC. 1983. Estudio biológico del chinche de encaje (*Vatiga manihotae*) y uno de sus enemigos naturales. In *Yuca: Control Integrado de Plagas*, ed. JA Reyes, pp. 163–68. Cali, Colomb.: Cent. Int. Agric. Trop. (In Spanish)
39. Braun AR. 1986. *Contributions to the pest management of* Mononychellus progresivus *Doreste in cassava (Manihot esculenta Crantz).* PhD thesis. Univ. Calif., Davis. 97 pp.
40. Braun AR, Bellotti AC, Guerrero JM, Wilson LT. 1989. Effect of predator exclusion on cassava infested with tetranychid mites (Acari: Tetranychidae). *Environ. Entomol.* 18(4):711–14
41. Braun AR, Bellotti AC, Lozano JC. 1993. Implementation of IPM for small-scale cassava farmers. In *Crop Protection Strategies for Subsistence Farmers*, ed. MA Altieri, pp. 103–15. Boulder, CO: Westview
42. Byrne DH, Bellotti AC, Guerrero JM. 1983. The cassava mites. *Trop. Pest Manage.* 29(4):378–394
43. Byrne DH, Guerrero JM, Bellotti AC, Gracen VE. 1982. Yield and plant growth responses of *Mononychellus* mite resistant and susceptible cassava cultivars under protected vs. infested conditions. *Crop Sci.* 22(5–6):486–50
44. Caicedo AM, Bellotti AC. 1994. Evaluación del potencial del nematodo entomógeno *Steinernema carpocapsae* Weiser (Rhabditida: Steinernematidae) para el control de *Cyrtomenus bergi* Froeschner (Hemiptera: Cydnidae) en condiciones de laboratorio. *Rev. Colomb. Entomol.* 20(4):241–46 (In Spanish)
45. Caicedo AM, Bellotti AC. 1996. Reconocimiento de nematodos entomopatógenos nativos asociados con *Cyrtomenus bergi* Froeschner (Hemiptera: Cydnidae) en ocho localidades de Colombia. *Rev. Colomb. Entomol.* 22(1):19–24 (In Spanish)
46. Calvacante MLS, Ciociola AI. 1993. Variabilidade quanto au grau de resistência de cultivares de mandioca ao percevejo de renda em Pacajus - CE. In *Relat. Annu. Pesqui. 1980/92*. Fortaleza, Brazil: Empr. Pesqui. Agropec. Ceará (In Portuguese)

47. Carballo M, Saunders JL. 1990. Labranza del suelo e insecticidas: efecto sobre la incidencia de *Cyrtomenus bergi* Froeschner en maíz. *Turrialba* 40(2):165–68 (In Spanish)
48. Castaño O, Bellotti AC, Vargas O. 1985. Efecto del HCN y de cultivos intercalados sobre daño causado por la chinche de la viruela *Cyrtomenus bergi* Froeschner al cultivo de la yuca. *Rev. Colomb. Entomol.* 11(2):24–26 (In Spanish)
49. Castillo J. 1996. *Moscas blancas (Homoptera: Aleyrodidae) y sus enemigos naturales sobre cultivos de yuca (Manihot esculenta Crantz) en Colombia.* MS thesis. Univ. Valle, Cali, Colomb. 173 pp. (In Spanish)
50. Cedeño L, Salazar F, LaBerry R. 1997. Fluctuación poblacional de *Erinnyis ello* en yuca al sur del Estado Monagas. In *Resumen Cong. Venez. Entomol., 15th, Trujillo, Jul. 5–9*, p. 29. Trujillo, Venez.: Cent. "José Torrealba," Núcleo Univ. "Rafael Rangel"; Univ. Andes; Soc. Venez. Entomol. (In Spanish)
51. Centro Internacional de Agricultura Tropical. 1978. *Annual Report Cassava Program, 1977.* Cali, Colomb.: Cent. Int. Agric. Trop. 110 pp.
52. Centro Internacional de Agricultura Tropical. 1981. *Annual Report Cassava Program, 1980.* Cali, Colomb.: Cent. Int. Agric. Trop. 93 pp.
53. Centro Internacional de Agricultura Tropical. 1982. *Annual Report Cassava Program, 1981.* Cali, Colomb.: Cent. Int. Agric. Trop. 259 pp.
54. Centro Internacional de Agricultura Tropical. 1984. *Annual Report Cassava Program, 1982–83.* Cali, Colomb.: Cent. Int. Agric. Trop. 521 pp.
55. Centro Internacional de Agricultura Tropical. 1986. *Annual Report Cassava Program, 1985.* Cali, Colomb.: Cent. Int. Agric. Trop. 371 pp.
56. Centro Internacional de Agricultura Tropical. 1987. *Annual Report Cassava Program, 1986.* Cali, Colomb.: Cent. Int. Agric. Trop. 254 pp.
57. Centro Internacional de Agricultura Tropical. 1990. *Annual Report Cassava Program, 1989.* Cali, Colomb.: Cent. Int. Agric. Trop. 385 pp.
58. Centro Internacional de Agricultura Tropical. 1991. *Annual Report Cassava Program, 1990.* Cali, Colomb.: Cent. Int. Agric. Trop. 553 pp.
59. Centro Internacional de Agricultura Tropical. 1992. *Annual Report Cassava Program, 1987–1991.* Cali, Colomb.: Cent. Int. Agric. Trop. 475 pp.
60. Centro Internacional de Agricultura Tropical. 1994. *Annual Report Cassava Program, 1993.* Cali, Colomb.: Cent. Int. Agric. Trop. 325 pp.
61. Centro Internacional de Agricultura Tropical. 1995. *Annual Report, Cassava Program, 1994*, III:144–63. Cali, Colomb.: Cent. Int. Agric. Trop.
62. Centro Internacional de Agricultura Tropical. 1996. *Annual Report Project BP-1 Assessment of Past and Expected Impact of Research on Agriculture and Natural Resources Management.* Cali, Colomb.: Cent. Int. Agric. Trop. 60 pp.
63. Centro Internacional de Agricultura Tropical. 1997. *Annual Report Project BP-1 Assessment of Past and Expected Impact of Research on Agriculture and Natural Resources Management.* Cali, Colomb.: Cent. Int. Agric. Trop. 101 pp.
64. Chakupurakal J, Markham RH, Neuenschwander P, Sakala M, Malambo C, et al. 1994. Biological control of the cassava mealybug, *Phenacoccus manihoti* (Homoptera, Pseudococcidae), in Zambia. *Biol. Control* 4(3):254–62
65. Chaves MM, Pereira JS. 1992. Water stress, CO_2 and climate change. *J. Exp. Bot.* 43:1131–39
66. Cock JH. 1978. A physiological basis of yield loss in cassava due to pests. *Proc. Cassava Prot. Workshop, Cali, 1977*, pp. 9–16. Cali, Colomb.: Cent. Int. Agric. Trop.
67. Cock JH. 1982. Cassava, a basic energy source in the tropics. *Science* 218:755–62
68. Cock JH. 1991. The development context. In *Integrated Cassava Projects,* ed. CA Perez-Crespo, pp. 4–16. Cali, Colomb.: Cent. Int. Agric. Trop.
69. Cock JH, Porto MCM, El-Sharkawy MA. 1985. Water use efficiency of cassava. III. Influence of air humidity and water stress on gas exchange of field grown cassava. *Crop Sci.* 25:265–72
70. Cooke RD, Coursey DG. 1981. Cassava: a major cyanide containing food crop. In *Cyanide in Biology,* ed. B Vennesland, EE Conn, CJ Knowles, J Westley, F Wissing, pp. 93–114. London: Academic
71. Costa AS, Russell LM. 1975. Failure of *Bemisia tabaci* to breed on cassava plants in Brazil (Homoptera, Aleyrodidae). *Cienc. Cult.* 27:388–90
72. Cudjoe AR, Neuenschwander P, Copland MJW. 1993. Interference by ants in biological control of the cassava mealybug *Phenacoccus manihoti* (Hemiptera: Pseudococcidae) in Ghana. *Bull. Entomol. Res.* 83(1):15–22
73. Delalibera I Jr, Sosa-Gomez DR, de

Moraes GJ, Alencar JA, Farias-Araujo W. 1992. Infection of the spider mite *Mononychellus tanajoa* (Acari: Tetranychidae) by the fungus *Neozygites* sp. (Entomophthorales) in Northeast Brazil. *Fla. Entomol.* 75(1):145–47

74. de Moraes GJ, de Alencar JA, Wenzel-Neto F, Mergulhao SMR. 1990. Explorations for natural enemies of the cassava green mite in Brazil. *Proc. Symp. Int. Soc. Trop. Root Crops, 8th, Bangkok, Oct. 30–Nov. 5, 1988*, pp. 351–53. Bangkok: Dept. Agric. Thai., Centr. Int. Agric. Trop., Centr. Int. Papa
75. de Moraes GJ, Delalibera I Jr. 1992. Specificity of a strain of *Neozygites* sp. (Zygomycetes: Entomophthorales) to *Mononychellus tanajoa* (Acari: Tetranychidae). *Exp. Appl. Acarol.* 14:89–94
76. Diehl-Fleig E, Silva ME, Pacheco MRM. 1988. Testes de patogenicidade dos fungos entomopatogênicos *Beauveria bassiana* e *Metarhizium anisopliae* em *Atta sexdens prirventris* (Santsch, 1919) em diferentes temperaturas. *Cienc. Cult.* 40:1103–5 (In Portuguese)
77. Diehl-Fleig E, Silva ME, Specht A, Valim-Labres ME. 1993. Efficiency of *Beauveria bassiana* for *Acromyrmex* spp. control (Hymenoptera: Formicidae). *An. Soc. Entomol. Bras.* 22:281–85
78. Doreste E. 1984. Avance en la búsqueda de variedades de yuca (*Manihot esculenta* Crantz resistentes a los ataques de ácaros tetraníquidos. *Rev. Fac. Agron. (Maracay)* 13(1–4):85–90 (In Spanish)
79. Douro-Kpindou OK, Godonou I, Housson A, Lomer CJ, Shah PA. 1995. Control of *Zonocerus variegatus* with ULV formulations of *Metarhizium flavoviridae* conidia. *Biocontrol Sci. Technol.* 5:131–39
80. Dubern J. 1994. Transmission of African cassava mosaic geminivirus by the whitefly (*Bemisia tabaci*). *Trop. Sci.* 34:82–91
81. Dufour DL. 1988. Cyanide content of cassava (*Manihot esculenta*: Euphorbiaceae) cultivars used by Tukanoan Indians in Northwest Amazonia. *Econ. Bot.* 42:255–66
82. El-Sharkawy MA. 1993. Drought-tolerant cassava for Africa, Asia, and Latin America. *BioScience* 43:441–51
83. El-Sharkawy MA, Hernández ADP, Hershey C. 1992. Yield stability of cassava during prolonged mid-season water stress. *Exp. Agric.* 28:165–74
84. Ezulike TO. 1987. Resistance of cassava cultivars to green spider mite (*Mononychellus tanajoa*) infestation (abstract). *Proc. Trop. Root Crops; Root Crops Afr. Food Crisis, Triennial Symp. Int. Soc. Trop. Root Crops—Africa Branch, 3rd, Owerri, Nigeria, 1986*, p. 118. Ottawa, Ont., Can.: Int. Dev. Res. Cent.
85. Farias ARN. 1985. *Hyaliodes vitreus* (Hemiptera: Miridae), un predador de *Vatiga illudens* (Drake, 1773) (Hemiptera: Tingidae) em mandioca, na Bahia. *Rev. Bras. Mand.* 4(1):123–24 (In Portuguese)
86. Farias ARN. 1987. Biologia de *Vatiga illudens* (Drake, 1922) (Hemiptera: Tingidae) em laboratório. *Rev. Bras. Mand.* 6(1):17–19 (In Portuguese)
87. Farias ARN. 1990. Especies de "mosca blanca": situação atual e perspectiva de controle. Cruz das Almas, Brazil: Empr. Bras. Pesqui. Agropec., Cent. Nac. Pesqui. Mand. Fruticult. (In Portuguese)
88. Farias ARN. 1990. *O mandorová da mandioca. Problemas e meios de controle.* Cruz das Almas, Bahia, Brazil: Empr. Bras. Pesqui. Mand. Fruticult. 17 pp. (In Portuguese)
89. Farias ARN. 1994. Flutuação poblacional de *Aleurothrixus aepim* em mandioca, em São Miguel das Matas, Bahia. *Rev. Bras. Mand.* 13:119–22 (In Portuguese)
90. Farias ARN, Santos Filho HP. 1987. Ocorrência de *Cladosporium* sp. infectando a mosca blanca *Aleurothrixus aepim* (Goeldi, 1886) (Homoptera: Aleyrodidae) em mandioca no Estado da Bahia. *Rev. Bras. Mand.* 6(1):79–80 (In Portuguese)
91. Farias ARN, Sousa JDS, Silveira JRS. 1991. Flutuação populacional de *Bemisia tuberculata* em Maragogipe, Bahia. *Rev. Bras. Mand.* 10(1–2):103–7 (In Portuguese)
92. Fishpool LDC, Burban C. 1994. *Bemisia tabaci*: the whitefly vector of African cassava mosaic geminivirus. *Trop. Sci.* 34:55–72
93. Food and Agricultural Organization of the United Nations. 1997. FAOSTAT database http://apps.fao.org/default.htm
94. França FH, Villas-Boos GL, Branco MC. 1996. Occurrence of *Bemisia argentifolli* Bellow & Perring (Homoptera: Aleyrodidae) in the Federal District. *An. Soc. Entomol. Brasil* 25(2):369–72
95. Frison EA, Feliu E, eds. 1991. *FAO/IBPGR Technical Guidelines for the Safe Movement of Cassava Germplasm.* Rome: Food Agric. Org. United Nations, Int. Board Plant Genet. Res. 48 pp.
96. Froeschner RC. 1993. The Neotropical lace bugs of the genus Vatiga (Heteroptera: Tingidae), pests of cassava: new synonymies and key to species. *Proc.*

Entomol. Soc. Wash. 95(3):457–62

97. Fukuda WMG, Magalhães JA, Cavalcanti J, Pina PR, Tavares JA, et al. 1997. *Pesquisa Participativa em Melhoramento de Mandioca: Uma experiência no Semiárido do Nordeste do Brasil.* Cruz das Almas, Bahia, Brasil: Empr. Bras. Pesqui. Agropec.-Cent. Nac. Pesqui. Mand. Fruticult. 46 pp. (In Portuguese)
98. Garcia CA, Bellotti AC. 1980. Estudio preliminar de la biología y morfología de *Cyrtomenus bergi* Froeschner. Nueva plaga dc la yuca. *Rev. Colomb. Entomol.* 6(3–4):55–61 (In Spanish)
99. Gold CS. 1992. Ecology of cassava whiteflies in Colombia. *Proc. Root Crops Food Secur. Afr., Int. Soc. Trop. Root Crops—Afr. Branch, IITA.* Ibadan, Niger.: Int. Inst. for Tropical Agric.
100. Gold CS. 1993. Effects of cassava intercropping and varietal mixtures on herbivore load, plant growth, and yield: applications for small farmers in Latin America. In *Crop Protection Strategies for Subsistence Farmers*, ed. MA Altieri, 5:117–42. Boulder, CO: Westview
101. Gold CS. 1994. The effects of cropping systems on cassava whiteflies in Colombia: implications for control of African cassava virus disease. *Afr. Crop Sci. J.* 2:423–36
102. Gold CS, Altieri MA, Bellotti AC. 1989. Cassava intercropping and pest management: a review illustrated with a case study from Colombia. *Trop. Pest Manage.* 35(4):339–44
103. Gold CS, Altieri MA, Bellotti AC. 1989. The effects of intercropping and mixed varieties on predators and parasitoids of cassava whiteflies in Colombia: an examination of the "natural enemy hypothesis." *Bull. Entomol. Res.* 79:115–21
104. Gold CS, Altieri MA, Bellotti AC. 1989. Effects of intercrop competition and differential herbivore numbers on cassava growth and yields. *Agric. Ecosyst. Environ.* 26:131–46
105. Gold CS, Altieri MA, Bellotti AC. 1989. Relative oviposition rates of the cassava hornworm, *Erinnyis ello* (Lep.: Sphingidae), and accompanying parasitism by *Telenomus sphingis* (Hym: Scalinidae) on upper and lower leaf surfaces of cassava. *Entomophaga* 34(1):73–6
106. Gold CS, Altieri MA, Bellotti AC. 1990. Effects of intercropping and varietal mixtures on the cassava hornworm, *Erinnyis ello* L. (Lepidoptera: Sphingidae), and the stemborer, *Chilomima clarkei* (Amsel) (Lepidoptera: Pyralidae), in Colombia. *Trop. Pest Manage.* 36(4):362–67
107. Gold CS, Altieri MA, Bellotti AC. 1991. Survivorship of the cassava whiteflies, *Aleurotrachelus socialis* and *Trialeurodes variabilis* (Homoptera: Aleyrodidae) under different cropping systems in Colombia. *Crop Prot.* 10:305–9
108. Gutiérrez J. 1987. The cassava green mite in Africa: one or two species? (Acari: Tetranychidae). *Exp. Appl. Acarol.* 3(2):163–68
109. Hahn SK, Leuschner K, Ezeilo W, Carpenter AJ, Khatibu AI, et al. 1980. Resistance of cassava clones to cassava green mite, *Mononychellus tanajoa*. *Trop. Pest Manage.* 26(3):265–67
110. Henry G. 1995. *Global Cassava Sector Constraints and Estimated Future R&D Benefits.* Cali, Colomb.: Cent. Int. Agric. Trop. 39 pp.
111. Henry G, Gottret MV. 1995. *Global Cassava Sector Trends: Reassessing the Crop's Future.* Cali, Colomb.: Cent. Int. Agric. Trop. 45 pp.
112. Herren HR, Neuenschwander P. 1991. Biological control of cassava pests in Africa. *Annu. Rev. Entomol.* 36:257–83
113. Hershey CH. 1987. Cassava germplasm resources. *Proc. Cassava Breeding: Multidiscip. Rev., Manila, March 4–7, 1985,* pp. 1–24. Cali, Colomb.: Cent. Int. Agric. Trop.
114. International Institute of Tropical Agriculture. 1996. Ibadan, Niger.: Int. Inst. Trop. Agric.
115. Iziquel Y, Le Ru B. 1989. Influence de l'hyperparasitisme sur les populations d'un hymenoptere encyrtidae, *Epidinocarsis lopezi*, parasitoide de la cochenille du manioc *Phenacoccus manihoti* introduit au Congo. *Entomol. Exp. Appl.* 52(3):239–47 (In French)
116. Janzen DH. 1986. Biogeography of an exceptional place: What determines the saturniid and sphingiid moth fauna of Santa Rosa National Park, Costa Rica, and what does it mean to conservation biology? *Brenesia* 25/26:51–87
117. Janzen DH. 1987. When and when not to leave. *Oikos* 49:241–43
118. Jiron LF, Soto-Manitiu J, Norrbom AL. 1988. A preliminary list of the fruit flies of the genus Anastrepha (Diptera: Tephritidae) in Costa Rica. *Fla. Entomol.* 71(2):131–37
119. LaBerry R. 1997. La aplicación de un programa MIP en producción industrial de yuca. In *Mem. Congr. Fitopatol., Biodivers., Micorrizas*, CIAT, Cali, 18, pp. 136–37. Cali, Colomb.: Asoc. Colomb. Fitopatol. Cienc. Afines (In Spanish)
120. Lal SS, Pillai KS. 1981. Cassava pests and

their control in Southern India. *Trop. Pest Manage.* 27(4):480–91
121. Le Ru B, Iziquel Y, Biassangama A, Kiyindou A. 1991. Variations d'abondance et facteurs de regulation de la cochenille du manioc *Phenacoccus manihoti* (Hom.: Pseudococcidae) cinq ans apres l'introduction d'*Epidinocarsis lopezi* (Hym.: Encrytidae) parasitoide neotropical au Congo en 1982. *Entomophaga* 36(4):499–501 (In French)
122. Lohr B. 1983. Biología, ecología, daño económico y control de *Chilomima clarkei* barrenador de la yuca. In *Yuca: Control Integrado de Plagas*, ed. JA Reyes, pp. 159–61 (In Spanish)
123. Lomer CJ, Bateman RP, Godonou I, Kpindou D, Shah PA, et al. 1990. Field infection of *Zonocerus variegatus* following application of oil based formulation of *Metarhizium flavoviridae* conidia. *Biocontrol Sci. Technol.* 3:337–46
124. Lozano JC, Bellotti A, Reyes JA, Howler R, Leihner D, et al. 1981. *Field Problems in Cassava*, 2nd ed. Cali, Colomb.: Cent. Int. Agric. Trop. 205 pp.
125. Lozano JC, Bellotti AC, Vargas O. 1986. Sanitary problems in the production of cassava planting material. *Proc. Global Workshop Root Tuber Crops Propagation, Cali, 1983,* pp. 73–85. Cali, Colomb.: Cent. Int. Agric. Trop.
126. Lozano JC, Byrne D, Bellotti AC. 1980. Cassava/ecosystem relationships and their influence on breeding strategy. *Trop. Pest Manage.* 26(2):180–87
127. Maddison P. 1979. *Pests Associated with Cassava in the Pacific Regions; Auckland: Pacific Islands Pest Survey.* Auckland: Entomol. Div. Dept. Scientific & Industrial Res. 16 pp.
128. Martin CA. 1985. *Biología y comportamiento de* Polistes erythrocephalus *Ltr. Hymenoptera: Vespidae, predador del gusano cachón de la yuca*, Erinnyis ello *L. Lepidoptera: Sphingidae.* BS thesis. Univ. Nac. Colomb., Palmira. 121 pp. (In Spanish)
129. Modder WWD. 1994. Control of the variegated grasshopper *Zonocerus variegatus* (L.) on cassava. *Afr. Crop Sci. J.* 2(4):391–406
130. Montagnini F, Jordan CF. 1983. The role of insects in the productivity decline of cassava (*Manihot esculenta* Crantz) on a slash and burn site in the Amazon Territory of Venezuela. *Agric. Ecosyst. Environ.* 9:293–301
131. Munthali DC. 1992. Effect of cassava variety on the biology of *Bemisia afer* (Priesner & Hosny) (Hemiptera: Aleyrodidae). *Insect Sci. Appl.* 13(3):459–65
132. Mustaers HJW, Ezumah HC, Osiru DSO. 1993. Cassava-based intercropping: a review. *Field Crops Res.* 34 (3–4):431–57
133. Mwanza F. 1993. South American wasp comes to the rescue of cassava growers in Africa. *Bioscience* 43(7):452
134. Neuenschwander P. 1994. Control of cassava mealybug in Africa: lessons from a biological control project. *Afr. Crop Sci. J.* 2:369–83
135. Neuenschwander P. 1994. Spiralling whitefly *Aleurodicus dispersus*, a recent invader and new cassava pest. *Afr. Crop Sci. J.* 2(4):419–21
136. Ngeve JM. 1995. Progress report 1992–1994 on cassava regional research project for maize and cassava. *Proc. RRPMC/WECAMAN Joint Workshop, Cotonou, Benin, May 28–June 2, 1995,* pp. 1–15. Ibadan, Niger.: Int. Inst. Trop. Agric.
137. Norgaard RB. 1988. The biological control of cassava mealybug in Africa. *Am. J. Agric. Econ.* 70:366–71
138. Palaniswami MS, Pillai KS, Nau RR, Mohandus C. 1995. A new cassava pest in India. *Cassava Newsl. (CIAT)* 19(1):6–7
139. Pardo-Locarno LC, Franco-Cruz MP, Alarcón-Gaviria AA. 1995. Estudios preliminares de las chizas (Coleoptera: Lamellicornia) de San Antonio, Cauca. Registros y observaciones en Laparosticti y Pleurosticti. *Rev. Colomb. Entomol.* 21(1):51–57 (In Spanish)
140. Parker BI, Booth RH, Bellotti AC. 1978. *Bemisia tabaci* identified on *Manihot esculenta* in Malaysia. *Appl. Biol.* 7(1):85–86
141. Peairs FB, Carballo M. 1987. Effects of cropping system and insecticide protection on insects associated with maize (*Zea mays* L.) and cassava (*Manihot esculenta* Crantz). *Turrialba* 37:147–54
142. Pegoraro RA, Bellotti AC. 1994. Aspectos biológicos de *Pseudococcus mandio* Williams (Homoptera: Pseudococcidae) em mandioca. *An. Soc. Entomol. Bras.* 23(2):203–7 (In Portuguese)
143. Peña JE, Waddill V. 1982. Pests of cassava in South Florida. *Fla. Entomol.* 65(1):143–49
144. Perez-Crespo CA, ed. 1991. *Integrated Cassava Projects.* Cali, Colomb.: Cent. Int. Agric. Trop. 242 pp.
145. Perring TM, Cooper AD, Rodriguez RJ, Farrar CA, Bellows TS. 1993. Identification of a whitefly species by genomic and behavioral studies. *Science* 259:74–77
146. Renvoize BS. 1973. The area of origin of *Manihot esculenta*, as a crop plant—a

review of the evidence. *Econ. Bot.* 26:352–60

147. Riis L. 1997. *Behaviour and population growth of the burrower bug,* Cyrtomenus bergi *Froeschner: effects of host plants and abiotic factors.* PhD thesis. R. Vet. Agric. Univ., Copenhagen. 167 pp.
148. Rodríguez NS, Hershey C. 1990. Dos nuevas variedades de yuca de alta productividad para los Llanos Orientales. Cali, Colomb.: Cent. Int. Agric. Trop. 6 pp. (In Spanish)
149. Salick J. 1983. *Agroecology of the cassava lacebug.* PhD thesis. Cornell Univ., Ithaca, NY
150. Samways MJ. 1980. O complexo de artropodas da mandioca (*Manihot esculenta* Crantz) em Lavras, Minas Gerais, Brasil. *An. Soc. Entomol. Bras.* 9(1):3–10 (In Portuguese)
151. Sanico RT. 1990. Status of integrated pest management in root crops. In *Natl. Conf. Workshop Integr. Pest Manage. in Rice, Corn and Selected Major Crops, Laguna (Philipp.), Mar. 1–3.* College, Laguna (Philipp.). 17 pp.
152. Schaefers GA. 1978. Grasshoppers (*Zonocerus* spp.) on cassava in Africa. *Proc. Cassava Protection Workshop, Cali, Nov. 7–12, 1977,* pp. 221–26. Cali, Colomb.: Cent. Int. Agric. Trop.
153. Schmitt AT. 1983. Ocorrência de inimigos naturais de *Erinnyis ello* (L.) no estado de Santa Catarina. *Rev. Bras. Mand.* 2(2):59–62 (In Portuguese)
154. Schmitt AT. 1988. Uso de *Baculovirus erinnyis* para el control biológico del gusano cachón de la yuca. *Yuca Bol. Inf. (Colomb.)* 12:1–4 (In Spanish)
155. Schulthess F. 1987. *The interactions between cassava mealybug* (Phenacoccus manihoti *Mat-Ferr.*) *populations as influenced by weather.* PhD thesis. Swiss Fed. Inst. Technol., Zurich. 136 pp.
156. Seigler DS. 1992. Cyanide and cyanogenic glycosides. In *Herbivores, Their Interactions with Secondary Plant Metabolites,* ed. GS Rosenthal, MR Berenbaum, pp. 35–77. San Diego: Academic
157. Skovgård H, Tomkiewicz J, Nachman G, Münster-Swendsen M. 1993. The effect of the cassava green mite *Mononychellus tanajoa* on the growth and yield of cassava *Manihot esculenta* in a seasonally dry area in Kenya. *Exp. Appl. Acarol.* 17(1–2):41–58
158. Smith L, Bellotti AC. 1996. Successful biocontrol projects with emphasis on the neotropics. *Proc. Cornell Commun. Conf. Biol. Control, April 11–13, 1996, Cornell Univ.* Ithaca, NY: Cornell Univ. Press. 12 pp. http://www.nysaes.cornell.edu/ent/bcconf/talks/bellotti.html
159. Thresh JM, Fargette D, Otim-Nape GW. 1994. Effects of African cassava mosaic geminivirus on the yields of cassava. *Trop. Sci.* 34:26–42
160. Thresh JM, Fishpool DC, Otim-Nape GW, Fargette D. 1994. African cassava mosaic virus disease: an underestimated and unsolved problem. *Trop. Sci.* 34:3–14
161. Umeh EDN. 1991. Natural enemies in the control of pests of cassava, *Manihot esculenta* Crantz. *Insect Sci. Appl.* 12 (1–3):43–49
162. Urias López MA, Bellotti AC. 1989. Parasitismo y desarrollo de *Trichogramma exiguum* Pinto & Platner y *Telenomus sphingis* (Ashm) sobre *Erinnyis ello* (L.). *Agric. Tec. Mex.* 15(1):93–97
163. Urias López MA, Bellotti AC, Bravo Mojica H, Carrillo Sánchez JL. 1987. Impacto de insecticidas sobre tres parasitoides de *Erinnyis ello* (L.), gusano de cuerno de la yuca. *Agrociencia* 67:137–46
164. Van Driesche RG, Bellotti AC, Castillo JA, Herrera CJ. 1990. Estimating total losses from parasitoids for a field population of a continuously breeding insect, cassava mealybug, *Phenacoccus herreni* (Hemoptera: Pseudococcidae) in Colombia, S.A. *Fla. Entomol.* 73:133–43
165. Van Driesche RG, Castillo JA, Bellotti AC. 1988. Field placement of mealybug-infested potted cassava plants for the study of parasitism of *Phenacoccus herreni. Entomol. Exp. Appl.* 46:117–23
166. Van Schoonhoven A. 1974. Resistance to thrips damage in cassava. *J. Econ. Entomol.* 67:728–30
167. Vargas O. 1978. The white scale (*Aonidomytilus albus* Ckll) on cassava. *Proc. Cassava Prot. Workshop, Cali, Nov. 7–12, 1977,* pp. 199–202. Cali, Colomb.: Cent. Int. Agric. Trop.
168. Vargas O, Bellotti AC. 1981. Pérdidas en rendimiento causadas por moscas blancas en el cultivo de la yuca. *Rev. Colomb. Entomol.* 7(1–2):13–20 (In Spanish)
169. Vargas O, Bellotti AC, Arias B. 1986. Control de *Cyrtomenus bergi,* chinche de la viruela de la yuca. *Yuca Bol. Inf.* (Colomb.) 10(1):7–10 (In Spanish)
170. Vásquez LL, de la Iglesia M, López D, Jiménez R, Mateo A, et al. 1995. Moscas blancas (*Homoptera: Aleyrodidae*) detectadas en los principales cultivos agricolas en Cuba. *Manejo Integr. Plagas* 36:18–21 (In Spanish)
171. Veiga AF de SL. 1985. *Aspectos bioecologicos e alternativas de controle do acaro verde da mandioca* Mononychellus

tanajoa *(Bondar, 1938) (Acarina, Tetranychidae) no Estado de Pernambuco.* PhD thesis. Univ. São Paulo. 152 pp. (In Portuguese)

172. Vides OL, Sierra OD, Gómez HS, Palomino AT. 1996. El barrenador del tallo de la yuca *Chilomima clarkei* (Lepidoptera: Pyralidae) en el CRECED Provincia del Rio. *Bol. CORPOICA.* (Colomb.). 12 pp. (In Spanish)
173. Villegas GA, Bellotti AC. 1985. Biología, morfología y hábitos de *Laocheirus araneiformis* Linne (Coleoptera: Cerambycidae) barrenador de la yuca en Palmira, Valle del Cauca. *Acta Agron.* 35(4):56–67 (In Spanish)
174. Wallner WE. 1987. Factors affecting insect population dynamics: differences between outbreak and new outbreak species. *Annu. Rev. Entomol.* 32:317–40
175. Wheatley CC, Chuzel G. 1992. Cassava (*Manihot esculenta*) use as a raw material. In *Encyclopedia of Food Science, Food Technology and Nutrition*, ed. R Macrae, RK Robinson, MJ Sadler, pp. 734–43. London: Academic
176. Williams DJ. 1985. *Pseudococcus mandio* sp. (Hemiptera: Pseudococcidae) on cassava roots in Paraguay, Bolivia and Brazil. *Bull. Entomol. Res.* 75:545–47
177. Williams DJ, Granara de Willink MC. 1992. Mealybugs of Central and South America. Wallingford, UK: CAB Int. 635 pp.
178. Wilson WM. 1998. *Why bitter cassava* (Manihot esculenta *Crantz*): *productivity and perception of cassava in a Tukanoan Indian settlement in the northwest Amazon.* PhD thesis. Univ. Colo., Boulder. 206 pp.
179. Wongkobiat A. 1988. Insectos plaga de la yuca en Tailandia. *Yuca Bol. Inf. (Colomb.)* 12(1):5–7
180. Wool D, Calvert L, Constantino LM, Bellotti AC, Gerling D, et al. 1994. Differentiation of *Bemisia tabaci* (Genn.) populations in Colombia. *J. Appl. Entomol.* 117:122–34
181. Wool D, Gerling D, Nolt BL, Constantino LM, Bellotti AC, et al. 1989. The use of electrophoresis for identification of adult whiteflies (Homoptera: Aleurodidae) in Israel and Colombia. *J. Appl. Entomol.* 107:344–50
182. Yaninek JS. 1988. Continental dispersal of the cassava green mite, an exotic pest in Africa, and implications for biological control. *Exp. Appl. Acarol.* 4:211–24
183. Yaninek JS, Animashaun A. 1987. Why cassava green mites are dry season pests. *Proc. Semin. Agrometeorol. Crop Prot. in Lowland Humid and Sub-humid Tropics, World Meteorol. Org./Int. Inst. Trop. Agric., Contonou, Benin, July 7–11, 1986,* pp. 59–66. Ibadan, Niger.: Int. Inst. Trop. Agric.
184. Yaninek JS, Herren HR. 1988. Introduction and spread of the cassava green mite, *Mononychellus tanajoa* (Bondar) (Acari: Tetranychidae), an exotic pest in Africa, and the search for appropriate control methods: a review. *Bull. Entomol. Res.* 78:1–13
185. Yaninek JS, Mégevand B, de Moraes GJ, Bakker F, Braun A, et al. 1991. Establishment of the Neotropical predator *Amblyseius idaeus* (Acari: Phytoseiidae) in Benin, West Africa. *Biocontrol Sci. Technol.* 1(4):323–30
186. Yaninek JS, Onzo A, Ojo JB. 1993. Continent-wide releases of neotropical phytoseiids against the exotic cassava green mite in Africa. *Exp. Appl. Acarol.* 17(1/2):145–60
187. Yaninek JS, Saizonou S, Onzo A, Zannou I, Gnanvossou D. 1996. Seasonal and habitat variability in the fungal pathogens, *Neozygites* c.f. *floridana* and *Hirsutella thompsonii*, associated with cassava mites in Benin, West Africa. *Biocontrol Sci. Technol.* 6(1):23–33
188. Yaseen M. 1986. Exploration for natural enemies of *Phenacoccus manihoti* and *Mononychellus tanajoa*: the challenge, the achievements. *Proc. Biol. Control HPR Control Cassava Mealybug Green Mite Afr.; Int. Workshop IFAD/OAU/ISTRC/IITA, Dec. 1982,* pp. 81–102. Ibadan, Niger.: Int. Inst. Trop. Agric.