

SUSTAINABLE PRODUCTION

GUIDE



COLEAD

CASSAVA (*MANIHOT ESCULENTA*)



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1

INTRODUCTION

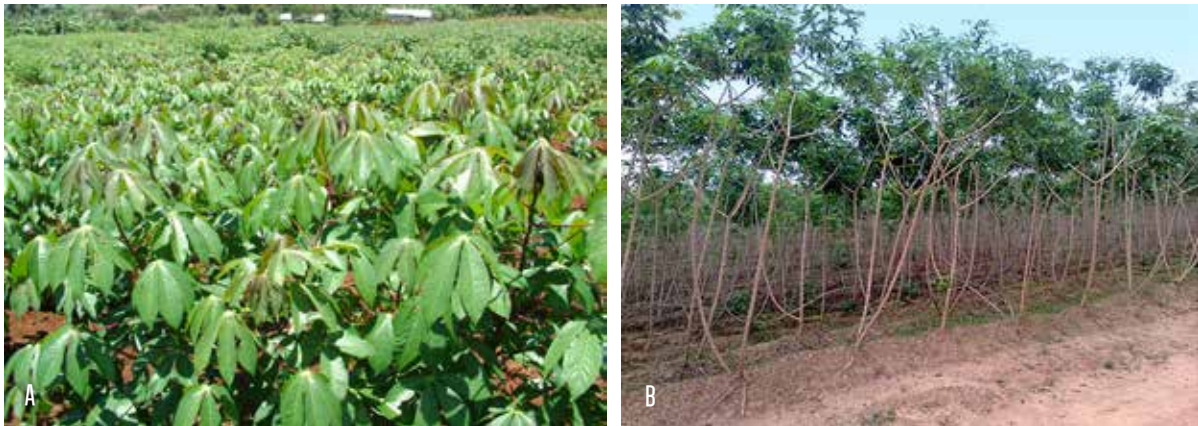


Figure 1 — (A) Young cassava plants; (B) mature cassava crop
 Source: PlantVillage, <https://plantvillage.psu.edu/topics/cassava-manioc/infos>

Cassava (*Manihot esculenta*, Crantz.), a plant (Figure 1A & B) in the family *Euphorbiaceae*, originated in South America, in the north-east of Brazil, and Central America (Onwueme, 1978). The crop was introduced in western Africa in the 16th century by the portugese, and later to east Africa at the end of the 17th century (Carter *et al.*, 1997). Currently, cassava is extensively cultivated in more than 100 countries in tropical and subtropical regions of the Americas, Africa and Asia (Lynam, 1991). It is primarily grown for its edible starchy root and is the third-largest source of food carbohydrates in the tropics after rice and maize. Cassava is a staple food for more than 800 million people worldwide (Waisundara, 2018). In addition, cassava leaves and young shoots are a good source of protein and vitamins.

There are both sweet and bitter varieties of cassava. The sweet varieties are mainly consumed fresh and can also be processed into various products. However, the roots and leaves of the bitter varieties contain anti-nutritional factors and toxins (cyanide) of up to 250 mg per kilogram of fresh root (Grace, 1977). These varieties must be processed (see chapter 9) before consumption to avoid acute poisoning and even death.

Cassava is vegetatively propagated and has a moderate to long growth cycle of 8–24 months. It is a pluriannual but usually grown as annual or biennial. Cassava is drought tolerant, with the ability to grow well even on marginal soils, partly following its strong associations with endomycorrhizae (Bellotti, 2008). The edible roots are usually cylindrical and tapered (Figure 2) and are white, brown or reddish in colour. The roots of late maturing (>9–24 months) varieties can remain in the soil without spoiling until needed, which makes these varieties suitable for commercialisation, requiring setting harvesting dates and use of large-scale storage and processing facilities. However, the early (6–9 months) maturing varieties tend to rot quickly if left in the soil after they reach maturity.



Figure 2 — crop yield

In Africa, cassava is mostly grown by small-scale farmers, for home consumption (90%) and sale (10%). The crop can potentially produce yields of 70 to 80 tons per hectare (Cock, 1985). However, most smallholder farmers achieve a much lower (7–9 tons) yield, because the crop is mainly grown on marginal and acidic soils and under unreliable rainfall conditions (Onayemi, 1982; Hahn, 1978).

Commercial farming of cassava involves simple farm operations, including: selection of suitable land, land preparation, planting, weeding, fertilisation, harvesting and processing. In general, the level of production inputs (fertilisers, plant protection and propagation) required is relatively low.

Processed cassava can be used to make several food forms (Figure 3) with diverse health benefits (Onwueme, 1978). As food, the fresh tubers contain approximately 62% water, 35% carbohydrate (starch), 1–2% protein, 0.3% fat, 1–2% fibres and 1% minerals (Onwueme, 1978). It is therefore:

- High in carbohydrates, a source of energy to the body.
- A source of vitamins and minerals such as manganese, calcium, phosphorus, potassium, and Iron.
- Rich in fibres and dietary fibre that helps prevent constipation.
- Filling when eaten, which helps easily feel full and achieve weight loss in weight management programs.
- Helpful in boosting the immune system and lowering blood pressure.



Figure 3 — by-products of cassava

Source: https://www.cassavaprocessingplant.com/faq/Uses_of_cassava_flour_processing_by_products_1450.html

In addition, cassava can be used in industry for the production of paper, ethanol, pharmaceuticals, biofuels, starch and flour. Cassava flour is increasingly used in baking bread, cookies and other confectioneries and in brewing beer (Figure 3). The leaves are used as vegetables and livestock (sheep, goats and cattle) feed (Onwueme, 1978).







2



CASSAVA REQUIREMENTS

2.1. CLIMATE

Cassava is a tropical crop that requires a humid-warm climate and flourishes in tropical and subtropical (latitudes of up to 30° north and 30° south) regions. Cassava grows best at temperatures ranging from 20 - 30°C and in areas with annual rainfall of 1,000 - 1,500 mm, which should be evenly distributed throughout the year (Onwueme, 1978). Cassava plants require at least 8 months of warm weather and regular rainfall, and are very sensitive to shading, which leads to poor growth and yield. The crop can adapt to adverse weather conditions, including drought spells, lower temperatures (17 - 20°C) at altitudes of 2,000 m and slight frosts (Cock, 1985). Figure 4 presents the Köppen-Geiger's climate classification (Table 1; Kottek *et al.*, 2006) and map of world dominant climate types (Figure 4; Chen *et al.*, 2013). The classification code defines the climate (precipitation, temperature and seasons) associated with ecological conditions, and serves as a good guide for identification of suitable areas to grow crops, including cassava.

Table 1 — Köppen and Geiger' classification code for global climatic and ecological conditions

TYPE	DESCRIPTION	CRITERION
A	Equatorial climates	$T_{\min} \geq +18^{\circ}\text{C}$
Af	Equatorial rainforest, fully humid	$P_{\min} \geq 60 \text{ mm}$
Am	Equatorial monsoon	$P_{\text{ann}} \geq 25 (100 - P_{\min})$
As	Equatorial savannah with dry summer	$P_{\min} < 60 \text{ mm in summer}$
Aw	Equatorial savannah with dry winter	$P_{\min} < 60 \text{ mm in winter}$
B	Arid climates	$P_{\text{ann}} < 10 P_{\text{th}}$
BS	Steppe climate	$P_{\text{ann}} > 5 P_{\text{th}}$
BW	Desert climate	$P_{\text{ann}} \leq 5 P_{\text{th}}$
C	Warm temperate climates	$-3^{\circ}\text{C} < T_{\min} < +18^{\circ}\text{C}$
Cs	Warm temperate climate with dry summer	$P_{\text{smin}} < P_{\text{wmin}}, P_{\text{wmax}} > 3 P_{\text{smin}}$ and $P_{\text{smin}} < 40 \text{ mm}$
Cw	Warm temperate climate with dry winter	$P_{\text{wmin}} < P_{\text{smin}}$ and $P_{\text{smax}} > 10 P_{\text{wmin}}$
Cf	Warm temperate climate, fully humid	neither Cs nor Cw
D	Snow climates	$T_{\min} \leq -3^{\circ}\text{C}$
Ds	Snow climate with dry summer	$P_{\text{smin}} < P_{\text{wmin}}, P_{\text{wmax}} > 3 P_{\text{smin}}$ and $P_{\text{smin}} < 40 \text{ mm}$
Dw	Snow climate with dry winter	$P_{\text{wmin}} < P_{\text{smin}}$ and $P_{\text{smax}} > 10 P_{\text{wmin}}$
Df	Snow climate, fully humid	neither Ds nor Dw
E	Polar climates	$T_{\text{max}} < +10^{\circ}\text{C}$
ET	Tundra climate	$0^{\circ}\text{C} \leq T_{\text{max}} < +10^{\circ}\text{C}$
EF	Frost climate	$T_{\text{max}} < 0^{\circ}\text{C}$

Note: Only precipitation parameters are provided for polar climates (E), but temperature conditions are presented (Kottek *et al.*, 2006)

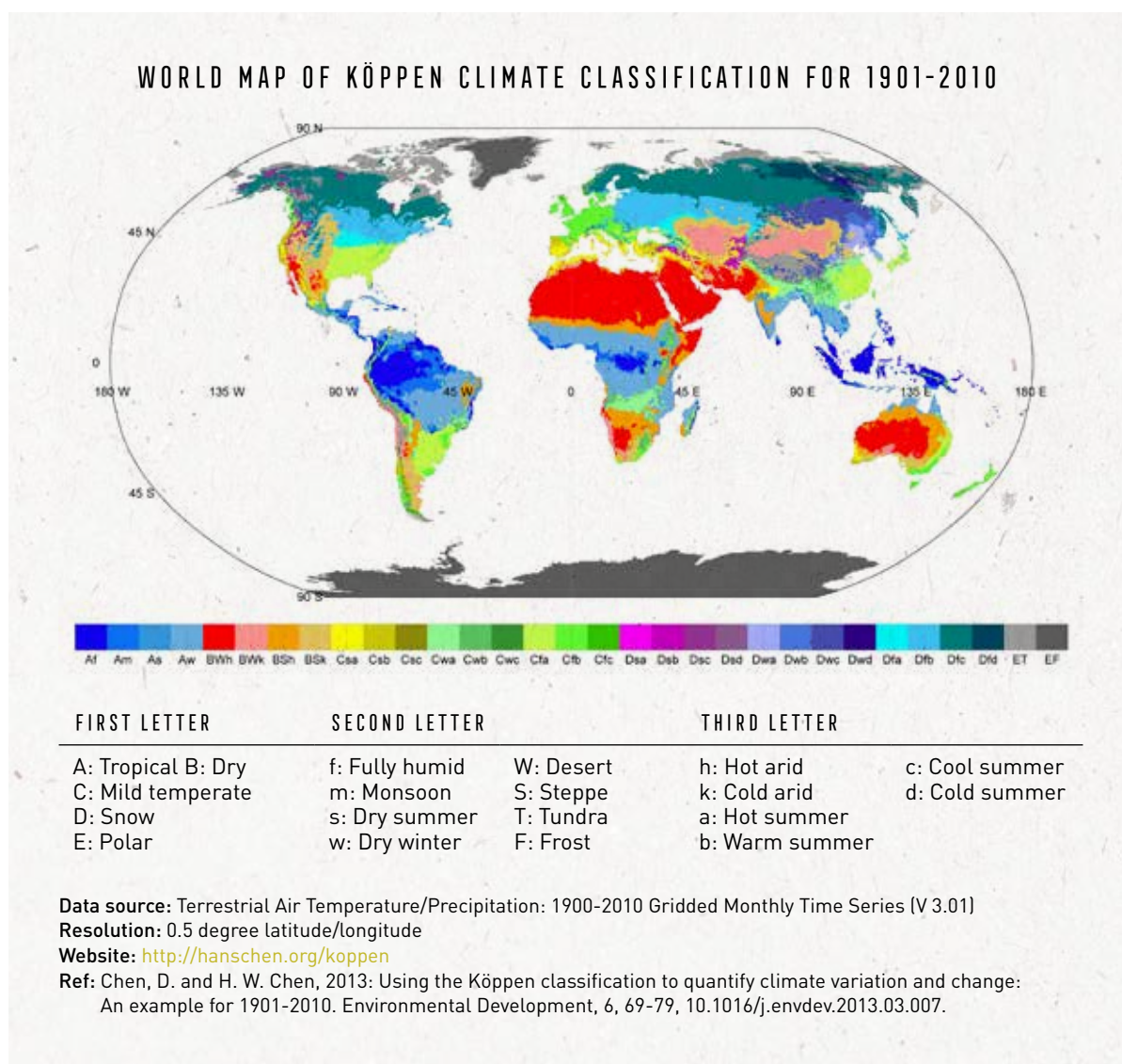


Figure 4 — World map of Köppen climate classification
Source: Chen *et al.* (2013); <http://hanschen.org/koppen>

2.2. WATER

Although the climate (rainfall and temperature) differs widely within and between African, Caribbean and Pacific (ACP) countries, for optimum growth and tuberisation cassava plants require an average of 1000 mm annually, or a minimum of 6 months with not less than 50 mm of rainfall per month (Hauser *et al.*, 2014). However, cassava can tolerate low soil moisture, but does not grow well in waterlogged conditions (Onwueme, 1978; Cock *et al.*, 1985). Irrigation water may be obtained from natural sources such as lakes, rivers, springs and valley dams to ensure water quality.

2.3. SOIL

Cassava can be grown in many types of soil but is most productive in well-draining, light sandy clay loam soils with a pH between 5.5 and 7.5. It prefers a slightly acidic soil. Stony soils inhibit the formation of the root tubers, while soils with stagnant water are unsuitable for the cultivation of the crop. Cassava needs medium soil fertility, and hence can grow well in marginal environments (Buyinza and Kitinoja, 2018), partly because of its strong associations with arbuscular mycorrhiza (Habte and Osorio, 2001; Okon *et al.*, 2010; Straker *et al.* 2010; Begoude *et al.*, 2016).

2.4. NUTRITION

Like all plants, cassava needs balanced nutrition for proper growth and development of root tubers (Jansson, 1980; Buyinza and Kitinoja, 2018). The primary macronutrients: nitrogen (N), phosphorus (P) and potassium (K) are required in large quantities. In contrast, the secondary macro- and micronutrients: magnesium, manganese, sulfur, zinc, iron, copper, boron, calcium and aluminium are needed in much lower amounts. Each of these nutrients has a unique function in supporting plant growth and development. Therefore, a shortage or excess in any of them could result in negative effects on the plant's growth and yield.

Table 2 shows an example of the general nutrient uptake and removal by cassava plants. The harvested tubers were found to contain large amounts of K compared to N and P in the ratio of 5:1:10 for NPK as opposed to the normal ratio of 7:1:7 in other crops (Vanlauwe *et al.*, 2008). It is important that these nutrients (NPK) are replaced following harvesting to restore soil fertility, using good soil management practices (Howeler, 1981; Susan John *et al.*, 2011). On average, cassava extracts about 4.91, 1.08, and 5.83 kg of N, P, and K respectively per ton of harvested tuber (Howeler, 1981)

Table 2 — Example showing general nutrient uptake and removal by cassava

YIELD	PLANT	N	P ₂ O ₅	K ₂ O
MT HA ⁻¹		KG HA ⁻¹		
37	Fresh roots	67	38	122
	Whole plant	198	70	220
18	Fresh roots	32	8	41
	Whole plant	95	23	77

Source: IFA, 1992. <https://www.worldcat.org/title/ifa-world-fertilizer-use-manual/oclc/802748292>

Several fertilisers are available on the market that could be used to provide the required nutrients needed for commercial cassava production (Table 3). More information on appropriate, site-specific soil fertility management may be obtained from the AKILIMO online resource [<https://akilimo.org/>].

Table 3 — Nutrient content (%) of fertilizers commonly used in cassava production

FERTILIZER		N	P ₂ O ₅	K ₂ O	MgO	CaO	S	OTHER
Urea	-	46						
Ammonium chloride	AC	25						66 Cl
Ammonium nitrate	AN	34						
Calcium nitrate	CN	15				26		
Calcium ammonium nitrate	CAN	27			2	4		
Ammonium sulphate	AS	21					24	
Monoammonium phosphate	MAP	11	48-55		0.5	2	1-3	
Diammonium phosphate	DAP	18-21	46-53				1-1.5	
Rock phosphate	RP		25-41			25-50		
Fused magnesium phosphate	FMP		12-20		10-15	12-16		
Single superphosphate	SSP		16-22			28	11-14	
Double superphosphate	SP36		32-36				5-6	
Triple superphosphate	TSP		44-53		0.5	12-19	1-1.5	
Potassium chloride	KCl			60-62				47 Cl
Potassium sulphate	SOP			50-53			17-18	
Potassium nitrate	KN	13		44	0.5	0.5	0.2	
Kieserite	Kies				27		22	
Langbeinite	SKMg			22 22	18		22	
Dolomite	GML				10-22	35-45		
Agrilime (calcite)	-					47		
Gypsum	-					22-30	13-16	
N-P-K 15-15-15	-	15	15	15				
N-P-K 16-16-8	-	16	16	8			1	
N-P-K 13-13-21	-	13	13	21				
N-P-K 12-12-17+2(Mg)+(TE)	-	12	12	17	2			Micro
N-P-K 15-15-6+4(Mg)	-	15	15	6	4			
N-P-K 5-18-10		5	18	10			8	
N-P-K 5-17-15		5	17	15				
N-P-K 8-14-7		8	14	7	34			

Source: Hauser *et al.* (2014)

2.4.1. NUTRITION DEFICIENCY SYMPTOMS

Inadequate amounts of macro - and micronutrients impair the growth and yield of cassava plants in diverse ways, and produce the deficiency symptoms described below.

NITROGEN

Nitrogen plays a major role in vegetative growth, leaf development and increasing the number of tuberous roots (Howeler and Spain, 1978). However, cassava plants respond better to N-fertilisers in the presence of high amounts of K, which is the reason why the two minerals should be well-balanced during application (Howeler and Spain, 1978; Muthuswamy and Rao, 1979). Nitrogen deficiency (Figure 5) results in uniform chlorosis of leaves beginning with the lower leaves and spreading throughout the plant (Howeler, 2012), cessation of leaf elongation (Marschner and Marschner, 2012), constrained photosynthesis (Gregoriou *et al.*, 2007), reduced chloroplast size (Li *et al.*, 2013), and slow plant growth. However, excess N application supports shoot growth at the expense of storage root formation (Cenpukdee and Fukai, 1991) and starch content (Nguyen *et al.*, 2002; Cruz *et al.*, 2003; Kaweewong *et al.*, 2013; Uwah *et al.*, 2013). Therefore, N should be applied with caution.



Figure 5 — Example of N-deficiency symptoms express as chlorosis/yellowing of older (lower) leaves, light green colour of the rest of the leaves and stunted growth:

Source: Nutritional disorders of cassava. file:///C:/Users/Owner/OneDrive%20-%20AgShare.Today/Private/Consultancy/COLEACP%20Consultancy/Calls%20WITH%20FRAMEWORK%20contracts/Call%20191134_Analytical%20and%20Technical%20Support%20Services/COLEACP_Cassava%20guide/Literature/Nutritional%20disorders%20in%20cassava.pdf [consulted on 5 January 2023]

PHOSPHOROUS

Phosphorus (P) is required during photosynthesis, metabolism of sugars, energy storage and transfer, cell division, cell enlargement, and transfer of genetic information (Malhotra *et al.*, 2018). However, due to soil fixation and/or adsorption of P, its availability in soil is usually low. Deficiency symptoms of phosphorus (Figure 6) appear as chlorotic lower leaves with the upper leaves looking healthy green. The lower yellow leaves eventually turn purple and shriveled. The plants have thin stems, narrow leaf lobes and poor root growth (Asher *et al.*, 1980).



Figure 6 — Example of P-deficiency symptoms: chlorotic lower leaves with healthy green-looking upper leaves. The lower yellow leaves eventually turn purple and shrivelled.

The plants also develop thin stems, narrow leaf lobes and poor root growth

Source: International Plant Nutrition Institute (IPNI), Better crops with Plant Food, Vol. XCIV (94) 2010, No. 1.
[http://www.ipni.net/publication/bettercrops.nsf/0/D0B1B69535D6EB7A8525797C0079C64A/\\$FILE/Better%20Crops%202010-1%20\(lo%20res\).pdf](http://www.ipni.net/publication/bettercrops.nsf/0/D0B1B69535D6EB7A8525797C0079C64A/$FILE/Better%20Crops%202010-1%20(lo%20res).pdf)

POTASSIUM

Potassium is required in high amounts for carbohydrate synthesis and translocation (Malavolta *et al.*, 1955). In addition, K also helps regulating the balance between nutrient assimilation and respiration, which are prerequisites for vigorous growth and the formation of reserve assimilates in the storage roots of cassava plants (Jansson, 1980). Potassium increases the diameter and weight of the tubers, storage cell size and number, and dry matter allocation. In addition, increase in K also improves

the starch and its quality parameters, i.e., amylose content, granule size, pasting temperature, viscosity and swelling volume (Nair and Aiyer, 1986). Potassium helps regulate stomatal opening and tolerance to water deficiency under drought conditions (Hasanuzzaman *et al.*, 2018).

K deficiency is considered as the most limiting nutritional constraint in cassava production, as K-deficient plants have lower starch content quality than those with sufficient K (Malavolta *et al.*, 1955). According to Howeler (1996), K-deficient plants show a variety of symptoms such as necrosis of leaf tips and margins (Figure 7A) and as stunted growth with drying and shedding of lower leaves (Figure 7B).



Figure 7 — K-deficiency symptoms: (A) necrosis of the tips and margins of cassava leaves and (B) stunted growth with drying and shedding of lower leaves

Source: K. Susan John, CTCRI. <https://www.ipipotash.org/publications/eifc-284>

Other K-deficiency symptoms express as:

- Short plants with many branches growing along the ground but with upwards turned tips.
- Very short zigzagging prematurely lignified upper internodes.
- Small and chlorotic leaves with a few lower yellow leaves that have black spots and boarder necrosis.
- Curling of leaf borders during drought and severe die-back in wet months due to induced anthracnose (*Colletotrichum* sp.).
- Where there is absence of recognizable symptoms, K-deficiency might be expressed as shorter plants with smaller leaves than plants with sufficient amounts of K.

MAGNESIUM

Magnesium (Mg) forms a basic component of chlorophyll and is essential for photosynthesis. Magnesium deficiency expresses itself most clearly as interveinal chlorosis in the lower leaves (Figure 8A). The chlorosis starts at the tips and margins of the leaf lobes and extends inwards between the veins towards the midrib with an evident bright yellow chlorotic tissue (Figure 8B). The chlorotic areas can be pale green to rather yellow (Figure 8C). However, under severe magnesium deficiency, the leaves become necrotic and develop a whitish or brownish colour (Figure 8D). Deficiency of magnesium is attributed to high concentrations of potassium and/or calcium, which tends to inhibit magnesium uptake via its antagonistic uptake mechanism (Spear *et al.*, 1987). This leads to shortage of chlorophyll, which results in poor and stunted plant growth. The symptoms are commonly observed in cassava grown on acid oxisols, ultisols, inceptisols and entisols (USDA soil taxonomy) (Howeler, 2012b).



Figure 8 — Example of magnesium deficiency symptoms: (A) interveinal chlorosis in lower leaves, (B) chlorotic tips and margins of leaf lobes and midrib veins, (C) pale green areas on leaves and (D) necrosis and whitish or brownish colouration of severely deficient leaves. Source: Asher *et al.* (1980)

MANGANESE

Manganese (Mn) plays an important role in plant growth and development: photosynthesis, respiration and pathogen defense (Mousavi *et al.*, 2011; Alejandro *et al.*, 2020). Symptoms of manganese deficiency appear as small dark brown spots along the veins of the lower leaves (Figure 9A). On severely affected plants the leaves show yellow or greenish colour (Figure 9B). The leaves hang soft and loosely and finally fall off the plant, affecting the growth of both the shoots and roots.



Figure 9 — Example of manganese deficiency symptoms: (A) small dark brown spots on veins of lower leaves, (B) yellowish and greenish colour on severely affected plants that are flaccid and may fall off
Source: Asher *et al.* (1980)

SULPHUR

Sulphur (S) is an important component of amino acids during protein synthesis (Narayan *et al.*, 2022). Sulphur deficiency symptoms express as uniform necrosis; first on the young leaves (Figure 10A) due to its limited mobility in the plant phloem (Krochmal and Samuels, 1968), but can also spread to the older leaves (Ngongi *et al.*, 1977) in the field (Figure 10B). The deficiency symptoms are similar to those of N deficiency and can be reduced by application of K_2SO_4 and other sources of sulphur.



Figure 10 — Example of sulphur deficiency symptoms:
(A) necrosis on young leaves and (B) necrosis on older leaves in the field
Source: Asher *et al.* (1980)

ZINC

Zinc (Zn) is a key component of various enzymes that catalyze metabolic reactions in plants. Deficiency of Zn causes interveinal chlorosis in the young leaves. This is followed by development of small white or light-yellow chlorotic patches between the veins (Figure 11A & B). The plants subsequently develop smaller and more chlorotic narrow leaves, with light green to white colour and upward curled margins (photo 8C). Under severe zinc deficiency conditions, the leaf tips become necrotic with the basal lobes pointing away from the stem (Figure 11B & C). The effects can occur on both young and older plants (Figure 11D). If the deficiency occurs early, plant growth is inhibited leading to stunted growth. However, when the situation is corrected by adding zinc, some plants can recover, with normal leaves, but with the older leaves indicating the symptoms suffered during deficiency period.



Figure 11 — Example of zinc deficiency symptoms: (A, B & C) interveinal chlorosis on young leaves, (C & D) light green to white colour and upward curled leaf margins on both young and older leaves
Source: Asher *et al.* (1980)

IRON

Iron (Fe) is required for metabolic processes like DNA synthesis, respiration and photosynthesis during synthesis of chlorophyll and maintenance of chloroplast structure and function (Rout and Sahoo, 2015). Iron deficiency expresses initially as chlorosis of the interveinal tissue but with green veins in the young leaves (Figure 12A). However, under more severe iron deficiency conditions, the veins lose the green colour and the whole leaf together with the petiole becomes yellow and later white (Figure 12B). The iron deficient plants develop distinct green veins against a yellow background, as opposed to manganese deficiency, which has a more diffuse boundary between the green and chlorotic tissues.

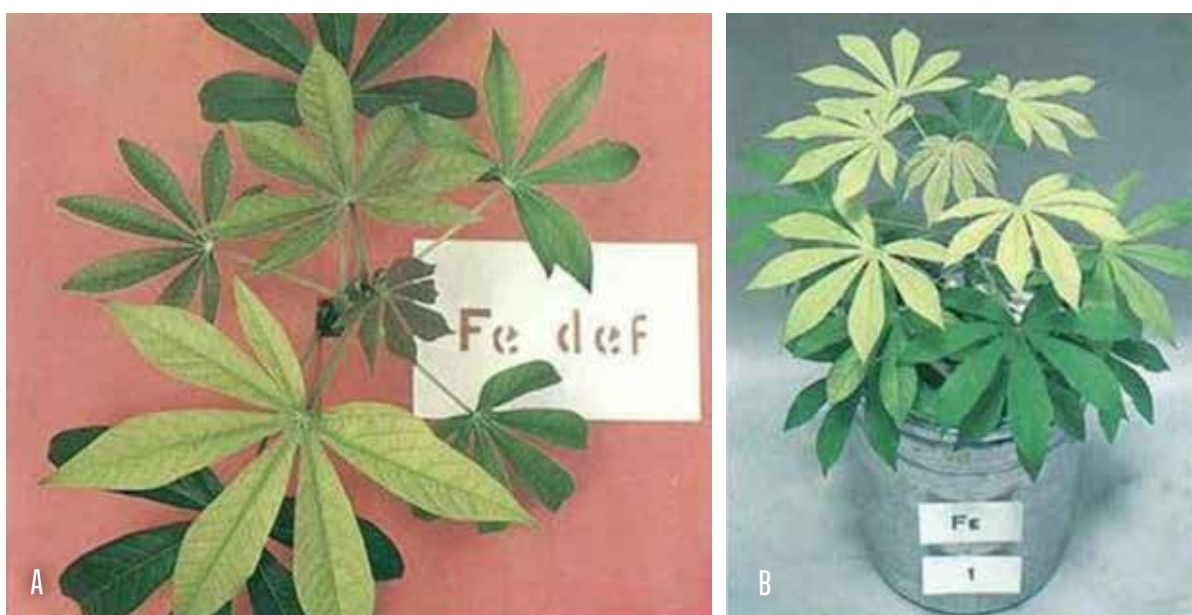


Figure 12 — Example of iron deficiency symptoms:
(A) chlorosis of interveinal tissue with green veins in young leaves
and (B) loss of green colour with yellow and white petioles
Source: Asher *et al.* (1980)

COPPER

Copper (Cu) plays a key role in several enzymatic activities in plants, including photosynthesis, respiration and the formation of lignin in cells (Pietrini *et al.*, 2019). Copper deficiency is expressed as chlorosis and deformity of the young leaves sometimes with necrotic leaf tips that are curled upwards (Figure 13A) or downwards (Figure 13B). The internodes are elongated on stems, with abnormally drooping petioles (Figure 13B & C).



Figure 13 — Example of copper deficiency symptoms: (A) chlorosis and deformed necrotic and curled young leaves, and (B & C) elongated and abnormally drooping petioles
Source: Asher *et al.* (1980)

BORON

Boron is important in plant cell wall formation and stability, structure and function of biological membranes, movement of sugar in phloem, pollination and seed set (Whittington, 1957). Hence it is a vital nutrient in enhancing agricultural productivity (Saleem *et al.*, 2011). In cassava, Boron (B) deficiency (Figure 14A) manifests itself as whitish or brownish necrotic spots on the lower leaves, with darker tissues around them (Figure 14B). The affected leaves develop ragged edges (Figure 14C) and the plants are stunted (Figure 14D).



Figure 14 — Example of boron deficiency symptoms: (B) whitish or brownish necrotic spots on the lower leaves, (C) ragged edges and (D) stunted plants. Source: Asher *et al.* (1980)

CALCIUM

Calcium is important for supplying and regulating water in the plant since it is a phloem immobile element. However, in the field, Ca deficiency symptoms are rarely noticeable unless in very acid soils. In such soils, severe Ca deficiency causes curling and puckering of leaftips and margins on young leaves; yellowing of leaf margins on older leaves, as well as limiting the fibrous root system and stunting of plants (Howeler, 2012b).

ALUMINIUM

Aluminium may increase plant growth by enhancing phosphorus availability when in combination with other minerals (Muhammad *et al.*, 2018). Aluminium deficiency bears similar symptoms as calcium deficiency, which include chlorotic lower leaves (Figure 15A), interveinal chlorosis and necrosis and pointed deformities of the young leaves (Figure 15B), believed to be due to aluminium-induced calcium deficiency (Asher *et al.*, 1980).

Cassava is tolerant to high levels of aluminium and manganese in the soil, but does not thrive well in extremely sandy, salt affected, clayey or waterlogged soils. Therefore, organic matter, including poultry manure, cow dung, household waste and compost should be used to provide both nutrients and organic carbon, which improve the soil's physical properties, water retention and microbial activity, all leading to a 'healthy soil' with fewer pests and diseases.

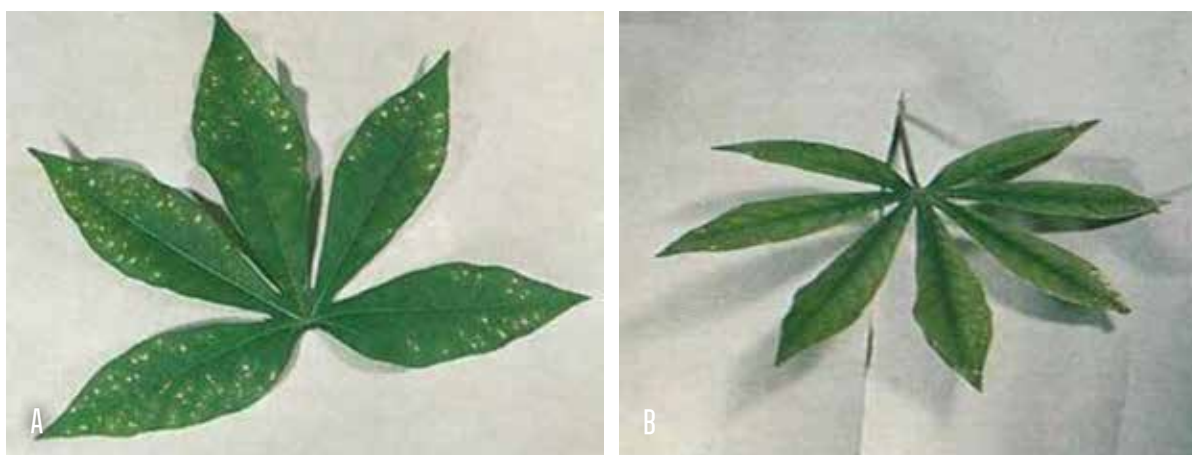


Figure 15 — Example of aluminium and calcium deficiency symptoms: (A) chlorotic lower leaves and (B) interveinal chlorosis and necrosis and pointed deformities of the young leaves
Source: Asher *et al.* (1980)



CASSAVA PRODUCTION REQUIREMENTS

KEY POINTS SUMMARY

Although cassava grows well in most tropical agro-ecologies, it has specific climatic & environmental, water, soil and nutritional requirements that should be observed for high yields and sustainable production, including:

CLIMATIC AND ENVIRONMENTAL FACTORS

- Annual rainfall: 750 - 1500 mm;
- Relative humidity: humid-warm;
- Temperature: 20 - 30°C;
- Altitude: <1500 masl.

NB: Consult the Köppen and Geiger' classification code for global climatic and ecological conditions (Table 1) and geographical distribution of world vegetations (Figure 1).

WATER

- Mean annual rainfall - approx. 1000 mm;
- 6 months with not less than 50 mm of rainfall per month;
- Avoid waterlogged conditions;
- Requires qualitative irrigation water from natural sources such as lakes, rivers, springs and valley dams.



CASSAVA PRODUCTION REQUIREMENTS

SOIL

- Well-draining, light sandy clay loam soils;
- pH between 5.5 and 7.5;
- Stones inhibit the formation of the root tubers;
- saline soils not suitable for cassava growth.

NUTRITION

- Balanced application of N, P & K;
- Essential minerals: Boron, Manganese, Magnesium, Iron, Copper, Zinc, Sulphur, Aluminium and Calcium.





3

PRE-PLANTING CHOICES

CHOOSE FARMING PRACTICES THAT REDUCE THE IMPACT ON THE ENVIRONMENT

1. Higher tubers yields can be obtained with **improved varieties or cultivars adapted to local conditions** on the condition that sufficient inputs (fertilizers & water) are used and that **GAPs and IPM** strategies are implemented.
2. Maintain species **diversity** in cultivated areas by keeping parcels of land surrounded by **hedges** and by adopting **crop rotation, intercropping** and **associated crops**.
3. Avoid destroying natural habitats by land clearance and the use of fire to manage weeds, which lead to the disappearance of beneficial fauna and flora.

To know more: browse the [COLEAD brochures](#) on the subject.

3.1. CHOOSING A PLOT

Although cassava can be grown on most soils, it grows best in well-drained sandy clay loam without a fluctuating water table. Look out for uncultivated or fallow land (2-5 years) or natural indicators of good soil fertility such as presence of leguminous plant species for nitrogen fixation or earthworm casts for good soil aeration. However, often cassava is placed as the last stage after clearing forests and before fallow in shifting cultivation. In such systems restoring soil fertility is crucial. Areas with steep slopes should be avoided since they are prone to soil erosion (Hauser *et al.*, 2014).

3.2. PLANTING MATERIAL

It should be noted that cassava is originally a cross polinator producing seeds. However, germinating seeds to propagate cassava is solely used in improvement programmes. Hence, in commercial production it is vegetatively propagated using stem cuttings. Therefore, select quality cassava planting material by considering: age (maturity) of stems, number of nodes, thickness quality, health of the stems (free from pests and diseases), quantity and preparation of stem cuttings.

3.2.1. AGE OR MATURITY OF STEMS

Stem cuttings should be taken from freshly harvested mature plants, which are between 8 and 18 months old (Figure 16A). The much younger (<8 months) stems with green bark or still bearing leaves (Figure 16B) are susceptible to pests and diseases and dry rapidly during drought, which leads to poor sprouting and crop establishment, and hence should be avoided. The mature parts of the stem at the middle and bottom (Figure 17) have better formed nodes than the younger/green parts at the top of the shoot (George *et al.*, 2000). However, avoid much older (>18 months) stems that are more lignified and contain small amounts of nutrients which cannot support sprouting of the young shoots. The young shoots may be used in rapid multiplication (see section 3.6.2, on rapid cassava multiplication) to quickly produce large numbers of planting materials for newly released varieties or varieties in short supply.



Figure 16 — Showing (A) mature and (B) immature/green cassava stems
 Source: (A) <https://www.sciencedirect.com/science/article/abs/pii/B9780444525123001510>;
 (B) <https://www.sciencedirect.com/science/article/abs/pii/B9780128000182000224>

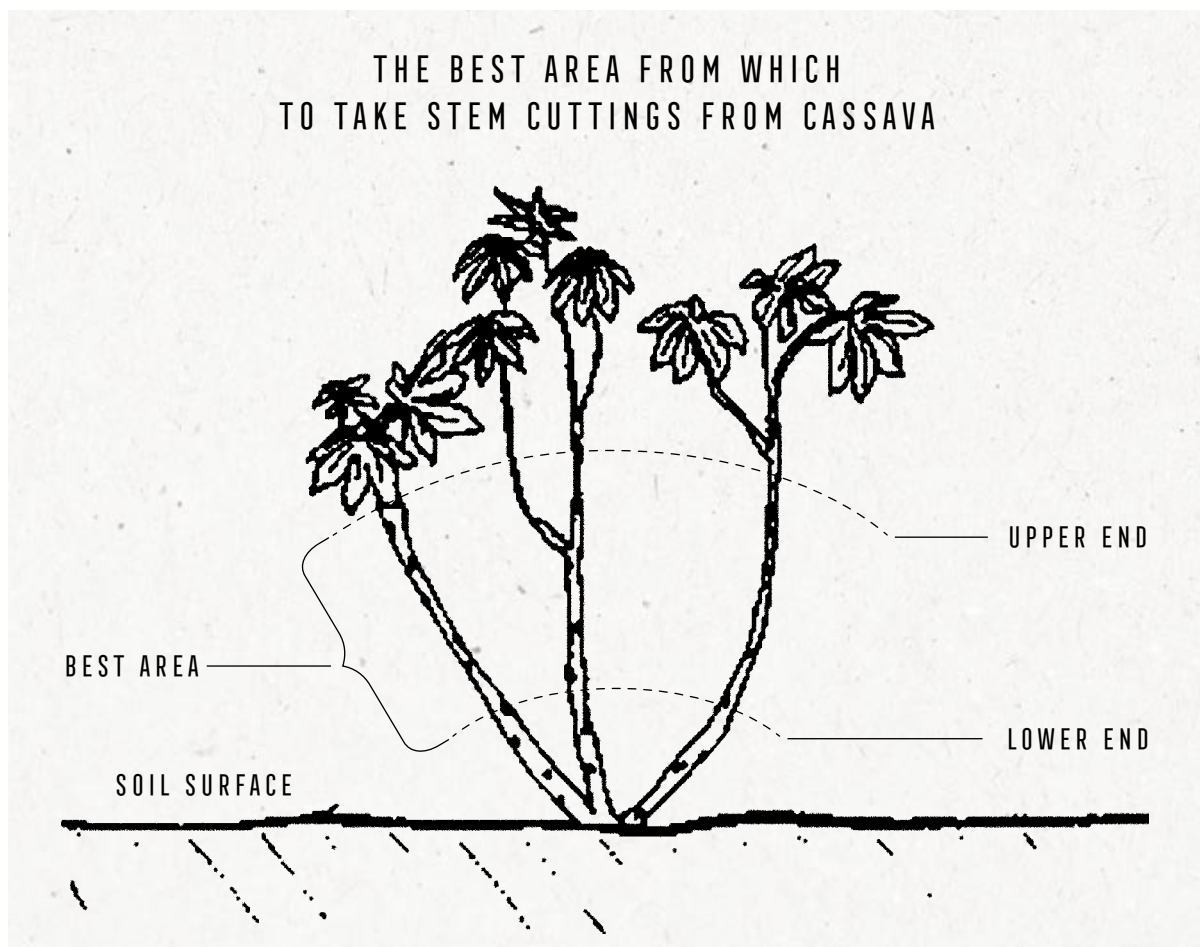


Figure 17 — Showing the best parts of the stem to take cuttings for proper germination of cassava plants
 Source: <https://nwdistrict.ifas.ufl.edu/phag/files/2017/06/Cassava-Cutting-Selection-1.png>

3.2.2. NUMBER OF NODES

Select stems with well-spaced and at least 5-7 nodes per 15-30 cm long cuttings. It is the nodes that grow into shoots and origin of the roots that form the tubers (Figure 18A). Avoid stem cuttings with few or too many nodes.

3.2.3. THICKNESS QUALITY

The thickness of the stems determines the amount of nutrients and moisture to support proper sprouting. Therefore, obtain cuttings from medium-large sized (2.5 to 3 cm diameter) stems (Figure 18A) that may produce more healthy (strong) sprouts and tuberous roots than thin-sized stems (Figure 18B) (Hauser *et al.*, 2014).



Figure 18 — Showing (A) medium to thick good quality and (B) thin poor quality cassava stem cuttings

Source: https://cropgenebank.sgrp.cgiar.org/images/cassava/cassava_stakes.jpg;

<https://image.shutterstock.com/image-photo/cassava-stem-cuttings-ready-be-600w-1499120570.jpg>

3.2.4. HEALTH OF CASSAVA STEMS

Select planting material from healthy plants while still in the field by carefully looking for absence of the key pests (mites, mealybugs, scales) and diseases (cassava bacterial blight, cassava viral diseases – mosaic and brown streak) that affect stem quality, to control further spread. Check the plants for physical presence of insect developed stages (nymphs and adults) and the typical disease symptoms. Cassava stem cuttings may also be dipped in Deltamethrin (2.5%) or Cypermethrin (150g/L) to kill insect pest eggs, nymphs and adults.

3.2.5. QUANTITY OF CUTTINGS

Using the agronomic recommendations for cassava in a monocropping system, a spacing of 1 m x 1 m between plants and between rows, 1 acre (4,000 m²) of branching (Figure 19A) cassava varieties need 4,000 cuttings, while 1 hectare (10,000 m²) requires 10,000 cuttings. In contrast, non-branching (Figure 19B) cassava varieties can be planted at a spacing of 0.8 m between plants and 1 m between rows, i.e., 0.8 m x 1 m, requiring at least 5,000 and 12,500 cuttings per acre and hectare (Abass *et al.*, 2014), respectively. The number of cuttings required for high density planting for 1 acre are 5,333 and 8,000 at a spacing of 1 m x 0.75 m and 1 m x 0.5 m, respectively. For high density planting in 1 hectare, 13,333 and 20,000 cuttings are needed at plant spacing of 1 m x 0.75 m and 1 m x 0.5 m (Hauser *et al.*, 2014), respectively. Always prepare extra cuttings to cater for the damaged and misplaced cuttings during planting.

Therefore, the number of cuttings to prepare depends on:

- Cropping system (mono or intercrop);
- Method of planting;
- Type of cassava variety (branching or non-branching);
- Plant spacing;
- Method of land preparation (flat field, ridges).



Figure 19 — Showing (A) branching and (B) non-branching/straight/upright growing cassava varieties

Source: <https://pbs.twimg.com/media/FbpVyHWXgAMh1sR?format=jpg&name=large>;
<https://encrypted-tbn0.gstatic.com/images?q=tbn:ANd9GcQ-5mE5LMFUKpowW8VfQte75TmMft1-ozhtsE906GcMLhtR9X5bFuGG3A3CW17vbvUtRd8&usqp=CAU>

3.2.6. VARIETIES

Globally, there is a wide diversity in cassava varieties, both developed for commercial and subsistence production. The diverse biotic and abiotic stresses demand that breeding programs continuously develop and produce new varieties that meet the production challenges and market demanded traits. Local varieties are generally lower yielding and may be more susceptible to pests and diseases, which impact yields (<35 tonnes per Ha). However, there is an increase in the development and distribution of improved cassava varieties with higher yield potential and resistance to biotic stresses and market demanded traits (Table 4). Therefore, it is always advisable to seek the advice of a local extension agent on the most suitable varieties to grow in each specific area (Howeler, 2007).

In general, the following characteristics are regarded as beneficial:

- Tolerance to pests and diseases;
- Early maturity/bulking (<8 months);
- High yielding;
- Suitability for inter cropping production systems (i.e, high branching varieties);
- High in dry matter (>30 %) content;
- Mealiness, i.e., the ability of the roots to cook well;
- Long in-ground storage without deterioration. This helps prolonged piece-meal harvesting;
- Long shelf life that prolongs the period over which cassava can be sold;
- Market acceptability;
- Use as sweet (fresh consumption) or bitter (processing) markets.

Table 4 (below) presents the selected varieties promoted for production in the last 10 years in Africa, Caribbean and the Pacific (not reported).

Table 4 — Improved cassava varieties promoted in selected ACP countries

AFRICA	CARIBBEAN AND SOUTH AMERICA	PACIFIC
NIGERIA (IITA BULLETIN_2556, 2020)	CARRIBEAN (CIAT, 2017)	Not reported
• IBA961632 (Farmer's Pride) • IBA980581 (Dixon) • CR36-5 (Ayaya) • IBA070593 (Sunshine) • IBA980505 (Fine Face) • TME 419 • TMS13F1160P0004 (Game Changer) • TMS13F1343P0022 (Obasanjo-2) • NR130124 (Hope) • TMEB693 (Poundable).	• Corpoica Belloti • Corpoica Sinuana • Corpoica Ropain	
	BARABADOS (LOPEZ ET AL., 2016)	
	• White stick • Butterstick (Brown stick) • M. Mex 23 • M. Mex 55 • Tacana	

AFRICA	CARIBBEAN AND SOUTH AMERICA	PACIFIC
GHANA (ACHEAMPONG, 2021)	GUYANA (LOPEZ <i>ET AL.</i> , 2016)	Not reported
• Ampong • Broni Bankye • Sika bankye • Otuhia • CRI-Duade Kpakpa • CRI-Amansan bankye • CRI-AGRA bankye • CRI-Dudzi • CRI-Abrabopa • CRI-Lamesese • BetaCarotene	Uncle Mack, Bad woman, MMex 59, MMex 52, Butter stick, Mexican, Brown stem, Kairuni Cassava, White stick, Smokey Prolific, Santa Rosa Magenta, Yankee	
TANZANIA (TARAWALI <i>ET AL.</i> , 2013)	JAMAICA (LOPEZ <i>ET AL.</i> , 2016)	Not reported
• TME 419 • 96/1632 • 98/0505 • 92/0326	Blue Bud, Cuba Sweet, Prison Farm, Real Sweet, Rockwood, Manson, Sweet cassava, Bitter cassava, CM516, CM849, CM3306-4, CM3299, CM6119, CM523-7, CM2772-3, CM2776-5, MCOL 22, MCOL 1505, BRA383, PER183	
UGANDA (ABACA <i>ET AL.</i> , 2021)	GRENADA (LOPEZ <i>ET AL.</i> , 2016)	Not reported
• NASE 19 • NAROCASS 1 • NAROCASS 2 • NASE 14 • NASE 19	MCOL 1468, Señorita Blanca, CMC-40, Valencia	
	DOMINICA (LOPEZ <i>ET AL.</i> , 2016)	
	Bitter, Butter Stick, White Stick, 100 pounds	
MOZAMBIQUE (RT-IIAM, 2014)	TRINIDAD AND TOBAGO (LOPEZ <i>ET AL.</i> , 2016)	Not reported
• Orera • Eyope • Colicanana • Mokhalana • Nziva	Blue bud, Butter stick, CIAT hybrid, Maracas black stick, White stick, Pickney mumma, sweet cassava, Bitter cassava, MMex 59, MMex 60, MMex 61, CM3306-4, CM3299, CM6119-5, CM523-7, BRA383, M PER183, CM2772-3, CM2766-5, MCOL 2215, MCOL 1505	

3.3. GROWING PERIOD

Cassava can be grown all year round following the rainfall patterns and agro-ecologies of the different countries (Howeler, 2007). The growing period varies from 8 to 12 months for early maturing varieties and may continue to 24-36 months for the late varieties. However, the market is key in determining the length of growth of cassava. Markets for fresh/sweet varieties mainly demand early maturing varieties (8 to 12 months), while markets for processed cassava need the tubers to reach the full potential to maximise the starch content, which starts at 12 to 36 months, depending on the variety (Howeler, 2007).

Most ACP countries experience distinct dry and wet seasons and a mono-modal rainfall pattern. Although cassava is able to grow well in drought prone areas (El-Sharkawy, 2012), it is advisable to plant the crop during the wet season to enable good germination (80-90%) and root formation (Howeler, 2007). However, in areas with bi-modal rainfall or uncertain rainfall patterns, staggered planting, where new crops are planted while older crops are still in the field, can be practiced to maximise crop yields and mitigate risks of adverse rainfall patterns. Doing so, a more continuous supply of cassava tubers can be achieved. This practice is still prevalent among subsistence farmers, who harvest piecemeal for home consumption, and replant on the same place they removed mature plants. This has the risk of exposing the young plants to pests and diseases, which could impede their growth. The best plan would be to establish entirely new cassava plots at least 200 m away from the older plots, and have both young and older crops growing alongside each other in a single season. However, where farmers can afford irrigation, staggered planting could also be practiced in single growing season areas. More information on staggered planting of cassava and site specific advice can be found here (<https://akilimo.org/>)

3.3.1. AFRICA

In Africa, rainfall patterns differ between the eastern, western, central and southern zones. In East Africa, there are two rainy seasons that run from March to May/June (long rains) and November to December (short sporadic rains). The long dry season runs from July to October and a short dry season from January to February each year. In West Africa, there are two rainy seasons in many countries; the long rains running from April to mid-July, and shorter rains in September and October, and the dry season running from November to April. Northern parts of West Africa experience less rainfall with only one rainy season, which runs from July to September. Countries in Central Africa such as Gabon, the Democratic Republic of Congo (DRC) and the Republic of the Congo, which near the equator experience heavy rainfall all year round, with no clear dry season. In Southern Africa, the rainy season runs from November to April, which is the main growing season. The dry season runs from June to October. Across Africa, the annual average rainfall ranges between 775 -1,700 mm and above in central Africa's tropical rainforest (Alahacoon *et al.*, 2021).

3.3.2. CARIBBEAN

In the Caribbean, the wet season runs from May to November and a dry season from December to April. Caribbean countries are divided into six rainfall zones, based on the pattern of rainfall received. In most zones, the wet season has a clear bi-modal pattern with a rainfall peak in coming in May or June and another in September and December. The region has four rainfall periods that define the Caribbean climate, including: early rainfall season (May-June), the mid-summer dry season (July-August), the late rainfall season (September-November) and the early dry season (December-April). The rainfall peaks come at different times in each zone. Zones 1 and 6 (far western and southern Caribbean), receive at least 2 to 27 mm/month of rainfall. Furthermore, part of zone 6 (south Caribbean) has rainfall peaking in December/January and in June. Zones 3 and 4 (central Caribbean) receive less rainfall (2 to 17 mm/month), while zone 5 (Lesser Antilles) has a unimodal pattern of rainfall which peaks in July/August (Climate Studies Group Mona, 2020).

3.3.3. PACIFIC

Pacific countries experience hot (June - July) and wet summers (August and November) with relatively long daylight and cold and dry winters (April to July), with occasional frost at higher latitudes. The highest yields were obtained for cassava planted early in the wet season and the highest dry starch content in the middle of the dry season (Howeler, 2007).

3.4. PLACEMENT OF CASSAVA CUTTINGS DURING PLANTING

There are three methods of placement of cassava stem cuttings (Figure 20A-C) during planting, including: horizontal, vertical and slanting/tilted or inclined placement (Amponsah *et al.*, 2017).

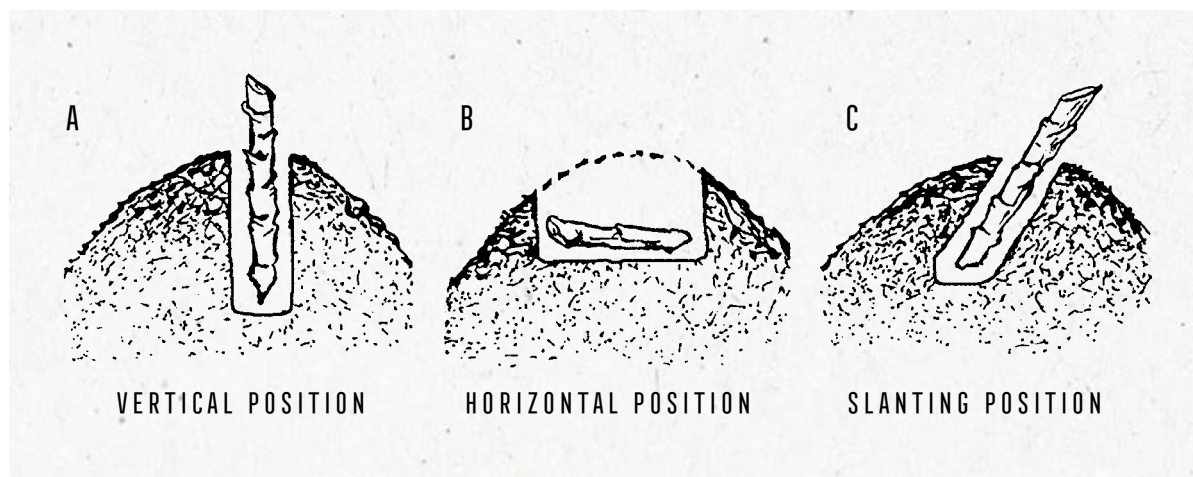


Figure 20 — Showing cassava stem cuttings placement methods:

(A) vertical; (B) horizontal and (C) slanting, tilted or inclined on ridges during planting

source: https://www.researchgate.net/profile/Shadrack-Amponsah/publication/313667385/figure/fig1/AS:461433566699520@1487025743956/Different-cassava-planting-positions_W640.jpg

3.4.1. VERTICAL PLACEMENT

Vertical planting (Figure 20A) is used mostly in soils with high water levels or in areas of high rainfall to avoid rotting due to constant dampness. However, in the dry season, vertical planting is prone to rapid dehydration and drying of the cuttings and young shoots.

3.4.2. HORIZONTAL PLACEMENT

With horizontal planting (Figure 20B), the cuttings are buried at least 5–10 cm below the soil surface. These cuttings produce multiple stems and many small tuberous roots. Horizontal planting is often used in the dry season.

3.4.3. SLANTING/TILTED/INCLINED PLACEMENT

With the slanting/tilted/inclined (Figure 20C) planting method, cuttings are placed in the ground at a 45° angle, leaving two or three nodes above ground level. This type of planting is suitable for areas with a moderate amount of rainfall. Slanted planting usually allows for easier harvesting of cassava tubers compared to horizontal or vertical planting. However, the horizontal (Figure 21A) planting method kills off the early developmental stages of pests attached to the stems when buried in the soil, while both the vertical and slanted/tilted/inclined methods (Figure 21B, below) expose the young cassava plants to early infestation and/or infection by pests and diseases.

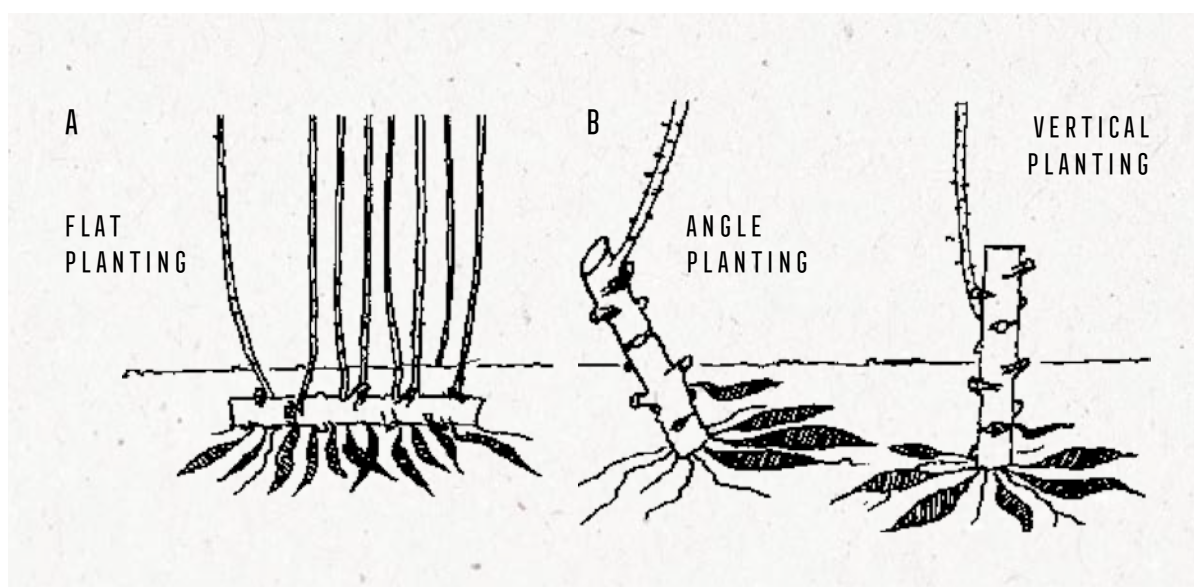


Figure 21 — Showing cassava cuttings placement methods:
(A) flat or horizontal; (B) angle or tilted and (C) vertical and their effect on development of tubers in the ground
Source: http://www.uq.edu.au/_School_Science_Lessons/64.15.GIF

3.5. MULTIPLICATION OF CASSAVA PLANTING MATERIAL

Multiplication of cassava planting materials can be achieved using three methods: conventional, rapid multiplication and micropropagation/tissue culture techniques.

3.5.1. CONVENTIONAL MULTIPLICATION

Conventional multiplication of cassava planting material is carried out in open fields using regular cuttings (15-35 cm, with 5 - 8 nodes) of the selected improved varieties, similarly to the general practice of cassava production for consumption. Close spacing of 0.5 m x 0.5 m is used to achieve high density to encourage rapid stem growth. The block should be at least 200 meters away from other cassava crops for breeder seed and 100 m for basic seed and certified seed to keep the plants from infection by pests and diseases. However, this method has a low multiplication ratio (1:10) compared to the rapid multiplication techniques (see section 3.5.2) which yields X60-100 more cuttings (Moyo *et al.*, 2004).

a. Plant health management

- Multiplication fields/blocks for producing healthy cassava planting materials should be situated in isolated areas from high pest and disease infection, i.e., with no or low infection (<5% infection) by pests (mites, mealybugs, scales) and diseases (cassava bacterial blight, cassava viral diseases – mosaic and brown streak). The block should also be at least 200 m away from old cassava crops.
- The multiplication blocks should be divided into well-labelled quadrants (Figure 22) to ease monitoring and sampling for pests and diseases, and control.
- Monitor for pests and diseases every two weeks in the first 4 months after planting (MAP), and once a month beginning 5 MAP until 9-12 MAP.
- Randomly sample at least 10 plants per row using a quadrant to look out for pests and disease symptoms. Count and record the pests and symptomatic disease-affected plants, and collect insect specimen and leaf samples for morphological and laboratory identification by phytosanitary inspectors.
- If viral infections are detected in a sample, resample every plant in the source quadrant to determine the level of spread and identify the infected plants. Remove or rogue out and burn the virus-infected plants to avoid further spread of the disease in the planting materials (Sseruwagi *et al.*, 2014).

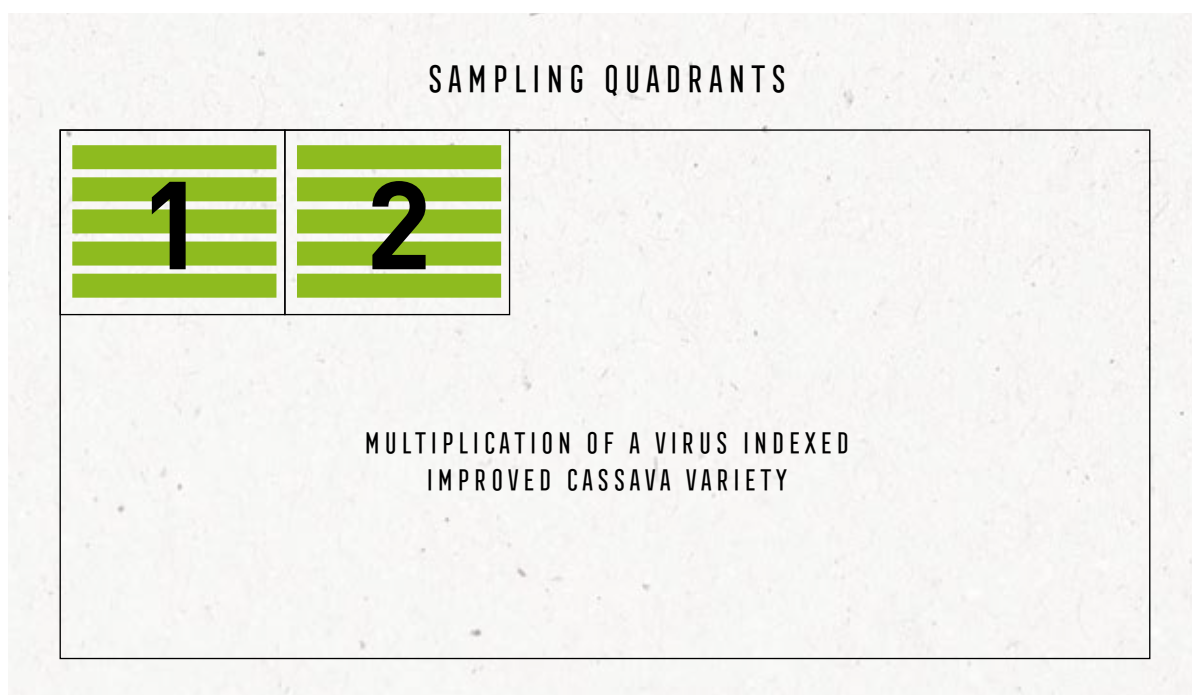


Figure 22 — Showing layout of a cassava multiplication block with sampling quadrants (inset 1 & 2) used for plant health management
Source: Sseruwagi *et al.* (2014)

b. Preparation of cuttings

Stem cuttings should be prepared when one is ready for planting and the following should be observed:

- It is better to take cuttings from the base than the top of the stem (Abass *et al.*, 2014).
- Use a sharp knife, secateurs, clipper, panga/cutlass or saw (Figure 23A) or (Figure 23B) engine-driven saw.



Figure 23 — Showing tools (A) secateurs, saw, panga clippers and (B) engine-driven saw used to prepare cassava stem cuttings
Source: Jorge (2008); https://plantvillage-production-new.s3.amazonaws.com/images/pics/000/002/891/original/9320787942_cf3a03cc57_z.jpg

- Care must be taken not to damage the nodes (Figure 24) or stem, which would reduce sprouting percentage.



Figure 24 — Showing nodes on a cassava stem cutting
Source: <https://i.ytimg.com/vi/1yEJgBvlnyw/maxresdefault.jpg>

- Using sharp tools, cut pieces of 15-35 cm long (Figure 25A) with a minimum of 5-8 nodes (Figure 24) (Hauser et al 2014);
- Avoid bruising or damaging cuttings (Figure 25B & 25C) as the wounds can be easy entry points for microorganisms;
- Dip the stem cuttings in ethanol for 5 minutes to kill any pests (mites, mealy bug, whitefly nymphs and thrips) and to avoid spread of viral diseases on the tools across cuttings. Expose the cuttings to dry under shade (Abass et al., 2014).



Figure 25 — Showing (A) recommended cutting size;
(B) well cut ends with sharp tools and (C) badly cut ends with blunt tools
Source: Hauser et al (2014)

c. Ratooning

During conventional cassava stem multiplication, the plants are ratooned (i.e., harvesting stems but not the tubers) initially at 9-12 MAP. Subsequently, the ratoon crop has the stems harvested every 6 months, depending on the season (International Institute of Tropical Agriculture, 1990). The following activities are carried out during ratooning and management of ratoon crops:

- Harvest the mature stems using sharp tools such as pangas/cutlasses or an engine-driven saw, leaving at least 15-20 cm (above the ground) of the stalks to allow for regrowth of the next season ratoon crop.
- Bundle up the stems full length or cut each into 15-35 cm long cuttings, and carefully place in gunny bags. It is estimated that for ratoon crops, cassava stems multiplication has a factor of at least X10, i.e., each 150-350 cm long stem produces 10 cuttings of 15-35 cm length.
- Keep the harvested stems or cuttings in a shade until transportation for distribution to farmers or commercial fields for planting.
- After harvesting the stems, the field should be carefully weeded to avoid disturbing the unharvested tubers, and fertilisers (N, P and K) should be applied (see chapter 6.2 on crop management) around each stand to initiate rapid vegetative regrowth.
- Normally, ratooned cassava crops tend to regrow several shoots (Figure 26A) since there is a ready supply of nutrients from the tubers that were not harvested. Therefore, thinning should be carried out to remove excess and weak-looking shoots to maintain only 2-3 strong well-spaced shoots per stand. This enables the development of good node intervals (Figure 26B), a major quality trait for good cassava planting materials.
- Repeat the above processes every 6 months after harvesting for at least 3 to 4 times more. After the 4-ratoon crops, the shoots tend to be weak and short due to depletion of nutrients in the over-grown cassava tubers.
- The over-grown tubers may be harvested for feeding livestock (cattle) or to be processed, although the accumulation of sugars means that the tubers have less starch and are of poor quality. Low quality tubers may be ploughed back into the field to replenish soil fertility. However, when the tubers are finally harvested, you may use the stems to plant new fields.



Figure 26 — Showing (A) ratooned cassava regrowth with overcrowded shoots and wide internode gaps – poor stems and (B) high quality cassava stems produced with good multiplication practices

Source: https://encrypted-tbn0.gstatic.com/images?q=tbn:ANd9GcTabLRP0vUHXmaGL0iRepHB7pl76_YzmDQ5sw&usqp=CAU; https://leafplanttech.com/wp-content/uploads/2020/06/cassava-_cuttings.jpg

d. Management of ratoon crop

- Prune or remove extra shoots during the early months (3-4) to maintain a maximum of 3 shoots per plant. This helps the growth of stronger stems with normal internode spacing.
- Weed the ratoon crop at least twice every month for the first 3 months, until the plants develop a closed canopy that is able to smother the weeds. Subsequent weeding should be carried out monthly or bi-monthly as deemed necessary.
- Apply ca. 50 kg N and 40 kg P₂O₅/ha to boost crop growth and production of strong stems.
- Apply K to support root formation for the ratoon crop.
- Remove/rogue disease-infected or pest-affected plants and burn them.

3.5.2. RAPID MULTIPLICATION

Rapid multiplication differs from conventional multiplication in the following ways:

- a. It uses a high multiplication ratio of 1:60-100 compared to 1:10 for conventional multiplication;
- b. The period of raising the planting materials is much shorter, and ranges between 4-6 weeks compared to 9-12 months (initial crop) and every 6 months (ratoon crops) in conventional multiplication;
- c. All the stem parts: shoot tips (immature green parts), semi-hard wood (semi-mature stem parts) and hard wood (mature stem parts) can be used for raising seedling compared to the use of regular mature stem parts (hard wood) of 15-35 cm, with 5-8 nodes (Moyo *et al.*, 2004).

Rapid multiplication can be carried out as follows:

a. Preparation of mini-stem cuttings

The number of nodes per mini-stem depends on plant part; internode length and stem diameter. Obtain mini stem cuttings using secateurs, a sharp knife/cutlass, or saw as follows:

- Hard wood (mature stem parts): 2-3 nodes;
- Semi-hard wood (semi-mature stem parts): 4-6 nodes
- Shoot tips (immature green parts): 6-10 nodes.

b. Rapid multiplication of immature green parts and shoot tips

1. Treat the mini-cuttings and shoot tips for at least 10 minutes using organic Neem (1kg in 5L of water) in large containers to kill of the pests. Immersing the mini-cuttings in hot water (about 60°) for 5 to 10 minutes helps treat against viral infections;
2. Drain the solution and plant the mini-cuttings and shoots in a nursery bed with heat-treated soil in a humid chamber. The humid chamber may be made of transparent polythene covers on the roof and walls (Figure 27). This helps to keep rain water out and allows heat accumulation, which facilitates rapid growth of the buds into shoots (sprouting);

3. Keep the immature plants in the humid nursery for 4-6 weeks, after which they are moved to an open section of the nursery to harden off, until transplanting into the multiplication block.
4. To maintain high humidity, apply water 2-3 times every day, avoiding too much water which might rot the tender immature cuttings and shoot tips, including those sprouting;



Figure 27 — Showing seedling nursery/humid chamber made of transparent polythene that can be used for rapid multiplication of cassava mini-cuttings

Source: <https://cassavadiasesolutionsasia.files.wordpress.com/2019/10/tunels.jpg?w=1024>

a. Rapid multiplication of semi-mature and mature mini-cuttings

1. Repeat step 1, above to treat the semi-mature and mature mini-cuttings against pests and diseases;
2. Pour the solution and put the cuttings in perforated transparent polythene bags for pre-sprouting;
3. Keep the polythene bags with the treated cuttings under shade for 7–10 days to sprout fully.

The polythene bags serve as humid chambers, which accumulate heat and moisture, which initiate the fast development of the buds/nodes to sprout;

1. After the 10 days, remove the polythene bags with the sprouted cuttings, and expose them to sunlight to harden before transplanting in the field.
2. Transplant the hardened sprouted cuttings into the field using spacing of 0.5 m x 0.5 m (Figure 28), and manage the crop as described for conventional multiplication (in section 3.5.1 above). Transplanting should be carried out when there is sufficient moisture in the soil, or immediately after a rain shower.
3. Mature/hard wood mini-cuttings with 3 nodes may ONLY be treated in step 1 (above), and directly moved for planting in the open field, skipping steps 2-4 for the semi-mature cuttings.

Sprouting the mini-cuttings in polythene bags is convenient, cheaper and ensures production of healthy plants, compared to using nursery beds, which is more expensive and prone to adverse weather and soil-borne disease.



Figure 28 — Showing cassava multiplication of improved variety established with mini-cuttings raised using rapid multiplication

Source: <https://i0.wp.com/nobowa.com/wp-content/uploads/2016/07/transplant.jpg?ssl=1>

3.5.3. MICROPROPAGATION USING CASSAVA PLANTING MATERIALS USING TISSUE CULTURE

Micropropagation is a means of raising plants using vegetative/clonal propagation using tissue culture techniques (Shiji *et al.*, 2014). The technique is normally used by plant breeders during crop improvement and large-scale commercial seedling producing companies. It uses asexual methods of propagation where the apical meristem is planted. A single cell and protoplast culture are used to produce a large number of plants in a very short period. For example, a single explant can produce 1,000,000 plants in a period of 6 months only (Karki, 2020). According to Murashige (1974), micropropagation can be achieved in four major stages (Figure 29 & 30).

STAGE 0: SELECTION OF STOCK PLANT/S

- Select healthy cassava stock plants of a suitable variety to get explants.

STAGE I: ESTABLISHMENT OF EXPLANTS

- Select and cut 5-10 mm long explants from healthy stock plants using a sterile blade;
- Place the explants in a watch glass and add 70% alcohol, sodium hypochlorite (bleach) and calcium hypochlorite to sterilize for 5-10 minutes;

NB: Avoid keeping the explants in the sterilizing agent/solution for longer than 10 minutes because it kills the cells.

- Wash the explants for several minutes using distilled water to remove the excess sterilizing solution;
- Transfer the sterilized explants to nutrient media for 2-3 month to establish.

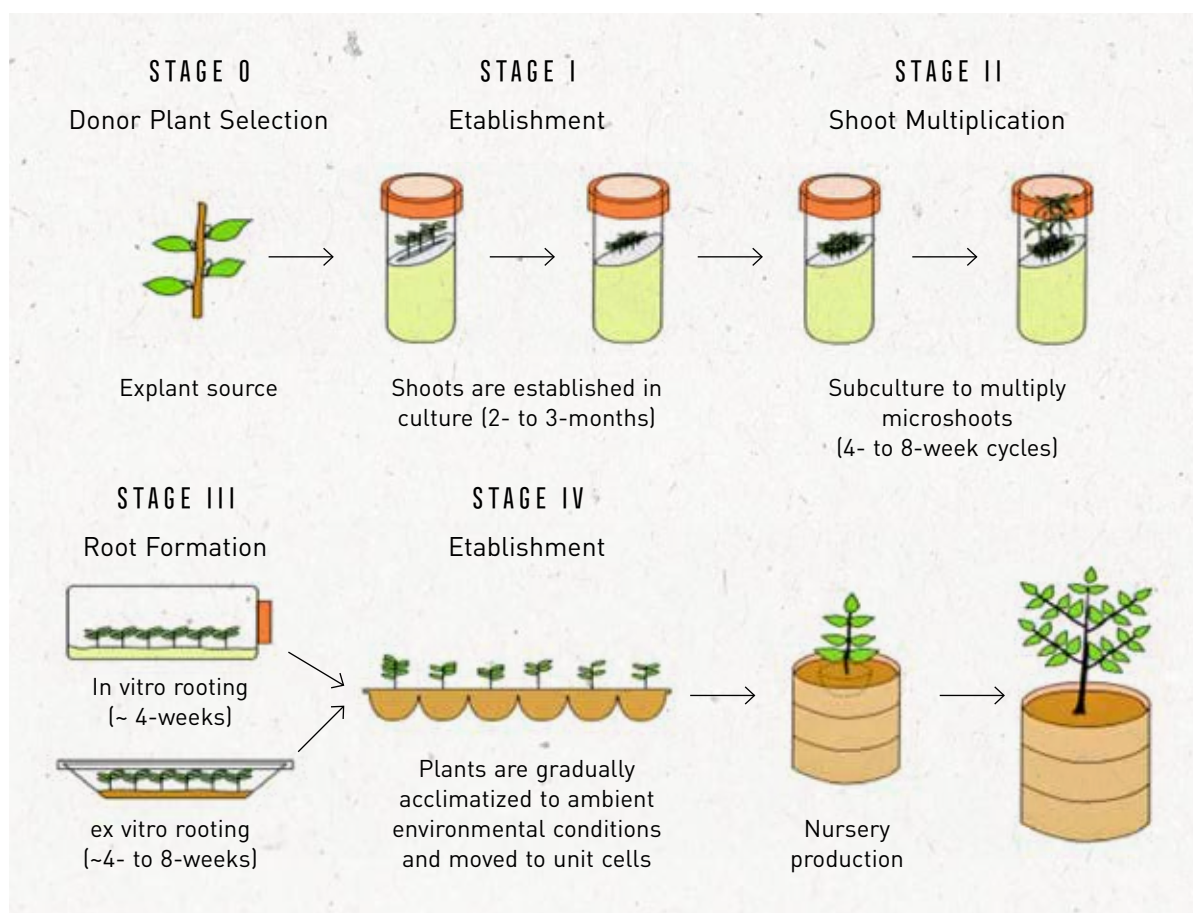


Figure 29 — Stages in micro-propagation of cassava plants using tissue culture techniques
Source: <https://propp.ifas.ufl.edu/images/09-tissue-culture/01-types/tctypesmicropropagation/image5.jpg>

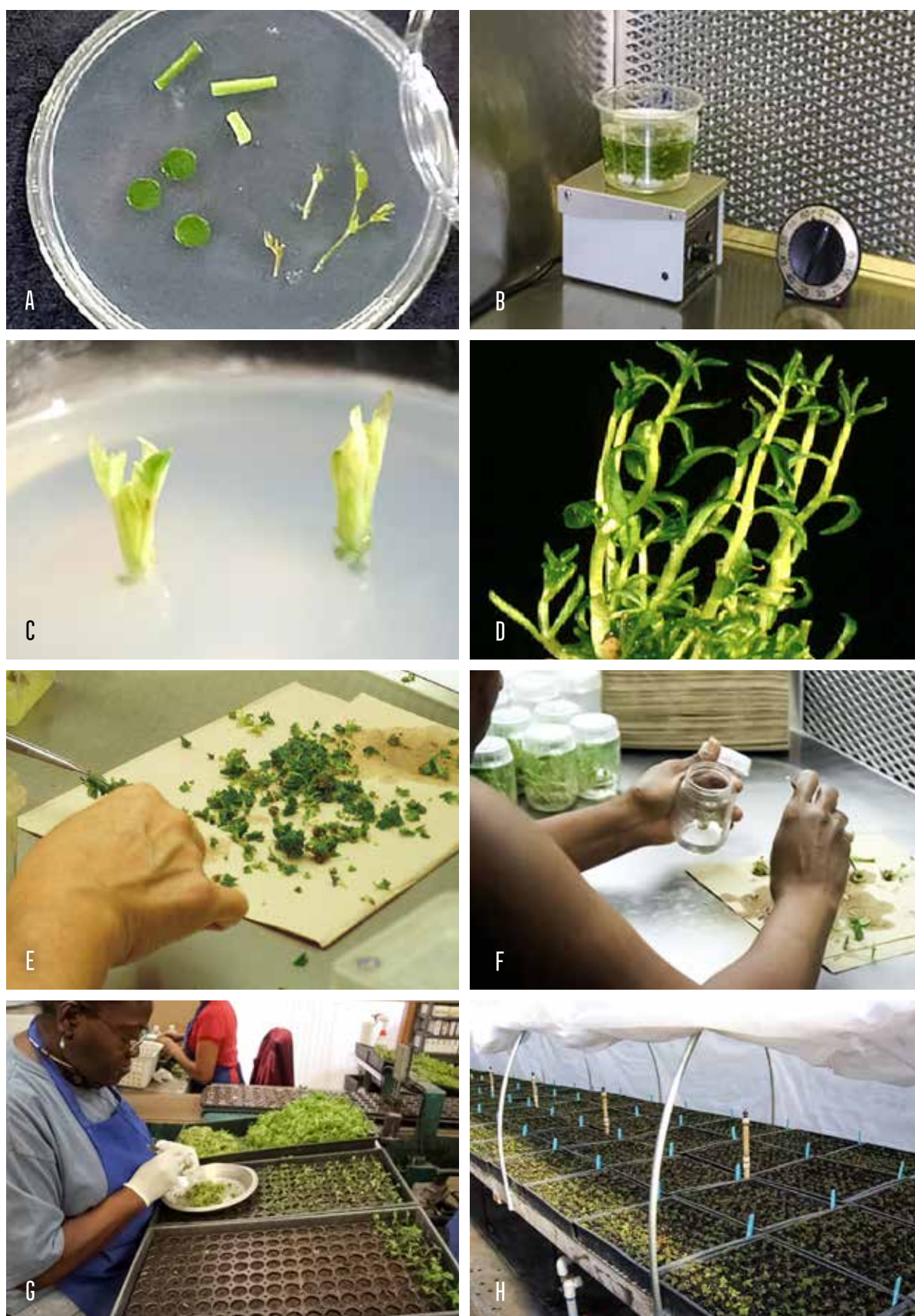


Figure 30 — Showing stages in micro-propagation of cassava plants using tissue culture techniques: stage I (A&B), stage II (C&D), stage III (E, F & G) and stage IV (H)

Source: <https://progp.ifas.ufl.edu/09-tissue-culture/01-types/04-tctypes-micropropagation.html>

STAGE II: SHOOT MULTIPLICATION

- Multiply the shoots from the explant by using a scalpel and forceps to cut and separate larger culture into smaller pieces, and subculture them on Musashige and skoog (MS) medium;
- Incubate for 4–8-week (2 months) cycles at 20–25° C to initiate the shoots;
- Transfer the shoots into inducing media and incubate until multiple shoots grow.

STAGE III: ROOT FORMATION

- Separate individual shoots on rooting medium, followed by planting into jars or trays with soil for 4–8 weeks to grow into small plantlets with well developed roots and shoots;

STAGE IV: ACCLIMATIZATION

- Acclimatize or harden off the plantlets for 1–2 weeks prior to planting in the open field. This helps the laboratory raised plants to adjust to external environmental conditions and developing functional leaves and roots.

3.6. STORAGE OF CASSAVA PLANTING MATERIAL

Except in cases where cassava is harvested piecemeal (in bits for home consumption), and the cuttings are immediately replanted where the harvesting was carried out or in a new field, the stems must be carefully stored for 2 to 5 days (preferably), and not more than 14 days until they are cut and used for the next growing season (Howeler, 2007). This helps to avoid exposing the stems to adverse climatic conditions, such as scorching sunshine that may dry the materials/cuttings leading to loss of viability or excessive moisture which may cause early sprouting or rotting (Tarawali *et al.*, 2013). Long storage periods could lead to loss of sprouting vigor (Ekanayake *et al.*, 1997) and hence poor crop establishment.

To store cassava stems, the following should be done:

- Choose a good shade, like under a tree (Figure 31A) or a grass thatched-shade (Figure 31B)
- Slightly dig the soil and stand place (upright) the bundles of cassava stems with the mature (lower parts) end parts touching the ground;
- Water the stems regularly to keep them moistened and alive.
- Keep the surroundings weed-free to remove potential alternative host plants for cassava-damaging insect pests and diseases.

Caution:

- Take care not to damage the nodes by handling the stems by the ends;
- Avoid keeping the stems in the open sun, such as in figure 31, which could dry them up resulting in poor sprouting.
- At planting cut off the top and basal parts of the stems, and only use the middle part for cuttings to be planted.



Figure 31 — Showing cassava stems

(A) under tree shade. Source: <https://gh-f.org/wp-content/uploads/2021/07/growing-cassava-a-training-manual-from-production-to-postharvest-adebayo-b.-abass.pdf>,
(B) in a grass thatched-shade; https://www.researchgate.net/publication/268523868_Pheneas_Ntawuruhunga_Maria_Andrade_Paul_Demo_and_CC_Moyo_2011_Brief_on_Achievements_of_the_%27Improving_the_Rural_Livelihoods_in_Southern_Africa_project_2004_to_2011/figures
and (C) storage in the open/exposure to sunlight; <https://rtb.crop-diversity.org/crops/cassava>

3.7. IRRIGATION

Irrigation supplements scarce or irregular rainfall and enables the production of cassava during the offseason months. It is important to select the equipment for the method of irrigation you plan to use for cassava production, before planting the field (Pushpalatha *et al.*, 2020).

The three most common methods of irrigation used for cassava production by small and large-scale (commercial) farmers are: furrow, drip and sprinkler irrigation.

3.7.1. FURROW IRRIGATION

Furrow irrigation involves making small channels between ridges to carry water for irrigating crops that are usually grown on the ridges (Figure 32A & B). The method is suitable for a wide range of soil types, crops and land slopes. The water is let to flow until the ground between the ridges is fully soaked, which requires large volumes of water. Water is pumped into the field by use of pump and flows by gravity until the last ridge. Not only is the method expensive, requiring the use of much fuel or electricity, but also very wasteful, since a lot of the water goes into the ground before reaching the crops. The method also risks spreading soil-borne diseases in the field, and if used during more mature stages of cassava, the tubers rot easily. The frequency of irrigation can be once or twice per day, in the morning and/or evening, when the weather is cool enough to reduce evaporation. The equipment required include: a petrol/diesel/electricity/solar-powered water pump, long horse pipes, jerricans to keep fuel and a water tank.

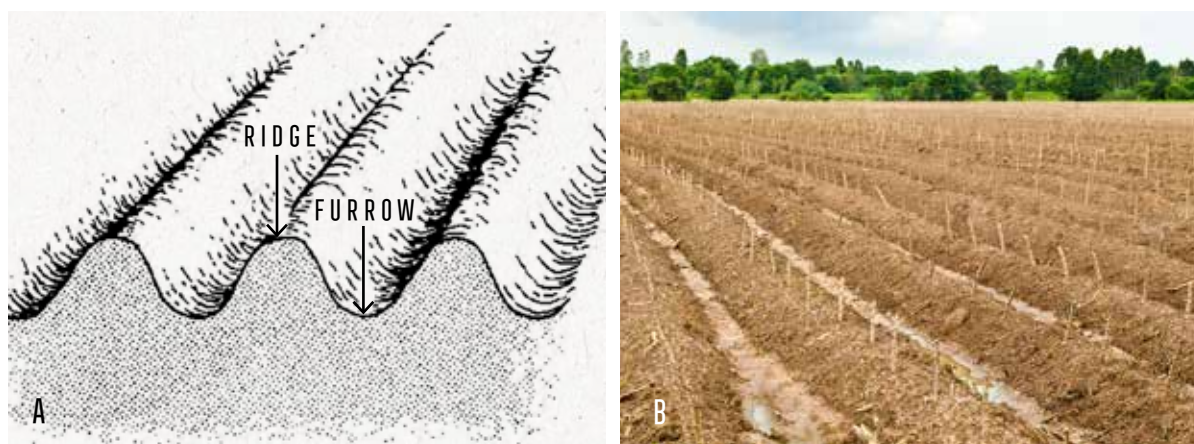


Figure 32 — Showing (A & B) furrow irrigation of cassava planted on ridges
Source: <https://www.fao.org/3/s8684e/s8684e0j.gif>

3.7.2. DRIP IRRIGATION

Drip irrigation involves the use of specially calibrated driplines with drips spaced at 1 m intervals to either fit in between the cassava plants or at each plant. The drip lines are laid on the length of the ridges (see figure 33, below) and are connected to a big water pipe that feeds them. The water source could be a large tank fitted on a raised (>4m high) metallic platform or ground to allow water to flow by gravity or from a water dam, using a petrol, diesel, electricity or solar powered engine pump. The drip line method is precise and economic in dispensing water (Odubanjo *et al.*, 2011). Drip irrigation can also be applied twice each day; in the morning and evening to ensure sufficient water for production.



Figure 33 — Showing drip irrigation equipment laid on ridges before planting cassava
Source: <https://image.shutterstock.com/image-photo/connection-drip-irrigation-system-cassava-260nw-2107736897.jpg>

3.7.3. SPRINKLERS

With sprinkler irrigation, the water is distributed above the crop canopy using a spray (Figure 34). However, continuous spraying of water on the cassava canopy by sprinkler irrigation systems tends to create a humid environment which favours pest and disease development and spread. There is also great water loss due to wind drift, evaporation of droplets in air, crop canopy and soil surface (Irmak *et al.*, 2011).



Figure 34 — Showing use of sprinkler irrigation in a cassava field
Source: <https://image.made-in-china.com/202f0j00fUkYiIHdSLq0/Cassava-Farm-Agricultural-Irrigation-Walking-Sprinkler.jpg>

3.8. ASSOCIATIONS

3.8.1. FAVOURABLE ASSOCIATED CROPS

Cassava is produced in a variety of favourable associations or cropping systems including: monocropping and intercropping (Hauser *et al.*, 2014).

a. Monocropping

Monocropping involves planting a single cassava crop in a field (Figure 35). It is mostly practiced by large-scale (>5 hectares) commercial farmers who seek to either market fresh or processed cassava products with standardised food quality characteristics. However, smallholder farmers that grow cassava as a monocrop tend to do so on smaller plots (<2 acres) or by mixing several cassava varieties in a single field.



Figure 35 — Showing cassava planted as a mono crop

Source: <https://thelensnews.com/aibom-disburses-interest-free-loans-totalling-n1-2b-to-cassava-farmers-others-to-raise-output/>

b. Intercropping

Intercropping involves planting a main crop together with another crop or crops. Many crops can be grown in association with cassava. For example, the common practices include intercropping cassava with maize, sweetpotato, beans or pumpkins (Eastern Africa and the Congo basin) and multicropping cassava with sesame, roselle, yams, beans and groundnuts (Cooler tropical regions). However, the most suitable intercrops to grow with cassava according to Devos and Wilson (1979), include shorter-

duration crops (2-5 months) such as maize (Figure 36A), cowpeas (Figure 36B), cocoyam (Figure 36C), yam and okra (Figure 36D), which must be planted at least 1-2 months earlier than cassava to avoid unfair competition that would affect either of the intercrops (Hauser *et al.*, 2014).



Figure 36 — Showing different cassava intercrops: (A) cassava and maize; (B) maize, cassava and jack bean; (C) cassava and cocoyam and (D) cassava, yam and okra

Source: Stefan Hauser, IITA, <https://africasoilhealth.cabi.org/wpcms/wp-content/uploads/2016/06/French-Cassava-A5-colour-Lowres.pdf>; Feedipedia, <https://www.feedipedia.org/content/field-maize-cassava-and-jack-bean>; Kikuno, IITA, <https://www.flickr.com/photos/iita-media-library/6075756511>; Damian Asawal, <https://africasoilhealth.cabi.org/wpcms/wp-content/uploads/2014/06/254-Cassava-A5-colour-lowres.pdf>

It must be noted, however, that the intercropping method can limit intensification of any of the intercrops, including cassava. For example, intercropping cassava with maize was reported to reduce the spread and impact of pests and diseases to cassava crops in traditional cropping systems (Fargette & Fauquet, 1988), but growth and yield were not improved (Mohan & Hrishi, 1978; Mutsaers *et al.*, 1993; Hauser *et al.*, 2014).

3.8.2. UNFAVOURABLE ASSOCIATIONS

Cassava may not do well when combined with tall foliage plants that could shield access to sunlight due to shades by the canopies. The lack of sunlight encourages elongation of shoots and internodes, resulting in weak stems and poor development of tuberous roots.

3.9. CROP ROTATION

Crop rotation involves growing different crop species in sequence on the same land each season or year, and may also involve resting the field (Tanveer *et al.*, 2019). Cassava is usually grown as the last crop in a rotation (following its robustness and tolerance to poor soil conditions), which has earned it the somewhat unjust reputation of being a soil-exhausting crop. Though sometimes it can be the first in a rotation when soil has become exhausted or when there is a great demand for cassava.

Crop rotation has several advantages that help increase crop yields, including:

- Mobilizing or rejuvenating soil nutrients and other beneficial micro-organisms;
- Improving soil structure to retain more water for plants and control soil erosion;
- Reducing the build up of pests and diseases;
- Reducing costs and increasing profitability by alternating the crops grown for different markets

3.9.1. CROPPING HISTORY

It is important to know the following crop history about the selected plot for growing cassava crops.

- Whether the land has not been previously planted with any crops, i.e., virgin land with natural vegetable (trees or shrubs);
- Whether the land has been rested for a short (<3 years) or longer (3-5 years and more) fallow period;
- Which crops were grown, including the trees;
- Freedom from soil-borne pests and diseases.

There are two forms of crop rotation that should be distinguished when planning crop rotation: crop rotation under continuous cropping and crop rotation nested in fallow.

- **Crop rotation under continuous cropping** is the continued use of the same field, while changing the crops grown from season to season or year to year.
- **Crop rotation nested in fallow:** involves resting the field for some time (fallow) to enable soil fertility to build back before growing another crop on it. A cover crop such as legumes may be grown on the fallow land to boost soil fertility or the land could be left to grow natural vegetation.

3.9.2. SUITABLE CROPS AND MIX

While there is a wide diversity of cassava growing practices across ACP countries, here are some of the common suitable crops to use in a cassava rotation calendar (Figure 37) and their mix.

- Cereals: maize, sorghum, millet, rice;
- Roots: yam, sweet potatoes;
- Legumes: cowpea, soybean, groundnuts, pigeon peas;
- Vegetables: peppers, melons, *Solanum* spp., okra, cauliflower, cabbage.

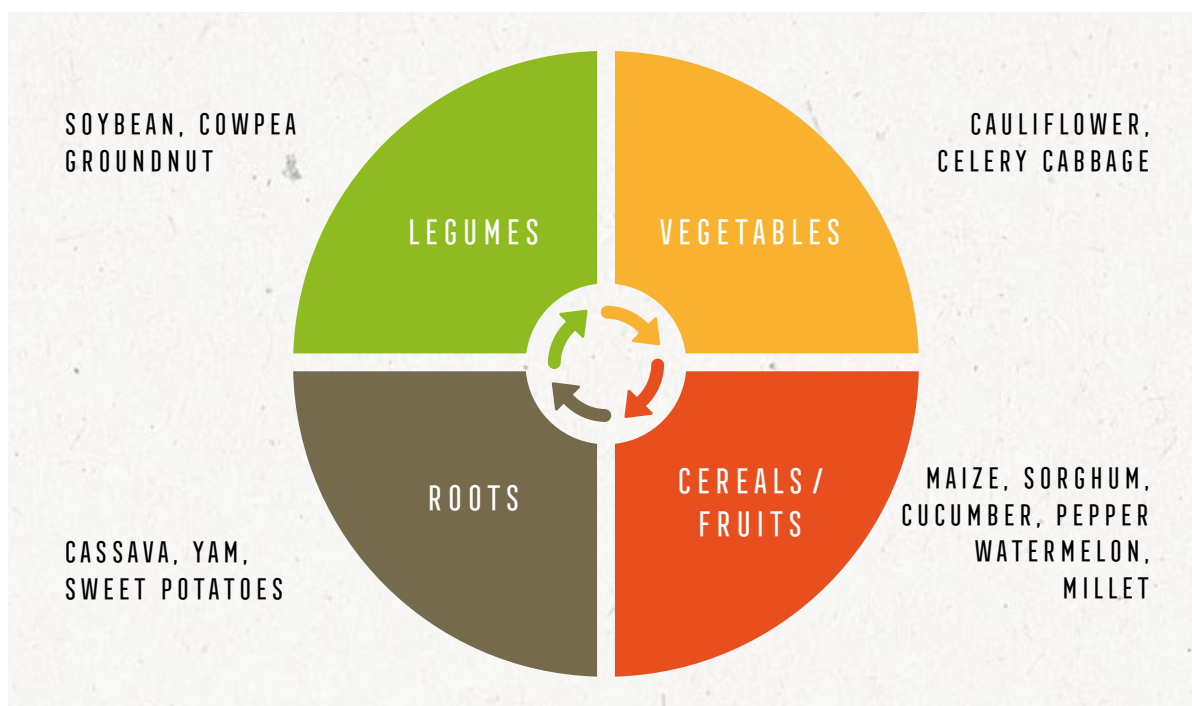


Figure 37 — Showing a cassava crop rotation calendar with the appropriate sequence of growing different crops to sustain the productivity of the soil

Source: <https://i0.wp.com/justagric.com/wp-content/uploads/2018/12/crop-2Brotation-2Bdiagram.jpg?ssl=1>

Below are the basic principles that should be followed by cassava farmers to select the correct crop and time to grow in a crop rotation calendar.

- Avoid continuously growing crops of the same family each season or year as they deplete soil nutrients and build up pests and diseases;
- Long growing (>6 months) crops should be followed by short growing (<6 months) crops;
- Crops with deep roots, such as sweet potato or yam should be followed by shallow rooted crops like maize, millet, rice or sorghum to allow for efficient use of soil nutrients;
- Legumes such as cow pea, ground nuts etc., should be grown after non-leguminous crops such as maize, rice etc. The legumes help to fix atmospheric nitrogen and build up organic matter content in the soil;
- More soil exhaustive crops like sunflower should be followed by crops that restore soil nutrients (nitrogen) such as legumes;
- Crops that require much irrigation should be grown after crops that need less water.



PRE- PLANTING CHOICES

KEY POINTS SUMMARY

CHOOSING A PLOT

- Select locations that are not close to other cassava fields (where possible); take note of the cropping history; and the plot should be well drained.

PLANTING MATERIAL

- Select planting materials that are free from pests & diseases, maturity (6-12 months old), 5-8 nodes, at least 2.5-3 cm diameter with no damages.
- Use 4,000 to 5,000 cuttings per acre with spacing of 1 m x 1 m and 0.8 m x 1 m, respectively, and 10,000 to 12,500 cuttings per hectare at spacing of 1 m x 1 m and 0.8 m x 1 m, respectively.

VARIETIES

- Select improved cassava varieties with tolerance or resistance to pests and diseases, and/or other abiotic stresses relevant in the area.
- Where improved varieties are lacking, select local cultivars that are more robust to abiotic stresses.

GROWING PERIOD

- Seasonality depends on region, but cassava can be harvested as early as 6 MAP until 12-24 months for late maturing varieties. Staggered planting and harvesting are viable methods for risk mitigation and ensuring a more continuous supply of tubers.

METHODS OF PLACEMENT OF CUTTINGS

- Cassava cuttings may be planted using 3 placement methods: horizontal, vertical or tilted/slanting.



PRE- PLANTING CHOICES

PLANTING MATERIALS MULTIPLICATION METHODS

- Conventional, rapid multiplication and micropropagation/tissue culture techniques

MULTIPLICATION BLOCK MANAGEMENT

- Regularly monitor for pests and diseases in the cassava multiplication blocks with the aid of a phytosanitary inspector to maintain infections below 5%;
- Rogue and destroy the infected plants;
- Ratoon the stems at 6-12 MAP in 1st ratoon, and subsequently after every 6 months for 4 times more.
- Keep the multiplication blocks free of weeds, and apply N, P, K fertilisers to improve growth and vigour.

IRRIGATION

- Irrigation can be achieved with use of furrows, drip lines or sprinkling where there is scarcity of rain or during offseason.

ASSOCIATIONS

- Grow cassava intercrops with favourable associated crops such as cereals, legumes etc.

ROTATION

- Make a crop rotation calendar after studying cropping history of the field and use suitable crops such as shallow rooted and restorative crops including legumes, vegetables and cereals, to rejuvenate soil fertility and break the cycle of pests and diseases.





41

**PREPARING
THE PLOT**

During preparation of the field/plot prior to planting a cassava crop, several activities should be carried out, including: landscaping, weed management, soil preparation, soil amendments and transformation of an existing plot. The activities are described in detail in the subsequent sections.

4.1. LANDSCAPING

Landscaping involves preparing the land by carrying out the following activities: clearing vegetation, planting hedges/windbreaks, grass strips and firebreaks, making terraces, drainage, laying irrigation, making paths and planting or erecting fences.

4.1.1. CLEARING VEGETATION

- Begin to clear the vegetation at least 1-2 months before the start of the growing season, to allow enough time for the debris to dry and decompose;
- It is advisable to uproot the shrubs and tree stumps, and either spread them or put them at the boundary of the plot to dry, rather than to burn them in the plot as many smallholder farmers tend to do. Burning kills beneficial micro-organisms for agricultural production in the soil, although it introduces wood ash, which is comprised of many major (potassium, phosphorus, calcium and magnesium) and minor (micronutrients) elements needed for cassava growth (Risse and Gaskin, 2010). However, it should be noted that mineral nutrients tend to be flushed after one rainfall event.

4.1.2. HEDGES, WINDBREAKS, FIREBREAKS, GRASS STRIPS

- To avoid destruction of the young crop by stray animals, including livestock (goats, cattle and sheep etc.), and small wild animals, it is advisable to plant hedges around the field.
- If the selected area is prone to strong winds, plant the recommended tree species for agro-forestry in your location to serve as windbreaks to protect the crop, especially for tall cassava varieties grown.
- Firebreaks are open spaces that are left between crop blocks or fields and a nearby forest or shrub area to safeguard against fires, especially during the dry season.
- If the site is situated on a slope, it is advisable to plant grass strips between the production blocks to control soil erosion.

4.1.3. TERRACES, DRAINAGE AND IRRIGATION

- Make terraces if the land is on a sloping place to enable control of soil erosion;
- If the land is flat or in a swampy area, make drainage channels to reduce the risk of flooding the field for growing cassava crops. Making ridges could further help to drain excess water;
- Select, procure and lay in the field the equipment needed to irrigate cassava as advised by the most suitable method of irrigation (see sections 3.7.1 and 3.7.2 above) that fits both your budget and terrain, including access to water.

4.1.4. PATHS AND FENCES

- It is advisable to make paths (Figure 38) between the blocks in a field that are wide enough for walking in or driving through with a tractor, vehicle or other farm machinery and equipment. A field without a path or paths risks destroying the crops;
- To keep away trespassers, including humans, livestock and wild animals, it is advisable to fence the field either using live plants (hedges) or fencing materials such as barbed wire, wire mesh or chain link. Fencing material can increase the cost of land preparation.



Figure 38 — Showing a path/road between cassava fields

Source: https://news.cornell.edu/sites/default/files/styles/full_size/public/2020-03/0304_cassava3.jpg?itok=TpYhG0cl

4.2. WEED MANAGEMENT

Weeds are undesirable plants (including crops) that affect the growth and development of the crop of interest. This is because the weeds compete for space, water, nutrients and light, which reduces the crop yields (Moyo *et al.*, 2004). In addition, weeds also serve as alternative hosts for pests, diseases and vectors for viral diseases, and small rodents that damage tuberous crop such as cassava. It is estimated that weed control takes up not less than 40% of the total labour cost of growing cassava in sub-Saharan Africa (Abass *et al.*, 2014; Joint FAO, 2018).

4.2.1. COMMON (INVASIVE) WEEDS IN CASSAVA FIELDS

Although there is a wide diversity of weeds species that occur in cassava growing areas, globally, and specifically in the ACP countries, there are generally two broad categories: annual and perennial weeds. The two categories are further sub-divided into three groups, including: broad leaved weeds, grasses and sedges (Table 5).

A list of some of the notorious invasive weed species that affect cassava production is presented in the table below by category and illustrated with photos.

Table 5 — Common (invasive) weeds in cassava production areas

BROAD LEAVED WEEDS	
SIAM WEED – <i>CHROMOLAENA ODORATA</i>	
	Perennial Seeds, wind Source: https://encrypted-tbn0.gstatic.com/images?q=tbn:ANd9GcRZKQs1kF3lPvsvpXd-nrK3YKE4TZQDRZ9hQaQ&usqp=CAU
WANDERING JEW – <i>COMMELINA BENGHALENSIS</i>	
	Perennial cuttings Source: https://keyserver.lucidcentral.org/weeds/data/media/Images/commelina_benghalensis/thumbs/commelinabenghalensis10_sml.jpg

WILD POINSETTIA/MEXICAN FIRE PLANT - *EUPHORBIA HETEROPHYLLA*



Annual and Perennial
seed

Source: https://keyserver.lucidcentral.org/weeds/data/media/Images/euphorbia_heterophylla/euphorbia_heterophylla_and_euphorbia_cyathophora4.jpg

WILD SUNFLOWER - *ASPILIA AFRICANA*



Perennial
seeds

Source: <https://www.researchgate.net/profile/George-Koffuor/publication/255921174/figure/fig1/AS:311972970156032@1451391560869/Aspilia-africana-growing-in-the-wild.png>

SENSITIVE PLANT OR TOUCH-ME-NOT - *MIMOSA* SPP



Annual and Perennial
seed

Source: https://encrypted-tbn0.gstatic.com/images?q=tbn:ANd9GcTjzvqDcJIM-Meh_Z0w2NJP97AwEitOrUZt7jNxEsuY2mTz9bVJu03Alc47SF-Sj8g-YYM&usqp=CAU

GRASSES

COGON GRASS - *IMPERATA CYLINDRICAL*



Perennial
rhizomes

Source: <https://indiabiodiversity.org/files-api/api/get/raw/observations//b90c75f3-2d86-41bf-95fb-6d41818bde22/949.JPG>

BERMUDA GRASS - *CYNODON DACTYLON*



Perennial
rhizomes, stolons and seed

Sources: <https://indiabiodiversity.org/files-api/api/get/raw/observations//b7b348d2-2773-4f21-999f-50a13f008310/185.jpg>

GUINEA GRASS - *PANICUM MAXIMUM*



Perennial
seeds and tillers

Sources: https://keyserver.lucidcentral.org/weeds/data/media/Images/megathyrsus_maximus_var._pubiglumis/megathyrsusmaximuspubiglumis24.jpg

MISSION GRASS - *PENNISETUM POLYSTACHION*



Annual and perennial
seeds

Sources: <https://plants.ifas.ufl.edu/site/assets/files/1855/penpol1.400x400.jpg>

SPEAR GRASS - *HETEROPOGON CONTORTUS*



Perennial
stolons or rhizomes, and seed

Sources: https://upload.wikimedia.org/wikipedia/commons/2/25/Heteropogon_contortus.jpg

SEDGES

NUTSEDGE - *CYPERUS* SPP.



Perennial
stolons with tubers
annual no tubers or rhizomes

Source: <https://floraofgibraltar.myspecies.info/sites/floraofgibraltar.myspecies.info/files/Cyperus%20rotundus%2006%20North%20Front%20Cemetery%2021-4-2016.jpg>

<https://www.nzpcn.org.nz/site/assets/files/0/46/360/cyperus-esculentus-01.1200x0-u0i1s1q90f1.jpg>

UMBRELLA SEDGE - *MARISCUS ALTERNIFOLIUS*



Perennial and annual
rhizomes

Source: <https://indiabiodiversity.org/files-api/api/get/raw/observations//b90c75f3-2d86-41bf-95fb-6d41818bde22/949.JPG>

Source: Moyo *et al.* (2004)

4.2.2. WEED CONTROL METHODS

While preparing the land, it is important to look out for and control the invasive weed species (see Table 5, above) that spread by seed, rhizomes or cuttings, since incorporating the propagation parts in the soil will make it difficult to control them when the crop is planted. These weeds should be removed during land clearing, tilling, harrowing and ridging using manual/cultural, mechanical and chemical control methods (where necessary) as follows:

a. Manual/cultural weed control

- Remove the weeds manually by hand and hoe/cutlass during bush clearing by digging up the rhizomes and moving the stems to boundaries of the field. This can be slow and ineffective and requires patience and keenness.

b. Mechanical weed control

- During tilling with a disc plough mounted on a tractor, deep ploughing helps to dig up the weeds together with their rhizomes. Following further exposes the weeds on the soil surface where they dry or could be collected and burnt.

c. Chemical weed control

- Apply approved post-emergence non-selective systemic herbicides to control the weeds at least 1 month before tilling the land in heavily infested areas.

4.3. SOIL PREPARATION

Soil preparation for cassava growing involves several processes, including: subsoiling, tillage, harrowing, ridges, mounds, zero-tillage and digging holes.

4.3.1. SUBSOILING AND TILLAGE

Subsoiling and tilling involve digging deep (30-45 cm) when preparing a new field. The large soil pieces are broken up to allow for better soil aeration, water infiltration, access to nutrients by plant roots and to remove any tree stump roots left behind during bush clearing (in section 4.1.1). There are three methods of tilling agricultural land, including: flat tilling (Figure 39A), ridging (Figure 39B) and making mounds (Figure 42).



Figure 39 — Showing (A) flat and (B) ridged land tillage methods

Source: Tarawali *et al.* (2013)

<https://cgspace.cgiar.org/bitstream/handle/10568/81177/U13ManTarawaliComprehensiveNothomDev.pdf-1427d7b5f67a7faa38df85af027128c0.pdf?sequence=1&isAllowed=y>

a. Flat tilling

Flat tilling (Figure 40A) is the most common ancient method of land preparation, especially in areas where the soils are deep. The process involves subsoiling, which may be carried out in two shifts, commonly referred to as 1st and 2nd ploughing. The 1st ploughing involves breaking the hard soil pan coupled with deep digging and turning the large soil clods. This is followed by a 2nd ploughing, which further breaks up the large soil clods and turns them. Both the 1st and 2nd ploughing (subsoiling) can be carried out with a tractor driven disc plough (Figure 40A), an oxen-pulled plough (Figure 40B) or a hand-held hoe (Figure 40C).



Figure 40 — Showing (A) tractor ploughing, (B) oxen-ploughing and (C) hand-held hoe ploughing
Source: https://www.mdpi.com/agronomy/agronomy-07-00040/article_deploy/html/images/agronomy-07-00040-g010.png

b. Ridging

Ridging (Figure 41A-C) is practiced on shallow poorly drained soils, and requires the availability of a big labour force and money to meet the cost of making the ridges. Heaps of soil are made at a height of 30 to 60 cm, and normally spaced 75–100 cm apart (Ekanayake *et al.*, 1997), except in very poorly drained areas, where wider ridges and spacing may be used. According to Tarawali *et al.* (2013), ridging was shown to produce relatively lower yields than flat cultivation, although weeds and harvesting were better managed on ridged farms.

The ridges can be made using specialised ridge-making implements mounted on a tractor (Figure 41A), oxen-pulled ridging (Figure 41B) or a hand-held hoe (Figure 41C).



Figure 41 — Showing (A) a tractor-mounted and (B) oxen-pulled ridgers and (C) a hand hoe to make ridges
Source: <https://www.businessonline.it/articoli/scatti-anzianita-contratto-agricolo-ogni-quanto-avvengono-e-di-quanto-aumenta-stipendio.html>;
<https://i.ytimg.com/vi/-gypmij8Tfo/hqdefault.jpg>; https://live.staticflickr.com/8465/8138261818_caa3f76f70_b.jpg

c. Mounds

Mounding is also an ancient ploughing method involving the use of a hand-held hoe (Figure 42, below), which consists in heaping soil in circular formations. The practice is mainly used to grow sweet potato in East Africa, although some farmers use mounds to grow cassava in swampy land with poor drainage. The heaps can be 30 – 60 cm high with a spacing of 60 – 200 cm between each other. The method is wasteful of space and is not good for commercial farming (Ekanayake *et al.*, 1997).



Figure 42 — Showing a female farmer making mounds to plant sweet potato
Source: <https://borgenproject.org/wp-content/uploads/Digital-Solutions-e1578722215507.jpg>

4.3.2. ZERO-TILLAGE

Zero/minimum tillage is used when cassava is planted on a new piece of land with the vegetation cleared but with no ploughing or burning of vegetative residues (Figure 43A & B). The method can also be used on land previously planted to another crop. However, depending on the type of soil, the method has the disadvantage of not enabling good enlargement of cassava tubers in areas with hard soil (Ekanayake *et al.*, 1997).



Figure 43 — Showing [A & B] fields cleared of vegetation and ready for planting using zero-tillage
 Source: https://i0.wp.com/nobowa.com/wp-content/uploads/2015/12/IMG_20150708_151802ed.jpg?resize=696%2C281&ssl=1

4.3.3. HARROWING

Harrowing is the activity that follows tilling the ground. It involves breaking up and smoothening the soil clods into smaller pieces to prepare for planting or making ridges or mounds. Like tilling or ridging, harrowing can also be carried out by tractor (Figure 44A) or oxen-pulled (Figure 44B) harrowers and hand-held rakes (Figure 44C). Harrowing has the advantage of further loosening the soil to enable sprouting of young cassava plants and formation of large tubers as compared to cassava grown on harder soil. However, the practice adds extra costs to farming, and should be only carried out if the ground was not adequately broken down during the 1st and 2nd ploughing.



Figure 44 — Showing (A) a tractor-mounted and (B) oxen-pulled harrowers, and (C) hand-held rake to harrow land for crop growing
 Source: <https://sc04.alicdn.com/kf/UT8n8gSXMpXXXagOFbXX.jpg>; <https://ak.picdn.net/shutterstock/videos/1023557581/thumb/1.jpg>; <https://ak.picdn.net/shutterstock/videos/6409403/thumb/2.jpg>

4.3.4. DIGGING HOLES

- Holes to plant in cassava cutting should be dug at least 10-15 cm deep and 30-40 cm wide. But the depth of the holes must be regulated depending on the weather and soil conditions (International Institute of Tropical Agriculture, 1990).
 - Make deep holes in sandy soils, although this might expose the cuttings to termites or make harvesting very difficult;
 - Make shallow holes on heavy and moist loamy soils. However, shallow planting could expose the cuttings to extreme weather conditions (high temperature or flooding), resulting in poor crop establishment with gaps and with low yields;
- Holes can be made by using hand-held hoes or pangas/cutlass and tractor planters.
- The holes should be dug using the recommended plant spacing, following the markings on a string/tape measure (manually) or with the aid of calibrated planters at 1 m x 1 m; 0.8 m x 1 m; 1 m x 0.75 m or 1 m x 0.5 m (Abass *et al.*, 2014; Hauser *et al.*, 2014).

4.4. SOIL AMENDMENTS PRIOR TO PLANTING

Add animal manure, poultry droppings, coffee husks or compost manure during land preparation. This is because these sources are slowly releasing nutrients and therefore need to be applied at least 1 month before planting cassava. In addition, they contribute to improved soil structure, and buffer supply of soil nutrients.

To apply fertiliser prior to planting, one must first carry out soil analysis to know the nutrient levels and soil pH of the field. If the soil has a pH of less than 5.5, it is considered acidic, which affects the uptake of nutrients by plants. It is advisable to apply garden lime to raise the pH level to 6-6.5 above which more lime will also affect the uptake of other nutrients.

4.5. TRANSFORMATION OF AN EXISTING PLOT

Existing plots tend to have less vegetation and softer soil, which may only require harrowing and making new ridges or remoulding old ones by breaking them up and heaping back the soil for better aeration and water infiltration. Look out for invasive weed species (see section 4.2.2. above) that spread by seed or rhizomes or cuttings to control them during clearing, tilling, harrowing and ridging. Herbicides may be used for zero-tillage cultivation, where pre-emergence herbicides (see section 4.2.3 'c', above) should be applied at least 1 month before planting, while post-emergence herbicides may be applied 1 month after planting.



PREPARING THE PLOT

KEY POINTS SUMMARY

LANDSCAPING

- Clearing vegetation by cutting down trees and shrubs, and moving the residues to the boundaries.
- Plant live hedges and windbreaks around the field to protect the crop against strong winds and stray animals such as livestock and wild animals;
- Establish grass strips to control soil erosion on terraces and boundaries of the field;
- Establish unplanted strips to serve as firebreaks near a forest of shrub that may catch wild fires;
- Erect a fence around the field using barbed wires, chain link, etc. to protect against stray animals and people that may destroy or steal the crop;
- Construct drainage channels to control flooding in swampy areas and choose an irrigation method. Leave room for unplanted areas to serve as paths.

WEED MANAGEMENT

- Use physical/cultural or mechanical methods to dig up deep rooted invasive weeds during land preparation;
- Only apply pre-planting non-selective systemic herbicides to control invasive weeds before ploughing if in high abundance in the area.

SOIL PREPARATION

- Till or plough the land using deep ploughing implements if the land has not been planted to crops before and is hard to open up for improved aeration, water infiltration and weed control. Harrow the fields only if you need to break up the soil clods after tilling or to make ridges;
- Practice zero-tillage where the vegetation does not require ploughing and there is low risk of presence of invasive weeds. Dig the holes using the agronomic recommended spacing for planting cassava.



PREPARING THE PLOT

SOIL AMENDMENTS PRIOR TO PLANTING

- Use manure, including chicken droppings and compost to improve soil fertility by incorporating them in the soil during land preparation. You may also use lime that helps reduce acidity if pH is below 5.5.

TRANSFORMATION OF AN EXISTING PLOT

- Existing plots tend to have minimum vegetation, which only need harrowing, making ridges and digging holes. Look out and control invasive weeds in the existing plots using manual, mechanical or approved herbicides at the recommended rates.







5



PLANTING
OF CASSAVA

Planting cassava crops involves carrying out several processes, including: timing the season; using the right plant density/population; using the right plantation layout, especially for commercial farming and appropriate planting methods. These processes are described in details in the following sections.

5.1. TIMING

Plan to plant cassava crops at the beginning of the growing season to get the maximum yield possible, since the first 100 days are most crucial for yield determination. Usually, cassava should be planted at least 14 days before the onset of rain, especially when using fresh cuttings obtained directly from the field or under good storage. Timely planting ensures higher sprouting percentage, good plant establishment/growth and early formation of tubers, as a result of receiving sufficient moisture for the duration of the rainy season (Howeler, 2007). Well established plants can tolerate adverse biotic (pests and diseases) and abiotic (drought, wind etc) stresses when the rains have reduced (International Institute of Tropical Agriculture, 1990).

The suitable seasons for growing cassava crops are described in detail in section 3.4 (on cassava growing period). Briefly, begin to plant cassava from:

- March/April to May/July (long rains) and September to December (short sporadic rains) in east, west and central Africa.
- November to April for the only main growing season in southern Africa (Alahacoon *et al.*, 2021).
- May/June to September/November in the Caribbean (Climate Studies Group Mona, 2020).
- July/August to November in the Pacific countries (Howeler, 2007).
- Staggered planting could be practiced by planting new cassava crops 2 to 3 months after the first crop, especially during the long rain season. However, in areas with a single rain season, staggered planting might only be feasible under irrigation to avoid the risk of exposing the young plants to dry weather.

5.2. PLANT DENSITY

5.2.1. SPACING AND PLANT POPULATION IN MONOCROPPING SYSTEMS

Optimal cassava plant spacing is 1 m by 1 m (Figure 45A) between and within the rows (including ridges or mounds), with 4,000 and 10,000 plants per acre and hectare, respectively in monocrops planted with both branching and non-branching varieties (Abass *et al.*, 2014; Hauser *et al.*, 2014). However, according to Hauser *et al.* (2014), higher plant density could be attained with reduced inter-plant spacing of 0.8 m x 1 m (5,000 and 12,500 plants) (Figure 45B) and 0.5 m x 1 m (8,000 and 20,000 plants) (Figure 45C) per acre and hectare, respectively.

Planting at high density is usually practised on soils with low fertility, but does not necessarily lead to increased yields. Moreover, overcrowding plants increases competition for the limited nutrients, moisture and sunlight, resulting in weak plant growth and low yields.



Figure 45 — Showing (A) planting cassava with spacing of (A) 1 m x 1 m; (B) 0.8 m x 1 m and (C) 0.5 m x 1 m
Source: Hauser *et al.* (2014); https://plantvillage-production-new.s3.amazonaws.com/images/pics/000/002/893/original/8098830644_f9659a4cac_z.jpg

5.2.2. SPACING IN CASSAVA INTERCROPPING SYSTEMS

Spacing of 1 m x 1 m may be used where cassava is intercropped with slow growing crops such as legumes (beans, cow peas, etc.), vegetables, cereals (maize, millet and sorghum) and ground yams. However, intercrops involving high-growing crops such as bananas or coffee etc., need much wider spacing that ranges from 1-4m between plants (International Institute of Tropical Agriculture, 1990).

The following are examples of recommended optimal plant densities for intercrops with upright growing cassava varieties planted at a spacing of 1 m x 1 m per hectare:

- **Cassava-maize intercrop:** 40,000–50,000 maize plants per hectare planted at spacing of 20 cm between plants for single rows between cassava rows (Figure 46A;
- **Cassava and legumes (e.g., cow pea, soybean etc.):** 80,000-100,000 plants per hectare planted in 2 rows at a spacing of 20 cm x 20 cm between plants and rows (Figure 46B).

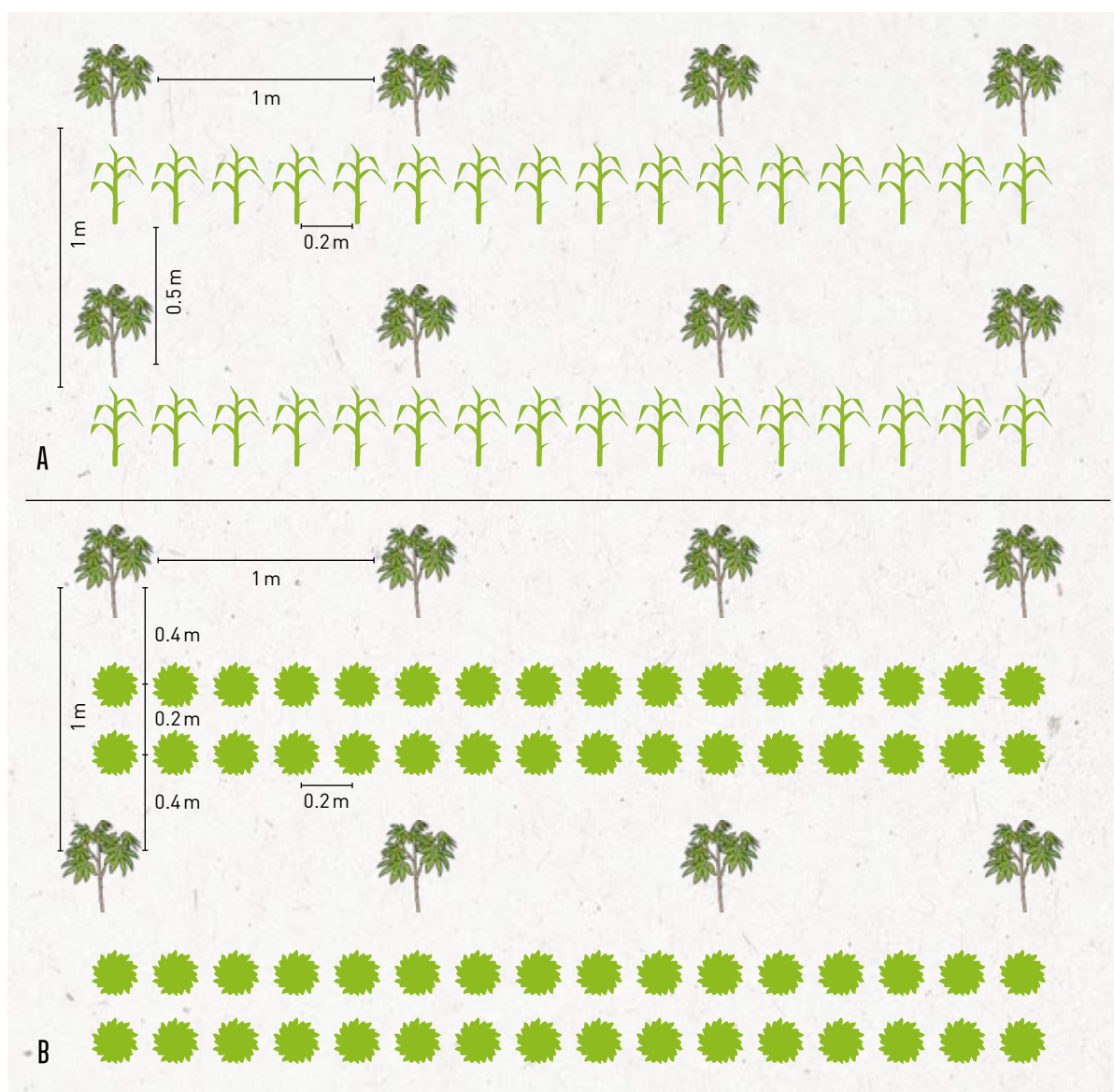


Figure 46 — Showing spacing for intercrops (A) cassava with maize and (B) cassava with cow peas
Source: Hauser *et al.* (2014)

5.3. PLANTATION LAYOUT

Before planting it is advisable to mark the field/plot, especially to facilitate mechanisation and attainment of the desired plant density. To mark the field, the following is required:

5.3.1. EQUIPMENT

- Strings – 200 m long (at least 50 pieces);
- Measuring tape (at least 100 m);
- Poles – 2 m long, not less than 10 pieces;
- Pegs – 30 cm long <100 pieces;
- Markers – (not less than 10)

5.3.2. MARKING A PLOT

Follow the **steps** below to mark an acre (ca. 4,000 m²) of land to be planted with cassava:

1. Stand at the left-hand corner of the plot (**BLACK** mark) and firmly fix the **1st pole** into the ground. Measure 5 m using a measuring tape and fix the **2nd pole** at 5 m mark (**RED** marks) along the length (Figure 47).



Figure 47 — Showing step 1 in marking a plot

2. Move the 3rd pole **anti-clockwise** until you get a 90° angle to make a **perfect triangle** that fits the length of the plot, to be able to **define the direction the width** will take. Firmly fix the 3rd pole in the ground to mark the spot for the perfect triangle (Figure 48).



Figure 48 — Showing step 2 in marking a plot

3. Place the measuring tape along the **length** and **width** of the plot, and fix pegs (**GREEN** marks) in the ground at 1 m intervals until the 100 m (101 pegs) and 41 m (41 pegs) marks, respectively (Figure 49).



Figure 49 — Showing step 3 in marking a plot

4. Mark the strings at 1 m intervals for the whole 200 m length of each string using a clear marker or brightly coloured tags (**YELLOW** marks) (Figure 50).



Figure 50 — Showing step 4 in marking a plot

5. Fix one 1 m marked string at each one of the 41 pegs (1 m apart) along the width on the **left side** of the plot.
6. Stretch the strings across the plot to the **right end**, and fix them on a corresponding peg parallel to each other at the 1 m spacing. When completed, all the 41 strings should run parallel to each other in the plot (Figure 51).

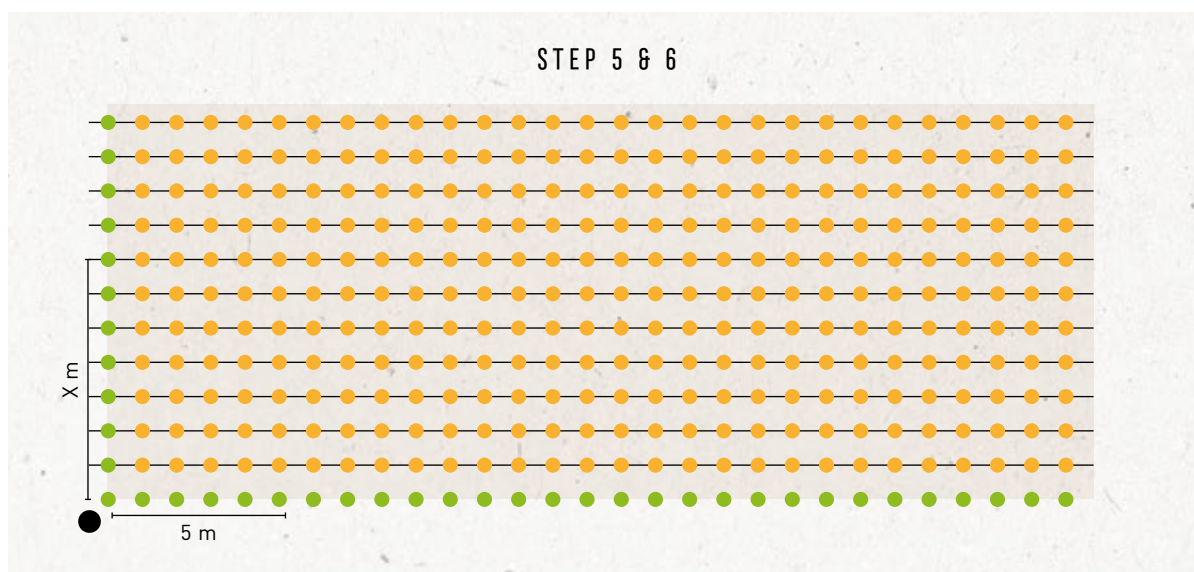


Figure 51 — Showing step 5 & 6 in marking a plot

7. Dig holes at the 1 m marks along the strings to prepare for planting on **FLAT GROUND**, or if you are using **RIDGES**, heap the soil along the string with the mid-point of the ridge running exactly under the string.

Note 1: If you do not have enough material to make 41 strings, use one (1) marked 200 m string, and keep moving it to the next peg along the width as you mark and dig planting holes at the 1 m intervals.

This technique is most applicable for manual land preparation. Mechanised planters are calibrated for making ridges at the required spacing. However, STEP 1 must be carried out to align the plot and the ridges or rows at the correct angle to maximise the space.

8. Make a boundary around the plot of 2 – 4 m wide, where you may plant a hedge or fence or rows of cassava plants of the same variety to act as a buffer for trapping pests and diseases.

5.4. METHODS OF PLACEMENT AND PLANTING CASSAVA CUTTINGS

5.4.1. PLACEMENT OF CUTTINGS

Cuttings may be placed into the holes on flat fields or ridges by use of three (3) methods, including: horizontal vertical and slanting/tilting/inclining placement (See section 3.5 for details and photos of each placement method).

5.4.2. NUMBER OF CUTTINGS PER HOLE

The agronomic recommended practice is to plant only 1 cutting per hole. However, traditional practices use 2 or 3 cuttings per hole, hoping that the multiple cuttings will produce higher yields or as a safe guard against the failure of one of the cuttings to germinate. Cassava stem cuttings produce the main tubers at the ends of the cutting, with only few side roots developing into sizeable edible tubers.

5.4.3. METHODS OF PLANTING

Cassava cuttings can be planted manually by hand and mechanically using specialised planters mounted and operated on a tractor, especially for large scale (>5 hectares) farming. See section 3.5 for details and photos of cutting placement methods. At planting, apply potassium (K) in the planting holes at 10–15 cm deep to support root establishment.

a. Manual planting

- Manually dig a hole at the marked spacing along a string using a hoe if planting on flat ground or a panga/cutlass when planting on ridges (Figure 52A) or mounds (Hauser *et al.*, 2014);
 - The planting hole should be made in centre of the ridge or mound to increase the stability of the cassava stand during rainy or windy weather.
- Insert 1 cutting vertically or slanted (45°) into the hole (Figure 52B) and fill back soil to cover 2/3 of the cutting, with 1/3 of the cutting remaining above the ground;
- Alternatively, push the cutting directly into the soil by hand (Figure 52C), if the soil is soft.



Figure 52 — Showing (A) making a hole using a panga/cutlass; (B) inserting the cutting vertically or slanting into the hole; (C) pushing the cutting directly into the soil on a ridge without making a hole and (D) the length of cuttings
Source: Stefan Hauser, IITA



Figure 53 — Showing farmers planting cassava cuttings manually by 'pushing' the cuttings directly without making holes into the soil on ridges
Source: https://plantvillage-production-new.s3.amazonaws.com/images/pics/000/002/892/original/9412141939_cca7394e77_z.jpg

Manual planting is labour intensive and slow, requiring a large workforce (Figure 53) to plant a sizeable quantity of cuttings per day.

b. Mechanised cassava planters

Mechanised planting involves the use of several designs (Figure 54A and 54B) of cassava planters globally. All the current models only place the cuttings horizontally in the soil, which is the method that has proved to achieve the highest yields in commercial cassava production.

The cuttings are kept in small or large containers, depending on the model of the planter and scale of operations, and placed into the planters by the help of a human seated at the back of the tractor. Mechanised planters can seed large acreages in a single day compared to manual planting by use of hands. The mechanized planters are also more precise and uniformly bury the cuttings. These machine planters are even able to make ridges and plant the cuttings at the same time. However, the method is expensive as it requires buying a tractor, planter/s, use of fuel and specialized expertise.



Figure 54 — Showing mechanised two models (A & B) of cassava planters mounted on tractors

Source: <https://tractorexport.com/wp-content/uploads/2017/07/Cassava-planter-1000x724.jpg>;
<https://image.made-in-china.com/43f34j00cUPYkBWqYrbV/Farm-Equipment-Cassava-Planter.webp>

5.4.4. REPLACEMENT OF FAILURES

If the quality of the cuttings is strictly observed, the recommended agronomic practices followed to the letter and the weather conditions are perfect, 90% of the cuttings will sprout (Howeler, 2007; Hauser *et al.*, 2014). The cuttings should sprout after 7-14 days after planting (DAP), and develop fully open leaves at 30 DAP.

- Look out for cuttings that fail to sprout by day 15. Unbury and move them to the boundaries for burning.
- Replant all the gaps (same holes) manually using healthy cuttings by the 3rd week (in 7 days) to maintain the required plant population. The small divergence between the planting dates will not expose the new plants to unfair competition from the initial plantings.
- If drought is the cause of sprouting failure, wait until the weather conditions have improved, or else, the replacements may dry up, which might be both costly and frustrating. Otherwise, dry cuttings tend to have less moisture, which affects sprouting, while poorly kept cuttings could have some nodes sprouting before they are planted, and such shoots get destroyed with poor handling during planting, which may also reduce percentage of sprouting.
- Seek the advice of an extension/agricultural officer or agronomist should a higher rate (>10%) of the cuttings fail to sprout.



PLANTING OF CASSAVA

KEY SUMMARY POINTS

TIMING

- Cassava should be planted at least 14 days before the rains begin to establish well;
- The main seasons are:
 - March/April to May/July (long rains) and September to December (short sporadic rains) in east, west and central Africa;
 - November to April for the only main growing season in southern Africa;
 - May/June to September/November in the Caribbean;
 - July/August to November in the Pacific countries.
- Staggered planting can be practiced, especially during the long rain season in areas with bi-annual rainfall pattern, and where irrigation is used in areas with a single growing season.

DENSITY

- Use the recommended spacing of 1 m x 1 m (10,000 plants per hectare);
- High density may be achieved with reduced spacing of 0.8 m x 1 m (12,500 plants) or 0.5 x 1 m (20,000 plants) for non-branching varieties;
- Low-growing intercrops such as legume, maize and vegetables may be planted with spacing of 1 m x 1 m, but wider spacing of up to 4 m between plants is needed for high-growing intercrops such as bananas and coffee.

PLANTATION LAYOUT

- Mark the field to obtain maximum plant density and to ease mechanized field operations.

METHOD

- Cuttings may be placed horizontally, vertically or by slanting at 45° in the ground during planting;
- Planting may be done manually using a hand-held hoe or cutlass, which is labour intensive and costly, or by use of mechanical planters mounted and operated by a tractor.
- Replace all failures within 21 days to maintain uniformity and the required plant density.





6

**CROP
MANAGEMENT**

6.1. WATER

The Food and Agricultural Organisation – FAO (2017) report on “Water for Sustainable Food and Agriculture” for the G20 Presidency of Germany, emphasized the importance of conserving water in efforts seeking to improve agricultural productivity. Key definitions regarding water use, including: water-use efficiency, irrigation efficiency and water productivity as they relate to sustainable agricultural productivity were made. According to the FAO (2017) report, the following parameters were defined as follows:

Water-use efficiency measures the “efficiency of crops (irrigated or rainfed) to produce biomass and/or harvestable yield per unit water”, which agrees with Pereira *et al.* (2012)’s definition.

Irrigation efficiency is “the ratio of the volume of water required for irrigation; including water needed for crop transpiration, soil leaching to prevent salinization, weed control, etc., over the volume of water diverted from the source of supply”.

Water productivity is “the ratio of the actual crop yield over the volume of water that is beneficially consumed by the crop”. According to Van Halsema and Vincent (2012), water productivity could also be measured in monetary units such as US dollars per cubic meter of water.

The FAO (2017) report recommended the use of ‘water productivity’ as the most inclusive unit of measure, because it is not scale-dependent and applies to a wider sense unlike ‘irrigation efficiency and ‘water-use efficiency’. Relating to cassava production and water use, the following observations and recommendations were based on empirical studies obtained in related literature.

6.1.1. WATER USE EFFICIENCY AND WATER PRODUCTIVITY

The quantity of water needed to produce cassava varies depending on the weather conditions, production methods, soil type and varieties grown. Although cassava can tolerate drought conditions and can do well on marginal land unsuitable for growing other crops (International Institute of Tropical Agriculture, 1990), it has the highest water requirement per hectare (ca. 7,827 m³) after rice (ca. 5,354 m³) and maize (ca. 3,756 m³). For example, the irrigation requirements of cassava crops ranged from 362 to 703 mm in India (Pushpalatha *et al.*, 2020) and 651.13 mm to 1,701.3 mm in Nigeria (Odubanjo *et al.*, 2011), with clear impact on yield (7.38 Kg/ha and 32.93 kg/ha), respectively. Therefore, adequate water must be provided to cassava crops to achieve its maximum yield potential of 80 t/ha (International Institute of Tropical Agriculture, 1990).

For good growth and formation of tubers, cassava plants need an average of 800 - 1000 mm annually with a minimum of 50 mm of rainfall per month (Hauser *et al.*, 2014; Silva *et al.*, 2021). However, excess supply of water should be avoided during irrigation as it may cause rotting of tubers as well as increasing the cost of production, leading to high economic losses.

Of the three irrigation methods described in section 3.7 (above), including: furrow, drip and sprinkler irrigation, drip irrigation was reported to be the most effective and had the highest efficiency (Pawar *et al.*, 2015) in delivering water for proper growth and increased yields and quality of cassava (Muhumed *et al.*, 2014).

6.1.2. MANAGEMENT OF WATER

The following recommendations help to manage water use in cassava farming to reduce wastage and achieve maximum benefits.

- a. Manage land parcels to reduce water requirements and loss
 - Install wind breaks to reduce evaporation and produce a bedding.
 - On flat ground, arrange the surface to foster the infiltration of water.
 - On sloping ground, arrange the slope to slow down run-off.
 - Make ridges or mounds to improve the drainage if the area to grow the crop is in or near a swamp, river banks or lake shores. The furrow between the ridges helps to drain the excess water.
- b. Water retention practices
 - Increase the organic matter content and maintain the lumpy structure of the soil.
 - Reduce water loss through evaporation by covering the soil with mulch.
 - Hoe the soil surface to break up the crust that contributes to water evaporation.
- c. Reduce wastage of irrigation water
 - Roughly estimate the soil humidity (the soil water content).
 - Adapt input strictly to the needs of the plant, on the basis of its state of growth and the crop density.
 - Adapt the surface areas to be irrigated to the water resource available and use the most efficient technique to avoid over-exploiting this resource.
- d. Diversify and protect water resources
 - To avoid water loss, carefully maintain equipment and irrigation systems (clean filters, channels and dripper, replace worn or defective items).
 - Diversify and exploit different water sources (including rain water and water used to wash fruit and vegetables); making sure that water contaminated with microbes (faecal contamination) or hazardous products is not used. Never use used domestic waste water: it contains bacteria that are dangerous to human.

6.1.3. IRRIGATION WATER QUALITY

Irrigation can boost the growth and yield of cassava during scarcity of rainfall and off season, which ensures that farmers can continue to have food security and income generation. Refer to section 3.7, for the description of the three main irrigation types: furrow, drip and sprinkler irrigation used in cassava farming. However, less attention is put on the quality of water used for irrigating cassava, although many farmers grow cassava in valleys, which are prone to contamination by physical, chemical and biological substances that affect yield and quality of cassava and human health (Ayers and Westcot, 1985; Arshad and Shakoor, 2017).

Important parameters to check for the quality of irrigation water:

- Physical:
 - Small stones and impurities that may block irrigation systems and equipment (drip lines, pumps, pipes etc.), reducing the efficiency of operations and increasing production costs.
- Chemical:
 - Soluble salts that cause salinity or toxicity which may affect soil fertility, water infiltration, resulting in reduced crop yields;
 - Chemicals that might corrode or clog irrigation systems and equipment;
 - Heavy metals like lead or mercury which are poisonous to humans and animals;
- Biological contaminants:
 - Bacteria and other micro-organisms harmful to human and animal health as well as for the soil and plants.

Therefore, before using water from any source for irrigation, take samples for analysis to a nearby accredited laboratory in your country, to analyse its quality. The US and FAO standards for irrigation water (Arshad *et al.*, 2017) are presented in Table 6.

Table 6 — Irrigation water quality standards
(US Regional Salinity Laboratory and FAO)

WATER QUALITY CLASSIFICATION	SALINITY HAZARD			
	EC AT 25 °C (MICROMHOS/CM)	TDS (MG/L)	SAR (MEQ/L)	RSC (MEQ/L)
Excellent	<250	<160	Up to 10	<1.25
Good	250-750	160-500	10-18	1.25-2.5
Medium	750-2250	500-1500	18-26	>2.5
Bad	2250-4000	1500-2500	>26	-
Very Bad	>4000	>2500	>26	-
WATER QUALITY CLASSIFICATION	SALINITY HAZARD			
	EC AT 25 °C (MICROMHOS/CM)	TDS (MG/L)	SAR (MEQ/L)	RSC (MEQ/L)
Usable water which can be used directly for irrigation without dilution	Up to 1500	1000	Up to 10	Up to 2.5
Marginal water useable after dilution with canal water with 1:1 ratio	1500-2700	100-1700	10-18	2.5-5.0
Hazardous water that is difficult to use without damaging crop or soil	>2700	>1700	>18	>5.0

Source: Arshad *et al.* (2017)

6.2. FERTILISER

Cassava is a heavy feeder and quickly depletes soil nutrients if produced on the same plot continuously. For example, each ton of cassava tubers harvested removes approximately 2.3 kg of nitrogen (N), 0.4 kg of phosphorus (P) and 3.0 kg of potassium (K) from the soil (Buyinza and Kitinoja, 2018). This is worsened with the harvesting and removal of other plant parts, including the stems and leaves from the plot. These plant parts were traditionally left in the garden, where they decomposed to form manure for the new crops. Therefore, it is important to apply fertilisers to reverse declining soil fertility, replace the depleted or rejuvenate the poor soils, and to achieve the full yield potential of cassava.

Ideally, soil and leaf analyses should be conducted by growers before making a decision on type and quantity of fertiliser to apply and fertilizer programmes should be adjusted to site specific edaphic conditions. To optimize the 'fertilizer use efficiency' (i.e. crop output per unit of nutrient applied), growers should consider the **4R strategy on nutrient stewardship**. This concept highlights the need to apply fertilizers at the

right source, right rate, right time, and right place. To this end, several methods of fertilizer applications can be considered for cassava, like fertigation, foliar sprays, broadcasting, and placements. This is the only way of ensuring that the trees are receiving the correct nutrition.

However, if the facility to conduct detailed soil and/or plant analyses or request site specific fertilizer recommendations is not available, it is recommended that the farmer should add a balanced fertiliser containing nitrogen, potassium and phosphorous (NPK), starting the year the trees are planted. Generally, organic manures, such as poultry manure and cattle dung, and leguminous plants and/or their debris can be used to further enhance soil fertility in cassava crops, especially where the nutrient levels are not known (Hauser *et al.*, 2014). In general, combining mineral fertilizers with organic inputs has most beneficial outcomes. Also, consider the the type of soils, timing (crop age & demand), cropping system (mono or intercrop), type, availability and cost of fertilisers. More information and advices to derive site-specific fertilizer recommendations can be found here. (www.akilimo.org)

4R PRINCIPLES

The 4R nutrient stewardship principles are the same globally, but how they are used locally varies depending on field and site specific characteristics such as soil, cropping system, management techniques and climate. The scientific principles of the 4R framework include:

- RIGHT SOURCE – Ensure a balanced supply of essential nutrients, considering both naturally available sources and the characteristics of specific products, in plant available forms.
- RIGHT RATE – Assess and make decisions based on soil nutrient supply and plant demand.
- RIGHT TIME – Assess and make decisions based on the dynamics of crop uptake, soil supply, nutrient loss risks, and field operation logistics.
- RIGHT PLACE – Address root-soil dynamics and nutrient movement, and manage spatial variability within the field to meet site-specific crop needs and limit potential losses from the field

IMPROVE SOIL FERTILITY

1. Apply **rational inputs** of **organic manures** and **minerals** to offset losses arising from harvesting.
2. Opt for **associations** that are beneficial to cultivated plant nutrition (development of bacteria and fungus on roots, certain bacteria fixing nitrogen from the air in the soil).
3. Adopt **sustainable agricultural practices** that protect the soil and foster microbial life.

PROTECT SOIL FERTILITY

1. **Avoid salinisation** of the soil due to irrigation with salt water and the use of certain forms of fertiliser (KCl).
2. **Avoid soil loss through erosion**. Retain trees in cultivated land parcels to exploit deep mineral resources and cover the surface.
3. **Avoid soil compaction** through trampling or the passage of machinery that destroys the structure and prevents air and water circulating.

OPTIMISE SOIL FERTILITY

1. Before applying the appropriate inputs, **estimate the need for nutrients** in the soil by observing plants indicative of low fertility and through laboratory **analysis**.
2. **Avoid all shortages and over-applications** of mineral and organic inputs since this favours the development of crop pests (white flies, leaf diseases).
3. **Combine mineral inputs with organic inputs** (combine forms of nutrient inputs to avoid excess and leaching).

To know more: browse the [COLEAD brochures](#) on the subject.

6.3. GROUND COVER

6.3.1. MULCHING

Mulching involves the use of crop residues or grass to cover the soil surface (Figure 55). It is an age-long practice that has been used by smallholder farmers. Mulching can be used in cassava crops with the following benefits (Aye *et al.*, 2013; Joint FAO, 2018):

- Reduces soil erosion;
- Suppresses weed growth;
- Enhances infiltration of rainwater into the soil;
- Reduces evaporation of soil moisture, especially in the dry season (summer);
- Conserves organic matter in the soil;
- However, weeds could be a problem in zero till fields, and thus weed control is critical to get high yields;



Figure 55 — Showing use of mulch in a cassava plot

Source: <https://plantvillage.psu.edu/blogposts/100-benefits-of-mulching-to-a-smallholder-farmer>

6.3.2. COVER CROPS

Cover crops, unlike mulch which is dead plant (including crop) residues, are live crop species such as *Pueraria javanica*; (legume) etc., that tolerate shade covers when grown together with the main crops, including cassava to confer several benefits (Asih, 2021), including:

- Protecting soil from erosion;
- Conserving soil moisture;
- Suppressing the growth of weeds;
- Enhancing soil structure with their root network of the legume cover crop *P. javanica*;
- Increasing organic matter;
- Providing nitrogen by legume cover crops;
- Increasing biodiversity;

6.4. WEED MANAGEMENT

Cassava is prone to weeds during the first three (3) months of growth before the plants develop a canopy that suppresses them. The weeds continue to affect plant growth and development, and greatly reduce yields.

6.4.1. WHY WEEDS ARISE IN A FIELD

Weeds arise as a result of the following factors:

- Failure to remove weeds with mature seed or propagation materials (rhizomes and cuttings) in the field during land preparation;
- Delaying to planting the field with the desired crop;
- Planting the crop using wider spacing than is recommended by agronomists. The open spaces encourage the growth of weeds between the crop;
- Weeding late, which allows the weeds to flower and disperse their seeds into the field;
- Poor weed management practices such as improper manual weeding; use of ineffective herbicides; poor application of herbicides with regards to timing and method of spraying etc.;
- Presence of allelopathic weeds that encourage the growth of other unwanted plant species in the field;
- Lack of crop rotation, which builds up the density of weeds such as spear grass and sedges in the field;
- Poor soil fertility, which tends to encourage the thriving of plant species that require limited nutrition such as weeds;

6.4.2. WEED CONTROL METHODS

Weeds should be managed using an integrated weed management (IWM) strategy that employs a combination of manual, cultural, mechanical and/or chemical control methods (International Institute of Tropical Agriculture, 1990; Moyo *et al.*, 2004; Abass *et al.*, 2014). The management options are described in detail here below.

a. Manual weed control

- Remove the weeds by hand and hoe/cutlass before fruiting or seed dispersal stage. Manual weeding is most practiced by smallholder farmers. It is slow and ineffective, and tends to build up perennial weeds, such as spear grass (*heteropogon contortus*) and sedges such as Nutsedge - *Cyperus* spp.; which become endemic in the field. To be of effect, manual weeding should be carried out regularly and as soon as the weeds sprout.

b. Cultural weed control

Observe good agricultural practices such as:

- Timely planting immediately after the land is ready, before the weeds establish.
- Use recommended spacing to attain high plant density to smother the weeds;
- Grow early branching cassava varieties to outcompete the weeds;
- Plant cover crops such as legumes between rows to suppress weed growth (Ekeleme *et al.*, 2003);
- Mulch cassava fields using crop residues from the previous season. The mulch cover helps to suppress weed growth;
- Apply nitrogen (N) fertiliser immediately after weeding to boost the vigour of cassava plants to outcompete the weeds.
- Practice intercropping by planting fast growing crops such as beans, soybeans, legumes, cowpea, groundnut that quickly cover the ground, while enriching the soil with nitrogen that supports cassava growth;
- Practice crop rotation to break the build-up of notorious weeds such as spear grass (*heteropogon contortus*) and sedges such as Nutsedge - *Cyperus* spp.;
- Minimum or zero tillage may not kill off the invasive weed species that propagate by rhizomes that remain underground or cuttings that tend to spread during bush clearing.

c. Mechanical weed control

- Disc ploughing during land preparation with a tractor helps to dig up deep-rooted weed species. After tilling, fallow the field to expose the weeds on the soil surface to dry or be collected and burnt;

d. Chemical weed control

- Use chemical herbicides only when deemed necessary such as in areas of the field with persistent perennial weeds that cannot be destroyed by the other methods in a-c above.
- Use post-emergence non-selective systemic herbicides to control perennial weeds that re-emerge when the crop is planted. The herbicides may be applied to areas that are affected and not to the entire field to limit associated dangers.

Note: When using herbicides to control weeds, rotate and use them correctly to avoid development of herbicide resistance in the weeds (Joint FAO, 2018).



CROP MANAGEMENT

KEY SUMMARY POINTS

WATER

- Water use efficiency and water productivity
 - Quantity of irrigation water depends on production methods, weather conditions, soil type and varieties grown;
 - Amount of water supplied to cassava impacts yield;
 - Excess supply of water may cause rotting of tubers and increased cost of production.
- Management of water in cassava farming
 - Manage land parcels to reduce water requirements and loss; perform water retention practices; reduce wastage of irrigation water; diversify and protect water resources.
- Irrigation water quality
 - Physical impurities such as stones may block irrigation systems;
 - Chemicals such as soluble salts cause salinity or toxicity affecting soil fertility, water infiltration or corrode or clog irrigation systems and equipment;
 - Heavy metals like lead or mercury are poisonous to humans and animals;
 - Biological contaminants such as bacteria and other micro-organisms are harmful to soil, plants, man and animals.

FERTILISER

- Consider soil type, timing (crop age & demand), cropping system (mono- or intercrop), fertilizer availability and cost of fertilisers;
- Ideally, soil and leaf analyses should be conducted by growers before making a decision on type and quantity of fertiliser to apply and fertilizer programmes should be adjusted to site specific edaphic conditions.
- To optimize the 'fertilizer use efficiency' (i.e. crop output per unit of nutrient applied), growers should consider the **4R strategy on nutrient stewardship (i.e. application at right source, time, rate, and place)**
- Site-specific fertiliser recommendations may be tailored here AKILIMO [www.akilimo.org] where applicable.



CROP MANAGEMENT

GROUND COVER

- Grow cover plants or crops;
- Mulching using crop residues or grass to cover the soil surface.

WEED MANAGEMENT

- Cassava is most prone to weeds during the first three (3) months of growth;
 - Manual weed control using hand and hoe before fruiting or seed dispersal stage;
 - Cultural weed control by observing GAPs such as timely planting, recommended spacing, planting cover crops, mulching, growing early branching varieties, mulching, applying nitrogen fertiliser to boost plant vigour, intercropping, crop rotation;
 - Mechanical weed control using disc ploughing during land preparation to dig up deep-rooted weed species and tilling and fallowing to expose weeds to dry or be collected and burnt.
 - Chemical weed control only if absolutely necessary such as where there is persistent perennial weeds that cannot be destroyed by the other methods.





7

CASSAVA PESTS AND DISEASES

7.1. KEY PESTS AND DISEASES

Table 7 — Summary of key pests and diseases affecting cassava and the growth stages impacted

PESTS	SPECIES	STAGE OF CROP GROWTH							
		FIELD/NURSERY	PLANTING MATERIAL	CROP ESTABLISHMENT	VEGETATIVE STAGE	TUBER DEVELOPMENT	HARVESTING OF STEMS & TUBERS	STORAGE OF STEMS & TUBERS	POST-HARVEST
INSECT PESTS									
Whiteflies	<i>Bemisia tabaci</i> , <i>Bemisia afer</i> , <i>Bemisia tuberculata</i> , <i>Aleurodicus disperses</i> , <i>Aleurotrachelus socialis</i> , <i>Tetraleurodes acacia</i> , <i>Trialeurodes variabilis</i>	0	0	+	+++	++	+	0	0
Mealybugs	<i>Phenacoccus manihoti</i>	+	++	++	+++	++	++	0	0
Mites	<i>Mononychellus tanajoa</i> , <i>Mononychellus progresivus</i> , <i>Oligonychus gossypii</i>	+	+	+	+	0	0	0	0
Scales	<i>Aonidomytilus albus</i> , <i>Parasaissetia nigra</i> , <i>Saissetia Miranda</i> , <i>Strictococcus vayssierei</i>	+	+	+	+	0	0	0	0
Thrips	<i>Retithrips syriacus</i> , <i>Scirtothrips manihoti</i>	+	+	+	+	0	0	0	0
Hoppers	<i>Zonocerus variegatus</i> , <i>Zonocerus elegans</i> , <i>Gryllotalpa Africana</i> , <i>Gryllotalpa</i> sp., <i>Gryllus assimilis</i> , <i>Empoasca bispinata</i> , <i>Scaphytopius fuliginosus</i> , <i>Scaphytopius marginelineatus</i>	+	+	+	+	0	0	0	0
Termites	<i>Isoptera: Rhinotermitidae</i> , <i>Heterotermes tenuis</i> , <i>Coptotermes paradoxis</i> , <i>Coptotermes</i> spp.	+	+	+	+	++	+	++	0
White grubs	<i>Coleoptera: Scarabeidae</i>	+	+	+	+	++	0	0	0
Caterpillars	<i>Erynnis ello: Hesperidae</i>	+	0	+	+	0	0	0	0

PESTS	SPECIES	STAGE OF CROP GROWTH							
		FIELD/NURSERY	PLANTING MATERIAL	CROP ESTABLISHMENT	VEGETATIVE STAGE	TUBER DEVELOPMENT	HARVESTING OF STEMS & TUBERS	STORAGE OF STEMS & TUBERS	POST-HARVEST
Nematodes	<i>Meloidogyne</i> spp.	+	+	+	+	+	0	0	0
Leaf-cutter ants	<i>Atta sexdens</i> , <i>Atta cephalotes</i> , <i>Acromyrmex landolti</i>	+	+	+	+	0	0	0	0
Cassava shoot flies	<i>Neosilva perezii</i> , <i>Lonchaea chalybea</i> , <i>Silva pendula</i>	+	+	+	+	0	0	0	0
Stem borers	<i>Coelosternus</i> spp., <i>Lagocheirus</i> spp., <i>Eubolus</i> sp., <i>Eulecrops manihoti</i> , <i>Chilomima clarkei</i> , <i>Chilozela bifilalis</i> , <i>Sinoxylon brassai</i> , <i>Heterobothrychus brunneus</i>	+	+	+	+	0	0	0	0
Cassava lace-wing bug	<i>Vatiga</i> spp., <i>Amblystira machalana</i>	+	+	+	0	0	0	0	0
Cassava root/Burrower bugs	<i>Cyrtomenus bergii</i>	0	0	0	+	+	+	0	0
Army worm	<i>Spodoptera</i> spp.	+	+	+	++	+	0	0	0
Tiger moth	<i>Phoenicoprocta sanguinea</i> , <i>Agrotis ipsilon</i>	+		+	+	0	0	0	0
Gall midge	<i>Iatrophobia brasiliensis</i>	+	+	+	+	0	0	0	0

Key: 0 = No impact, + = Low impact, ++ = Medium impact, +++ = High impact

DISEASE	CAUSAL PATHOGEN/ SPECIES	STAGE OF CROP GROWTH							
		FIELD/NURSERY	PLANTING MATERIAL	CROP ESTABLISHMENT	VEGETATIVE STAGE	TUBER DEVELOPMENT	HARVESTING OF STEMS & TUBERS	STORAGE OF STEMS & TUBERS	POST-HARVEST
BACTERIAL DISEASES									
Cassava bacterial blight (CBB)	<i>Xanthomonas axonopodis</i> pv. <i>manihotis</i>	+	+	+	+++	+++	0	0	0
Cassava leaf spot/Bacterial angular leaf spot	<i>Xanthomonas campestris</i> pv. <i>Cassava</i>	+	+	+	+	+	0	0	0
Bacterial stem gall	<i>Agrobacterium tumefaciens</i> Biovar 1	+	+	+	++	++	0	0	0
Bacterial stem rot and Bacterial wilt	<i>Erwinia carotovora</i> subsp. <i>Carotovora</i> ; <i>Erwinia herbicola</i>	+	+	+	+++	+++	0	0	0
FUNGAL DISEASES									
<i>Cercospora</i> leaf spots	Cassava brown leaf spot - <i>Passalora henningsii</i> / <i>Cercosporidium henningsii</i>	0	0	+	+	+	0	0	0
	Cassava leaf blight - <i>Cercospora vicosae</i>	0	0	+	+	+	0	0	0
	White leaf spot - <i>Cercospora caribaea</i>	0	0	+	+	+	0	0	0
Cassava anthracnose disease	<i>Colletotrichum gloeosporioides</i> f. sp. <i>Manihotis</i> / <i>Glomerella cingulata</i>	0	+	+	++	++	0	0	0
Concentric ring leaf spot	<i>Phyllosticta manihotae</i> , <i>Phyllosticta manihoticola</i>	0	0	+	+	+	0	0	0
Super elongation	<i>Sphaceloma manihoticola</i> , <i>Elsinoë brasiliensis</i>	+	+	+	+	+	0	0	0

DISEASE	CAUSAL PATHOGEN/ SPECIES	STAGE OF CROP GROWTH							
		FIELD/NURSERY	PLANTING MATERIAL	CROP ESTABLISHMENT	VEGETATIVE STAGE	TUBER DEVELOPMENT	HARVESTING OF STEMS & TUBERS	STORAGE OF STEMS & TUBERS	POST-HARVEST
Cassava root rot diseases	Soft root rot/ Phytophthora root rot - <i>Phytophthora cryptogea</i> , <i>Phytophthora drechsleri</i> , <i>Phytophthora erythroseptica</i> , <i>Phytophthora nicotianae var. parasitica</i> & <i>Pythium root rot</i> (<i>Pythium</i> spp., <i>Fusarium</i> spp.)	0	0	+	+	+++	+++	+	0
	Dry root rot - <i>Rigidoporus lignosus</i> , <i>Rosellinia necatrix</i> , <i>Lasiodiplodia theobromae</i> , <i>Armillariella mellea</i> , <i>Sphaerotilbe repens</i>	0	0	+	+	+++	+++	+	0
	Dematophora root rot - <i>Rosellinia root rot</i> , <i>Dematophora necatrix</i> <i>Rosellinia necatrix</i>	0	0	+	+	+++	+++	+	0
	Diplodia root and stem rot - <i>Diplodia manihoti</i>	0	0	+	+	+++	+++	+	0
	Black root and stem rot - <i>Scytalidium</i> sp., <i>Hendersonula toruloidea</i>	0	0	+	+	+++	+++	+	0
Cassava tuber rot disease	Sclerotium/Tuber rot (southern blight)	0	0	+	+	+++	+++	+	0
	Rust	0	0	+	+	+++	+++	+	0
	Verticillium root and stem rot	0	0	+	+	+++	+++	+	0
Cassava ash		0	0	+	+	+	0	0	0

Key: 0 = No impact, + = Low impact, ++ = Medium impact, +++ = High impact

DISEASE	CAUSAL PATHOGEN/ SPECIES	STAGE OF CROP GROWTH							
		FIELD/NURSERY	PLANTING MATERIAL	CROP ESTABLISHMENT	VEGETATIVE STAGE	TUBER DEVELOPMENT	HARVESTING OF STEMS & TUBERS	STORAGE OF STEMS & TUBERS	POST-HARVEST
VIRAL DISEASES									
Cassava mosaic disease	<i>African cassava mosaic virus (ACMV)</i> , <i>East African cassava mosaic virus (EACMV)</i> & <i>South African cassava mosaic virus (SACMV)</i>	++	++	++	+++	0	+	+	0
Cassava brown streak disease	<i>Cassava brown streak virus (CBSV)</i> & <i>Uganda cassava brown streak virus (UCBSV)</i>	++	++	++	+++	+++	+++	++	++
Cassava common mosaic disease	<i>Cassava common mosaic virus (CCMV)</i> , <i>Brazilian cassava common mosaic virus</i> or <i>Cassava common mosaic potexvirus</i>	+	+	++	+++	+++	+	0	0
Cassava leaf vein mosaic disease	<i>Cassava vein mosaic virus (CsVMV/CVMV)</i> , <i>Cassava vein mosaic caulimovirus</i>	+	+	++	++	++	+	0	0
Other viral diseases/ viruses	<i>Cassava virus X (Potexvirus) (CsXV)</i>	+	0	+	+	++	0	0	0
	<i>Cassava Colombian symptomless virus (Potexvirus) (CCSpV)</i>	0	0	+	+	++	0	0	0
	<i>Cassava symptomless infections (Cassava American latent virus - CsALV, Cassava Ivorian bacilliform virus - CIBV)</i>	0	0	+	+	++	0	0	0
	<i>Cassava frogskin disease (Cassava frogskin Associated Virus - CsFSaV)</i>	+	0	+	+	++	0	0	0

DISEASE	CAUSAL PATHOGEN/ SPECIES	STAGE OF CROP GROWTH							
		FIELD/NURSERY	PLANTING MATERIAL	CROP ESTABLISHMENT	VEGETATIVE STAGE	TUBER DEVELOPMENT	HARVESTING OF STEMS & TUBERS	STORAGE OF STEMS & TUBERS	POST-HARVEST
PHYTOPLASMA DISEASES									
Cassava Antholysis	Phytoplasma	+	+	+	+	0	0	0	0
Cassava Witches' broom	Candidatus Phytoplasma aurantifolia	+	+++	+++	+++	+++	0	0	0
POST-HARVEST PESTS									
Red flour beetle	Tribolium castaneum	0	0	0	0	0	0	+++	++
Lesser grain borer	Rhizopertha dominica	0	0	0	0	0	0	+++	++
POST-HARVEST DISEASES (ROTS) AND CAUSAL PATHOGENS									
Post-harvest root rot	Botryodiplodia theobromae	0	0	0	0	+	+	+++	+++
Post-harvest root rot	Aspergillus flavus	0	0	0	0	+	+	+++	+++
Post-harvest root rot	Fusarium solani & Rhizopus sp.	0	0	0	0	+	+	+++	+++

Key: 0 = No impact, + = Low impact, ++ = Medium impact, +++ = High impact

7.2. INTEGRATED PEST MANAGEMENT

7.2.1. BACKGROUND

Integrated pest management (IPM) involves the use of two or more control options to effectively manage the pests and diseases that limit crop production, while safeguarding the quality and quantity of agricultural produce through minimizing the impact of pesticide use on human health and the environment (including biodiversity (Wijnands and Baur, 2012).

Among the control options used in an IPM programme, are: cultural practices that include the manipulation of crop, husbandry and good agricultural practices (GAPs); biological control which acts through proven natural enemies to crops pests and disease-causing pathogens; mechanical techniques and plant protection products that include organic and inorganic pesticides that are considered as the last option. The use of pesticides should be selective, with approval and knowledge of the Maximum Residue Limits (MRLs) for the target markets (see recent guidelines from CODEX Alimentarius: <https://www.fao.org/fao-who-codexalimentarius/codex-texts/dbs/pestres/pesticides/en/> for EU markets: https://food.ec.europa.eu/plants/pesticides/eu-pesticides-database_en; and the UK market: <https://secure.pesticides.gov.uk/MRLs/search> and COLEAD Crop Protection Database: <https://resources.colead.link/en/vue-substance-active-culture>). Even then, pesticides should be used only in circumstances where GAPs, cultural, biological and mechanical control options are either inaccessible or the pest situation is out of control. Nevertheless, to ensure food and environmental safety, only approved pesticides should strictly be used with a good knowledge of the approved and regulated active ingredients for the target pest/diseases. (consult lists of pesticides authorised by competent authorities)

Farmers globally and specifically in the African, Caribbean and Pacific (ACP) countries generally use traditional knowledge acquired over many years to manage pests and diseases. However, local knowledge alone may not be sufficient to sustainably manage the major pests and disease currently limiting agricultural production. This is due to the ever-increasing emergence of new pest and disease epidemics with devastating impact on food production. This has forced majority of the farmers to rely on the use of chemical pesticides to control agricultural pests and diseases. However, apart from a general lack of knowledge of the chemical pesticides, i.e., active ingredients, application rates and stage of crop growth, MRLs, and mode of action etc, the farmers also lack knowledge of target pest biology. Therefore, overuse of chemical pesticides has resulted in development of pesticide resistance among some pests such as whiteflies (Ellsworth *et al.*, 2001). Failure to control a certain pest has led some farmers to mix multiple chemical pesticides (and sometimes together with fertilisers with a view of saving time), which reduces the efficacy of the pesticides on one hand, and risks the health of farmers/workers and the environment. However, the use of IPM strategies is still low among most farmers, due to the following critical factors:

- Lack of knowledge of IPM;
- Lack of knowledge of pest/disease identity, biology (cause, symptoms, means of spread/dispersal, damage caused, life cycle), favourable climatic conditions and ecology;

- Low awareness of control options, their effectiveness and method of application;
- Limited knowledge of stage of crop growth to achieve maximum efficacy;
- Cost of IPM options.

To increase the adoption and use of IPM practices there must be deliberate creation of awareness and training of farmers (Sseruwagi *et al.*, 2019). As indicated in the results of the IPM adoption study (Figure 56), majority of the good agricultural practices (GAPs) and cultural pest/disease control strategies are affordable (low cost) and require only basic knowledge. However, the **integration of the control practices into an effective IPM package (no. 16)** for a specific pest/disease situation requires **increased knowledge** of both the pest/disease and practices.

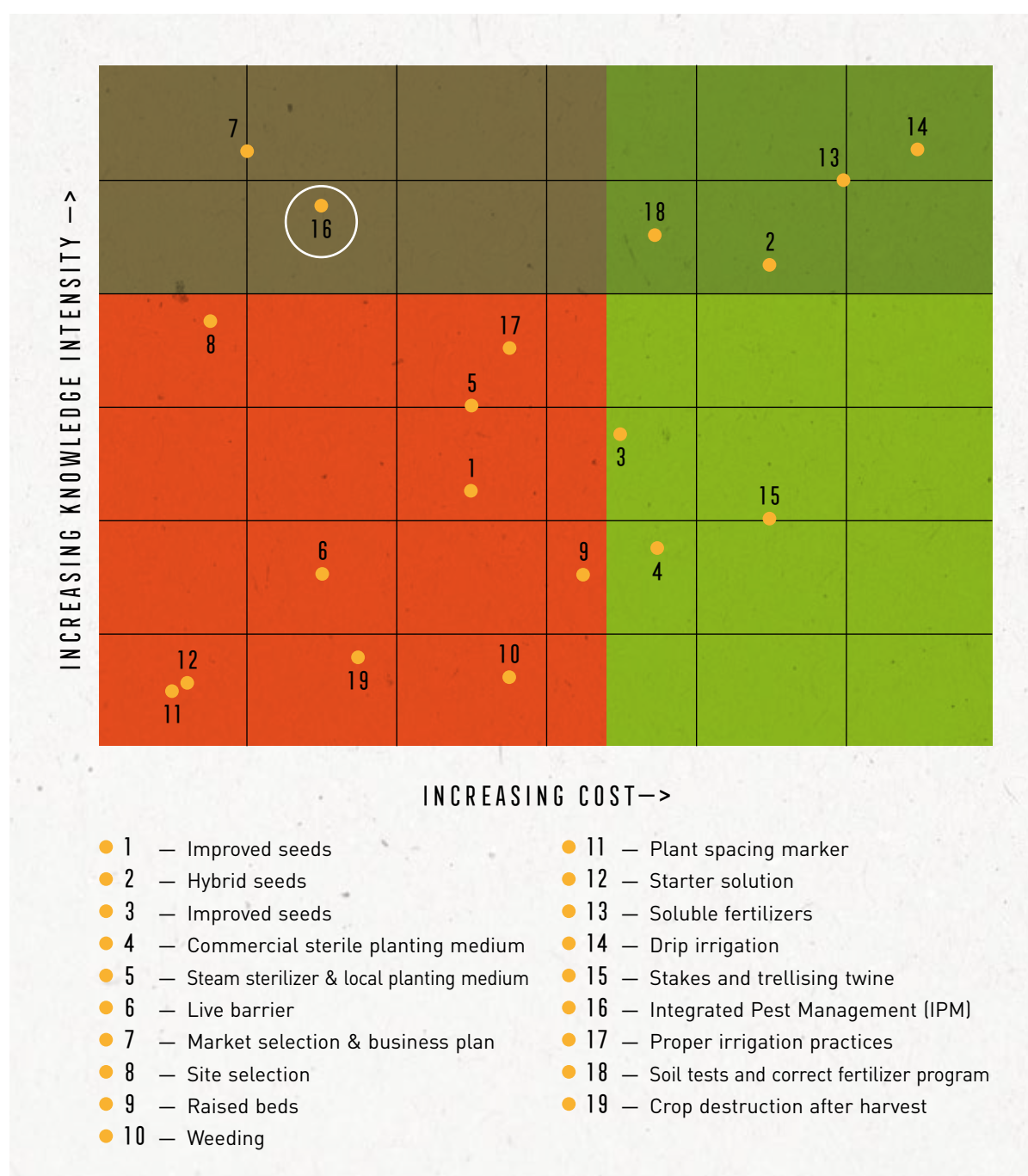


Figure 56 — Adoption of integrated pest management knowledge and technologies
Source: Sseruwagi *et al.* (2019)

Therefore, an integrated pest management (IPM) programme aims to:

- Ensure sustainable pest and disease control;
- Increase crop yields through effective control of pests and diseases;
- Increase agricultural productivity through reduced costs;
- Improve production of healthy/safe food;
- Minimise reliance on chemical pesticides to control pests and diseases;
- Reduce risks to human health and the environment.

7.2.2. COMPONENTS OF AN IPM PROGRAMME

The following are the component control options (with examples) that may be used in an IPM programme for sustainable management of cassava pests and diseases.

a. Scouting and surveillance

Scouting and surveillance for pests and diseases should be carried out one month after planting (MAP) at sprouting and in the early stages of crop growth (2-6 months) as described in Sseruwagi *et al.* (2003; 2004), to detect pest and/or disease entry. Thereafter, scouting/surveillance should be conducted at regular intervals (bi-weekly) and the pest population or number of disease-infected plants recorded to guide the control measures to deploy. This should prevent further build-up of pests and diseases in the crop, and spread to other cassava fields. For example, surveillance data were used to model the deployment of cultural control measures such as fallow crops and whitefly-resistant cassava varieties in the management of cassava whiteflies (*Bemisia tabaci*) according to Parry *et al.* (2020) and other cassava pests and diseases (Alonso *et al.*, 2021). Diagnosis of cassava diseases may be facilitated by the recently developed Plant Village Nuru application.

b. Early and deep ploughing

During field preparation, early and deep ploughing exposes life stages (larvae, pupae and adults) of pests to the surface where they could die due to heat shock by sunlight or be eaten up by birds and small animals.

c. Selection and use of healthy (pest and disease-free) planting materials

Selection of pest and disease-free cassava planting materials is key to ensuring good crop establishment and plant vigour. For example, the progress of cassava mosaic disease (CMD) infection and spread (incidence) in a local or unimproved susceptible cultivar Ebwanatereka obtained from healthy source was slower than in planting materials of the same cultivar obtained from CMD-infected source crops (Osiru *et al.*, 1999). More recently, the advent of biotechnology and molecular biology tools and techniques can help avoid the spread of cassava diseases. For example, tissue culture allows the production of high numbers of planting materials of the desired cultivar, or enzyme-linked immunoassay (ELISA) and polymerase chain reaction (PCR) (Silva *et al.*, 2011; Bulubulu *et al.*, 2015) make possible the screening of cassava plants for absence of viral diseases before harvesting, distribution and planting of cassava planting materials.

d. Method of planting (burying vs staking)

The method of planting cassava cuttings can be used to control pests and diseases. For example, using the horizontal/burying method can kill pests such as cassava green mites (CGM), scales etc., on the stems. On the other hand, the vertical or inclined/slanting/tilted planting method promotes early infestation of cassava plants upon sprouting (Ezulike and Egwuatu, 1990).

Planting cassava on ridges rather than flat ground was reported to increase plant vigour and root yield by 22% (Capinera, 2008) as a result of reduced infestation by scales.

e. Planting date choice

The adequate choice of the planting date can help reduce the spread of pests and diseases such as the incidence and population of *S. vayssierei* that reduced in plantings made in the dry season (Ambe *et al.*, 1999) in central and southern Cameroon. Similarly, delayed planting at the end of the rainy season helped reduce the spread of cassava bacterial blight (CBB) disease (Zinsou *et al.*, 2004).

f. Use of traps and baits

Use of traps such as sticky yellow and blue traps helps attract whiteflies (Idris *et al.*, 2012) and thrips, which eventually die without application of chemical pesticides. Bright colours such as white, blue and yellow attract more insects than darker ones and are used for mass trapping of pests in greenhouses too. On the other hand, formicide baits containing sugary substrates and an insecticide were used to trap leafcutter ants (Lewis and Norton, 1973; De Britto *et al.*, 2016). Hydrolyzed maize, balsamic and red wine vinegar are used as baits for trapping fruit flies (Enkerlin and Reyes-Flores, 2018). However, care must be taken to limit the destruction of beneficial insects such as bees and others.

g. Isolation from other cassava crops (young and old)

The common practice of growing new cassava crops with or next to old (mature) crops exposes them to pest and disease infection. It is recommended that where land is available, the new plantings should be isolates from the older fields to reduce the chances of early pest and disease entry. This practice was used to safely produce cassava multiplication blocks of varieties with moderate resistance to CMD for distribution to farmers ahead of the severe CMD-pandemic affected zones in East Africa (Burundi, DRC, Kenya, Rwanda, Tanzania and Uganda) (Thresh and Cooter, 2005 ; Legg *et al.*, 2006).

h. Sanitation, including roguing or removal of affected plants or plant parts

Keeping cassava fields clean, including removing pest and disease-infected plants, weeds and pruning infected leaves, shoots, branches can reduce the pest populations and disease inoculum. For example, removing debris from a previously army worm-infested crop and clean weeding reduced army worm abundance and its impact on the crop (Bista *et al.*, 2020; Tambo *et al.*, 2020). Removing and burning cassava crop debris/residues infested with whitefly eggs or nymphal instars slowed down the buildup of whitefly populations and the spread of cassava mosaic disease (CMD) in cassava crops (Thresh and Cooter, 2005). Ploughing crop debris into soil after harvest helped to manage bacterial diseases (Fanou *et al.*, 2017).

i. Removal of known and potential alternative host plants

Each of the numerous pests and diseases that limit cassava production have other plant species that serve as alternative hosts where they survive during seasons when cassava is not in production. These may be other crops, weeds, bushes or trees in and next to the cassava fields. If not removed or pruned, these plants, some of which are known alternative hosts for whiteflies (Sseruwagi *et al.*, 2006; cassava green mites (Moraes *et al.*, 1995); scales (Tindo *et al.*, 2009) and cassava mosaic geminiviruses (Horowitz *et al.*, 2011), e.t.c., can be a source of pest infestation or disease infection in newly planted cassava crops. Therefore, farmers must clean cassava fields and their immediate surroundings of any weeds and bushes to remove the alternative host plant species that may harbour the pests and diseases that limit cassava production.

j. Hot water treatment of cassava stem cuttings

Use of hot water therapy (25–60°C) and two rounds of meristem tip culture reduced the cassava brown streak virus (CBSV) viral loads by 100% in the cassava brown streak disease (CBSD)-infected planting materials (Nakabonge *et al.*, 2020). Where possible, soil could be sterilised with hot water before planting to treat termites.

k. Use of fumigants for treating cassava stem cuttings

Fumigants such as dazomet can be used as pre-plant treatments to control root-knot nematodes, though care should be taken to the hazardousness of these products. It is important to cover the soil to avoid rapid spread of fumigant vapour through the air.

l. Physical measures for pest and disease control

- Shaking trees helped to fall adult scarab beetles off the tree, which were collected and killed by burning.
- Handpicking army worm egg masses, caterpillars, and immature (non-winged) grasshopper can help reduce the populations when infestation is still low (Wright *et al.*, 2015).
- Spraying plants with a strong jet of water can help to reduce build-up of mite populations.
- Pouring charcoal dust or ash on the affected plants or plant parts also helps reducing pest infestation.
- Applying plant extracts of chilli, neem, Tephrosia, Tithonia, Lantana and garlic can reduce entry of army worms into the field.
- Protect cassava plants by banding the stems with grease or polyisobutylene gel to avoid fruit flies settling and piercing the stems.

m. Change land use

Change type of land use, such as planting shade trees that create unfavourable conditions for the survival of pests such as fruit flies near cassava crops.

n. Barrier plants (and crops)

Barrier crops such as sorghum, and maize can be used to control the entry of whiteflies into new cassava crops. However, Fereres (2000) argues that the efficacy of barrier

crops/plants depends on the height of the crop to be protected, the timing of the intervention to maximise the effect, and whether or not the barrier crops/plants serve as alternative hosts for pests/disease that affect the protected crop.

o. Irrigation/flooding

Flooding resulting from irrigation and exposing fields to sunlight can be used to control nematodes, especially in the dry season (Olsen, 2011).

p. Mulching

Reflective mulches deterres thrips from settling on cassava plants (Lewis, 1997). Organic mulch, such as crop residues reduces thrips and leaf miner populations (Larentzaki *et al.*, 2008). Furthermore, organic mulch can also reduce water and soil splashes, which minimises the spread of fungal pathogens.

q. Cover crops and repellant plants

Use plant cover crops such as sudangrass and marigolds to create unfavourable conditions for nematodes. Marigold contains thiopenes which is unpleasant to nematodes, whiteflies and other pests. Other plants such as chives, garlic, tansy or catnip also serves as repellants and can be used to prevent adult beetles or grubs attacking cassava crops.

r. Crop rotation

Practice crop rotation with crops such as maize to disrupt the growth cycle of bacterial pathogens (Persley, 1978) and nematodes (The American Phytopathological Society, 2011) that infect cassava.

s. Intercropping

Intercropping cassava with cow peas or maize reduces whitefly populations on cassava (Fargette and Fauquet, 1988; Gold *et al.*, 1990; Islami *et al.*, 2011).

t. Mineral nutrient treatments

Use of manure increases the content of rutin an unfavourable substance for mealybug infestation in cassava plants (Tertuliano *et al.*, 1999). The increased mineral nutrition also helped to increase the parasitoid wasp population that controls mealybugs (Schulthess *et al.*, 1987). Heavy application of nitrogen fertiliser can kill first instar white grub larvae, while adding potassium (K) to soil helps reduce the severity of cassava bacterial blight (CBB) disease infection (Odurukwe and Arene, 1980). However, high levels of nitrogen (N) increases the symptom severity of cassava mosaic disease (Ogbe *et al.*, 1993; Sseruwagi *et al.*, 2003).

u. Resistant/tolerant varieties

Grow cassava varieties with proven resistance to or tolerance to pests and diseases. For example, presence of hairs on the leaf surface prevented the settling of mites on some cassava varieties (Leuschner, 1982). Natural genetic resistance is used by many cassava breeding programmes to manage cassava mosaic disease (Thresh *et al.*, 2005; Thuy *et al.*, 2021), cassava whiteflies (Gwandu *et al.*, 2019) and cassava bacterial blight disease (CBB) (Banito *et al.*, 2008; Zárte-Chaves *et al.*, 2021).

v. Early harvesting

Harvesting cassava crops early helps to avoid severe losses due to reduced pest and disease impact associated with cassava brown streak disease (CBSD) infections.

w. Biological control with natural enemies

Natural enemies such as the parasitic wasps: *Encarsia sophia* and *Eretmocerus mundus* have been used to effectively control whiteflies on cassava crops with parasitism rates of 34–58% of 4th instar whitefly nymphs (Bellotti *et al.*, 2012a). The mealybugs can be controlled by ladybird beetles (*Hyperaspis* spp., *Exochomus* spp. and *Diomus* spp.) (Neuenschwander *et al.*, 1987).

x. Plant protection products (PPPs), including inorganic and organic or botanical pesticides

As noted earlier, PPPs should be used as the last control strategy in an IPM programme. Always rotate pesticides with different modes of action to prevent pest-resistance development (Bista *et al.*, 2020). The following are some examples of PPPs used to control some pests on cassava crops :

- Organic neem or Azadrachtin reduces eggs, nymphs and adults of whiteflies.
- Use Deltamethrin to control grasshoppers by spraying every 7-14 days, depending on the level of infestation (Wright *et al.*, 2015). Cassava stem cuttings can be dipped in Deltamethrin or Cypermethrin to kill whitefly eggs, nymphs and adults.
- Neem extracts can control many pests including grasshoppers (Olaoifa and Adenuga, 1988).

Note: When using a pesticide, always wear protective clothing and follow the instructions on the product label, such as dosage, timing of application and pre-harvest interval. It is also important to use PPPs rationally to minimise possible harmful effects on people and the environment.

y. Creation of awareness and training

Create awareness and train farmers, extension officers and agro-dealers in demos, farmer field schools, agricultural shows, workshops, etc., on the biology (life cycle), effect, cause, symptoms and means of spread/dispersal and management of cassava pests and diseases (Bellotti *et al.*, 2012a).

z. Quarantine

Institute quarantine laws and regulations to restrict the movement of cassava planting materials and tubers infested with quarantine pests and diseases to minimise spread into uninfected areas (Frison and Feliu, 1991).



INTEGRATED PEST MANAGEMENT

KEY POINTS SUMMARY

An Integrated Pest Management (IPM) strategy involves multiple tactics and approaches to manage pest populations effectively, sustainably, and economically. The essential components of an IPM strategy include:

Pest Identification and Monitoring

- **Setting Action Thresholds:** Establishing threshold levels at which pest populations or damage indicate that control measures are needed to prevent economic harm.
- **Preventive Cultural Practices:** Implementing cultural practices that reduce pest establishment, reproduction, and survival. This can include crop rotation, intercropping, proper plant spacing, and sanitation.
- **Biological Control:** Using natural predators, parasites, and pathogens to manage pest populations. This includes introducing or conserving beneficial organisms like ladybugs, predatory mites, and parasitic wasps.
- **Mechanical and Physical Controls:** Employing physical methods to remove or exclude pests, such as hand-picking, traps, barriers, mulches, and tillage.
- **Chemical Control:** Applying pesticides judiciously and as a last resort, selecting products that are least harmful to beneficial organisms and the environment. This includes the use of biopesticides and selective chemicals.
- **Host Plant Resistance:** Growing pest-resistant crop varieties developed through breeding and genetic modification to reduce pest impact.
- **Legislative and Regulatory Controls:** Adhering to and supporting regulations that limit the spread of pests and promote safe pesticide use.
- **Education and Training:** Providing education and training to farmers, pest managers, and stakeholders on IPM principles, techniques, and tools.
- **Record Keeping and Evaluation:** Keeping detailed records of pest occurrences, monitoring data, control measures applied, and their outcomes to evaluate the effectiveness of the IPM strategy and make necessary adjustments.
- To safely use PPPs, it is important to use approved PPPs and have the knowledge of the active ingredients and the Maximum Residue Limits (MRLs) of the target markets.



INTEGRATED PEST MANAGEMENT

COLEAD'S CROP PROTECTION DATABASE PROVIDES INFORMATION ON GOOD AGRICULTURAL PRACTICES

It was put online in 2018 and is accessible to all its members and beneficiaries. To date, it is the only database of its kind to provide information specifically dedicated to supporting the horticultural sector in ACP countries. The GAP data are obtained from a combination of sources, including COLEAD field PPP trials, PPP manufacturer data and scientific literature. The Crop protection database brings together the MRLs set by the EU and Codex Alimentarius for key horticultural crops in ACP countries. It also brings together good agricultural practices (dose, interval between treatments, preharvest interval, etc.) that ensure compliance with these MRLs. Additional information such as the pesticide type, the authorisation status of the active substance in the EU and ACP countries, the classification recommended by the World Health Organisation (WHO) and the resistance group (FRAC code for fungicides); IRAC classification for insecticides) are also available. The COLEAD, Crop Protection Database can be accessed on our website: [here](#)

7.3. PESTS

7.3.1. WHITEFLIES

SCIENTIFIC NAME

- **Cassava whitefly** – *Bemisia tabaci* (Gennadius) (Hemiptera: Aleyrodidae)
- *Bemisia afer* (Gennadius) (Hemiptera: Aleyrodidae)
- **Spiralling whitefly** – *Aleurodicus dispersus* (Russell)
- *Bemisia tuberculata*
- *Aleurotrachelus socialis* Bondar
- *Trialeurodes variabilis* (Quaintance)
- **Acacia whitefly** - *Tetraleurodes acaciae* Quaintance, 1900

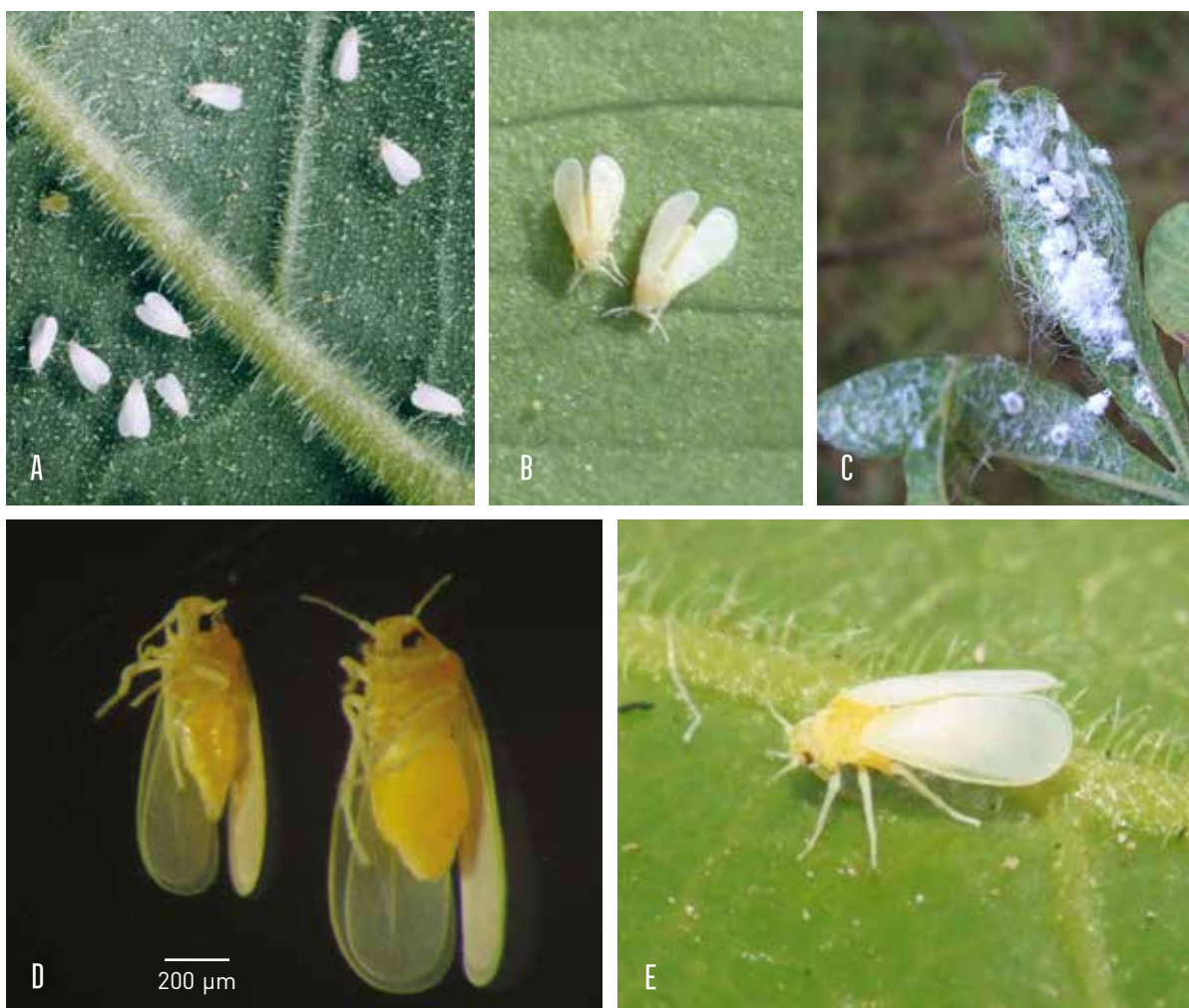


Figure 57 — Showing adult whitefly species that affect cassava production: (a) *Bemisia tabaci*; (b) *Bemisia afer*; (c) spiralling whitefly (*A. dispersus*) with fluffy substances; (d) *Bemisia tuberculata* and (e) Acacia whitefly (*Tetraleurodes acaciae*)

Sources: John C. French Sr, [Bugwood.org](http://bugwood.org); Pest and Diseases Image Library, bugwood.org, <https://www.ipmimages.org/browse/detail.cfm?imgnum=5502871>; CABI Plantwise Knowledge Bank, <https://www.plantwise.org/KnowledgeBank/pmdg/20167800170>; https://www.researchgate.net/publication/234083705_Biology_of_Bemisia_tuberculata_Bondar_Aleyrodidae_and_parasitism_by_Encarsia_porteri_Mercet_1928_Hymenoptera_Aphelinidae_on_cassava_plants/figures; https://www.inaturalist.org/taxa/493762-Tetraleurodes-acaciae/browse_photos

DESCRIPTION/IDENTIFICATION

PEST

Approximately 1,500 species of whitefly (Order Hemiptera, Family Aleyrodidae) infest agricultural crops globally (Legg *et al.*, 2003), among which *Bemisia tabaci* is the most important as a pest and as a vector of plant viruses in tropical and subtropical regions of the world (Polston and Anderson, 1997). In Africa, cassava crops are significantly affected by three whitefly species: *B. tabaci* (Figure 57A), *Bemisia afer* (Figure 57B) and *Aleurodicus disperses* (Figure 57C). Two other species *Trialeurodes variabilis* (Figure 57D) and *Aleurotrachelus socialis* (Figure 57E) are prevalent in the Americas (Angel *et al.*, 1989; Evans & Castillo, 1998), while *B. tabaci* and *A. disperses* are the most common in Asia and the Pacific (Bellotti *et al.*, 2012b; Bellotti *et al.*, 2012c). In the Caribbean, South America and Pacific, *Bemisia tuberculata* (Oliveira and Lima, 2006; Andrade *et al.*, 2012) and the Acacia whitefly (*Tetraleurodes acaciae*, Quaintance, 1900) (Dooley, 2011) were also reported on cassava.

BIOLOGY

Adult whiteflies are tiny white insects about 1 mm long. Their whole body, including wings, has a bright white powdery appearance (Smith, 2009). The nymphal stages are tiny, flattened, oval scales that have no obvious legs, do not crawl (except immediately after egg hatch for a day or so), and with no obvious head, thorax, or abdomen, they do not look like a 'typical' insect. On average, the females are larger (~1 mm) than the males (~0.8 mm) (Azab *et al.*, 1970; Gill, 1990). The males are also thinner and have more tapered abdomens, with a pair of claspers at the tip (Fishpool and Burban, 1994). *B. tabaci* have piercing and sucking mouthparts and the adults usually colonise the underside of plant leaves, where they feed and lay eggs. Female *B. tabaci* and *B. afer* lay the eggs in small circular or semicircular groups. *B. tabaci* have tapered wings, but it is difficult to distinguish between the numerous species due to similarity in their physical characteristics. Therefore, molecular techniques and sequencing are necessary to confirm the identity of the species (Sseruwagi *et al.*, 2005). Unmated females produce only male progeny, while mated females produce a mixture of male and female progeny. Adults are most active in the morning, but mate in the afternoon (Waterhouse and Norris, 1989). Large numbers are easily seen by the naked eye, and fly off the plant leaves when disturbed. *Bemisia afer* are similarly tiny white insects but have wide open wings. Spiralling whitefly, *A. dispersus*, derives its name from its characteristic spiral egg-laying pattern (Martin, 1990). The first instar nymphs (crawlers) of *Bemisia tuberculata* have three pairs of legs and move around. Successive instars remain immobile till growth into adults (Hoddle and Soliman, 2001). During the fourth instar growth stage, also called the pupae, feeding is paused while the insect develops morphologically (Gallo *et al.*, 2002). Like other whiteflies, the females (916.12 µm - length and 338.99 µm - width) are bigger in size than the males (797.16 µm - length and 200.81 µm - width) (Andrade *et al.*, 2012). The acacia whitefly, *Tetraleurodes acaciae* nymphs and pupae are shiny black with a white wax fringe and range in size from about 0.1 mm to 0.7 mm.

Figure 58A-F show *B. afer* developmental stages. In Table 8 is a brief description of the appearance of the development stages of *B. tabaci* (Byrne and Bellows, 1991), *B. afer* (Gamarra *et al.*, 2016) and *A. dispersus* (Russell, 1965; Waterhouse and Norris, 1989; Martin, 1987, 1996; Wen *et al.*, 1994a).



Figure 58 — Showing developmental stages of the whitefly, *B. afer*:

(A) egg; (B & C) I-III nymphal instars; (D & E) IV nymphal instar (pupa) and (F) adult

Source: Heidy Gamarra/ResearchGate, https://www.researchgate.net/figure/Photos-2-The-developmental-stages-of-the-whitefly-Bemisia-afer-A-egg-B-and-C_fig2_320676985

Table 8 — Description of morphological features of whitefly species affecting cassava

WHITEFLY SPECIES	EGG	NYMPHAL INSTARS	PUPA (4TH NYMPHAL INSTAR)	ADULT
<i>Bemisia tabaci</i> and <i>Bemisia afer</i>	Eggs are creamy to transparent (Figure 58A). Mature eggs turn to brown and black.	1 st nymphal instars (crawlers) are flat, oval and almost transparent (Figure 58B). 2 nd nymphal instars are translucent, oval with scalloped edges, and a yellow-greenish body (Figure 58C). 3 rd nymphal instars are similar to the 2 nd instar (Figure 58D).	The 4 th nymphal instars (pupal) are oval, flat and near-transparent. Eyes are visible towards adult emergence (Figure 58E).	Newly emerged adult males have wings. The females are clear and become covered with a white wax (Figure 58F).
<i>Aleurodicus dispersus</i>	Eggs are elliptical, smooth and yellow-to-tan in colour. Eggs have a short pedicel. The eggs are laid along with deposits of waxy secretions in a spiralling pattern.	The 1 st larval instar is a 'crawler'. The 2 nd larval instar is 0.5 mm long with a row of mid-back waxy tufts towards the front of the body. The 3 rd larval instar has glass-like waxy rods and pores on the side of the body.	Mature pupae (4 th instars) are colourless or yellowish and almost oval. The pupae have several large pores close to the edge of the body.	Adult <i>A. dispersus</i> have white bodies coated with fine dust-like waxy secretion. Both males and females have clear wings after emergence that turn white due to the wax coating after a few hours. The antennae have seven segments and eyes are dark reddish-brown.

WHITEFLY SPECIES	EGG	NYMPHAL INSTARS	PUPA (4TH NYMPHAL INSTAR)	ADULT
<i>Aleurodicus dispersus</i>	<i>A. dispersus</i> , 'spiralling whitefly', eggs on soyabean leaf.	Adult <i>A. dispersus</i> usually have plain white wings, but occasionally have pale or dark spots on the forewings. Pupae secrete copious amounts of white, cottony, flocculent wax.		
<i>Bemisia tuberculata</i>	Not reported	Not reported	Not reported	Not reported
<i>Aleurotrachelus socialis</i>	Not reported	Not reported	Not reported	Not reported
<i>Trialeurodes variabilis</i>	Not reported	Not reported	Not reported	Not reported
<i>Tetraleurodes acaciae</i> (Dooley 2011)		Nymphs are shiny black with a white wax fringe and range in size from about 0.1 mm to 0.7 mm.	Similar pupae are shiny black with a white wax fringe and range in size from about 0.1 mm to 0.7 mm.	

SYMPTOMS

Whitefly feed by extracting the contents of the cassava leaf cells. This results in chlorosis (removal of chlorophyll, the green pigment in leaves); mottling, leaf fall and in extreme cases death of affected leaves. Severe attack by whiteflies leads to death and shedding of terminal shoots, causing the characteristic 'candle stick' appearance when the plant has lost all or most of its leaves (Milenovic *et al.*, 2019).

Adult whiteflies lay their eggs on the lower surfaces of young succulent leaves at the top of cassava plants (Sseruwagi *et al.*, 2003). As the plants grow, the younger nymphs are found on the upper part of the plant, while the older nymphs are found on the lower, older parts of the plant. Therefore, the adults cluster in a few places on cassava leaves during the early stages (1–3 months after planting [MAP]) of crop growth (Sseruwagi *et al.*, 2003). The population peaks from 4–6 MAP, when the foliage is very well-formed and succulent, after which (from 7 MAP) it decreases drastically, as the plants grow taller, become woodier (less succulent) and shade the leaves.

On average, *B. tabaci* undergo 11–12 generations (Fishpool and Burban, 1994), while *B. afer* have 8–10 generations (Gamarra *et al.*, 2016) annually, depending on the environmental conditions (Asiimwe *et al.*, 2007a; b).

Whitefly population growth, dispersal and distribution depend on several factors (Macfadyen *et al.*, 2018), including:

- **Temperature:** Early in the day (6.00–11.00 a.m.), the adults are less active and tend to concentrate in a few places on the cassava leaves. However, as the temperature rises, the whiteflies become more active and spread widely over the whole crop and/or neighbouring bushes in search of shelter. Whitefly development appears to be faster during the dry season than in the wet/cold months.
- **Cassava varieties:** Improved cultivars have been reported to host higher populations of whiteflies than the local land races.
- **Poor cultural management practices:** If cassava plants are stressed due to under-watering, extreme temperatures, poor soil fertility, low light, incorrect pH of growing media, or crowding, they may be more susceptible to whitefly. High nitrogen levels have also been associated with spikes in whitefly population growth.
- **Leaf morphology and plant biochemistry:** Healthy cassava plants tend to support whitefly population growth, while unhealthy plants with reduced leaf surface area have reduced ability to support whitefly population buildup.
- **Proximity of other cassava crops and plant density:** Adult whitefly can easily infest and establish in areas heavily planted with cassava crops (Legg *et al.*, 2011).
- **Presence of alternative host plants:** The whiteflies that attack cassava, including *B. tabaci*, *B. afer* and *A. dispersus*, are polyphagous pests (i.e. they feed on many different plants) that infest more than 64 plant families – see Table 9, below (Russell, 1965; Kajita *et al.*, 1991; Wen *et al.*, 1994b; Sseruwagi *et al.*, 2006; Mware *et al.*, 2010). In addition to crop plants, ornamentals such as rose mallow (*Hibiscus* sp.) or rose (*Rosa hybrida* L.) can be whitefly hosts. The presence of any or all these host plants makes it possible for whiteflies to survive, become established and spread to new cassava crops (Table 9).

Table 9 — Host range and plants colonised by different whitefly species

WHITEFLY SPECIES	HOST PLANT SPECIES
<i>Bemisia tabaci</i> (Sseruwagi et al., 2006) & <i>B. afar</i> (Mware et al., 2010)	Asteraceae - Wild sunflower (<i>Aspilia africana</i> Pers. Adams) Convolvulaceae - Sweet potato <i>Ipomoea batatas</i> (L.) Lam Euphorbiaceae - Cassava (<i>Manihot esculenta</i> Crantz), tree cassava (<i>Manihot glaziovii</i> Muell. Arg.), Jatropha (<i>Jatropha gossypifolia</i> L.), Mexican fireplant (<i>Euphorbia heterophylla</i> L.) Fabaceae - Beans (<i>Phaseolus vulgaris</i> L.) Lauraceae - Laurel (<i>Laurus nobilis</i> L.) Malvaceae - Mexican cotton (<i>Gossypium hirsutum</i> L.), Pima cotton (<i>Gossypium barbadense</i> L.), okra (<i>Abelmoschus esculentus</i> L.) Myrtaceae - Apple guava (<i>Psidium guajava</i> L.) Rutaceae - Bitter orange (<i>Citrus aurantium</i> L.), lemon (<i>C. limon</i> (L.) Burnm. F.), sweet orange (<i>C. sinensis</i> Osbeck) Solanaceae - Pepino (<i>Solanum muricatum</i> Aiton), Chinese (<i>Capsicum chinense</i> Jacq.)
<i>Aleurodicus dispersus</i> (Mware et al., 2010; Boopathi, 2013)	<i>A. dispersus</i> has at least 147 host plants, including ornamentals, shade and fruit trees and annual crops.
<i>Bemisia tuberculata</i>	Not reported
<i>Aleurotrachelus socialis</i>	Not reported
<i>Trialeurodes variabilis</i> (Mound & Halsey 1978; Culik et al., 2004).	Main host plants are <i>C. papaya</i> and <i>M. esculenta</i> , while other hosts include <i>Acer</i> sp., <i>Citrus</i> sp., <i>Coccoloba</i> sp., and <i>Gardenia</i> sp.
<i>Tetraleurodes acaciae</i> (Dooley 2011)	Fabaceae family (legumes) Shower trees (<i>Cassia</i> sp.) Erythrina species (Fabaceae) Other plant hosts - Koa (<i>Acacia koa</i>), wiliwili (<i>Erythrina sandwicensis</i>), and uhiuhi (<i>Mezoneuron kavaensis</i>), Bauhinia, Bougainvillea, Melaleuca, Jatropha, Chamaedorea, and Pseudobombax.

DISPERSAL

The dispersal of whitefly adults is provoked by several factors, including (a) population growth, where large numbers can no longer stay comfortably on the same leaves due to reduced surface area; (b) senescence and falling of mature leaves that are low in nutrients; (c) wind, which may blow the winged adults from cassava plant leaves, transporting them several kilometres during the day but with less movement at night, depending on the wind speed and prevailing vegetation; and (d) moving nymphal instars on cassava leaves (Gamarra *et al.*, 2016).

DAMAGE

Whiteflies cause widespread damage to cassava crops globally (Otim-Nape *et al.*, 2000; Colvin *et al.*, 2004; Legg *et al.*, 2006; Patil *et al.*, 2015). Both nymphs and adults cause direct feeding damage to cassava by sucking the plant sap (Gamarra *et al.*, 2016). Severe infestation (Figure 59A) can lead to stunted plant growth, wilting and defoliation, which seriously reduces plant vigor and crop yield, and in extreme cases may kill the plants. Indirect damage occurs when adult whiteflies excrete a sugar-rich sticky honeydew, on which sooty moulds (Figure 59B) can grow, reducing the respiration and photosynthesis ability of cassava leaves. Whitefly feeding decreases plant vigor (Figure 59C). Infestations may result in leaf yellowing, wilting, and defoliation (Dooley, 2011).

On cassava, *A. dispersus* infestation causes yellowish speckling of the leaves, and in severe infestation the leaves are crinkled and curled. Infestation usually spreads from the bottom to the top leaves (Palaniswami *et al.*, 1995). *A. socialis* and *T. variabilis* were reported to reduce yield by 60-80% (Belloti *et al.*, 1983) in Colombia, with *A. socialis* transmitting the virus causing frogskin symptoms on cassava tubers (Angel *et al.*, 1987).

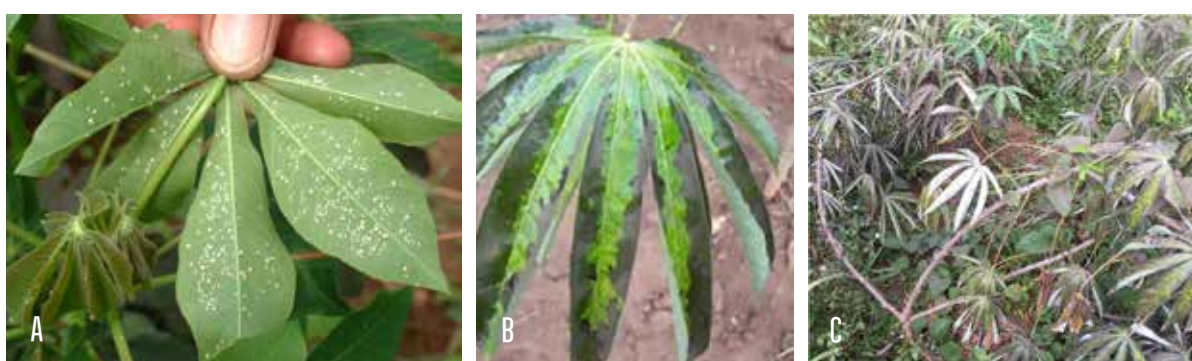


Figure 59 — Showing (A) a cassava leaf heavily infested with whiteflies; (B) symptoms of darkening due to sooty mould on cassava leaf; and (C) on plants

Source: Peter Sseruwagi (field photo); Omongo *et al.* (2018),

<https://www.sciencedirect.com/science/article/pii/S0085562618300669>

Adult whiteflies also cause indirect losses by transmitting plant viruses, the causal agents of two of the most damaging cassava diseases: cassava mosaic disease (CMD) (Figure 60A) and cassava brown streak disease (CBSD) (Figure 60B), which in combination lead to significant yield loss in cassava (Holt and Colvin, 2001; Maruthi *et al.*, 2002).

IMPACT ON YIELD AND QUALITY

Severe whitefly infestation can reduce plant growth, leading to yield loss of 40–50% in the affected plants (Thresh *et al.*, 1997; Legg *et al.*, 2014). The sooty mould reduces the quality of leaves, as the dark colouration is undesirable to consumers.



Figure 60 — Showing leaves with symptoms of (A) Cassava mosaic virus disease and (B) Cassava brown streak disease on leaves

Sources: AjayTvm/Shutterstock, <https://www.shutterstock.com/image-photo/cassava-mosaic-virus-disease-tapioca-manihot-1585752058>; Crop Crisis Control Project, <https://www.crs.org/sites/default/files/tools-research/c3p-new-spread-of-cassava-brown-streak-virus-disease.pdf>

GEOGRAPHICAL DISTRIBUTION

Whiteflies occur globally, including in sub-Saharan Africa (SSA), the Caribbean and the Pacific, where they are of high economic importance. The distribution pattern is given in Table 10, below.

Table 10 — Geographical distribution of whitefly species on cassava in the ACP countries

WHITEFLY SPECIES	AFRICA	CARIBBEAN AND SOUTH AMERICA	PACIFIC
<i>Bemisia tabaci</i>	Angola, Benin, Burkina Faso, Cabo Verde, Cameroon, Chad, Republic of the Congo, Democratic Republic of Congo, Ethiopia, Gabon, Ghana, Ivory Coast, Kenya, Madagascar, Malawi, Mauritius, Morocco, Mozambique, Nigeria, Réunion, São Tomé and Príncipe, Senegal, Seychelles, Sierra Leone, South Africa, South Sudan, Sudan, Tanzania, Togo, Uganda	Not reported	Not reported
<i>Bemisia afer</i>	Angola, Benin, Burkina Faso, Chad, Ethiopia, Ghana, Ivory Coast, Kenya, Madagascar, Malawi, Mozambique, Nigeria, Sierra Leone, South Africa, Sudan, Tanzania and Uganda	Not reported	Not reported
<i>Aleurodicus dispersus</i>	Benin, Cabo Verde, Cameroon, Republic of the Congo, Democratic Republic of the Congo, Gabon, Ghana, Kenya, Mauritius, Morocco, Mozambique, Nigeria, Réunion, São Tomé and Príncipe, Senegal, Seychelles, Tanzania, Togo and Uganda.	Cuba, Dominican Rep, Haiti, Barbados, Cayman Islands, Costa Rica, Cuba, Dominican Republic, Guadeloupe, Guatemala, Haiti, Martinique, Nicaragua, Panama, Puerto Rico, USA (Florida, Hawaii)	American Samoa, Australia Queensland, Cook Islands, Federated States of Micronesia, Fiji, French Polynesia, Guam, Kiribati, Marshall Islands, Nauru, New Caledonia, Northern Mariana Islands, Palau, Papua New, Guinea, Samoa, Solomon Islands, Tokelau and Tonga.
<i>Bemisia tuberculata</i>	Not reported	Brazil, Colombia, Ecuador and Guyana	Not reported

WHITEFLY SPECIES	AFRICA	CARIBBEAN AND SOUTH AMERICA	PACIFIC
<i>Aleurotrachelus socialis</i> Bondar	Not reported	Brazil, Colombia, Ecuador and Guyana	Not reported
<i>Trialeurodes variabilis</i> (Quaintance)	Not reported	Brazil, Colombia, Ecuador and Guyana	Not reported
<i>Tetraleurodes acaciae</i>	Not reported	Brazil, USA (Florida, California, Hawaii), Venezuela	Not reported

QUARANTINE ORGANISM

Whiteflies occur globally, especially in tropical and subtropical areas. They can fly long distances between locations and across natural geographical barriers, which makes them very difficult to control as a quarantine pest. In addition, whitefly can easily be transported or imported on cassava leaves as nymphs, which can emerge and infest other host plants (including cassava crops), posing a serious threat. Phytosanitary measures should be instituted to restrict the movement of cassava planting materials (especially the leaves, which are eaten as vegetables) from one region to another.

FAVOURABLE CONDITIONS

Whitefly may be present in cassava crops year-round, but infestations are worse during the dry season (summer), when it is hot, dry or sunny. High temperatures following the rainy season lead to an increase in whitefly populations, with a resultant increase in the spread of viral diseases such as CMD (Sseruwagi *et al.*, 2003). During winter the life cycle is longer, which makes it easier to control the whiteflies.

There is a strong potential for re-infestation to occur from crop to crop and from nearby weeds to crops where fields are grown closely together or near uncultivated bushes. The direction and speed of the prevailing wind also influences the side of the cassava field that is colonised by whiteflies (Macfadyen *et al.*, 2018).

PEST LIFE CYCLE

The life cycle of *B. tabaci* varies depending on the environmental conditions and the host. The development period of *B. tabaci* from egg to adult emergence ranges between 19 and 29 days (Table 11). The whitefly goes through four nymphal instars (Figure 61), before entering a pupal phase (Colvin *et al.*, 2006), with 11–12 generations per year (Asiimwe *et al.*, 2007a; b).

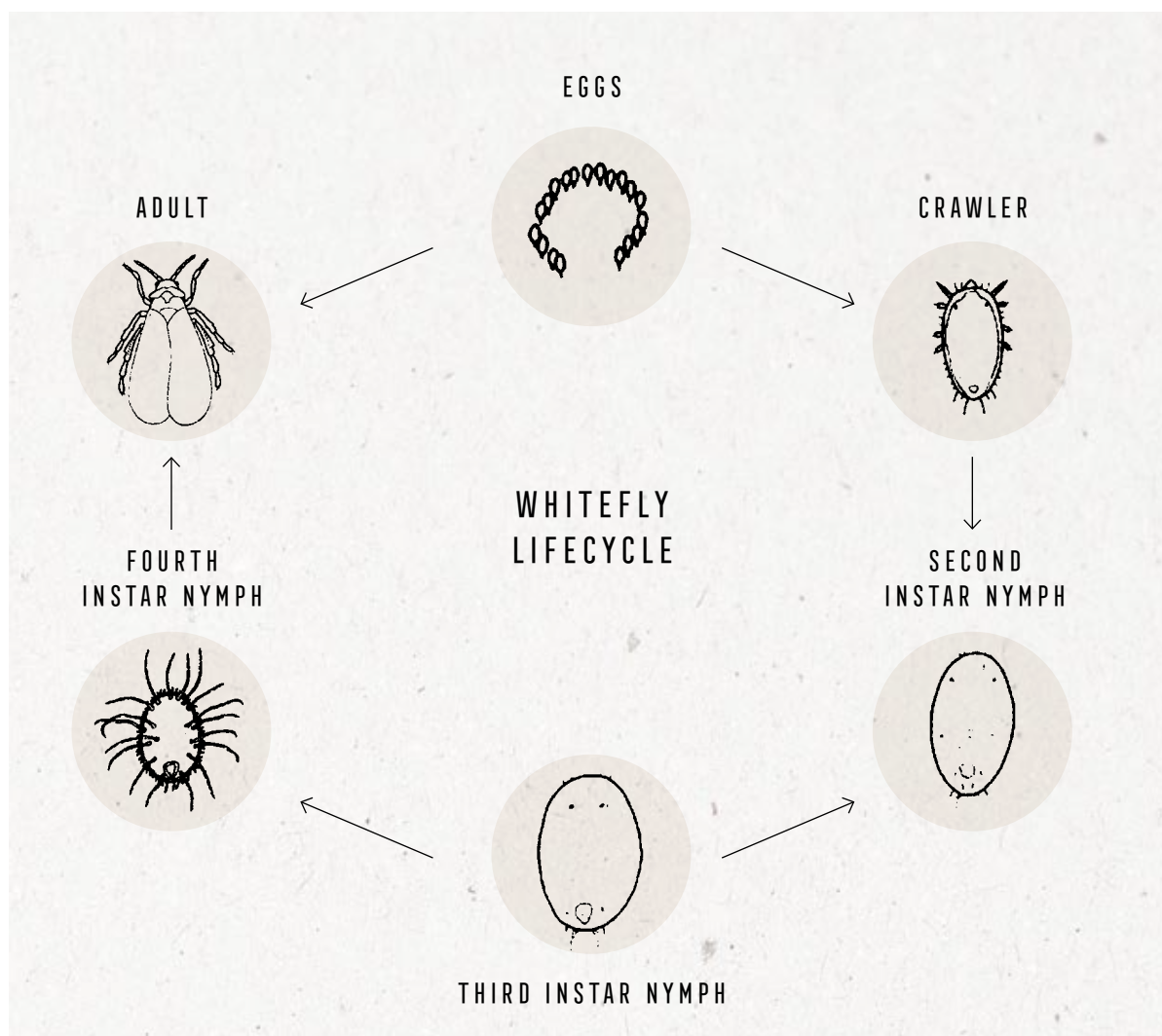


Figure 61 — Life cycle of whitefly showing the growth and development stages from eggs to adults
Source: <https://www.syngentaornamentals.co.uk/whitefly-life-cycle>

Table 11 — Description of whitefly developmental stages

WHITEFLY SPECIES	EGG	NYMPHAL INSTARS	PUPA (FOURTH NYMPHAL INSTAR)	ADULT
<i>Bemisia tabaci</i> and <i>Bemisia afer</i>	Females lay 70–400 eggs on the underside of cassava leaves, starting from around one day after emergence.	1 st nymphal instar (crawler) moves to a suitable position to feed.	The pupa is the last non-feeding stage of the 4 th instar.	Mated females live up to 41 days, but adult males live only for 23–29 days.
<i>Aleurodicus dispersus</i>	Females lay 14–26 eggs in a loose spiral on the underside of leaves.	The eggs hatch after 7–10 days. The 1 st and 2 nd larval instar stage lasts for 6–9 days, while the 3 rd instar lasts 5–13 days.	The 4 th instar (pupae) lasts 5–16 days.	Adult <i>A. dispersus</i> live for about 14 days.
<i>Bemisia tuberculata</i>	Each female lays an average of 6.3 eggs in 24 hours. The egg size is 163.22 µm in length and 72.39 µm in width.	Egg laying to fixed nymph stage takes 12–13 days	Fixed nymph stage and “pupae” last 14 days	It takes 26 days from egg laying stage until the adult stage.
<i>Aleurotrachelus socialis</i> Bondar	Female lays 20 to 119 eggs on average, with 3.4 to 19.7 eggs per day.	Not reported	Not reported	It takes 33.5 to 38 days to adulthood and may live 5 to 30 days depending on the cultivar.
<i>Trialeurodes variabilis</i> (Quaintance)	Not reported	Not reported	Not reported	Not reported
<i>Tetraleurodes acaciae</i>	Not reported	Not reported	Not reported	Not reported

Scouting for whiteflies on the underside of leaves should be carried out on cassava plants of 1 to 6 months old, in the mornings or evenings when it is cool and the whiteflies are less active than when it is hot (Sseruwagi *et al.*, 2003). Regular scouting is essential to detect whitefly and avoid economic damage. Crops must be inspected at weekly intervals to find infestations early.

It is also possible to monitor whitefly population levels and movement by trapping winged adults on yellow sticky cards (Idris *et al.*, 2012) placed randomly in and around the borders of a cassava field or by inspecting the leaves for the presence of adults and nymphal instars. To detect whiteflies on plants, randomly select 10 plants per field and thoroughly examine the underside of leaves for the presence of whitefly adults, nymphs and eggs. A 10x hand lens may be needed to see eggs and tiny nymphs.

Whitefly eggs are laid on new upper leaves of the plants, while the nymphs are usually found on the lower (older) leaves. A good population estimate of whiteflies can be made by sampling leaves from different parts of the plants. Also, look for plants (especially at the margins of fields) with sooty moulds and/or disease symptoms (from Cassava mosaic disease or Cassava brown streak disease). Scouting disturbs the plants, which makes some whiteflies take flight. If possible, count the adults that fly away and note where they settle (Sseruwagi *et al.*, 2004).

Whiteflies are difficult to control since they breed rapidly and produce multiple (11–12) generations in a year, develop rapidly, and are highly polyphagous with a wide range of host plants (Asiimwe *et al.*, 2007). However, some control has been achieved using selected control strategies, as described below.

CULTURAL CONTROL

Ensure crop hygiene at harvest by removing and burning cassava crop debris/residues infested with whitefly eggs or nymphal instars.

- Isolate new cassava crops from older fields infected with whiteflies or cassava viral diseases (CMD and CBSD).
- Intercropping (Legg *et al.*, 2003) cassava at high density with cow peas or maize can lower whitefly populations on cassava leaves (Fargette and Fauquet, 1990), with cowpeas being more effective than maize (Gold *et al.*, 1990; Njoku and Muoneke, 2008; Islami *et al.*, 2011). This is mainly due to changes in the microclimate and ecosystem (Fondong, 2002) or competition for nutrients, which alters cassava plant biochemistry to be less attractive to the whiteflies (Gold *et al.*, 1990). However, maize may reduce cassava yields (Olasantan *et al.*, 1996), while legumes such as cowpeas increased the yield (Njoku and Muoneke, 2008; Islami *et al.*, 2011).
- Plant architecture can influence whitefly populations, with the thin-leaved varieties having fewer whitefly eggs and nymphs than the wide-leaved varieties (Gold *et al.*, 1990; Njoku and Muoneke, 2008; Njoku, 2008).

- Roguing, or removal of cassava plants with viral disease infections (CMD and CBSD), in young crops (under 3 months old), followed by replanting healthy planting materials can help to control whitefly. Weed cassava crops to remove potential alternative host plant species for whitefly vectors and cassava-infecting viruses.
- The use of barrier crops such as sorghum, and maize can be used to control the entry of whiteflies into new cassava field. However, the barrier crops/plants should be planted at least 1-2 months before the cassava crops.
- Use other good agricultural practices, such as timely planting, method of planting, spacing, soil fertility (use of organic manure), weed and water management to ensure optimum plant growth and high yields.

BIOCONTROL

Biological control of whiteflies has been achieved with parasitoids, predators and entomopathogenic fungi.

Of these, the most widely used are the parasitoids, followed by the predators (ladybeetles) and fungi, which are slow acting. Figure 62 shows a parasitized nymph.



Figure 62 — Showing *Bemisia tuberculata* parasitized nymph

Source: https://www.researchgate.net/publication/234083705_Biology_of_Bemisia_tuberculata_Bondar_Aleyrodidae_and_parasitism_by_Encarsia_porteri_Mercet_1928_Hymenoptera_Aphelinidae_on_cassava_plants/figures

Biocontrol (the release and promotion of existing biological control agents or pathogens) can include:

- Natural enemies (Table 12): parasitoid species, such as the parasitic wasps *Encarsia sophia* and *Eretmocerus mundus*, can more effectively control whiteflies on cassava crops – with parasitism rates of 34–58% of 4th instar whitefly nymphs (Legg, 1995; Bellotti *et al.*, 2012b; Bellotti *et al.*, 2012c) – than whitefly predators, such as the beetle *Delphastus pusillus*.

Table 12 — A list of natural enemies and the whiteflies they parasitise

APPENDIX 2: NATURAL ENEMIES OF WHITEFLY ON CASSAVA (FROM BELLOTTI *ET AL.*, 2012B)

PRINCIPAL SPECIES	PARASITOIDS	PREDATORS	ENTOMOPATHOGENS (FUNGUS)
<i>Aleurotrachelus sociales</i>	<i>Amitus macgowni</i> <i>E. americana</i> <i>E. bellotti</i> <i>E. cubensis</i> <i>Encarsia hispida</i> <i>E. luteola</i> <i>E. sophia</i> <i>Encarsia sp. nr. variegata</i> <i>Encarsia sp.</i> <i>E. tabacivora</i> <i>Euderomphale sp.</i> <i>Eretmocerus spp.</i> <i>Metaphycus sp.</i> <i>Signiphora aleyrodis</i>	<i>Delphastus sp.</i> <i>D. quinculus</i> <i>D. pusillus</i> <i>Chrysopa sp. nr. cincta</i> <i>Condylostylus sp.</i>	<i>Beauveria bassiana</i> <i>lecanicillium lecani</i> <i>Aschersonia aleyrodes</i>
<i>Aleurothrixus aepim</i>	<i>Encarsia porteri</i> <i>E. aleurothrix</i> <i>E. hispida</i> <i>Eretmocerus sp.</i>		<i>Cladosporium sp.</i>
<i>Aleurodicus dispersus</i>	<i>Aleurotonus vittatus</i> <i>E. haitiensis</i> <i>Encarsia sp.</i> <i>Eretmocerus sp.</i> <i>Euderomphale sp.</i>		
<i>Aleuroglandulus similis</i>	<i>Encarsia guadeloupae</i> <i>Encarsia desantisi</i>	<i>Nephaspis namolica</i>	
<i>Aonidomytilus albus</i>	<i>Aspidophagus citrinus</i> <i>Signiphora sp.</i>	<i>Chilocorus distigma</i>	<i>Septobasidium sp.</i>

PRINCIPAL SPECIES	PARASITOIDS	PREDATORS	ENTOMOPATHOGENS (FUNGUS)
<i>Bemisia tuberculata</i>	<i>E. hispida</i> <i>E. pergandiella</i> <i>E. sophia</i> <i>Encarsia</i> sp. prob. <i>variegata</i> <i>E. tabacivora</i> <i>Eretmocerus</i> sp. <i>Euderomphale</i> sp. <i>Metaphycus</i> sp.	<i>Condyllostylus</i> sp.	
<i>Bemisia tabaci</i>	<i>Encarsia sophia</i> <i>E. lutea</i> <i>E. Formosa</i> <i>E. mineoi</i> <i>Encarsia</i> sp. <i>Eretmocerus mundus</i>	<i>Delphastus pusillus</i> <i>Condyllostylus</i> sp.	
<i>Trialeurodes variabilis</i>	<i>E. bellotti</i> <i>E. hispida</i> <i>E. luteola</i> <i>E. nigricephala</i> <i>E. pergandiella</i> <i>Encarsia</i> sp. <i>E. sophia</i> <i>E. strenua</i> <i>E. tabacivora</i> <i>Eretmocerus</i> spp.	<i>Chrysopa</i> sp. nr. <i>cincta</i> <i>Condyllostylus</i> sp.	<i>Aschersonia aleyrodes</i> <i>Beauveria bassiana</i> <i>Lecanicillium lecani</i>
<i>Trialeurodes vaporariorum</i>	<i>Encarsia tabacivora</i>		

Source: Bellotti *et al.* (2012c)

- Fungi such as *Verticillium lecanii*, *Paecilomyces fumosoroseus* and *Beauveria bassiana* can suppress whitefly (Faria and Wraight, 2001), especially when small numbers of whitefly are present (Bellotti *et al.*, 2012c). Others include *Aschersonia* and *Metarhizium*, which are infectious to whitefly (Horowitz *et al.*, 2011). *Beauveria bassiana* is a fungus used widely in South Africa and Zambia to control pests on beans, tomatoes, cucumbers and eggplant. However, fungal whitefly control is slow acting and relatively ineffective in killing adult whitefly. Fungal control can also be expensive, have negative interactions with other commonly used fungicides and a limited shelf life, and is dependent on good weather (Faria and Wraight, 2001), which may limit its application by small-scale cassava farmers. Some fungi also infect whitefly predators and parasites, interfering with other control options.

CONTROL WITH PLANT PROTECTION PRODUCTS

Plant protection product options for whitefly include:

- Organic neem or Azadirachtin can be sprayed on cassava leaf surfaces (including the undersides of leaves) until completely wet to kill eggs, nymphs and/or repel adults of whiteflies.
- Cassava stem cuttings may be dipped in Deltamethrin or Cypermethrin to kill whitefly eggs, nymphs and adults before distribution to farmers.
- More targeted insecticides and insect growth regulators (IGRs), including soil treatments, second-generation nicotinoids and non-neurotoxic IGRs such as buprofezin and pyriproxyfen (Palumbo *et al.*, 2001) target specific pests with increased effectiveness at lower application rates, and are non-persistent in the environment (Casida and Quistad, 1998). However, the IGRs have had limited application on cassava (Horowitz *et al.*, 2011).

Note: Insecticide resistance management strategies based on the structured and restricted use of non-neurotoxic IGRs, coupled with the use of cultural and biological pest management tactics (Carrière *et al.*, 2012), are currently thought to be the best way to combat insecticide resistance in *B. tabaci* (Ellsworth *et al.*, 2001).

OTHER CONTROL METHODS

- Use of elite cassava varieties with proven resistance to or tolerance of whiteflies (Gwandu *et al.*, 2019).
- Establish repellent crops such as marigold near or around cassava fields to repel whiteflies.
- Spraying insecticidal soaps under leaf surface kills the whiteflies.
- Use sticky traps with yellow and blue colours (Idris *et al.*, 2012) to trap whiteflies.

7.3.2. CASSAVA MEALYBUG

SCIENTIFIC NAME

— Cassava mealybug – *Phenacoccus manihoti*



Figure 63 — Showing (A) adult cassava mealybugs and (B & C) damage caused on cassava plants
Sources: CABI Plantwise Knowledge Bank, <https://www.plantwise.org/KnowledgeBank/pmdg/20177800433>

DESCRIPTION/IDENTIFICATION

PEST

Cassava mealybugs, *Phenacoccus manihoti* (Hemiptera: Pseudococcidae), are tiny, pink, wingless insects. Adult mealybugs are pink with white wax secretions (Figure 63A) and are 1.10–2.6 mm long and 0.5–1.40 mm wide. They have soft bodies covered by white wax. Mealybugs do not fly but colonize stems and leaf nodes and may only move slightly.

Female mealybugs can be produced from unfertilised eggs. Adult females lay eggs in an ovisac (Cox and Williams, 1981; Williams and Granara de Willink, 1992). The eggs are oblong, golden yellow and enclosed in woolly ovisacs located at the rear end of the adult females. They measure 0.30–0.75 mm long and 0.15–0.30 mm broad (Matile-Ferrero, 1978; Nwanze, 1978).

Larvae have antennae with six segments in the 1st instar and nine segments in subsequent instars. Body length and breadth are 0.40–0.75 mm and 0.20–0.30 mm for 1st instars/crawlers; 1.00–1.10 mm and 0.50–0.65 mm for 2nd instars; 1.10–1.50 mm and 0.50–0.60 mm for 3rd instars; and 1.10–2.6 mm and 0.50–1.40 mm for 4th instars/newly emerged adults (Matile-Ferrero, 1978; Nwanze, 1978).

Adult females are ovoid, rose pink and dusted with white, powdery wax; they have prominent eyes and well-developed legs (Matile-Ferrero, 1978). Body segments have very short filaments on the sides and near the tail end, which look like teeth on the body (Williams and Granara de Willink, 1992).

BIOLOGY AND ECOLOGY

Mealybugs are found around major leaf veins on the underside of cassava leaves and at low to high densities inside growing tips. When the population increases, the adults spread over the plant, while remaining underneath the leaves and on the shoot tips (Herren and Neuenschwander, 1991).

Mealybugs are most abundant in the shoot tips at low population densities (Schulthess *et al.*, 1987; Neuenschwander and Hammond, 1988). Ants that feed on the honeydew protect mealybugs from natural enemies that would otherwise attack them, which helps populations to increase (Cudjoe *et al.*, 1993).

Mealybugs reproduce asexually by laying undeveloped, unfertilised eggs and have nine reproduction generations, with the largest occurring during the dry season. However, numbers decline at the start of the rainy season, when many mealybugs are physically washed off the plant. The most favoured sites for egg laying are shoot tips, lower leaf surfaces and leaf stalks.

SYMPTOMS

On cassava and other plants of the *Euphorbiaceae* family, *P. manihoti* causes stunting, leaf distortion and loss, dieback and weakening of stems. However, the pest does not cause any significant damage to other host plants where it overwinters.

DISPERSAL

The 1st instar stage of the mealybugs, the crawler, moves to upper leaf surfaces and shoots, from where they naturally get blown away by wind. However, the main mode of dispersal is humans moving infested cassava stem planting material over long distances. Other modes of dispersal include vector transmission by grazing animals, workers attending to the crop carrying the pest on their clothes, unburnt pruned or harvested crop debris from infested plants, and farm tools and vehicles used by agricultural officers and traders.

DAMAGE

Cassava mealybugs feed by sucking sap from cassava shoots. They cause direct damage by contaminating the plant with their toxic saliva, leading to leaf loss, stunting (Figure 63B) and drying (Figure 63C) of cassava plants. Indirectly, the saliva encourages the development of sooty moulds that interfere with photosynthesis (Calatayud and Le Rü, 1997).

IMPACT ON YIELD AND QUALITY

Yield losses of up to 84% due to mealybugs have been reported (Nwanze, 1982). Leaf loss reduces the availability of cassava leaves as a food source. Heavy pest infestation reduces stem quality (for use in subsequent plantings) and tuber root quality (size), causing famine and food insecurity (Herren and Neuenschwander, 1991).

GEOGRAPHICAL DISTRIBUTION

P. manihoti is indigenous to South America. It was accidentally introduced to the Republic of the Congo in 1973 (Herren and Neuenschwander, 1991) and has spread throughout Africa to almost all countries where cassava is grown, in a broad belt from West - through to East Africa and down to the eastern edge of South Africa (Table 13).

Table 13 — Geographical distribution of mealybugs affecting cassava in the ACP countries

AFRICA	CARRIBEAN AND SOUTH AMERICA	PACIFIC
Angola, Benin, Cameroon, Central African Republic, Democratic Republic of the Congo, Republic of the Congo, Côte d'Ivoire, Equatorial Guinea, Gabon, Gambia, Ghana, Guinea, Guinea-Bissau, Kenya, Liberia, Madagascar, Malawi, Mali, Mozambique, Niger, Nigeria, Rwanda, Senegal, Sierra Leone, South Africa, Sudan, Tanzania, Zanzibar Island, Togo, Uganda, Zambia, Zimbabwe	Argentina, Bolivia, Brazil (Amazonas, Mato Grosso do Sul, Pará, Paraná, Rio Grande do Sul), Colombia, French Guiana, Guyana, Paraguay	Not reported

QUARANTINE ORGANISM

Accidental introduction of *P. manihoti* to new territories, through the transportation of infested cassava materials such as leaves and stems by shipping, air transport and/or mail, poses a significant threat to other agricultural crops. It is important for plant quarantine services to regulate trade and/or impose strict inspection and certification of cassava materials from Africa, South America and other areas where the pest is prevalent. A phytosanitary certificate should guarantee absence of the pest from consignments of cassava leaves and planting material.

FAVOURABLE CONDITIONS

P. manihoti thrives on cassava and other plants of the *Euphorbiaceae* family in the tropical and subtropical regions of the world (Williams and Granara de Willink, 1992). Other host plant species and crops are only marginally infested (Herren and Neuenschwander, 1991). Cassava mealybug populations increase during the dry

season, when they are more abundant and cause the most damage. Populations decline at the onset of the rainy season, when many mealybugs are washed from the plants. Optimum temperatures for mealybug development range from 14.7°C to 28°C, with no development occurring above 35°C (Herren and Neuenschwander, 1991).

PEST LIFE CYCLE

The life cycle of *P. manihoti* consists of an egg and four instar stages (Nwanze *et al.*, 1979; Iheagwam, 1981; Lema and Herren, 1985; Rü and Fabres, 1987; Schulthess *et al.*, 1987). Adult female mealybugs lay up to 500 eggs in an ovisac and produce only females (thelytokous parthenogenesis). Egg incubation lasts approximately 8 days and the insect usually dies 1–3 days after ceasing egg production (Nwanze, 1978).

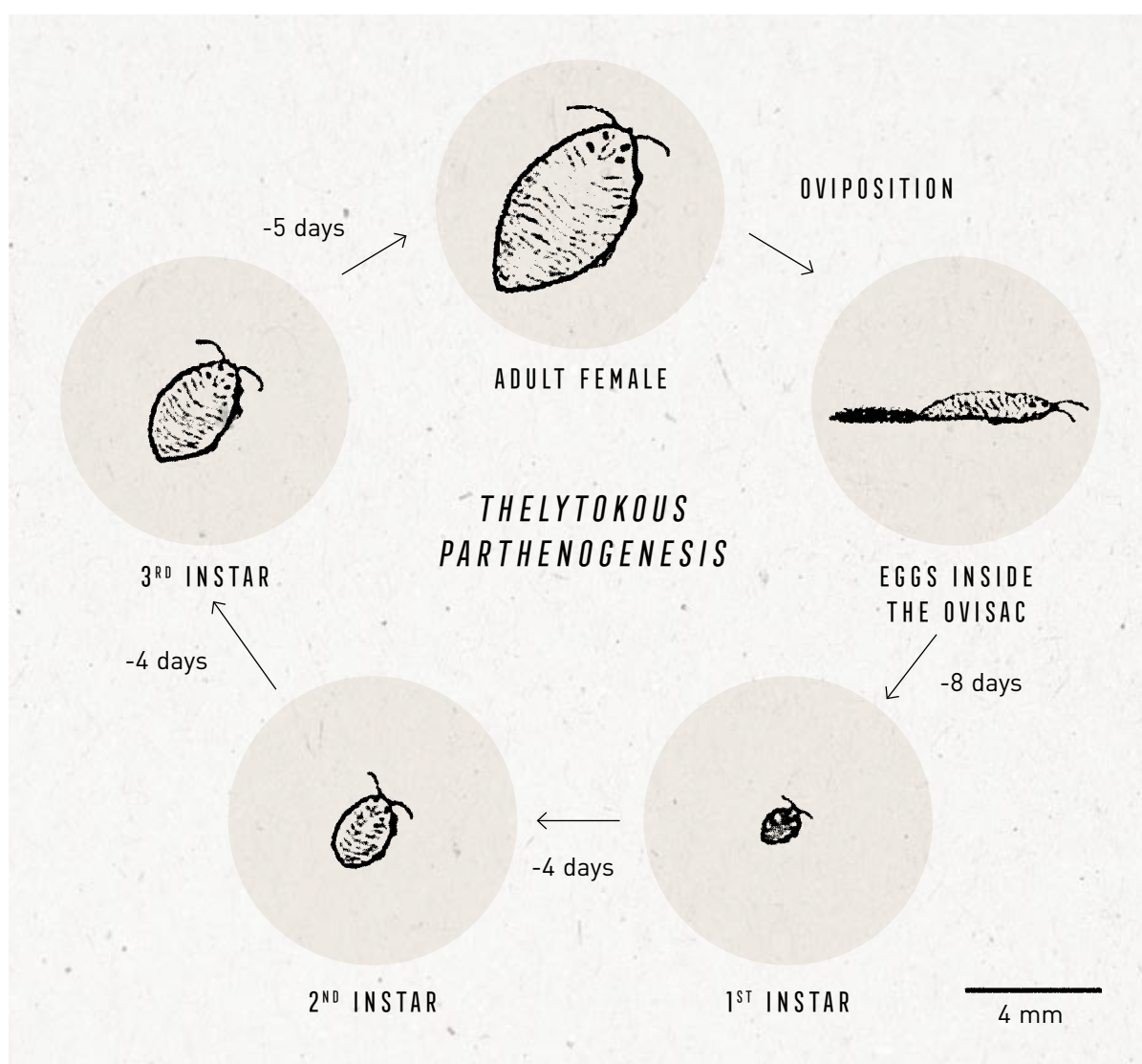


Figure 64 — Showing developmental stages of *P. manihoti* according to Nwanze (1977)
 Source: Catalyud and Le Ru (2006), Cassava-Mealybug Interactions,
<https://books.openedition.org/irdeditions/9875>

The eggs hatch into crawlers (1st instars), which are mobile (Nwanze, 1977) and are responsible for plant colonisation. The insect moults three times in its development (Figure 64) to the 4th instar (adult). The average development period from egg to adult is around 20-33 days (Fabres, 1980; Fabres and Boussiengue, 1981; Bellotti *et al.*, 1984).

SURVEILLANCE/MONITORING

Examine cassava fields for white immobile adult mealybugs on shoots, stems and leaves at least once per week. Also look out for the presence of ants, which are known to protect the mealybugs and feed on the honeydew they produce, and black sooty mould. Use control measures when at least 5–10 mealybugs are detected on 4–10 plants in 25% of an acre.

CULTURAL CONTROL

Use only healthy or pest-free planting materials.

- Avoid movement of plants or cuttings with mealybugs.
- If cassava has been produced continuously for more than 3 years on the same piece of land, rotate the fields to grow maize and legumes.
- Plant at the start of the rainy season, so that plants can become established and resist mealybug attack.
- Dip cassava planting materials in chemical pesticides before planting to inhibit the spread of mealybugs.
- Remove or prune infested plants or plant parts (leaves, shoots) and burn or bury to kill the mealybugs.
- Practice clean weeding of cassava crops to remove potential alternative host plant species for the mealybugs within the field and in the surrounding fields and bushes.
- Apply manure or other fertilisers to reduce the mealybug population – the nutrition helps to increase the parasitoid wasp population (Schulthess *et al.*, 1987).
- Mulching and use of manure enhanced the rutin (an unfavourable substance for insects) content in cassava plants with subsequent increased resistance to mealybug infestation (Tertuliano *et al.*, 1999).

BIOCONTROL

- *Apoanagyrus lopezi*, an exotic parasitic wasp, successfully and economically controlled cassava mealybug (Neuenschwander and Hammond, 1988; Hammond and Neuenschwander, 1990; Bellotti *et al.*, 2012b; 2012c).
- In Africa, mealybugs are attacked by predators such as the ladybird beetles (e.g. *Hyperaspis* spp., *Exochomus* spp. and *Diomus* spp.) (Fabres and Kiyindou, 1985; Boussienguet, 1986; Neuenschwander *et al.*, 1987; Neuenschwander and

Hammond, 1988; Stäubli Dreyer *et al.*, 1997a; 1997b), by parasitoids of mealybugs (Bartlett, 1978; Moore, 1988; Ben-Dov and German, 2003) and by fungi such as the *Neozygites fumosa*, which causes disease in the pest populations under warm and very humid conditions (Bartowski *et al.*, 1988).

- In Central Africa, the hyperparasitoid wasp *Prochiloneurus aegyptiacus* is usually most common (Fabres and Matile-Ferrero, 1980; Boussienguet, 1986; Neuenschwander *et al.*, 1987; Biassangama *et al.*, 1989).
- However, ants attending mealybugs for their honeydew tend to defend the pests from natural enemies, which interferes with biological control strategies in some countries (Cudjoe *et al.*, 1993).

CONTROL WITH PLANT PROTECTION PRODUCTS

- Organic Neem or azadrachtin can be applied on cassava leaf surfaces and undersides until completely wet to kill eggs and nymphs and to kill or repel adults of mealybugs.
- Immerse cassava cuttings in manipueira (a liquid extract from cassava roots) for 60 minutes to reduce infestation (Razafindrakoto *et al.*, 1999).
- Treat cassava leaves weekly with a 1% neem kernel water extract, which makes them less attractive to colonisation by the first instars.

OTHER CONTROL METHODS

- The use of cassava varieties with resistance to mealybugs helped to control the pest (Calatayud and Le Rü, 1997).
- Spray cassava plants infested with mealybugs with fermented, diluted cow urine, which irritates the soft body of the insect making them to move off the plants. However, this can only work in small plots with low infestation.
- The mealybugs can be washed off by hand or using a strong spray of water with a hose pipe on the affected plants during early infestation when the numbers are still low.
- Mealybugs are adapted to secondary compounds such as cyanide in cassava, which induces their sustained feeding, but their growth rate can be reduced by the presence of a flavonoid known as rutin (Calatayud, 2000), depending on cultivar, location and season.

7.3.3. MITES

SCIENTIFIC NAME

- Cassava green mite (*Mononychellus tanajoa*/*Mononychellus progresivus*)
- Red spider mite (*Oligonychus gossypii*)



Figure 65 — Showing (A) Cassava green and red mites; (B) typical symptoms of infestation; and (C) severe damage

Sources: Pacific Pests & Pathogens, https://apps.lucidcentral.org/ppp/text/web_full/entities/cassava_green_mite_438.htm; CABI Plantwise Knowledge Bank, <https://www.plantwise.org/KnowledgeBank/pmdg/20137804344>; Pheneas Ntawuruhunga/ResearchGate, https://www.researchgate.net/figure/Typical-symptoms-of-Cassava-Green-Mite-attack_fig15_325262375. See also predatory red mites at [infonet-biovision.org, http://69.63.68.22/infonet/export/print\\$ct\\$74\\$pests.html](http://69.63.68.22/infonet/export/printct74$pests.html)

DESCRIPTION/IDENTIFICATION

PEST

Cassava green mite (CGM; *Mononychellus tanajoa*) originated in the Neotropics (Central and South America, including the tropical southern part of Mexico and the Caribbean) and was accidentally introduced to Africa in 1971 (Nyiira, 1972). *M. tanajoa*, like other members of the Acari, are inconspicuous or have segmented bodies with the appearance of a single body unit. The mites do not have wings, compound eyes or antennae. Adult females are bigger than males, at around 0.8 mm.

BIOLOGY AND ECOLOGY

Cassava green mites are found on the underside of cassava leaves (Figure 65A). The developmental time, number of offspring produced and adult lifespan depend on temperature, humidity, the host plant and the sex of the mite (Byrne *et al.*, 1982a; Mesa *et al.*, 1987; Yaninek *et al.*, 1989a, 1989b). The optimum temperature is 27°C at a relative humidity of 70% and a photoperiod of 12 hours light and 12 hours dark. The developmental period for egg, larva, protonymph and deutonymph on cassava leaves was 5.4, 3.0, 1.1 and 2.8 days, respectively.

In sub-Saharan Africa, adult female mites live for up to 11.6 days and lay an average of 62.8 eggs over a period of 9.8 days, with a maximum net reproduction rate of 43.2 progeny. The egg to adult developmental periods were reported to be 21.3, 15.5, 12.3, 7.7 and 6.9 days at 20, 24, 27, 31 and 34°C, respectively (Yaninek and Gnanvossou, 1993). The presence of fresh foliage on cassava plants and rainfall are the most important factors in determining the population dynamics of *M. tanajoa* in the field (Yaninek *et al.*, 1989a, 1989b). The population increases to 20–200 mites/leaf on new cassava leaf growth during the early dry season but declines with defoliation of leaves during drought. Prolonged rainfall also reduces the CGM population by washing the mites off the leaves.

SYMPTOMS

Cassava green mites feed by inserting their piercing and sucking mouthparts (chelicerae) into individual cassava leaf cells and extracting the contents. This causes chlorosis (removal of chlorophyll), mottling, leaf fall and death of the affected leaves (Figure 65B). Severe infestation produces what is referred to as the 'candle stick' symptom (Figure 65C), with extreme leaf loss. Other symptoms include yellow striping of leaves; yellow or pale spots on leaves; yellowing of entire leaves; deformation and stunting of plants. On the stunted plants, damage is caused to stems, resulting in low quality planting material. In contrast, red spider mites cause the leaves to have a spotted appearance, with a yellowing, bronzed colour and webbing covering the leaves. The mites appear as tiny moving dots on the webs or underside of leaves when viewed under a hand lens.

DAMAGE

Green mites feed on the underside of young leaves, beginning along the veins and at the base. Damage is visible to the naked eye, but the eggs, crawlers and adults are usually only visible under a light microscope.

Scoring for CGM symptoms uses an arbitrary scale of 1–5, where: 1 = no obvious symptoms; 2 = moderate damage, no reduction in leaf size, scattered yellow spots on young leaves, 1–2 spots per cm²; 3 = severe spotting, some reduction in leaf size, stunted shoot, 5–10 spots per cm²; 4 = severe chlorotic symptoms and leaf size of young leaves severely reduced; and 5 = tips of affected plants defoliated, resulting in a candle stick appearance of shoot tips. A score of 1 indicates symptomless (healthy) leaves, and 5 severe damage (Chalwe *et al.*, 2015).

DISPERSAL

Green mites can disperse actively from plant to plant, or passively by wind and water (Bellotti, 1985; Moraes *et al.*, 1995). CGM were reported to disperse 200 m by wind in 13 weeks (Yaninek *et al.*, 1989b). The pest is mainly spread by human activity, through transportation of infested plant materials (leaves and stems) between different locations. The mites can survive on cassava leaves removed from the cassava plants for up to 5 days and can also persist on cassava cuttings for up to 60 days.

IMPACT ON YIELD AND QUALITY

Damage caused by *M. tanajoa* varies according to the cassava cultivar and the length of the dry season. Prolonged drought encourages the build-up of mites and increases yield losses of both the leaves and tubers to 10–80% (Shukla, 1976; Markham and Robertson, 1987; Pallangyo *et al.*, 2007). CGM can cause up to 50% reduction in leaf weight, and up to 80% tuber yield loss in cassava (Shukla, 1976; Gutierrez *et al.*, 1988; Pallangyo *et al.*, 2004). The dry matter of storage roots of infected plants can be reduced by 29% and the stems by 21% (Skovgard *et al.*, 1993). The loss of tuber and leaf yields can lead to household and income insecurity, and eventual malnutrition and increased unemployment.

GEOGRAPHICAL DISTRIBUTION

Cassava green mites are widely distributed in most cassava growing areas in Africa (Hicks, 2017), South and Central America (Bolland *et al.*, 1998) and the Caribbean (Table 14). However, a report by the National Plant Protection Organization of 2014 and 2016 (EPPO, 2014; 2016) confirmed their absence from Australia, Asian cassava-producing countries (Bellotti *et al.*, 2012c) and China (Parsa *et al.*, 2015).

Table 14 — Geographical distribution of mites affecting cassava in the ACP countries

AFRICA	CARIBBEAN AND SOUTH AMERICA	PACIFIC
Angola, Benin, Cameroon, Central African Republic, Republic of the Congo, Democratic Republic of the Congo, Ivory Coast, Equatorial Guinea, Gabon, Gambia, Ghana, Guinea, Guinea-Bissau, Kenya, Liberia, Madagascar, Malawi, Mali, Mozambique, Niger, Nigeria, Rwanda, Senegal, Sierra Leone, South Africa, Sudan, Tanzania, Zanzibar Island, Togo, Uganda, Zambia, Zimbabwe	Antigua, Argentina, Barbuda, Bolivia, Brazil, Chile, Colombia, French Guiana, Guyana, Northern Mariana Islands and Micronesia, Paraguay, Peru, Suriname, Uruguay	Not reported

QUARANTINE ORGANISM

M. tanajoa is considered a quarantine pest (Delalibera *et al.*, 1992) and calls for strict regulatory control. The pest's risk and impact factors include:

- Invasiveness (and the ability to survive outside its native range)
- Capable of securing and ingesting a wide range of food (a habitat generalist)
- Highly mobile locally
- Rapid growth and reproductive potential.

The pest CGM impacts the host plants, causing retarded growth and yield, which negatively impacts agriculture, human health, people's livelihoods and interaction with other invasive species in the environment. CGM are likely to enter other regions (including internationally) by accident, as they are hard to identify/detect, both in the field and on materials, and difficult and/or expensive to control. Strict inspection of cassava cutting materials and the use of clean certified cuttings are key to reducing the spread of CGM and delaying infestation of cassava crops and other hosts of economic importance.

FAVOURABLE CONDITIONS

Severe damage mostly occurs during the dry season, when water-stressed cassava plants are more susceptible to attack (Bolland *et al.*, 1998). Spider mites thrive in dusty conditions. Growth of new foliage on cassava plants following a rainy season determines the population dynamics of *M. tanajoa* in the field (Yaninek *et al.*, 1989a, 1989b). The population of *M. tanajoa* increases to 20–200 mites/leaf on new leaf growth early in the dry season, but declines when loss of leaves begins in the dry season. However, prolonged rainfall leads to a decrease in mite population (<1 mite/leaf) due to physical wash off from the plant. *M. tanajoa* has a wide host range (Moraes *et al.*, 1995), which means that it can thrive in the surrounds of cassava fields and is very difficult to manage because the pest overwinters in other plants that are not usually targeted by control measures.

PEST LIFE CYCLE

M. tanajoa has four life stages, including a larva with six legs, two nymphal instars and the adult (Figure 66), which takes at least 12 days at 27°C to emerge. Adult mites are green at first and later become yellowish green. The adult female lives for 12 days and lays about 60 eggs (Helle and Pijnacker, 1985).

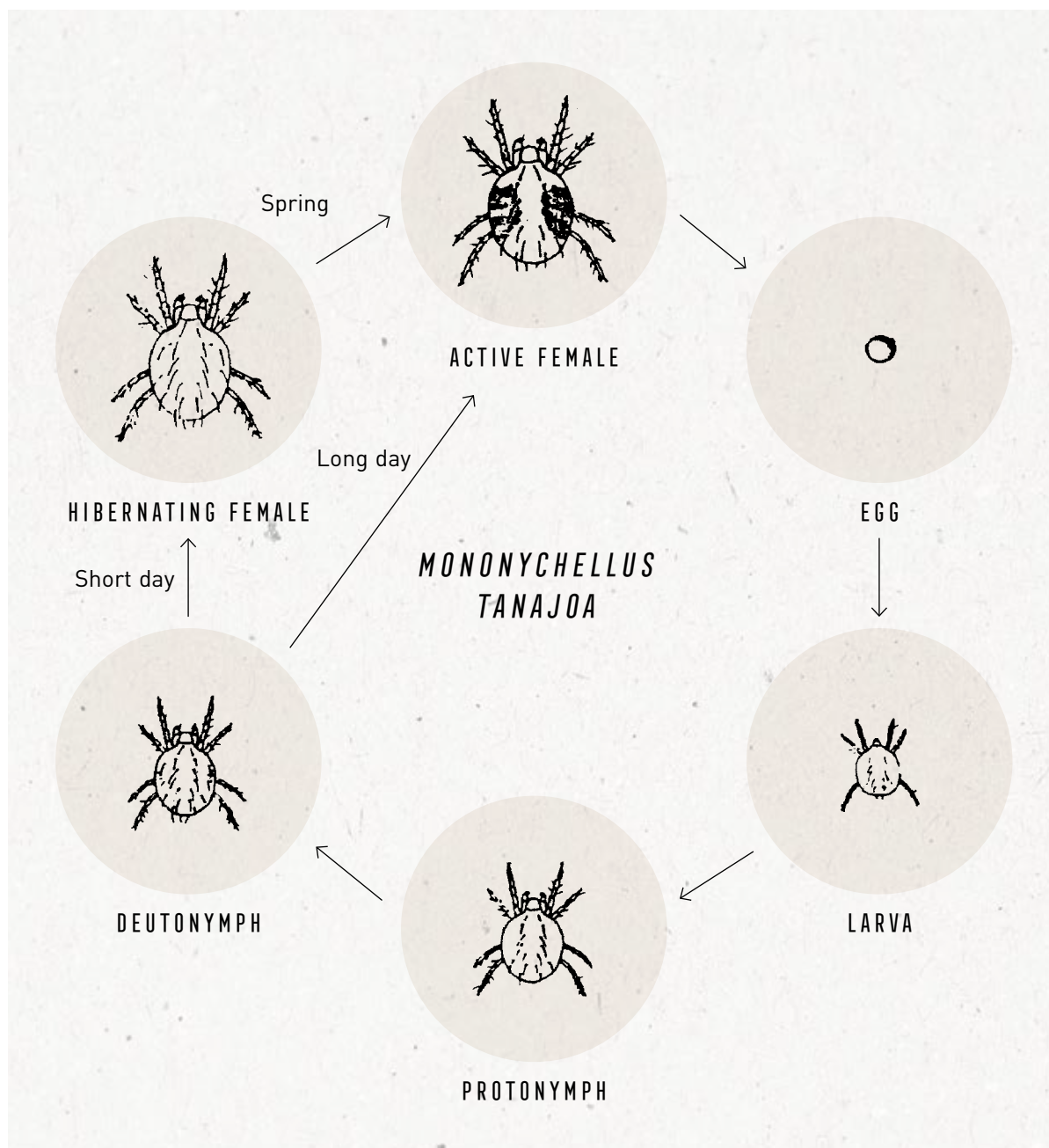


Figure 66 — Life cycle of *Mononychellus tanajoa* [Bondar, 1938] on cassava
Source: <https://www.semanticscholar.org/paper/Effect-of-Neozygites-floridana-infection-in-urticae-Bhattarai/bd5f779f627fec20a2fc4286204f884e3ff2e620>

SURVEILLANCE/MONITORING

Cassava plants should be examined for green mite infestation by checking for crawlers or adults on the shoot tips and the underside of leaves using hand-held magnifying lenses (x10) at least once per week. In addition, damage due to CGM is very clear to the naked eye and includes leaf yellowing and mottling, leaf fall and candlestick shaped plant shoots. The damage is scored using a 1–5 scale, where 1 is healthy and 5 indicates severe stunting and leaf fall. Fresh sprouts may also harbour the CGM and must be examined to prevent spread.

CULTURAL CONTROL

Isolate new cassava crops from older fields that may be infested with CGM.

- Plant cassava cuttings using the horizontal/burying method to kill CGM on the stems. The vertical or inclined/slanting/tilted planting method can promote early infestation upon sprouting for mites on the unburied parts of the stem cuttings (Ezulike and Egwuatu, 1990).
- Plant cassava crops at the beginning of the rainy season for vigorous plant growth and better tolerance of mite infestation.
- Intercrop with cowpeas, which can preferentially attract mites and reduce damage to cassava plants (Ezulike and Egwuatu, 1993).
- Spraying plants with a strong jet of water can help to reduce build-up of mite populations.
- Apply insecticidal soap to plants.
- Clean weed cassava crops to remove potential alternative host plant species for CGM within and around the fields.
- Provide sufficient water for irrigation to cassava crops to avoid stress, which encourages CGM infestation and physical damage.

BIOCONTROL

This involves releasing and promoting existing biological control agents/pathogens (including natural enemies) to control the mites.

- The predatory mite *Typhlodromalus aripo* has successfully controlled CGM in many regions of Africa (Bellotti *et al.*, 2012b; 2012c) with mortalities of 64.3–70% (Pallangyo *et al.*, 2004).
- Insect predators, including various beetles, lacewings and hoverflies (members of the families Coccinellidae, Staphylinidae, Lygaeidae, Chrysopidae, Syrphidae, Anthocoridae and Cecidomyiidae; Farias *et al.*, 1981; Byrne *et al.*, 1983) and thrips (Order Thysanoptera). The beetles *Stethorus* spp. and *Holobus* spp. have been used to control *M. tanajoa* in many countries.

- Mites of the family Phytoseiidae are the most important predators of *M. tanajoa* because they have a high capacity for predation, even at low prey densities, and are easy to rear. Bellotti *et al.* (1987) listed 46 phytoseiid species found in association with *M. tanajoa* on cassava in Colombia.
- The predator mite *Amblyseius manihoti* controlled 20% of *M. tanajoa* when released in an infested field at the beginning of the dry season (Toko *et al.*, 1996).
- A fungus (*Neozygites* spp.) has been used to control *M. tanajoa* in Venezuela (Agudelo-Silva, 1986), Brazil (Delalibera *et al.*, 1992), Colombia (Alvarez *et al.*, 1993), Benin (Yaninek *et al.*, 1996) and Kenya (Bartowski *et al.*, 1988).

CONTROL WITH PLANT PROTECTION PRODUCTS

- Organic Neem or azadirachtin can be sprayed on cassava leaf surfaces (including the undersides of leaves) until completely wet to kill eggs and nymphs and/or repel adult mite infestation.
- Cassava stem cuttings may be dipped in deltamethrin or cypermethrin to kill whitefly eggs, nymphs and adults before distribution to farmers.

OTHER CONTROL METHODS

- Plant tolerant cassava varieties where possible; plant at the beginning of the rainy season to encourage vigorous growth, which allows the plant to tolerate attack.
- Certain attributes, such as small hairs on the leaf surface (leaf pubescence; Leuschner, 1982), may improve tolerance of *M. tanajoa*. Mite-resistant cassava cultivars may be possible (Shukla, 1976; Santos *et al.*, 1977; Byrne *et al.*, 1982a & b; Markham and Robertson, 1987; Msabaha, 1984) but further studies including damage symptoms, tuber yields, and disease resistance of the selected cultivars need to be carried out. Immunity to *M. tanajoa* is not known.

7.3.4. SCALES

SCIENTIFIC NAME

- Cassava scale (*Aonidomytilus albus*)
- Black scale (*Parasaissetia nigra*)
- Black scale (*Saissetia miranda*)
- African root and tuber scale (*Strictococcus vayssierei*)

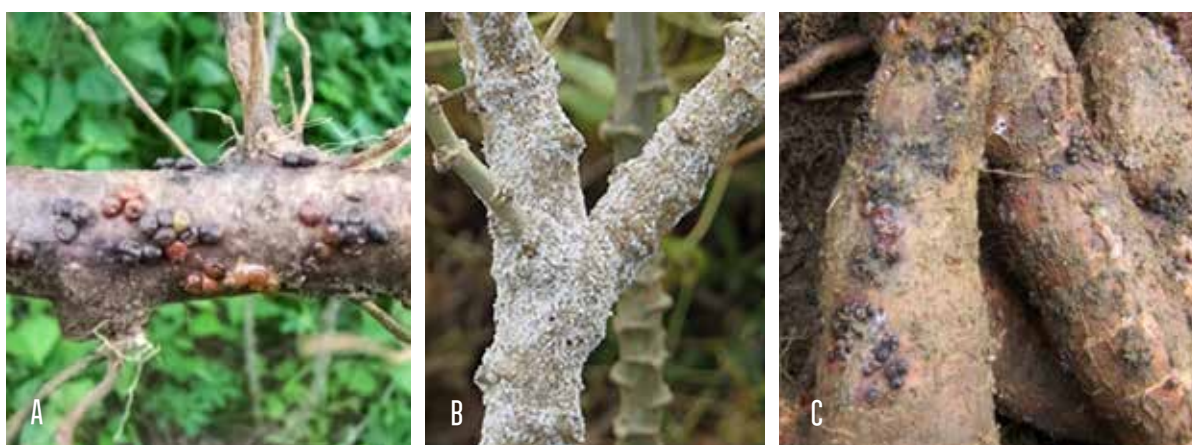


Figure 67 — Showing (A) black & red and (B) white peach scales on cassava stems and (C) tubers.
Source: International Institute of Tropical Agriculture, <https://www.flickr.com/photos/iita-media-library/4603107741>; <https://www.pestnet.org/white-peach-scale-on-cassava-within-tonga/>; and PlantVillage, <https://plantvillage.psu.edu/topics/cassava-manioc/infos>

DESCRIPTION/IDENTIFICATION

PEST

Adult male and female scales are black and red in colour, bag-shaped (Figure 67A) and do not have wings or legs. The males are usually shorter than the females. The white peach scales secrete a silvery-white waxy covering (Figure 67B), while the black and red scales also called armored scales (Figure 67C). Adults are approximately 2–3 mm in length with a mussel-shell shape. Scales have two golden-brown oval shaped patches on their upper surface, which are remains of the 1st and 2nd nymphal skins that are part of the adult scale. The underside has a soft cottonlike secretion that helps the scales to attach to the stems of plants.

The eggs are usually laid in wax-like threads secreted underneath the body, where they are protected, and hatch into cream-white 1st instar nymphs (Ngeve, 2003). The population of male and female scales is almost the same in a population (Swaine, 1950). The cassava root scale (*Stictococcus vayssierei*, or African root and tube scale) lives underground on the storage roots, feeder roots and submerged stems of cassava. These insects are reddish-purple or brown in colour, oval in shape and resemble ticks. Like the other scales, the adults lack wings and are attached firmly to the plant.

SYMPTOMS

Infected leaves lose their green colour and dry up, while a white waxy substance is present on stem surfaces and plants wilt or have stunted growth and poor yields under heavy infestation. Cassava cuttings from infected plants usually do not sprout. On cassava roots, there is tip dieback leading to eventual death of cassava plants (Ngeve, 2003; Williams *et al.*, 2010).

DISPERSION

The nymphs are crawlers and can move 18 inches per hour to disperse in the field. They can also spread by aid of wind and infested cassava cuttings used for planting. Improper disposal of cassava peels is also a likely means of spread of the pest. Ants (*Anoplolepis tenella Santschi*) were reported to be the key agents of dispersal of *S. vayssierei* in southern Cameroon (Dejean & Matile-Ferrero, 1996).

DAMAGE

The scales establish on the stems and leaf stalks of cassava plants. They lack mouth parts but cause damage by sucking when plants are heavily infested. When infestation occurs early in the plant's growth, the stem can be completely desiccated, leading to death of the plant. However, when infestation occurs later, the damage is mild (Swaine, 1950). Scales can cause deformation of tubers and reduce tuber root size and weight.

IMPACT ON YIELD AND QUALITY

During heavy infestation, yield loss of up to 100% may occur and the tuber quality is highly compromised and unpalatable (Swaine, 1950). *S. vayssierei* were reported to cause yield losses of up to 100% in Central Africa (Ngeve, 2003; Lema *et al.*, 2004).

GEOGRAPHICAL DISTRIBUTION

Cassava scales occur widely in Africa, Caribbean and South America (Table 15) but are not currently reported in the Pacific.

Table 15 — Geographical distribution of scales on cassava in the ACP countries

SCALE SPECIES	AFRICA	CARIBBEAN AND SOUTH AMERICA	PACIFIC
Cassava scale (<i>Aonidomytilus albus</i>)	Angola, Cabo Verde, Democratic Republic of the Congo, Republic of the Congo, Côte d'Ivoire, Gambia, Ghana, Guinea-Bissau, Kenya, Liberia, Madagascar, Malawi, Mauritius, Mozambique, Nigeria, Senegal, Somalia, Tanzania, Uganda, Zambia	Antigua and Barbuda, Argentina, Bahamas, Bahia, Barbados, Brazil (Amazonas, Ceara, Paraiba, Rio Grande do Sul), British Virgin Islands, Colombia, Cuba, Dominica, Dominican Republic, French Guiana, Grenada, Guadeloupe, Guyana, Haiti, Honduras, Jamaica, Martinique, Peru, Puerto Rico, Saint Kitts and Nevis, Saint Lucia, Saint Vincent and the Grenadines, Suriname, Trinidad and Tobago, U.S. Virgin Islands.	Not reported
Black scale (<i>Parasaissetia nigra</i>), Black scale (<i>Saissetia miranda</i>)	Angola, Cabo Verde, Democratic Republic of the Congo, Republic of the Congo, Côte d'Ivoire, Gambia, Ghana, Guinea-Bissau, Kenya, Liberia, Madagascar, Malawi, Mauritius, Mozambique, Nigeria, Senegal, Somalia, Tanzania, Uganda, Zambia	Antigua and Barbuda, Argentina, Bahamas, Barbados, Brazil (Amazonas, Ceara, Paraiba, Rio Grande do Sul), British Virgin Islands, Colombia, Cuba, Dominica, Dominican Republic, French Guiana, Grenada, Guadeloupe, Guyana, Haiti, Honduras, Jamaica, Martinique, Peru, Puerto Rico, Saint Kitts and Nevis, Saint Lucia, Saint Vincent and the Grenadines, Suriname, Trinidad and Tobago, U.S. Virgin Islands.	Southern Asia
African root and tuber scale (<i>Strictococcus vayssierei</i>)	Cameroon, Central African Republic, Democratic Republic of the Congo, Republic of the Congo, Gabon, Uganda.	Not reported	Southern Asia

QUARANTINE ORGANISM

The species are considered a quarantine threat to other parts of the world where cassava is produced, such as southern Asia.

FAVOURABLE CONDITIONS

Scales are more prevalent on 9-month-old cassava stems from fields where the leaf litter has not been cleared, compared with similar stems from well-cleared, burnt and/or ploughed fields (Dejean and Matile-Ferrero, 1996). The scales live underground (Tindo *et al.*, 2006). Infestations are more severe in the dry season (Tchuanyo *et al.*, 2000), rather than in the wet season in the tropical climate (Ngeve, 2003).

The scales also prefer flat ground to ridges (Tchuanyo *et al.*, 2000). The lateritic (rich in aluminium and iron) and clay soils are the most infested due to low fertility, shallowness or flatness (Ngeve, 2003). The pest does not thrive in freshly opened forest fields or monocropped cassava land (Ngeve, 2003). Scales feed and colonise the lower stems and, in severe infestations, the side shoots, petioles and underside of the leaves.

PEST LIFE CYCLE

Eggs are laid by adult females between the upper scale covering and lower cottony secretion. Eggs are laid from 2 weeks after mating, with about 100 eggs over 8–9 days. The female reduces in length as it lays the eggs and eventually shrivels up (Swaine, 1950). The first eggs laid become female, while those laid later become male.

‘Crawlers’ emerge after 3–4 days. The nymphs are oval shaped, 0.2 mm long and 0.15 mm wide. They have a depressed, pale-pink body with two conspicuous black eye spots on the anterior. The crawlers can move 18 inches per hour to disperse into the field. After a few days, the crawler settles on the nodes of cassava stems and produces some whitish threads, which cover the body within 2 days. The 1st moult occurs 12–15 days after egg hatching.

SURVEILLANCE/MONITORING

Monitor cassava plants for heavy infestations of whitish bugs on the stems. The leaves and fruits are not usually colonised by scales. If there are many scales but they are localised on the plant, remove the affected plants and burn them. However, if the infestation is widespread in the cassava field, consider not using the planting materials for subsequent planting.

CULTURAL CONTROL

Isolate new cassava crops from older fields that may be infected with scales.

- Use scale-free planting material, confirmed by visual inspection of absence of scales from the source plants or fields.
- Planting cassava on ridges rather than flat ground can increase plant vigour and root yield by 22% (Capinera, 2008) as a result of reduced infestation by scales.

- Practice crop hygiene at harvest by removing and burning crop debris/residues infested with scales, which could serve as sources of scales for further spread of the infestation.
- Weed cassava fields to remove potential alternative host plant species within the surrounding fields and bushes (Tindo *et al.*, 2009).
- Apply organic matter to soil to improve fertility.
- Manipulation of the planting date helped to reduce the incidence and population of *S. vayssierei* in the dry season (Ambe *et al.*, 1999) in central and southern Cameroon.

BIOCONTROL

- Using endomycorrhizae (fungi) as a biological control agent was recommended for scale management in cassava fields (Ngeve, 2003).

CONTROL WITH PLANT PROTECTION PRODUCTS

- Cassava stem cuttings may be dipped in deltamethrin to kill the scales before distribution to farmers.
- Cuttings can be treated with insecticidal bioproducts before planting to minimise the impact of the pest (Ngeve, 2003).

OTHER CONTROL METHODS

- Use of elite cassava varieties with proven resistance or tolerance to scales delayed infestation for 6 months after planting (Lema *et al.*, 2004; Ngeve, 2003).

7.3.5. THRIPS

SCIENTIFIC NAME

- Thrips (*Scirtothrips manihoti*)
- Black vine thrips (*Retithrips syriacus*) (New pest on cassava)



Figure 68 — Showing (A) Thrips eggs and (B & C) adults underneath a cassava leaf and the (D) injuries caused to leaves by *Corynothrips stenopterus*

Source: Nature&Garden, <https://www.nature-and-garden.com/gardening/thrips-eggs.html>;

http://https://idtools.org/citrus_pests/index.cfm?packageID=63&entityID=410;

Infonet biovision, <https://infonet-biovision.org/PlantHealth/Pests/Thrips>;

and BioOne Complete, <https://bioone.org/journals/florida-entomologist/volume-101/issue-4/024.101.0426/Toxicity-of-Different-Insecticides-Against-Two-Thrips-Thysanoptera--Thripidae/10.1653/024.101.0426.full>

DESCRIPTION/IDENTIFICATION

PEST

Thrips are common pests found on several plant species, including cassava, vegetables, herbs, ornamentals and some trees. The two most common species of thrips on cassava are *Scirtothrips manihoti* and the black vine thrips (*Retithrips syriacus*). The eggs are oval shaped (Figure 68A), the adults are very small, slender insects 1–2 mm in length, with two pairs of wings (Figure 68B & C). Thrips are very difficult to see with the naked eye but can be identified from the damage they cause on the leaves.

SYMPTOMS

Thrips feed by scratching the plant and leaf surfaces and sucking the sap, which results in wounds. Once the wounds heal, they appear like brown scars on the leaves, stems and flowers (Figure 68D). The cassava leaves affected by thrips have a characteristic silvery colour and small irregular punctures. Plants that are severely infested have twisted and curled leaves, elongated leaf stalks, dropping of flowers and stunting of plants.

DISPERSAL

Thrips multiply very rapidly in warm climates, in which they quickly grow to overwhelming numbers that cause severe damage to plants. By the time damage is seen, the responsible thrips may have already left. Thrips easily spread long distances by floating on the wind. Thrips can feed on a wide range of different host plants. They can also disperse long distances with the aid of human activity, such as transportation of infested planting material and leaves.

DAMAGE

Thrips cause elongation of the leaf stalks and narrowing of the entire leaf. Thrips (larvae and adults) feed on the lower surface of leaves, buds and flowers by piercing the plant tissue and sucking sap. Heavy infestation causes premature wilting, delay in leaf development, distortion of leaves and young shoots and abortion of terminal buds and flowers (Alvarez *et al.*, 2012a).

IMPACT ON YIELD AND QUALITY

Reports showed a yield reduction of 10–64.8%, depending on the cassava variety (Tata-Hangy *et al.*, 2012).

GEOGRAPHICAL DISTRIBUTION

Thrips have been reported to occur on cassava only in Africa but not the Caribbean and Pacific (Table 16).

Table 16 — Geographical distribution of thrips affecting cassava in the ACP countries

AFRICA	CARIBBEAN AND SOUTH AMERICA	PACIFIC
Angola, Benin, Cameroon, Central African Republic, Democratic Republic of the Congo, Republic of the Congo, Côte d'Ivoire, Equatorial Guinea, Gabon, Gambia, Ghana, Guinea, Guinea-Bissau, Kenya, Liberia, Madagascar, Malawi, Mali, Mozambique, Niger, Nigeria, Rwanda, Senegal, Sierra Leone, South Africa, Sudan, Tanzania, Zanzibar Island, Togo, Uganda, Zambia, Zimbabwe	Not reported	Not reported

QUARANTINE ORGANISM

Thrips occur widely in Africa but are not currently reported in the Caribbean and South America or in the Pacific Islands. Therefore, the pest is of quarantine importance and strict measures must be instituted to restrict the movement of cassava plant parts such as stems and leaves, where the thrips may harbour to avoid introducing them in areas they do not exist.

FAVOURABLE CONDITIONS

Thrips multiply very rapidly in warm weather conditions and their population quickly increases, which causes severe damage to plants. Unfortunately, damage is only recognised after the thrips that caused it have migrated from the plants. They easily spread long distances with the aid of wind.

PEST LIFE CYCLE

The life cycle of thrips (Figure 69) takes approximately 14–21 days under warm conditions. The females lay eggs on plants, followed by hatching of the eggs into larvae (nymphs), that feed on the plants before dropping to the soil to develop into adults (pupate). The adults emerge from the soil and fly up to the plant to repeat the cycle (Kumar, 2021).

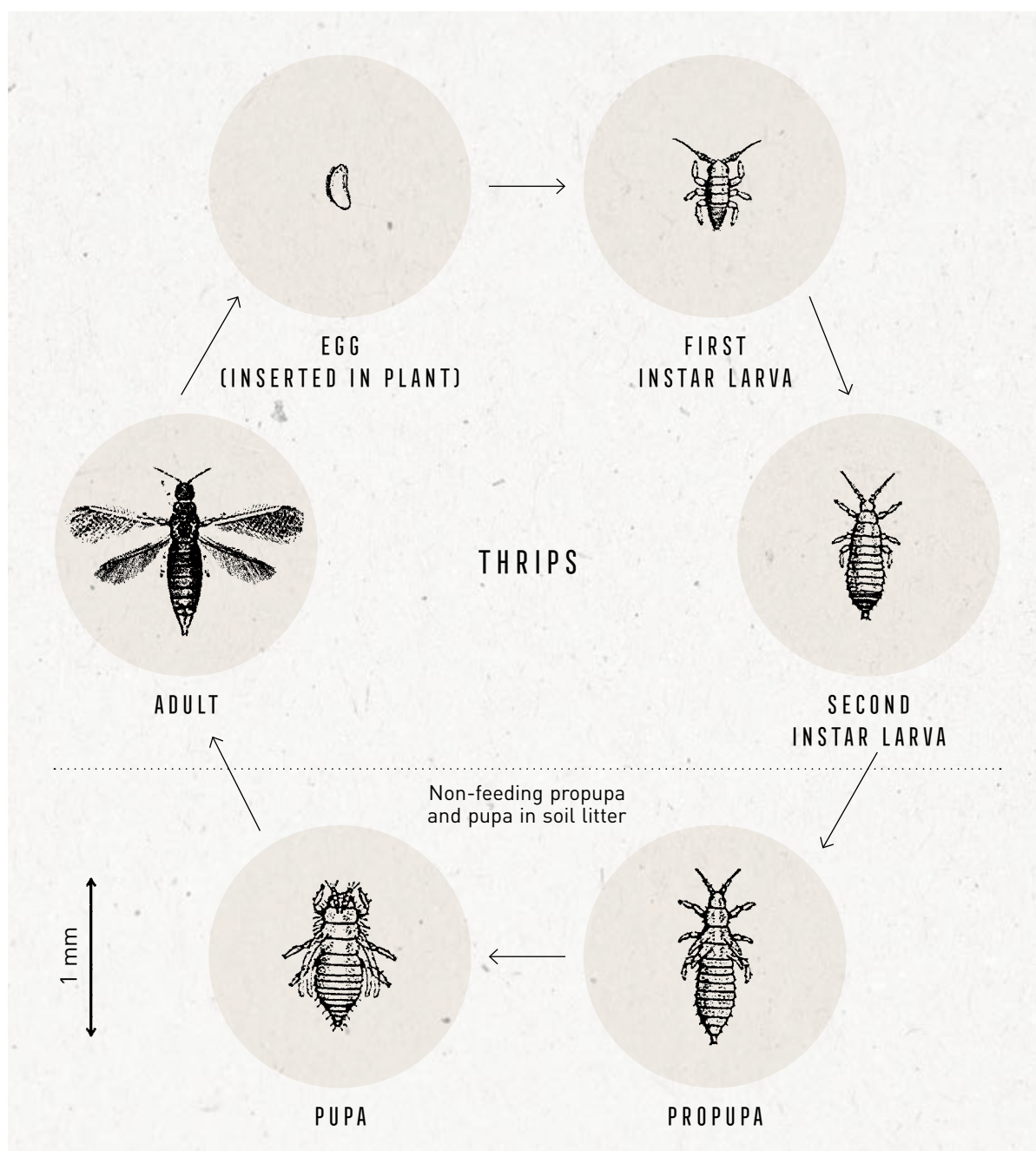


Figure 69 — Thrips life cycle

Source: Kumar, 2021, https://link.springer.com/chapter/10.1007/978-981-15-8075-8_9

Eggs are very tiny: A single egg is 0.25 mm long and 0.1 mm wide. Eggs are white when freshly laid but turn pale yellow toward maturation. Eggs are usually laid singly inside the plant tissue and are therefore not visible. Some thrips (e.g. *Haplothrips* spp.) lay eggs singly or in clusters on the plant surface (Kumar, 2021).

Larvae: The 1st and 2nd instar larvae are 0.5–1.2 mm in size, elongated, slender and can be pale yellow, orange or red, according to the species. They have piercing-sucking mouthparts and resemble miniature versions of the adults but lack wings (Kumar, 2021).

Pre-pupa and pupae: These comprise 2 or 3 instars as intermediate stages between the nymph and the adult. They lack functional wings and are also inactive – they do not feed and do not cause any damage to the plant. They may occur on a plant or in the soil beneath, depending on species.

Adult thrips: are 1–1.5 mm in size, slender and have long, narrow and fringed wings with long hairs, which during rest are tied on the back along the body. Their colour varies according to the species, with majority being black, brown or yellow (Kumar, 2021).

SURVEILLANCE/MONITORING

Monitor the crop regularly, as early detection of thrips is important in determining appropriate control strategies. Randomly sample the cassava plants and evaluate the adult thrips numbers and damage under the leaves. Sample at least 30 plants per field along two diagonals following an 'X' formation. Adult thrips can be collected by shaking the leaves and flowers on a white sheet of paper for further laboratory identification. Other methods include trapping with coloured (blue, yellow or white) sticky traps or water traps in the field.

CULTURAL CONTROL

Thrips are very difficult to control, as they are highly active and quickly fly away when plants are disturbed. Hence, management involves using different strategies to exploit the understanding of the insect's biology.

- Deep ploughing, harrowing and solarisation or exposure to the sun help to kill pupae in the soil from the previous season.
- Plant early to ensure proper establishment of cassava plants and prevent thrip infestation.
- Crop rotation avoids successive planting of susceptible cassava varieties and reduces the occurrence of thrip infestation.
- Intercropping with cowpea can reduce thrips populations (Parella and Lewis, 1997).
- Use thick layers of dried mulch on the soil to prevent larvae from easily entering the soil to pupate.
- Practice field sanitation by removing cassava and other plant debris from the previous crop and improving drainage. Use clean weeding to reduce the pest population.
- Use overhead irrigation to wash the insects off the cassava leaves helps to knock off the insects and interferes with the ability of the thrips to spread to the entire field. However, irrigation is rarely used by small-scale cassava farmers, so would only be beneficial in large-scale commercial farming, where flooding fields was found to reduce thrips damage by destroying a large proportion of pupae in the soil.

- Use sticky traps with yellow and blue colours to attract the thrips, which eventually die. The coloured sticky traps (blue, yellow or white) or water traps are useful for monitoring and in some cases reducing thrips by mass trapping them in the nursery or field. Bright colours attract more thrips than darker ones.
- Ensure good growing conditions, such as provision of sufficient water, to enhance rapid plant growth and to reduce stresses that make plants more susceptible to thrip infestation.
- Walk-in field tunnels made from ultraviolet-absorbing plastics have proved effective in protecting crops from western flower thrips.
- Reflective mulches deter thrips. Aluminium-surfaced mulch (e.g., coated plastic mulch) has considerably decreased thrips and virus infection in tomato, pepper and tobacco. The effectiveness of the mulch decreases with increased shading by lower leaves (Lewis, 1997).

BIOCONTROL

- Natural enemies, in particular predators, are often found feeding on thrips. They include predatory thrips, predatory mites (e.g., *Amblyseius* spp.), anthocorid bugs or minute pirate bugs (*Orius* spp.), ground beetles, lacewings, hoverflies and spiders. The parasitic wasp *Ceranisus menes* is an important natural enemy. The farmer can increase the number of these natural enemies by providing protective habitats for them.

CONTROL WITH PLANT PROTECTION PRODUCTS

- Organic Neem or azadirachtin can be sprayed on cassava leaf surfaces (including the undersides of leaves) until completely wet to kill eggs and nymphs and/or repel adults of thrips. The neem-based pesticides were reported to control young nymphs, inhibit growth and development of older nymphs, and reduce egg-laying by adult thrips. Adding 0.1–0.5% of soft soap enhances efficacy of neem-based pesticides.
- Cassava stem cuttings may be dipped in deltamethrin to kill thrips eggs, nymphs and adults before distribution to farmers.
- Spinosad, a microbial insecticide successfully controlled thrips. This biopesticide is derived from the fermentation of an *Actinomyces* bacterium, commonly found in the soil.

7.3.6. HOPPERS

SCIENTIFIC NAME

- **Grasshoppers** (variegated grasshopper, elegant grasshopper) [*Zonocerus variegatus*, *Zonocerus elegans*, *Gryllotalpa africana*, *Gryllotalpa* sp. & *Gryllus assimilis*]



Figure 70 — Showing (A) variegated grasshoppers on a cassava plant and (B & C) defoliated cassava plants
Source: Amateur Entomologists' Society, <https://www.amentsoc.org/insects/glossary/terms/brachypterous/>;
Project Noah, <https://www.projectnoah.org/spottings/1542536013>; International Institute of Tropical Agriculture, <https://www.flickr.com/photos/iita-media-library/5430536731>

DESCRIPTION/IDENTIFICATION

PEST

Adult variegated grasshoppers are dark green with yellow, black and orange markings on their bodies (Figure 70A). Nymphs are black with yellow markings on the body, legs, antenna and wing pads. Female grasshoppers lay many eggs just below the surface of the soil in the shade under evergreen plants, usually outside cassava fields. Eggs are tiny pods laid in masses of froth, which harden to form sponge-like packets (egg pods) (Sévior *et al.*, 2014).

SYMPTOM

Variegated grasshoppers defoliate plants; remove bark from stems; damage flowers, leaves, seeds and stems; and cause dieback of growing points and whole plants (Figure 70B & C). Leaf hoppers cause honeydew or sooty mould on cassava leaves (Bohlen, 1973).

DISPERSAL

The early instars (I-III) mainly feed on soft vegetables. When the plant is depleted, the older instars (V, VI) and adults move to other crops and shrubs. The adults move in groups by flying and with the aid of wind currents. Variegated grasshoppers feed on many different plants (Sévior *et al.*, 2014), affecting many crop and plant species, including cassava.

DAMAGE

Grasshoppers feed on cassava plants, chewing the leaves and debarking the stems, often resulting in dieback of growing points, with wilting of whole plants in extreme cases. The damage is most severe on the edge of the fields because grasshoppers avoid entering dense plant canopies. Cassava fields next to bushes in the dry season are the most affected.

IMPACT ON YIELD AND QUALITY

Grasshoppers destroy leaves and stems. If this occurs early in plant growth, it affects the yield and size of cassava tubers.

GEOGRAPHICAL DISTRIBUTION

The variegated grasshopper is found in Africa, south of the sahara. In contrast, leafhoppers are only widespread and of economic importance in Tanzania (Table 17).

Table 17 — Geographical distribution of hoppers affecting cassava in the ACP countries

SPECIES	AFRICA	CARIBBEAN	PACIFIC
Grasshoppers (variegated and elegant)	Angola, Benin, Burkina Faso, Cameroon, Central African Republic, Chad, Democratic Republic of the Congo, Republic of the Congo, Côte d'Ivoire, Gabon, Gambia, Ghana, Guinea, Guinea-Bissau, Kenya, Liberia, Malawi, Mali, Niger, Nigeria, Rwanda, Senegal, Sierra Leone and Togo.	Not reported	Not reported
Leaf hoppers	Tanzania	Not reported	Not reported

QUARANTINE ORGANISM

Variegated grasshoppers such as desert locusts are migratory pests that breed in one place and move short and long distances with the aid of wind to attack crops and other vegetation (Ebbels, 2003). National Plant Protection Organizations are not usually responsible for the control of migratory pests; instead, this falls under the remit of the different departments of the ministry of agriculture in many countries. Quarantine against such pests is very unlikely. Instead, measures can be put in place to manage an invasion when it arises.

FAVOURABLE CONDITIONS

Zonocerus variegatus is a major pest of cassava in the dry season (late September – early November) in the humid lowland forests and savannas of West and Central Africa. Forest clearing to grow crops such as cassava, maize and vegetables creates an ideal habitat for the pest. The eggs start to hatch at the beginning of the main dry season.

PEST LIFE CYCLE

The life cycle of a variegated grasshopper has three stages: egg, nymph and adult (Figure 71). However, the nymphs go through five moults before reaching adulthood. Grasshopper eggs are laid in the ground in pods that contain anything from very few to more than 100 eggs. The nymphs come out of their eggs, eat and moult five times, shedding their exoskeleton as they grow (Kpindou *et al.*, 2000).

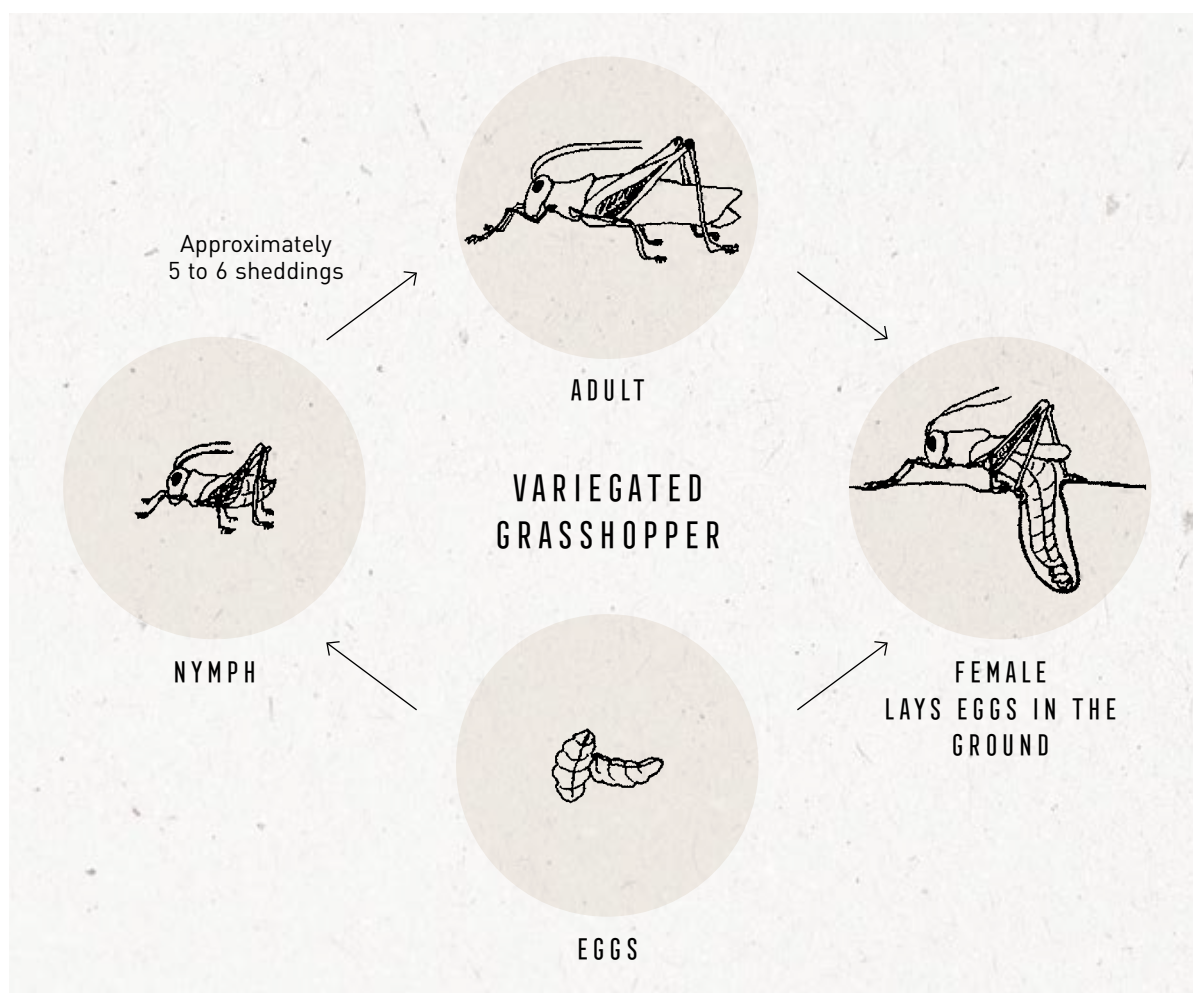


Figure 71 — Life cycle of a variegated grasshopper

Source: <http://www.nzdl.org/cgi-bin/library?e=d-00000-00---off-0fnt2.2--00-0----0-10-0---0--0direct-10---4-----0-1l--11-en-50---20-about---00-0-1-00---4---0-0-11-1-0utfZz-8-10&c-l=CL3.6&d=HASH0150ba4e9f73176fac50b5ae.7.11&gt=2>

SURVEILLANCE/MONITORING

As grasshoppers spread to crops from uncultivated bushes and grasslands, it is recommended that scouting and treatment are carried out in surrounding vegetation first. However, if the grasshoppers have already entered the cassava fields, sampling should be conducted to determine whether the population and/or damage warrant control interventions. If the populations are high, control should be applied on the borders and before the grasshoppers fully develop wings.

Use quadrats (1 m²) placed randomly at the field edges and in the surrounding varied vegetation to estimate the population of the grasshoppers. Recorded the number of grasshoppers that jump in and out of the quadrat. Repeat this at least 10 times. Work out the average of the grasshopper numbers by stage of development (nymphal stages and adults with fully developed wings) to determine if, when and what control measure to apply (Wright *et al.*, 2015).

CULTURAL CONTROL

Hand pick and kill any adult grasshoppers found on cassava and other plants in and around the fields. You may also look for grasshopper egg pods in and around cassava fields and destroy them to reduce populations. However, this is only feasible in small plots with low infestation. It is harder to control grasshoppers after they fully develop wings as they tend to fly away. Therefore, control measures should be used on the immature stages (Wright *et al.*, 2015).

BIOCONTROL

- Entomopathogenic fungi: *Beauveria bassiana* (Jaronski *et al.*, 1997) and *Entomophthora grylli fresenius* caused high mortality of locusts and grasshoppers (Roffey, 1968).

CONTROL WITH PLANT PROTECTION PRODUCTS

- Biopesticides such as the ones containing *Metarhizium anisopliae var. acridum* proved very effective at reducing the grasshopper population with 70 to 100% mortality rates after 8 to 28 days of application (Douthwaite, 2001).
- Apply deltamethrin on cassava plants and the bushes bordering the field to prevent further entry of grasshoppers. This should be done at least 7-14 days, depending on the level of infestation (Wright *et al.*, 2015).
- Use neem extracts to manage grasshoppers as they stop feeding when they are exposed to neem products, which affect their development (Olaoifa and Adenuga, 1988).

OTHER CONTROL METHODS

- None

7.3.7. TERMITES

SCIENTIFIC NAME

Isoptera: Rhinotermitidae

- *Heterotermes tenuis*
- *Coptotermes paradoxus*
- *Coptotermes* spp.



Figure 72 — Showing (A) an adult termite and (B & C) destruction caused to cassava plants

Source: Collins Dictionary, <https://www.collinsdictionary.com/dictionary/english/termite>;
<https://leafplanttech.com/understanding-attack-of-termites-on-cassava-plants/>; Zambia National Farmers' Union,
<https://www.betuco.be/manioc/Cassava%20-%20How%20to%20Plant%20Zambia.pdf> [consulted on 5 January 2023]

DESCRIPTION/IDENTIFICATION

PEST

Adult termites are ant-like, mainly wingless and 0.5–1 cm long (Figure 72A). Unlike ants, termites have straight antennae, broad waists, same-sized wings (if present) and multiple veins. Termite workers and soldiers are small insects with white or brown bodies and brown heads. They often have but sometimes lack wings (Mullins *et al.*, 2015).

SYMPTOMS

In newly planted cassava crops, termites chew and eat stem cuttings (Figure 72B & C), which causes poor growth, death and rotting of plants. In older cassava crops, termites chew the stems, weakening them to the point of breaking.

DISPERSAL

Long-distance dispersal of termites occurs by human activity during transportation of infested goods. Termites also disperse by nuptial (Higa and Tamashiro, 1983) flights of alates (the winged form of the insect) in ships infested with termites into new ports (Scheffrahn *et al.*, 2009; Hochmair and Scheffrahn, 2010; Scheffrahn and Crowe, 2011).

DAMAGE

Worker termites cause the main damage to cassava crops and tend to feed on planting material and roots of growing plants, which prevents the proper establishment of nodes and cuttings. When they attack established cassava crops, they may damage the stems and storage roots (Figure 72B & C), causing poor plant growth, death and rotting.

IMPACT ON YIELD AND QUALITY

Termites caused more than 5 metric tonnes/ha of yield loss when plants were harvested from termite-infested soil (Atu, 1993).

GEOGRAPHICAL DISTRIBUTION

Various species of termites occur in many countries around the world and cause economic loss to many crops, including cassava in Africa, the Caribbean and the Pacific.

QUARANTINE ORGANISM

Avoid moving cassava planting materials infested with termites.

FAVOURABLE CONDITIONS

Termites live in soil or in nests above the ground and tend to attack cassava in the lowland tropical areas. They also occur in tunnels on the surface of cassava stems and cause serious problems in the dry season.

PEST LIFE CYCLE

The life cycle of termites goes through several stages, all of which are able to move except the eggs (Figure 73).

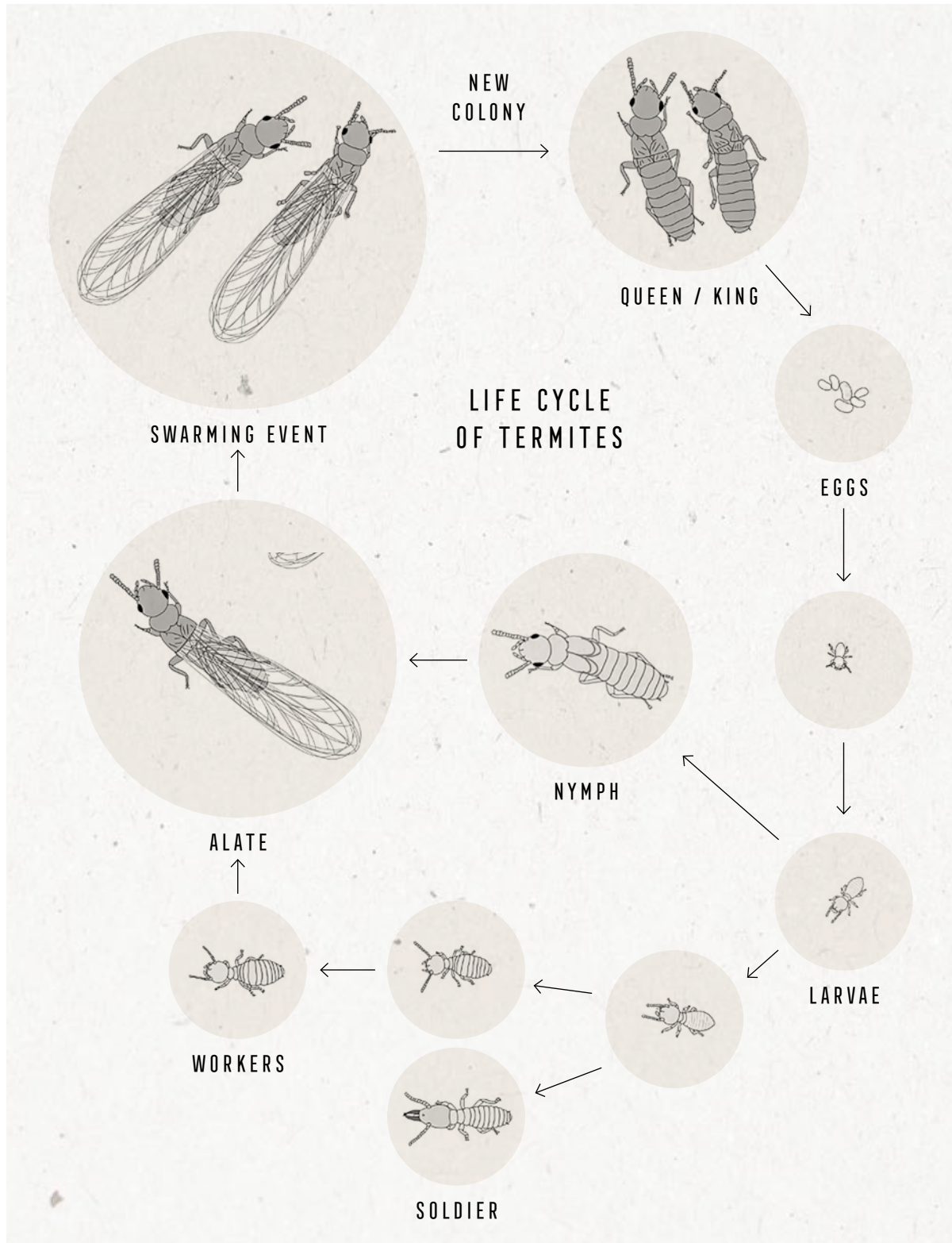


Figure 73 — Life cycle of termites

Source: University of Florida, <https://flrec.ifas.ufl.edu/termites-in-florida/life-cycle/>

SURVEILLANCE/MONITORING

Scout for the presence of termites once a week, increasing to twice a week in the dry season, when the most termite damage occurs. Look out for chewed stem cuttings, mud or cuts on stems, termite nests in the field and galleries in soil and wood. Consider control measures if 5–10 plants are affected. Termites build nests above and/or below ground, depending on the species. Mound structures can vary in size and be up to 5 m high.

CULTURAL CONTROL

Avoid fields infested with termites when selecting planting sites.

- Plant cassava on ridges to avoid termite establishment.
- Plant cassava cuttings during sufficient rains/during the rainy season.
- Uproot and destroy plants that fail to germinate due to termite damage.
- Introduce ‘driver ants’ (*Dorylus*, or safari ants) in areas affected by termites to kill the pest off.
- Dig out termite colonies and remove the white anthills/termite mounds.
- Put wood ash on the base of cassava plants.
- Where possible, sterilise the soil with hot water before planting (if infestation is localised).
- Remove all plant debris and burn to remove any suitable sites for termite colonisation.
- If mulch material is used in the field, avoid putting it too close to the cassava plant as it will attract termites.
- Irrigate the fields to keep soil moist in the dry season.
- Plant cassava cuttings at high density to cater for those that may be destroyed by termites.
- Establish barrier plants such as basil (*Ocimum* spp.) to repel termites.

BIOCONTROL

- Use of driver ants significantly reduces termite infestation.
- Expose the termites to predators such as ants, dragonflies, ground beetles, bats, birds and frogs.
- Deploy the fungus *Beauveria bassiana* as biological control if available in the country.

- Use of fungal cultures was reported to control termites (*Reticulitermes flavipes* Kollar) more effectively than exposure to spores alone or to extracted fungal toxins (Delate *et al.*, 1995; Grace; 1995). However, the fungi were of no consequence in open field conditions, even in large amounts (Boucias *et al.*, 1996; Ramakrishnan *et al.*, 1999; Osbrink *et al.*, 2001).

CONTROL WITH PLANT PROTECTION PRODUCTS

- Spray cassava plants with extract of neem leaves or fruits to keep termites away.
- Spray diazinon around the edges of the field to prevent termites from entering the crop, and wherever there is an anthill. A maximum of two applications per season is sufficient to control termites.
- You may also use deltamethrin.

7.3.8. WHITE GRUBS

SCIENTIFIC NAME

Coleoptera: Scarabeidae

- *Anomala obsoleta*
- *Euchlora viridis*
- *Euchlora pulchripes*
- *Heteronychus plebejus*
- *Lepidiota stigma*
- *Phyllophaga menetriesi*

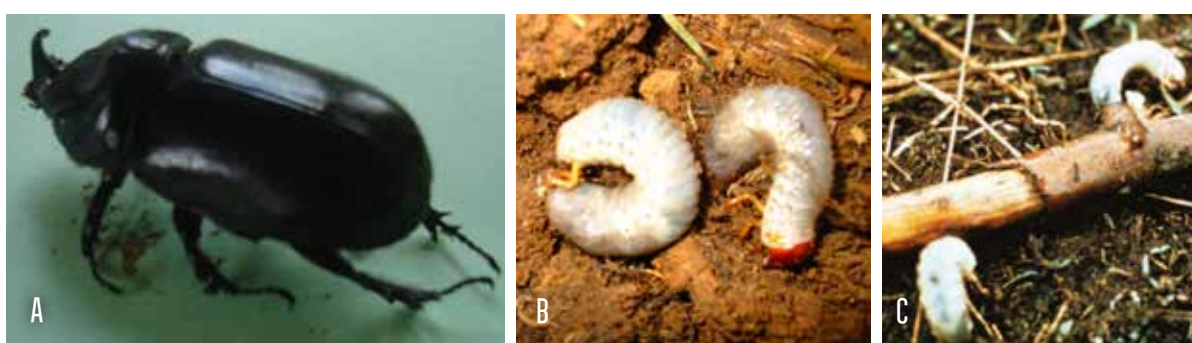


Figure 74 — Showing (A) an adult scarab beetle;

(B) the white grubs (larvae of scarab beetles); (C) damage caused on stems

Source: Purdue University, <https://extension.entm.purdue.edu/fieldcropsipm/insects/corn-whitegrubs.php>; [pests.org](https://www.pests.org), <https://www.pests.org/scarab-beetles>

DESCRIPTION/IDENTIFICATION

PEST

White grubs are the larvae of scarab beetles (Figure 74A) including *Cyclocephala* spp., *Heteronychus* spp., *Eulepida* spp. and *Phyllophaga* spp., which damage crops by feeding on the root systems. Freshly laid eggs are white, oval and approximately 3 mm long and 2 mm wide (Figure 74B). At 3–4 days after laying, eggs are spherical, smooth and 2 mm in diameter. Mature eggs are brownish-black and 4 mm wide. **Larvae** are creamy white, C-shaped (**Scarabaeiform**) and vary in size (2–3 mm to 20–40 mm) depending on age and species.

White grubs range from 1.5 to 3 cm in length and are around 1.5 cm wide. However, different species differ in the size, shape and colour of the adults. They have protruding brown heads and large, dark brown mouthparts, five-segmented antennae, three pairs of spiny legs and a shiny tip on the abdomen. The early stages of pupae are white to yellowish and slowly turn brown. The grubs pupate in the soil. The adult beetles have extra thick wing cases and a plate-like covering of the front segment of the **thorax**. Some species have brightly coloured, metallic green, patterned or plain brown to black structures (Bellotti *et al.*, 2012a).

SYMPTOMS

Damage symptoms on cassava plants (Figure 74C) include wilting, poor crop stands, tilted/curved/lodged plants, uneven growth, damaged plants that are easily pulled from the soil, dead heart, and purple coloration of the plants due to reduced phosphorus uptake (Nataraja *et al.*, 2015). Feeding of various adult scarab beetles occurs at night and takes on a characteristic form, as they tend to make running “zigzag” holes within the blade of the leaves, not at the leaf edges. Hence damage by the adults is not economically important.

DISPERSAL

White grubs feed on numerous plant species, including cassava, pasture grasses, napier grass, maize, tobacco, wattle saplings, potatoes, tomatoes, asparagus, sunhemp, groundnuts, strawberries and pineapples, as well as many vegetables, roses and many other ornamentals, including lawn grasses, which aids its survival and dispersal. The larvae can be moved in cassava tubers by humans, while the beetles fly during the night (Bellotti *et al.*, 2012a).

DAMAGE

White grubs damage crops by feeding on root systems, causing poor plant growth and death in severe cases. The larvae are the most damaging stage and cause most severe damage when feeding on young plants. The adults can also damage plants but rarely do so severely (Bellotti *et al.*, 2012a).

IMPACT ON YIELD AND QUALITY

Affected cassava plants have damaged tubers, which reduces the quality and quantity of edible cassava.

GEOGRAPHICAL DISTRIBUTION

Scarab beetles and white grubs occur worldwide.

QUARANTINE ORGANISM

The wide distribution of scarab beetles and white grubs means that no quarantine measures may be imposed in their control, although preventive measures must be observed to ensure that plant materials are free of these pests before export to other locations.

FAVOURABLE CONDITIONS

White grubs prefer light clay or sandy soils that are rich in organic matter with good aeration and moisture. Decaying organic matter is preferred by female beetles for laying eggs. The pest thrives at soil temperatures of around 25°C and has one generation per year.

PEST LIFE CYCLE

Eggs are deposited singly in weedy fields or grasslands several centimetres below the soil surface. The time taken for eggs to hatch varies from about 6 days to over 50 days.

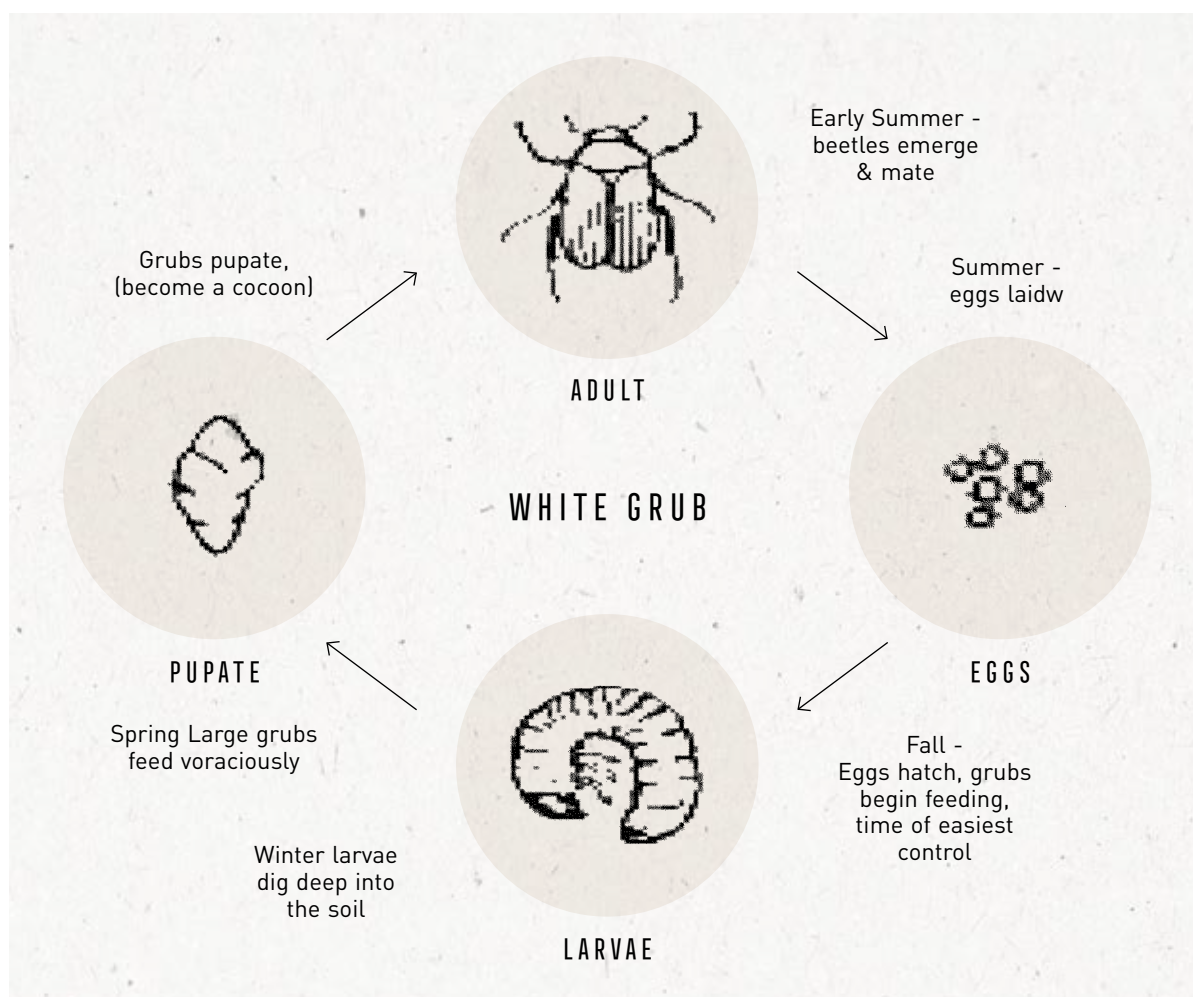


Figure 75 — White grub life cycle

Source: DeVries, <https://devrieslandscape.com/about/turf-tips/white-grubs>

Female adults can lay up to 60 eggs at a time for over 100 days. Larvae undergo three instar stages (Figure 75) and may undergo diapause, a period of dormancy triggered by climatic conditions such as dry seasons before pupation. Wet soil conditions during diapause cause high larval mortality by promoting fungal and bacterial infections.

Pupal development takes 30–40 days, while most species complete their lifecycle in one year. However, the most damaging species, *Phyllophaga implicita* (sometimes known as a May beetle or June bug) can take up to 3 years. Beetles emerge from pupae in the soil at the onset of rains or ploughing. The adults come out at dusk and fly actively in the night. The pest normally feeds on trees but comes down to grasses and crops to lay eggs after mating (Bellotti *et al.*, 2012a).

SURVEILLANCE/MONITORING

White grubs should be monitored whenever they come close to the surface (and are easy to see). Also, examine below grass thatches during the dry season, where the young 1st instar larvae are normally present. A threshold of 5–10 white grubs per square foot is deemed high enough to begin applying control measures.

CULTURAL CONTROL

Check for white grub infestation by early ploughing in the growing season, when the grubs are closer to the surface and expose the larvae, pupae and adults to the sun and predators such as birds.

- Practice crop rotation by inclusion of resistant or less susceptible deep-rooted legumes such as alfalfa, cowpea and pigeon peas.
- Use trap crops such as African marigold, sunflower or castor, or repellent plants such as chives, garlic, tansy or catnip to prevent adult beetles from attacking cassava crops.
- Improve drainage of crop fields to avoid the moist soil that helps the survival of grubs.
- Reduce or prune trees bordering cassava fields that may attract adult grubs.
- Shake trees so that adult scarab beetles fall off and can be collected and killed by burning.
- Heavy application of nitrogen fertiliser can kill first instar white grub larvae.

BIOCONTROL

- Inoculating soil with the bacteria *Bacillus popilliae*, *Bacillus lentimorbus* and *Metarhizium anisopliae* can help control white grubs.
- Parasitic wasps such as *Tiphia* spp., *Myzinum* spp. and *Pelecinus polyturator* and the parasitic fly *Pyrgota undata* have proven effective as biocontrol agents.
- Nematodes worms such as *Steinernema* can effectively control white grubs.
- Use of entomopathogenic fungi to control white grubs was investigated in Rwanda with significant success (Kajuga *et al.*, 2018).

CONTROL WITH PLANT PROTECTION PRODUCTS

Use of chemical pesticides (such as chlorantraniliprole) for white grubs' control is more effective when they are young, as older larvae and adults can move deeper into the soil, which makes it difficult to reach them with the pesticides.

Field infestations of white grubs can be sporadic and patchy, which makes the application of pesticides very expensive.

7.3.9. CASSAVA CATERPILLAR/CASSAVA HORNWORM

SCIENTIFIC NAME

— *Erynnis ello*: Sphingidae

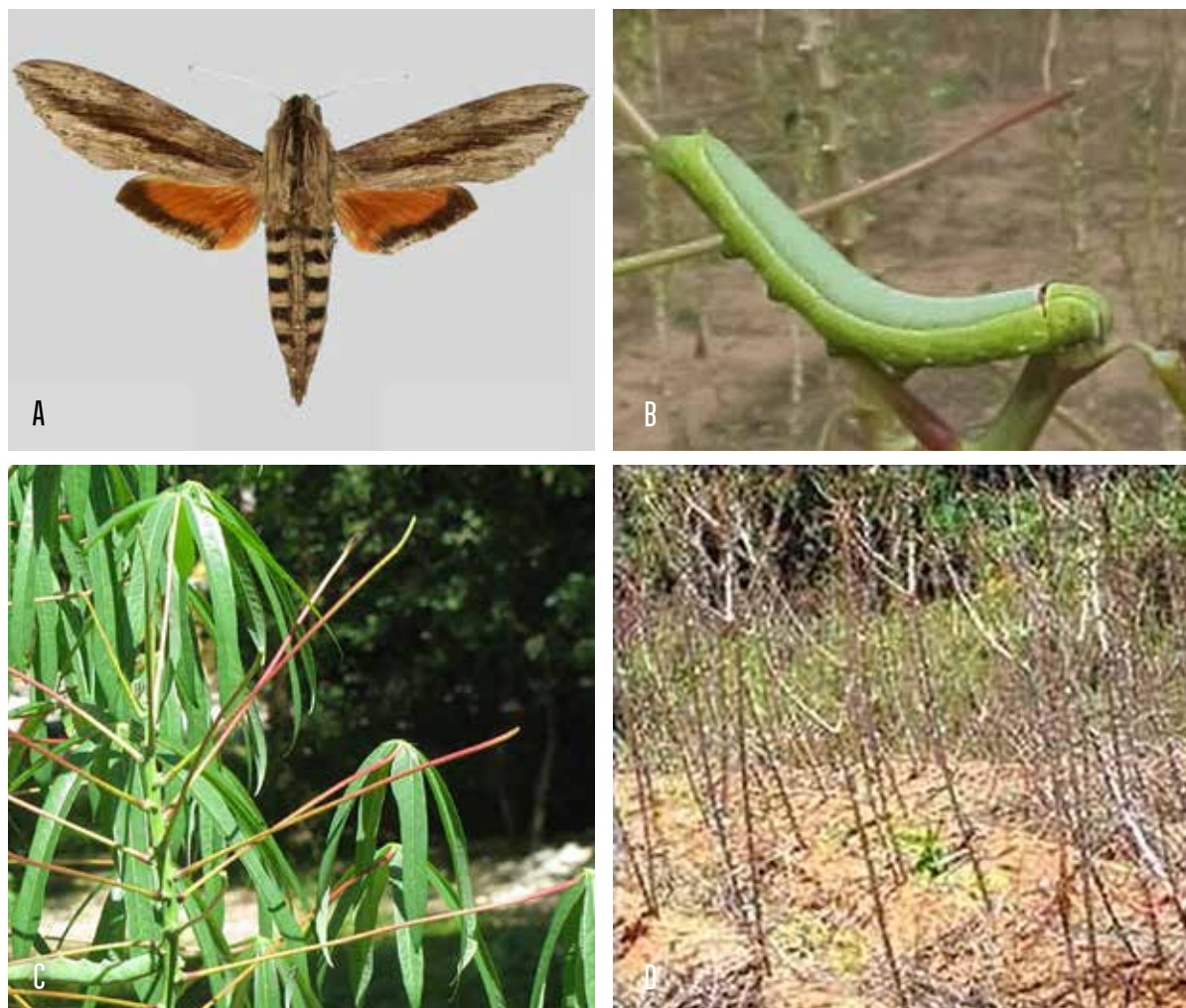


Figure 76 — Showing (A) adult cassava hornworm - moth; (B) larva; (C) damage to cassava plant leaves; and (D) heavily defoliated cassava crop

Source: Wikipedia, https://en.wikipedia.org/wiki/Erinnyis_ello;

CABI Plantwise Knowledge Bank, <https://www.plantwise.org/knowledgebank/factsheetforfarmers/20137804418>;

Jim Conrad, <https://www.backyardnature.net/yucatan/erinnyis.htm>;

CABI Plantwise Knowledge Bank,

<https://www.plantwise.org/KnowledgeBank/factsheetforfarmers/20127802690>

DESCRIPTION/IDENTIFICATION

PEST

Cassava caterpillar, also known as the cassava hornworm is a moth (Figure 76A), with the destructive stage being the larva, which have diverse colours: green (Figure 76B), yellow, brown, grey or black with red, black or white spots. The caterpillars grow to 10–12 cm in size and produce small eggs (1.0 mm) that are smooth and dark green to yellow in colour. The pest hides in crevices of buds, stems and barks of cassava plants, and deposits eggs on the upper part of the leaf. Mature 5th larval instars also vary in colour and can be whitish green with yellow, red or black spots in its abdomen. Mature larvae are very glossy and black with pale orange streaks. Adult moths have pale grey wings and red hind wings (70–90 mm) that have dark marginal bands (Bellotti *et al.*, 2012a).

SYMPTOMS

Young leaves are attacked first, followed by more mature leaves (Figure 76C). When severe, the cassava fields are completely defoliated (Figure 76D), and the stems are damaged too. The first signs of an infestation are multiple holes on the younger leaves, caused by larvae that stay on the underside.

DISPERSAL

The adults fly only at night and tend to migrate in swarms (Bellotti *et al.*, 2012a).

DAMAGE

Mature larvae (5th instar) are the most destructive stage of the cassava caterpillar (Adaime *et al.*, 2020). Frequent attacks result in complete defoliation of cassava plants; on immature cassava, this interferes with tuber formation, resulting in reduced crop yield. Mature cassava crops tend to be less affected because they have less foliage and have already formed edible tubers. Despite this, cassava caterpillars are considered minor pests of cassava, and tend to colonise several other plant species in the *Euphorbiaceae* family.

IMPACT ON YIELD AND QUALITY

The cassava caterpillar attacks the cassava leaves, leading to significant losses that are estimated to range from 10% to 64%, depending on the severity and number of attacks and the local ecology (Bellotti *et al.*, 2012a).

GEOGRAPHICAL DISTRIBUTION

The pest is native to tropical regions of the Americas, where it has been reported in Central America, the Caribbean, Florida (USA), Brazil, Columbia and Ecuador (Table 18). It has also been reported in Papua New Guinea. Cassava caterpillars are not known to cause economic damage on cassava in Africa.

Table 18 — Geographical distribution of cassava hornworm in the ACP countries

AFRICA	CARIBBEAN AND SOUTH AMERICA	PACIFIC
Not reported	Antigua and Barbuda, Bahamas, Barbados, Belize, British Virgin Islands, Cayman Islands, Costa Rica, Cuba, Dominica, Dominican Republic, El Salvador, Grenada, Guadeloupe, Guatemala, Haiti, Honduras, Jamaica, Martinique, Montserrat, Nicaragua, Panama, Puerto Rico, Saint Kitts and Nevis, Saint Lucia, Saint Vincent and the Grenadines, and Trinidad and Tobago	Papua New Guinea

QUARANTINE ORGANISM

Efforts must be made to control the movement of materials infected with the cassava caterpillar to areas where it is not yet reported.

FAVOURABLE CONDITIONS

Cassava caterpillars thrive in warm conditions where the life cycle is completed at a faster rate.

PEST LIFE CYCLE

Cassava caterpillars undergo complete metamorphosis, which involves five stages: egg, larva, prepupa, pupa and adult (Figure 77). This takes 32–49 days, depending on temperature. The cycle is complete in a shorter period under warm conditions. The eggs hatch within 3–5 days, after which the larva, which is the destructive stage of the moth, feeds for 12–15 days.

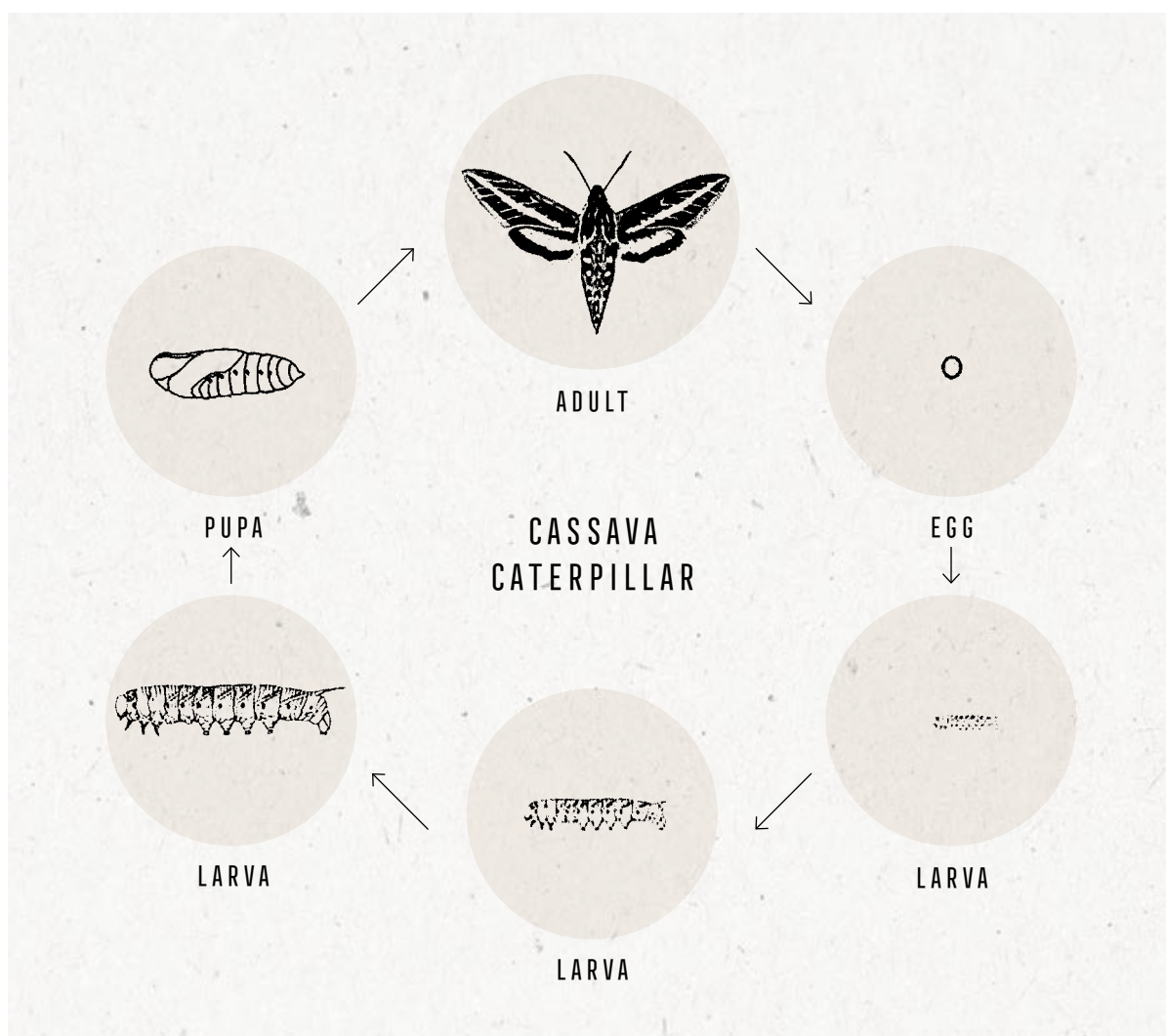


Figure 77 — Life cycle of cassava caterpillar

Source: University of Nebraska–Lincoln, <https://entomology.unl.edu/scilit/hornworms>

The newly hatched 1st instar is 6 mm long and pale green with a black horn on the last segment. It undergoes five larval instars, which feed and grow to 80–100 mm in length. Mature 5th larval instars vary in colour and can be whitish green with yellow spots and red and black spots in its abdomen. The mature larvae are very glossy and black with pale orange streaks. They are 55 mm long and crawl down to 4–8 cm into the soil to develop into the pupa stage. After 20–26 days, adult moths emerge from the pupae. Adults fly only at night and migrate in swarms. Adults lay at least 850 eggs in a lifetime.

SURVEILLANCE/MONITORING

Monitor cassava caterpillars and their damage by checking cassava fields for their presence, especially during the dry season. If the population of caterpillars is high, apply appropriate control measures, including use of insecticides such as organophosphates mixed with a pyrethroid or microbial insecticide.

CULTURAL CONTROL

Monitoring cassava fields for symptoms of damage should be carried out regularly, especially during the dry season.

- If the cassava caterpillars is detected early, it is important to rotate with other crops such as corn (maize) to decrease the number of caterpillars.

BIOCONTROL

- Use of a bio-rational pesticide such as *Bacillus thuringiensis*, which is environmentally friendly and effective against early instars of the pest (Gabrys, *et al.*, 2008).
- Other biocontrol agents that have proven effective in controlling cassava caterpillars include the parasitoid wasps *Trichogramma* spp., *Cotesia* spp. and *Sarcodesia* spp., the lacewings *Chrysopa* spp., and the common wasp *Polistes* spp.
- A virus, *Baculovirus erinnyis*, was successfully used to control cassava caterpillars in Brazil (Bellotti *et al.*, 1992).

CONTROL WITH PLANT PROTECTION PRODUCTS

- Adopt practices that conserve beneficial insects, including applying ecological products such as Methoxyfenozide when the caterpillar starts to cause damage.
- Spray deltamethrin or Cypermethrin to control the caterpillars when they are very small.

7.3.10. ROOT-KNOT NEMATODE

SCIENTIFIC NAME

— *Meloidogyne* spp.



Figure 78 — Showing (A) root-knot nematodes (under microscope);

(B & C) affected cassava roots and (D) tubers with characteristic knots or swellings

Source: NC State, <https://content.ces.ncsu.edu/management-of-root-knot-nematodes-in-bedding-plants>;

Infonet biovision, <https://infonet-biovision.org/PlantHealth/Pests/Root-knot-nematodes>;

Semantic Scholar, <https://www.semanticscholar.org/paper/SCREEN-HOUSE-RESPONSE-OF-SEVEN-ELITE-CASSAVA-TO-Akinsanya-Afolami/2c76b20ce1359efbdd5774c187f19a9c5f9511df>

DESCRIPTION/IDENTIFICATION

PEST

Root-knot nematodes (*Meloidogyne* spp.) are minute (microscopic), worm-like (Figure 78A) and live in soil, where they feed on roots. They have a wide host range and cause problems in many annual and perennial crops. Root-knot nematode juveniles are active, thread-like worms about 0.5 mm long and may not be easily seen by the naked eye.

SYMPTOMS

Galls (swellings) form on roots (Figure 78B) with sizes of up to 3.3 cm in diameter (but are usually smaller); reduction in plant vigour; yellowing plants that wilt in hot weather. Affected plants are stunted or may wilt with yellowing.

TRANSMISSION

Root-knot nematodes are most often introduced in a new field by planting seedlings, tubers or young plants that are already infested.

SPREAD

Root-knot nematodes are spread by running water, farm tools and machinery, as well as by animals.

DAMAGE

Root-knot nematodes cause typical galls on roots. These galls tend to interfere with the flow of fluids and nutrients to the plant and hence the infected plants are less vigorous than the healthy. Severely affected plants wilt and may rot away, leaving a poor root system with a few large galls on the tubers (Figure 78C & D).

IMPACT ON YIELD AND QUALITY

Severe infections by *Meloidogyne incognita* have resulted in reduced yields of between 41.8–88.4% (Akinsanya *et al.*, 2020).

GEOGRAPHICAL DISTRIBUTION

The root-knot nematode *M. incognita* is found worldwide. It is also widespread in Africa, Caribbean and the Pacific (Table 19) and usually occurs in warmer areas. In some countries, *Meloidogyne javanica* is dominant.

Table 19 — Geographical distribution of root-knot nematodes affecting cassava in the ACP countries

AFRICA	CARIBBEAN	PACIFIC
Algeria, Angola, Benin, Botswana, Burkina Faso, Cabo Verde, Cameroon, Central African Republic, The Democratic Republic of Congo, Côte d'Ivoire, Egypt, Eritrea, Ethiopia, Gambia, Ghana, Guinea, Kenya, Liberia, Madagascar, Malawi, Mali, Mauritania, Mauritius, Morocco, Mozambique, Niger, Nigeria, Réunion, Senegal, Seychelles, Sierra Leone, Somalia, South Africa, Sudan, Tanzania, Togo, Tunisia, Uganda, Zambia, Zimbabwe	Antigua and Barbuda, Costa Rica, Dominica, Dominican Republic, El Salvador, Guadeloupe, Mexico, Nicaragua, Panama, Puerto Rico, Trinidad and Tobago, United States	American Samoa, Australia, Fiji, Kiribati, New Caledonia, New Zealand, Niue, Norfolk Island, Papua New Guinea, Samoa, Solomon Islands, Tonga, Tuvalu, Vanuatu

QUARANTINE ORGANISM

As the pest is found worldwide, it is difficult to impose quarantine measures.

FAVOURABLE CONDITIONS

Nematodes prefer sandy soils and are most active in warm weather in moist, but well aerated, sandy soils in the presence of host plants (Olsen, 2011). In addition, infected volunteer plants, monoculture, weed and continuous production of susceptible crops/ or varieties without rotation favour nematode establishment.

PEST LIFE CYCLE

The life cycle of root-knot nematodes has three stages (Figure 79), including the egg, juveniles and adults, which lasts 4–8 weeks. The eggs hatch into juveniles that infect plant roots and take nutrients as they mature, causing the characteristic knots or swellings to form (Olsen, 2011). The juveniles undergo four stages and four moults.

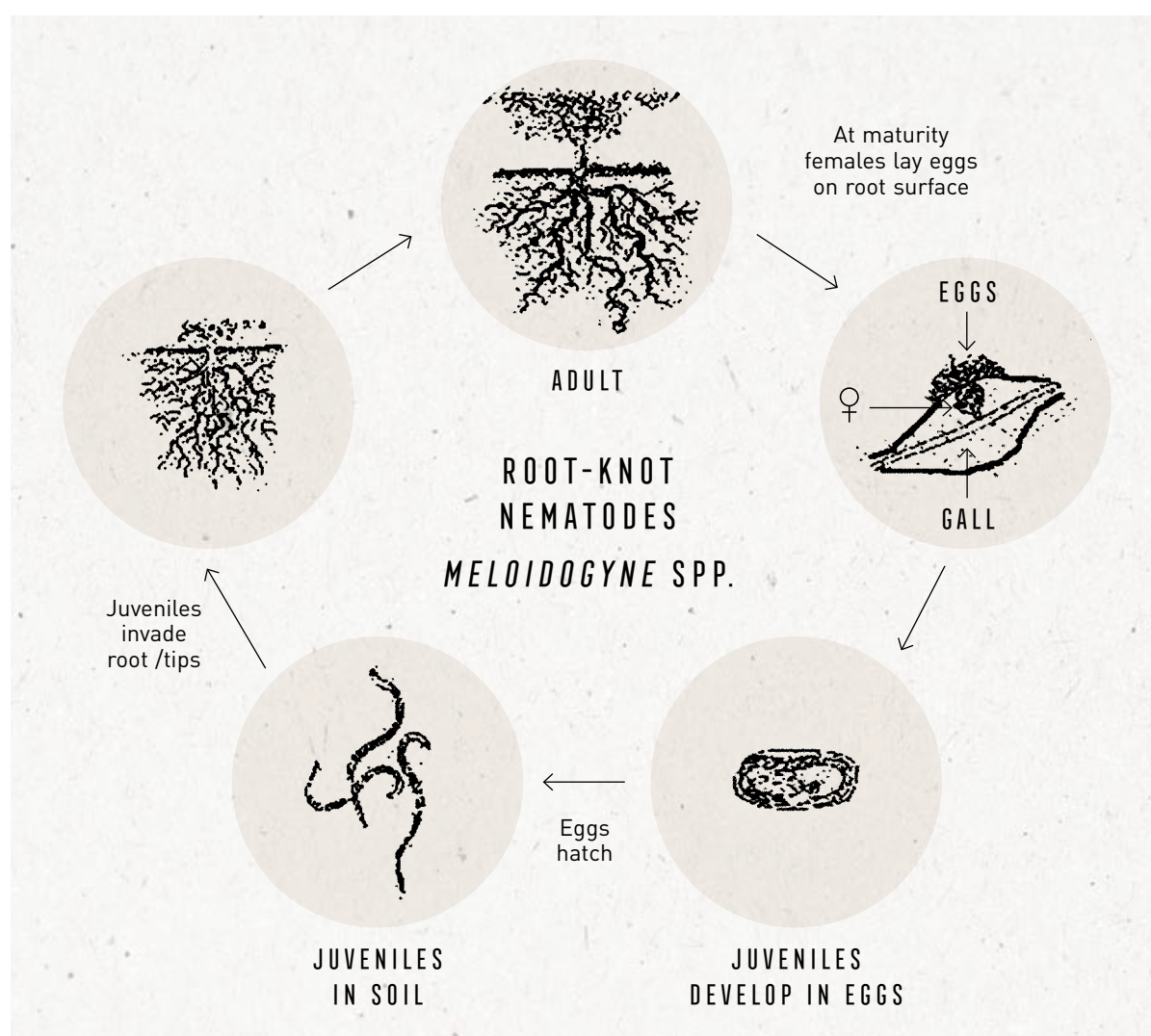


Figure 79 — Life cycle of root-knot nematodes

Source: Michigan State University,

<https://www.canr.msu.edu/resources/nematodes-in-commercial-vegetables>

The 1st stage juvenile develops in the egg, while the 1st first moult occurs within the eggshell, which forms into the 2nd juvenile stage that emerges into the soil or plant tissue. During feeding, the 2nd, 3rd and 4th moults develop, resulting in the 3rd, 4th and 5th or adult stages, respectively. This is followed by development of the reproductive systems in the male and female sexes (The American Phytopathological Society, 2011; Queensland Government, 2017).

SURVEILLANCE/MONITORING

As nematodes are not visible with the naked eye, they can only be detected or counted using a microscope. However, this process is very laborious, costly and may require technical knowledge and techniques not available to everyone. Hence, the number of samples to be collected must be kept to a minimum but must also be representative.

To monitor, collect soil samples randomly with a 2.5 cm-diameter sampling tube for 12–50 cores per plot, depending on the size of the field (Barker and Campbell, 1981; Barker, 1985). Submit the samples to a certified laboratory for nematode detection, identification and analysis. To assess the extent of damage caused at harvest, dig up 10–20 cassava plants, and use the root-knot index rating for galling on the roots, on a 0–10 scale (Zeck, 1971). If high numbers of nematodes are found, apply appropriate control measures.

CULTURAL CONTROL

Use clean planting materials with proven absence of nematode infestation.

- Obtain soil or growth media from sites that are not infested with nematodes.
- Practice crop rotation with non-host crop species such as maize to disrupt the growth cycle of nematodes (The American Phytopathological Society, 2011).
- Plant cover crops such as sudangrass and marigolds, which create unfavourable conditions for nematodes outside the normal growing season. Cover crops also help to stabilise the topsoil and improve soil quality.
- Flooding and exposing fields to sunlight can be used to control nematodes, especially in the dry season (Olsen, 2011).

BIOCONTROL

- Nematophagous (nematode-feeding) fungi such as mycelial traps or sticky spores can be used to capture nematodes.
- The fungi *Pochonia chlamydosporia* and *Paecilomyces lilacinus* parasitize eggs and female root-knot nematode females (Ghahremani *et al.*, 2019).
- *Pasteuria penetrans* and species of *Bacillus* are antagonistic bacteria that can control nematodes.

CONTROL WITH PLANT PROTECTION PRODUCTS

- Fumigants such as dazomet can be used as pre-plant treatments to control root-knot nematodes. However, it is important to cover the soil to avoid rapid spread of fumigant vapour through the air.

OTHER CONTROL METHODS

- Plant resistant varieties.
- Disinfesting by heating the soil to high temperatures of 104° to 130°F (40° to 54°C) will kill nematodes.
- Soil solarisation.

7.3.11. OTHER IMPORTANT ARTHROPOD PESTS

COMMON AND SCIENTIFIC NAMES

- A. Leafcutter ants (*Atta sexdens*, *Atta cephalotes*, *Acromyrmex landolti*)
- B. Cassava shoot flies (*Neosilva perezii*, *Lonchae chalibea*, *Silva pendula*)
- C. Fruitflies (*Anastrepha pickeli*, *Anastrepha manihoti*)
- D. Stemborers (*Chilomima clarkei*)
- E. Cassava lace bug (*Vatiga* spp., *Amblystira machalana*)
- F. Cassava root/Burrower bugs (*Cyrtomenus bergi*, *Pangaeus piceatus*, *Tominotus communis*)
- G. Army worm (*Spodoptera* spp.)
- H. Tiger moth (*Phoenicoprocta sanguinea*, *Agrotis ipsilon*)
- I. Gall midge (*Iatrophobia brasiliensis*)

A. LEAFCUTTER ANTS (*ATTA SEXDENS*, *ATTA CEPHALOTES*, *ACROMYRMEX LANDOLTI*)

SCIENTIFIC NAME

— *Atta sexdens*, *Atta cephalotes*, *Acromyrmex landolti*



Figure 80 — Showing (A) adult leafcutter ants and (B & C) damage caused to plant leaves

Source: <https://cdn.britannica.com/21/188721-050-3D55AD79/leafcutter-ant-species-fungus-substrate-Atta-cephalotes.jpg?w=400&h=300&c=crop>; https://www.futurity.org/wp/wp-content/uploads/2015/10/leaf_cutter_ant_1170.jpg; https://buglord.com/wp-content/uploads/2021/06/how_to_get_rid_of_leaf_cutter_ants.jpg

DESCRIPTION/IDENTIFICATION

PEST

There are 47 known species of leafcutters (also called parasol ants), which are leaf-chewing ants belonging to two genera: *Atta* and *Acromyrmex*. They are among the serious pests of tropical and subtropical America (Discover Life, 2012). Leafcutter ants have spiny reddish-brown bodies and long legs (Figure 80A). The soldier and worker ants each carry out different and precise functions. The ant colonies contain workers of different sizes, ranging 0.3–1.6 cm in length (Boulogne *et al.*, 2014).

SYMPTOMS

The leaves (Figure 80B) and young stems are most affected. The young plants are usually totally defoliated, with the damage resembling pruning. In mature plants only the leaves are removed. The cut pieces litter the ground (Figure 80C) (Boulogne *et al.*, 2014).

DISPERSAL AND SPREAD

Leafcutter ants move in a trail to suitable sites with new foliage and a favourable climate.

DAMAGE

In large numbers, leafcutter ants can destroy an entire crop overnight by cutting portions of a leaf in a characteristic crescent-shaped pattern. They then carry the cut portions with their jaws to their nest. In severe cases, the ants can completely defoliate plants.

IMPACT ON YIELD AND QUALITY

Atta spp. are major pests of many crops and plantations in Latin America, where they are considered to be among the five worst pests (Cherrett and Peregrine, 1976). Leafcutters can cause 20–30% yield loss of the affected plants (Cherrett and Simms, 1968).

GEOGRAPHICAL DISTRIBUTION

Leafcutter ants mostly affect cassava crops in the Caribbean (Table 20) and have not been reported to affect cassava in Africa and the Pacific.

Table 20 — Geographical distribution of leafcutters affecting cassava in the ACP countries

AFRICA	CARIBBEAN	PACIFIC
Not reported	Belize, Brazil, Carriacou, Costa Rica, Cuba, Curaçao, El Salvador, French Guyana, Grenada, Guadeloupe, Guatemala, Guyana, Honduras, Mexico, Nicaragua, Panama, Surinam, Trinidad and Tobago, Venezuela	Not reported

QUARANTINE ORGANISM

Ants are widely distributed globally. Nevertheless, efforts must be made to ensure that produce and plant/crop materials are checked for ants to avoid introducing new species to areas where they may not presently occur.

FAVOURABLE CONDITIONS

As social insects, ants are flexible to changing environmental conditions (Arenas and Rocés, 2017).

PEST LIFE CYCLE

The ants' life cycle takes from several weeks to several months, depending on the species and environment (Terminix, 2021). Ants go through metamorphosis– a complete change in body form – in four stages: egg, larva, pupa and adult.

1. Egg. The ant life cycle begins with an egg. Fertilised eggs become female ants, while unfertilised eggs become male ants.
2. Larva. The tiny egg hatches into a worm-shaped larva without eyes or legs. Ant larvae eat constantly, relying on adult workers to feed them. Larvae grow quickly, moulting several times as they get bigger.
3. Pupa. When the larva is big enough, it metamorphoses into a pupa. Pupae look more like adults, but their antennae and wings are folded against their bodies. In some species, the pupa spins a cocoon around itself for protection. In other species, the pupa remains uncovered. The pupal stage is one of rest and reorganisation. During this time, the pupa changes into an adult.
4. Adult. Finally, the pupa emerges as an adult. Their hard exoskeleton prevents them from getting any larger. Adult ants belong to one of three castes: queens, female workers or males.

SURVEILLANCE/MONITORING

To monitor for leafcutter ants, look for leaves damaged by cutting and the associated insects. Assess the type and number of the insects and apply appropriate control measures.

CULTURAL CONTROL

Change land use, for example by interchanging grasses with shade crops (Lewis and Norton, 1973; Lewis and Phillips, 1973).

- Look for trails of the leafcutter ants and locate their nest at night when they are most active.
- Mark areas with established nests by use of painted sticks or cloth.
- Set up barriers to prevent entry of leafcutter ants into the crop.
- Set up formicide baits that contain an attractive substrate and an insecticide to trap leafcutter ants (Lewis and Norton, 1973; Lewis and Phillips, 1973; De Britto *et al.*, 2016).

BIOCONTROL

- Pathogenic fungi such as *Beauveria bassiana* and *Metarrhizium anisopliae* may be useful in control of leafcutter ant nests (Kermarrec *et al.*, 1986).
- Arboreal ants were reported to protect citrus and cocoa trees against attack by *Atta* spp. (Leston, 1978; Jutsum *et al.*, 1981).
- A parasite fungus, *Escovopsis*, which attacks the leafcutter ants can be used as a control strategy (Boyd-Rice, 2015).

CONTROL WITH PLANT PROTECTION PRODUCTS

None approved for commercial use.

OTHER CONTROL METHODS

- Plant species or clones with resistance to attack by leaf cutter ants (Laranjeiro, 1994).

B. CASSAVA SHOOT FLIES

(*NEOSILBA PEREZI*, *LONCHAEID CHALIBEA*, *SILBA PENDULA* BEZZ)



Figure 81 — Showing adult cassava shoot fly (A); (B & C) damage they cause to cassava shoots

Source: Infonet biovision, <https://infonet-biovision.org/PlantHealth/MinorPests/Shoot-fly-0>;
CABI Plantwise Knowledge Bank, <https://www.plantwise.org/KnowledgeBank/pmdg/20167800984>;
Iain MacGowan, <https://lonchaeidae.myspecies.info/file-colorboxed/115>

DESCRIPTION/IDENTIFICATION

PEST

Cassava shoot flies are small flies measuring about 0.5–0.66 cm – the size of a house fly. They have larvae that feed solely on the shoots of cassava plants. The flies are a shiny black colour with dark-brown antennae and brownish black legs (Figure 81A) (Waddill and Weems, 1978; Bellotti, 2002; Hillocks *et al.*, 2002).

SYMPTOMS

Affected shoots have whitish larvae that kill the apical buds (those at the top of the plant), slow the growth of young plants and induce the growth of lateral (side) buds. Larvae produce a yellowish secretion that is visible by the naked eye (Gislotti and do Prado, 2011).

DISPERSAL AND SPREAD

Adult cassava shoot flies can fly, while larvae can be spread on planting materials.

DAMAGE

The females lay eggs in the apical leaves of the shoots, or in small holes made with the ovipositor. The white-coloured larvae make holes in the soft tissues of the plants, during which they damage the terminal buds (Figure 81B & C), leading to retarded plant shoot growth (Boza and Waddill, 1977; Gislotti and do Prado, 2011) and development of lateral budding and branches (Bellotti and van Schoonhoven, 1978; King and Saunders, 1984; Hogue, 1993).

IMPACT ON YIELD AND QUALITY

Damage caused by the larvae of *Neosilba perezii* to cassava crops is restricted to cassava shoots and stems and does not significantly impact leaf and tuberous root yield (Boza and Waddill, 1977; Lozano *et al.*, 1981; Bellotti, 2002).

GEOGRAPHICAL DISTRIBUTION

The distribution of *N. perezii* is restricted to the Americas and the Caribbean (Table 21) (Hershey, 1987; Bellotti *et al.*, 1999; McAlpine and Steyskal, 1982; Bellotti, 2004).

Table 21 — Geographical distribution of shoot flies affecting cassava in the ACP countries

AFRICA	CARIBBEAN AND SOUTH AMERICA	PACIFIC
Not reported	Antigua and Barbuda, Brazil, Dominica, Dominican Republic, Grenada, Guadeloupe, Haiti, Jamaica, Martinique, Puerto Rico, Saint Vincent and the Grenadines, Trinidad and Tobago, Suriname, Venezuela	Not reported

QUARANTINE ORGANISM

Cassava shoot flies are widespread in the Americas but are not well reported on in sub-Saharan Africa and the Pacific. Therefore, restrictions should be put in place to control the movement of cassava materials from the infected areas to new areas with no record of the pest.

FAVOURABLE CONDITIONS

Younger plants are more likely to be infested by shoot flies than the older plants (Gislotti and do Prado, 2011). The pest is more prevalent in warm than cold weather (Waddill and Weems, 1978).

PEST LIFE CYCLE

Shoot fly females lay eggs inside the leaves of the apical part of the shoot, or in small cavities made using their ovipositors (Boza and Waddill, 1977).

SURVEILLANCE/MONITORING

Look for cassava shoot flies (*N. perezii*) every two weeks, starting from 60 days after planting, when the buds are fully formed and oviposition by *N. perezii* is taking place. Monitor the cassava crop for 7 months to assess the infestation. Count and record the number of plants with damaged buds that have a yellowish secretion and the number of cassava shoot fly larvae.

CULTURAL CONTROL

Management of cassava shoot flies is unwarranted due to the insignificant impact of the pest on cassava leaves and tubers, which are the economic components of cassava.

BIOCONTROL

At least 24.36% of the larvae of *Neosilba perezii* shoot flies were parasitised by a parasitoid wasp, *Phaenocarpa neosilba*, in Brazil (Gislotti and Prado, 2012).

CONTROL WITH PLANT PROTECTION PRODUCTS

None applicable.

C. FRUIT FLIES (*ANASTREPHA PICKELI*, *ANASTREPHA MANIHOTI*)

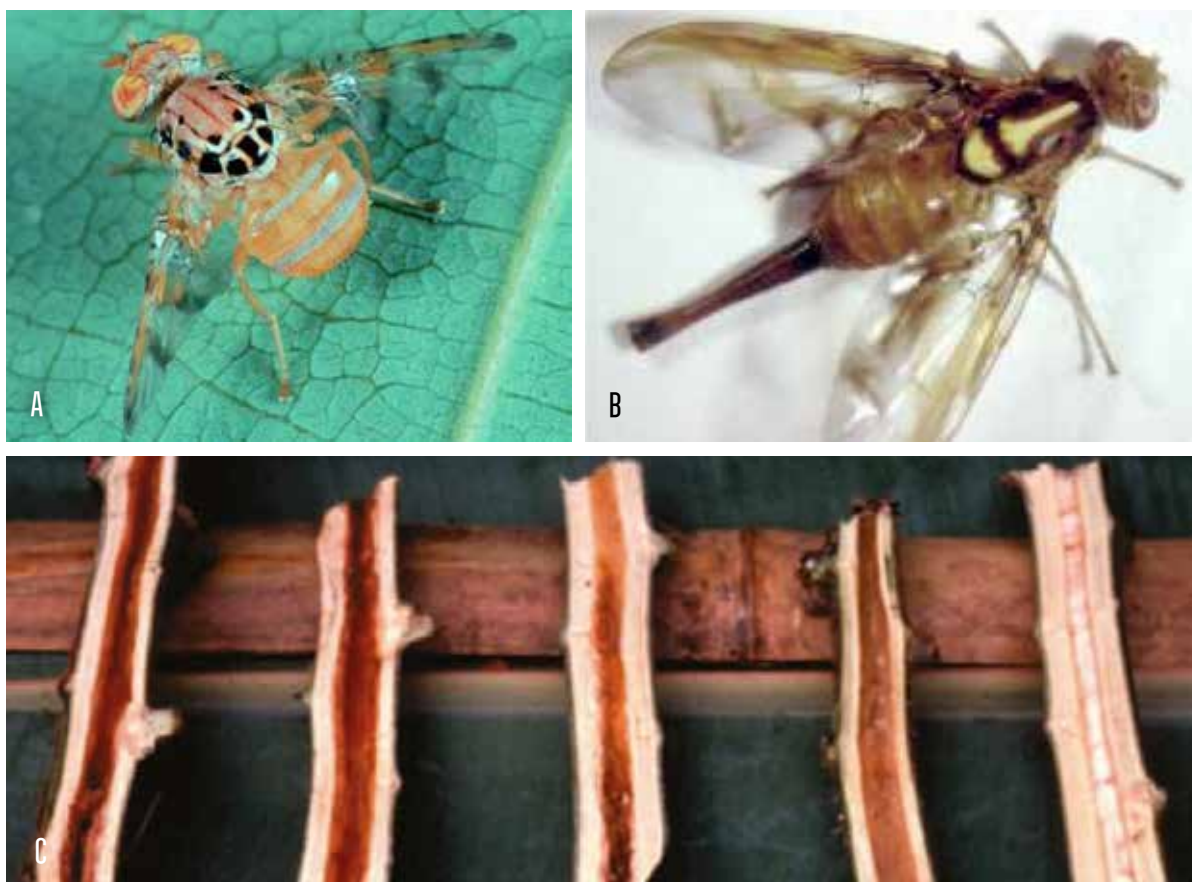


Figure 82 — Showing (A & B) adult fruit flies and (C) damage they cause to cassava stems
Source: Infonet biovision, <https://infonet-biovision.org/PlantHealth/Pests/Fruit-flies>;
file:///C:/Users/Owner/Downloads/Practical_Handbook%20(1).pdf

DESCRIPTION/IDENTIFICATION

PEST

Fruit flies are very small oval-shaped flies. Adults grow to around 0.3 cm in length with brightly coloured brown and yellow spotted wings (Figure 82A). They have a tan thorax and a black abdomen with a grey underbelly. Fruit flies usually have red eyes, but some can be dark, while the antennae have feathery bristles. Fruit flies have transparent wings with yellowish coffee-coloured bands. The female's abdomen has extended ovipositors (Figure 82B), while the male has a rounder abdomen (Pena and Bellotti, 1977; Bellotti *et al.*, 2012).

SYMPTOMS

Affected plants develop punctures, which appear necrotic, usually made by females during egg laying. In mature cassava plants, affected stems develop light to dark brown pith (Figure 82C), with a watery appearance due to the development of a bacterium, *Erwinia carotovora* (Pena and Bellotti, 1977; Bellotti *et al.*, 2012).

DISPERSAL AND SPREAD

Fruit fly adults can move long distances with the aid of wind. The pest may also be transported in planting materials and vegetation (cassava leaves) by humans.

DAMAGE

Fruit flies cause severe damage to cassava plants by puncturing and making tunnels in the stems, forming brown corridors in the pith, which could result in stem rot.

IMPACT ON YIELD AND QUALITY

Sprouting of cassava planting materials obtained from affected plants is greatly reduced due to loss of the capacity to produce viable shoots. However, when the infestation is limited, there is no risk of significant economic losses (Bellotti *et al.*, 2012).

GEOGRAPHICAL DISTRIBUTION

Fruit flies occur widely in Africa and the Caribbean (Table 22) on several plants, including cassava (Bellotti *et al.*, 2012), but have not been reported in the Pacific.

Table 22 — Geographical distribution of fruit flies affecting cassava in the ACP countries

AFRICA	CARIBBEAN	PACIFIC
Angola, Benin, Cameroon, Central African Republic, Democratic Republic of the Congo, Republic of the Congo, Côte d'Ivoire, Equatorial Guinea, Gabon, Gambia, Ghana, Guinea, Guinea-Bissau, Kenya, Liberia, Madagascar, Malawi, Mali, Mozambique, Niger, Nigeria, Rwanda, Senegal, Sierra Leone, South Africa, Sudan, Tanzania, Zanzibar Island, Togo, Uganda, Zambia, Zimbabwe	Antigua and Barbuda, Costa Rica, Dominican Republic, El Salvador, Guadeloupe, Mexico, Nicaragua, Panama, Puerto Rico, Trinidad and Tobago, United States	Not reported

QUARANTINE ORGANISM

There should be restrictions on movement of planting materials from infected areas, as they may transport the fruit flies to unaffected areas. Fruit flies have a wide host range and risk becoming economic pests on new plants (including crops) in the new areas of introduction.

FAVOURABLE CONDITIONS

Fruit flies thrive under precipitation/in the rainy season and in humid conditions brought about by crowded canopies and too much litter on the ground (Bellotti *et al.*, 2012).

PEST LIFE CYCLE

Eggs are laid under the skin of fruits or stems in groups of 3 to 8 eggs, depending on the species. The eggs hatch within 1–2 days. The larvae, which are white maggots, feed on the flesh, causing fruit or stem rot (Figure 83). After 4–17 days the maggots leave the fruit/stem through holes in the skin and drop to the ground to pupate. The pupae, which are white, brown or black and 4–12 mm long, develop in the soil, 2–5 cm beneath the host plant.

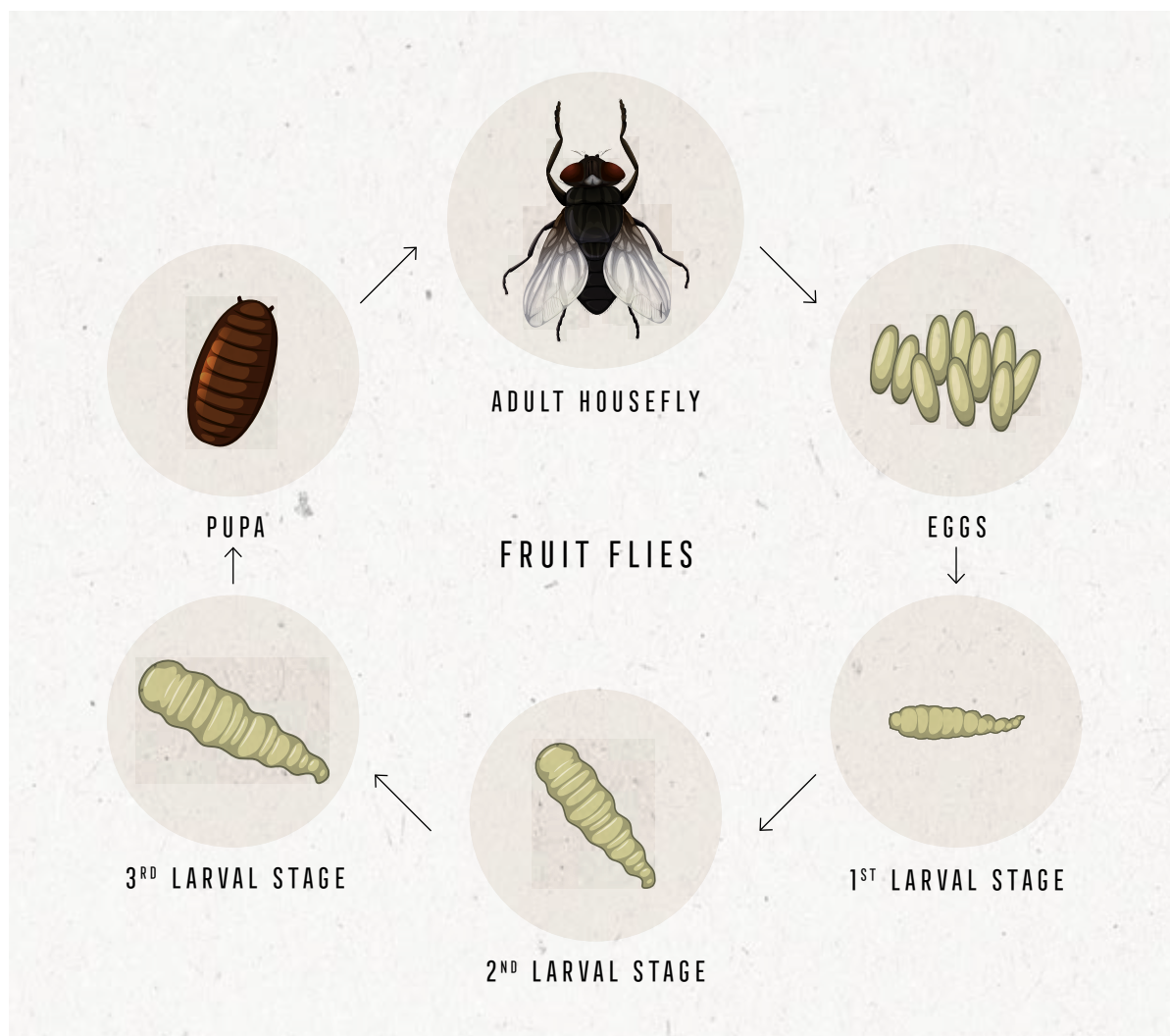


Figure 83 — Life cycle of a fruit fly

Source: Infonet biovision, <https://infonet-biovision.org/PlantHealth/Pests/Fruit-flies>

Adult flies emerge from the pupae 10–20 days after pupation, depending on climatic conditions. After oviposition, hatching takes place in the fruit (if available) but if not, the female looks for tender tissue in the stem where it lays the eggs. *A. pickeli* fruit flies live for an average of 39.5 days (Pena and Bellotti, 1977; Bellotti *et al.*, 2012).

SURVEILLANCE/MONITORING

Trap baits and male lures can be used to monitor fruit fly numbers, changes in populations and the impact on crops, as well as the timing and effectiveness of control measures. Lay at least three traps with protein or sugar baits or male lures (para-pheromones) randomly in the field. The protein traps are attractive to both males and females of most fruit fly species but may not capture all. However, the male lures are designed to capture specific fruit fly species and are more effective. The data is important in forecasting devastating invasions and guiding when to apply IPM strategies (Pena and Bellotti, 1977).

CULTURAL CONTROL

Select healthy plants with white piths, indicating freedom from fruit fly infestation.

- Change type of land use, such as planting shade trees to create unfavourable conditions for the survival of pests such as fruit flies near cassava crops.
- Practice sanitation to remove materials that serve to propagate the development of the pest.
- Protect cassava plants by banding the stems with grease or polyisobutylene gel to avoid fruit flies settling and piercing the stems.
- Use barriers crops such as sorghum or maize to deter fruit fly entry into young cassava fields. However, the sorghum or maize must be planted at least 1-2 months earlier than the cassava crop.
- Sugar baits such as hydrolyzed maize, balsamic and red wine vinegar (Enkerlin and Reyes-Flores, 2018) can be treated with pesticide to trap and kill the fruit flies. However, care must be exercised to limit the destruction of beneficial insects such as bees and others.

BIOCONTROL

- *Anastrepha manihoti* populations were controlled using the parasitoid wasps *Utetes anastrephae* and braconid entoparasitoid in Brazil (Leonel *et al.*, 1995).

CONTROL WITH PLANT PROTECTION PRODUCTS

- Use appropriate registered and approved pesticides for fruit fly management.

OTHER CONTROL METHODS

- Plant pest-resistant varieties if available.

D. STEM BORERS (*CHILOMIMA CLARKEI*)



Figure 84 — Showing (A) adult moth; (B) mature larva; (C & D) cassava stem with symptoms of infestation by larvae of stem borers; (E & F) damage due to pupa inside the stem
Source: Adilson Lopes Lima in Adaime et al. (2020) [https://www.semanticscholar.org/paper/First-record-of-Chilomima-clarkei-\(Amsel\)-in-Crantz-Adaime-Santos/537b9eac1b8b2ad9fb8f6a88a1b79a83d35b2221](https://www.semanticscholar.org/paper/First-record-of-Chilomima-clarkei-(Amsel)-in-Crantz-Adaime-Santos/537b9eac1b8b2ad9fb8f6a88a1b79a83d35b2221)

DESCRIPTION/IDENTIFICATION

PEST

Adults of stem borers of *Chilomima clarkei* develop into butterflies (Figure 84A), with white-, yellow- or cinnamon-coloured larvae (Figure 84B) which vary in size and can be as long as 30 mm. The adult has antennae that are longer than its body, are filiform and light brown in colour with 11 segments. Adult males are 1.60 cm long by 0.72 cm wide, while females have average body length of 1.64 cm and 0.69 cm wide. Mouth parts are used for masticating (Bellotti *et al.*, 2012). The adults lay eggs on the stems around the lateral buds of leaves that have already fallen. The first four larva instars feed around the buds. The life cycle varies from 62 to 68 days.

SYMPTOMS

Stem borers feed on cassava stems and branches, causing substantial damage mostly to adult cassava plants. Affected plants have visible holes when the stems and branches are cut or broken open (Figure 84C & D). In the dry season the affected stems and branches lose their leaves and may die (Bellotti *et al.*, 2012).

DISPERSAL

The pest is spread very rapidly through movement of planting materials between locations and among farmers (Bellotti *et al.*, 2012).

DAMAGE

The larvae are the destructive stage, that feed on the buds of cassava and by penetrating the stem to eat away the pith, making tunnels (Figure 84E & F) that weaken the stem and branches. As a result, plants may lose their leaves and die (Bellotti *et al.*, 2012).

IMPACT ON YIELD AND QUALITY

Severe infestation by the pest (e.g., >20 insects per stem) causes perforations in the stem leading to breaking, which reduces the quality and quantity of planting materials. In addition, severe infestation significantly reduces tuberous yield (by 45%–62%) in field crops (Bellotti *et al.*, 2012).

GEOGRAPHICAL DISTRIBUTION

Stem borers occur widely in South America in Colombia, Paraguay and Venezuela, and in the Caribbean in Trinidad and Tobago (Table 23), where they cause high economic damage on cassava stems and yield loss (60%) (Schotman, 1989).

Table 23 — Geographical distribution of stem borers on cassava in the ACP countries

AFRICA	CARIBBEAN	PACIFIC
Not reported	Colombia, Trinidad & Tobago, Paraguay and Venezuela	Not reported

QUARANTINE ORGANISM

The pest is not widely distributed, so restrictions should be imposed on the movement of infected cassava planting materials to control its spread.

FAVOURABLE CONDITIONS

Adults are most active in the dry season and less active in the cool months. The effect is worse during the dry season, where affected stems and branches lose their leaves and may die. Four to six cycles of the pest may occur during a cropping year, potentially increasing damage and making control much more difficult (Bellotti *et al.*, 2012).

PEST LIFE CYCLE

Not reported (fairly new pest on cassava)

SURVEILLANCE/MONITORING

Look for larvae or adults of the stem borer *Chilomima clarkei* in cracked parts of cassava stems and branches, remove and count them. If infestation is high, many plants will show typical symptoms with the young plants dying off. Implement IPM measures if the number of infested cassava plants exceeds a few plants.

CULTURAL CONTROL

Remove and burn infested planting cassava materials and plant residues.

- Plant healthy undamaged stakes.
- Intercrop with maize to reduce stem borer populations until the maize is harvested (Gold *et al.*, 1990).

BIOCONTROL

- The eggs are parasitised by the wasp *Trichogramma microhymenopterans*; and larvae by Bracon wasps, *Brachymeria conica* and *Apanteles* sp.
- Applications of the bacterium *Bacillus thuringiensis* and the fungus *Spicaria* sp., were sprayed over the *Chilomima clarkei* larvae and attained mortality rates of 99%, 88%, and 100%, respectively for the stem borer.

CONTROL WITH PLANT PROTECTION PRODUCTS

- Malathion with “polyspray” caused 100% larval mortality (Bellotti *et al.*, 2012).

OTHER CONTROL METHODS

- May use pest resistant varieties (where available).

E. CASSAVA LACE BUG (*VATIGA* SPP., *AMBLYSTIRA MACHALANA*)

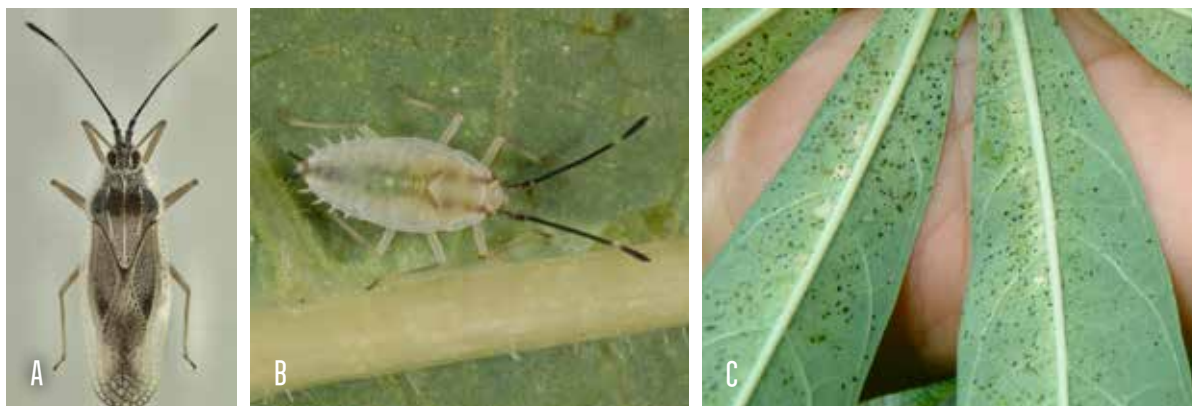


Figure 85 — Adult cassava lace bug (A); nymph (B); and damage caused on cassava leaves (C)
Source: Susan Halbert, https://www.researchgate.net/figure/Adult-cassava-lacebug-Photo-credit-Michael-Thomas_fig2_242684537

DESCRIPTION/IDENTIFICATION

PEST

The adult cassava lace bug is an elongate brown bug measuring 3–4 mm long, while the nymphs are spiny and pale. Adult *Vatiga* are grey and measure about 3 mm long and 1 mm wide (Figure 85A), while older nymphs have dark antennae with white bands near the tips (Figure 85B). The nymph is white and a little smaller than the adult. Both adults and nymphs live in large numbers on the underside of leaves (Neal and Schaefer, 2000; Halbert, 2010; Bellotti *et al.*, 2012).

There are two species (Halbert, 2010) that damage cassava: *Vatiga illudens*, which has two small horns on the front of the head and *Vatiga manihotae*, which has only one horn in the middle of the front of the head (Froeschner, 1993). Occasionally, *V. illudens* specimens have only one horn, with a single horn not in the middle of the head. The proximal antennal segments of *V. manihotae* are relatively longer than those of *V. illudens* (Bellotti *et al.*, 2012). Due to their predatory nature, lace bug larvae are known as ‘aphid lions’, while the adults mainly feed on nectar, pollen and aphid honeydew.

SYMPTOMS

Lace bugs usually occur in clusters underneath the leaves, where their feeding results in leaf mottling and necrosis; in the severe cases, the leaves fall off. Leaves become stippled in the middle of the leaf near the veins or on top and have dark spots (Figure 85C) on the undersides (Halbert, 2010).

DISPERSAL

Adults fly among and between plants to find suitable hosts on which they feed and develop (Bellotti *et al.*, 2012).

DAMAGE

Damage is caused to the leaves that is similar to that made by mites and includes small white spots of star-like appearance, which later turn reddish-brown. The damage can be differentiated from that made by mites by the black points underneath the leaves, which are faeces from the bugs. Foliage is damaged or defoliated leading to extensive reduction of photosynthesis and reduced plant growth and yield (Bellotti *et al.*, 2012).

IMPACT ON YIELD AND QUALITY

Leaf damage by cassava lace bugs can be severe, but with minimal impact on yield of marketable roots (Halbert, 2010).

GEOGRAPHICAL DISTRIBUTION

Lace bugs have only been reported on cassava in the Caribbean (Table 24) (Halbert, 2010).

Table 24 — Geographical distribution of lace bugs affecting cassava in the ACP countries

AFRICA	CARIBBEAN	PACIFIC
Not reported	Antigua and Barbuda, Barbados, Dominica, Grenada, Guadeloupe, Martinique, Montserrat, Saint Kitts and Nevis, Saint Lucia, Saint Vincent and the Grenadines, Trinidad and Tobago	Not reported

QUARANTINE ORGANISM

The pest has been reported mainly in the Americas and hence restrictions should be imposed on the movement of infected cassava planting materials to areas where it is not currently found.

FAVOURABLE CONDITIONS

Both *V. illudens* and *V. manihotae* attack cassava mainly during dry seasons, which worsens in seasons with prolonged droughts (Bellotti *et al.*, 1999; 2012; Halbert, 2010).

PEST LIFE CYCLE

The lace bugs go through three life stages – egg, nymph and adult – with several generations a year. Females insert tiny, oblong eggs in leaf tissue and cover them

with dark excrement. Nymphs develop through about five, increasingly larger, instars (growth stages) over a period of weeks before maturing into adults. Lace bugs can overwinter as eggs in leaves on evergreen hosts and as adults in protected locations, such as under bark plates and fallen leaves and other debris beneath host plants. All life stages can be present throughout the year on evergreen hosts in areas with mild winters (Bellotti *et al.*, 2012).

SURVEILLANCE/MONITORING

To monitor the population of lace bugs before they begin to cause damage and get out of control. Look for the symptoms such as damage on the upper side of leaves similar to that caused by spider mite feeding damage. Flip the leaves and use a hand lens to examine if there are black spots that confirm damage due to lace bugs.

CULTURAL CONTROL

Good field sanitation should clear the field of weeds and plant residues from previous crops.

- Practice integrated pest management.

BIOCONTROL

- Although biological control of lace bugs is difficult (Bellotti *et al.*, 1999), parasitic wasps, predatory assassin bugs, lace-wing larvae, lady beetles, jumping spiders, pirate bugs and mites were reported to reduce the populations.
- Lace-wings are biocontrol agents (predators) that have high potential for use in pest management (Mansoor *et al.*, 2013) for a wide range of pests, including aphids, chinch bugs, mealybugs, scales, whiteflies, leafhoppers, lepidopterous eggs and larvae, and mites (Hydorn, 1971; Satpathy *et al.*, 2012; Mansoor *et al.*, 2013).

CONTROL WITH PLANT PROTECTION PRODUCTS

- Continuous use of insecticides is expensive and may destroy the natural enemies of other pests.
- Deltamethrin, and lambda-cyhalothrin, act on the nervous system of insects, and are recommended for chemical control of cassava lace bugs.

OTHER CONTROL METHODS

- Preliminary studies and evaluations made in the cassava germplasm bank held at CIAT indicate that varietal resistance may exist, but that more research is needed to develop the technology (Bellotti *et al.*, 2012).

F. CASSAVA ROOT/BURROWER BUGS (*CYRTOMENUS BERGI*)

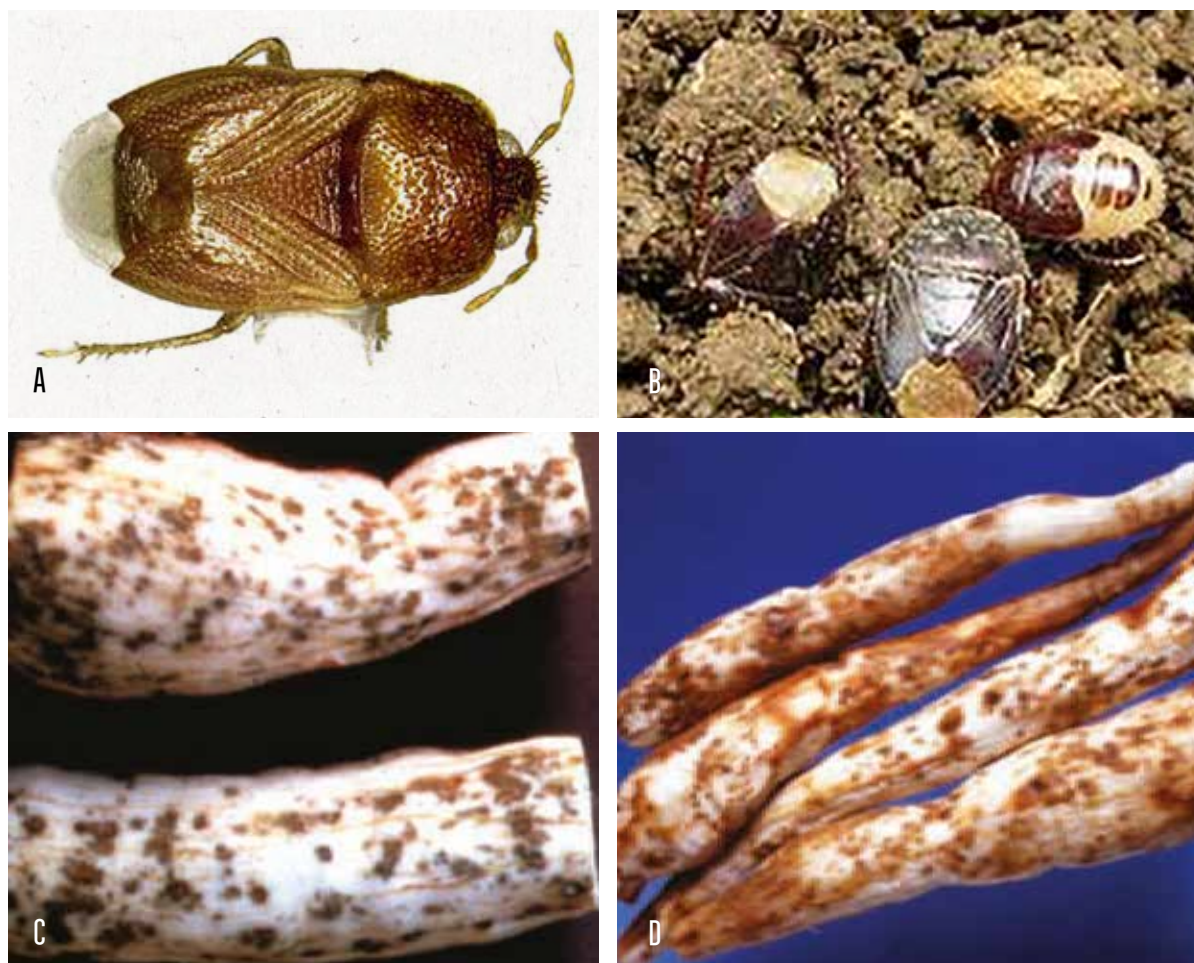


Figure 86 — Showing (A & B) adult cassava burrower bugs and (C) damage caused on peeled cassava tubers
Source: Schwertner and Nardi, https://link.springer.com/chapter/10.1007/978-94-017-9861-7_21;
file:///C:/Users/Owner/Downloads/Practical_Handbook%20(1).pdf

DESCRIPTION/IDENTIFICATION

PEST

Cyrtomenus bergi is an arthropod pest (Figure 86A & B) that feeds directly on cassava root and tubers causing a characteristic symptom called smallpox disease (Figure 86C) due to microbial rotting following feeding of the pest.

SYMPTOMS

Upon harvesting and peeling the tubers, there is a characteristic discolouration of coffee-black lesions (Figure 86C) known in Spanish as “chinche de la viruela”, meaning “smallpox bug” (Bellotti *et al.*, 2012).

DISPERSAL

Adults can move short distances and fly between plants, while the nymphs may move with cassava during transportation by man (Bellotti *et al.*, 2012).

DAMAGE

Nymphs and adults feed on the tubers, penetrating the peel, which allows soil-borne pathogens such as certain fungal species to enter the root cells (Arias and Bellotti, 1985; Bellotti and Riis, 1994). The entry of these pathogens causes coffee-black lesions, which appear in the roots 24 hours after the feeding of the pest. However, the damage cannot be detected until harvesting and peeling the roots (Bellotti *et al.*, 2012).

IMPACT ON YIELD AND QUALITY

Pest attack causes between 70% and 80% damage to tuberous roots, with 50% starch content reduction (Bellotti *et al.*, 2012).

GEOGRAPHICAL DISTRIBUTION

Cassava root burrower bugs occur in the Caribbean (Table 25) (Bellotti *et al.*, 2012) and have not been reported in Africa and the Pacific.

Table 25 — Geographical distribution of root burrower bugs affecting cassava in the ACP countries

AFRICA	CARIBBEAN	PACIFIC
Not reported	Costa Rica, Trinidad and Tobago, South America, Colombia	Not reported

QUARANTINE ORGANISM

Cassava root/burrower bugs occur in a few countries in South Americas and the Caribbean. Therefore, quarantine restrictions should be imposed on the movement of infected cassava tubers and soil to none-infested areas.

FAVOURABLE CONDITIONS

Populations of *C. bergi* are present in the soil throughout the cropping cycle, with the bug being strongly attracted to moist soils. Adults migrate when soil moisture content is less than 22% and will remain in the soil with moisture content of 31%. The rainy season favours the development and survival of adults and nymphs and dispersion (Bellotti *et al.*, 2012).

PEST LIFE CYCLE

Cyrtomenus bergi undergoes five nymphal instars. The nymphs and adults may live for more than 1 year, feeding on cassava roots (García and Bellotti, 1980). The adult life span was 66 in sweet cassava (CMC 40) and 64 days in bitter cassava (Bellotti *et al.*, 2012).

SURVEILLANCE/MONITORING

Regular check the soil for burrower bugs and count their number to keep them under check before they begin to cause economic damage to cassava crops. As damage cannot be recognised until after peeling the cassava, it is always advisable to control the population early in the crop's development.

CULTURAL CONTROL

Controlling *C. bergi* is difficult because of its polyphagous nature and adaptation to different soil environments.

BIOCONTROL

None applicable

CONTROL WITH PLANT PROTECTION PRODUCTS

- Use of chemical pesticides may lower pest populations and reduce damage. However, this would require frequent applications, which is expensive and damaging to the environment (Bellotti *et al.*, 2012).

OTHER CONTROL METHODS

- Natural resistance was confirmed in 15 varieties with low cyanogenic potentials (CNP) (Bellotti *et al.*, 2012).

G. ARMY WORM (*SPODOPTERA* spp.: *SPODOPTERA FRUGIPERDA*,
SPODOPTERA ERIDANIA, *SPODOPTERA SUNIA*)



Figure 87 — Army worm (caterpillar) (A), Moth (B), and damage to leaves (C)

Source: Infonet biovision, <https://infonet-biovision.org/PlantHealth/Pests/African-armyworm>;
<https://extension.entm.purdue.edu/fieldcropsipm/insects/corn-armyworm.php>; <https://blog.invasive-species.org/2018/07/26/southern-armyworm-identified-from-west-and-central-africa/>

DESCRIPTION/IDENTIFICATION

PEST

Army worm (*Spodoptera* spp.) is a lepidopteran pest that has a dark head with a pale Y-shaped marking that is upside-down and four elevated spots that look like a trapezium on each body segment when seen from above (Figure 87A). There are four spots that make a square on the second-to-last body segment. The skin is smooth when touched.

Moths have a wingspan of 32–40 mm, with the male moth having forewings that are shaded grey and brown with triangular white spots at the tip (Figure 87B) and near the centre of the wing. In contrast, the forewings of females are less distinctly marked, ranging from a uniform greyish brown to a fine mottling of grey and brown. The hindwing is silver-white with a narrow dark border in both sexes. Adults are nocturnal, and are most active during warm, humid evenings (Navasero *et al.*, 2019; Ramzan *et al.*, 2021).

SYMPTOMS

Affected plants have ragged-looking leaves due to intense feeding (Figure 87C) by the pest, causes damage similar to that of grasshoppers (Navasero *et al.*, 2019).

DISPERSAL

Moths are active in the night in warm and humid seasons. The caterpillars (army worms) can be spread on planting materials by people, while the moths can fly between plants and locations.

DAMAGE

Army worms feed in large numbers on crop leaves, stems and reproductive parts of more than 350 plant species. The young army worm larvae cause the most damage to the leaves/foilage of crops. Under high infestation, the larvae also burrow into the growing points (buds) of plants, which retards plant growth and affects yield (Navasero *et al.*, 2019).

IMPACT ON YIELD AND QUALITY

Army worm attacks of mean densities of 0.2 to 0.8 larvae per plant could reduce crop yield by 5–20% (James, 2017).

GEOGRAPHICAL DISTRIBUTION

Army worms are widely distributed in the ACP countries (Table 26) where they have also affected cassava production (James, 2017).

Table 26 — Geographical distribution of army worms on cassava in the ACP countries

AFRICA	CARIBBEAN	PACIFIC
Angola, Benin, Botswana, Burkina Faso, Burundi, Cabo Verde, Cameroon, Central African Republic, Chad, Democratic Republic of Congo, Republic of the Congo, Côte d'Ivoire, Egypt, Equatorial Guinea, Eritrea, Eswatini, Ethiopia, Gabon, Gambia, Ghana, Guinea, Guinea-Bissau, Kenya, Liberia, Madagascar, Malawi, Mali, Mayotte, Mozambique, Namibia, Niger, Nigeria, Réunion, Rwanda, São Tomé and Príncipe, Senegal, Seychelles, Somalia, South Africa, South Sudan, Sudan, Tanzania, Togo, Kara and Plateau regions, Uganda, Zambia, Zimbabwe	Argentina, Bolivia, Brazil, Chile, Colombia, Ecuador, French Guyana, Guyana, Paraguay, Peru, Puerto Rico, Saint Kitts and Nevis, Saint Lucia, Saint Vincent and the Grenadines, Trinidad and Tobago, Uruguay, Venezuela	American Samoa, Australia, Christmas Island, Cook Islands, Federated States of Micronesia, Fiji, French Polynesia, Guam, Kiribati, Marshall Islands, New Caledonia, New Zealand, Niue, Norfolk Island, Northern Mariana Islands, Palau, Papua New Guinea, Pitcairn, Samoa, Solomon Islands, Tokelau, Tonga, Tuvalu, Vanuatu, Wallis and Futuna

QUARANTINE ORGANISM

Army worms are native to the Americas and have recently spread to Africa (in 2016), where they have caused significant damage mainly to maize crops. The pest is on the EPPO A1 list of quarantine pests and is intercepted in Europe, where strict measures are currently in place to control the entry of imported plant materials from infested countries (Seymour *et al.*, 1985; EUROPHYT_ https://ec.europa.eu/food/plants/plant-health-and-biosecurity/europhyt_en).

FAVOURABLE CONDITIONS

Army worms develop and thrive at temperatures of 11–30°C, with the optimum temperature being 28°C. At lower (cooler) temperatures than these, the pest tends to reduce the number of generations per year. Heavy rains can wash away eggs and young caterpillars from leaves, which reduces the populations (Navasero *et al.*, 2019; Ramzan *et al.*, 2021).

PEST LIFE CYCLE

The life cycle undergoes four stages (Figure 88). The female moths lay 100–200 spherical cream, grey or whitish eggs underneath of leaves and often on top of the leaves when the numbers of the pest are high. A moth produces at least 1,500 eggs in her lifetime. The egg stage takes 2–3 days in warm weather to develop to the next stage.

- After 3–5 days, the eggs hatch into small caterpillars and locate to the funnel. Smaller caterpillars (green) may be carried to other plants with the aid of wind. After 14–22 days, the mature caterpillars (orange-green-black or brown) drop to the ground to pupate. The pest undergoes 12 overlapping generations annually.
- In the soil, the caterpillar develops into reddish-brown pupae, although this stage is difficult to notice. On hard soil, the caterpillars gather leaf debris and other material to form a “cocoon”, which is a protective covering on the soil surface to pupate in. Pupation may also take place on plant parts. The pupal stage takes 8–9 days during warm weather and can last between 20–30 days in cooler weather.
- Adults emerge after an average of 10 days and a range of 7–21 days (FAO and CABI, 2019).



Figure 88 — The life cycle of the African army worm
Source: University of Georgia, <https://extension.uga.edu/publications/detail.html?number=C1130&title=Armyworms%20in%20Sod>

SURVEILLANCE/MONITORING

Look out for symptoms of fall army worm eggs; feeding by tiny caterpillars, such as pinhole damage to leaves; droppings; ragged and torn leaves; and pupae in the soil.

CULTURAL CONTROL

Ensure good sanitation by removing debris from a previously army worm-infested crop and clean weeding (Bista *et al.*, 2020; Tambo *et al.*, 2020).

- Pore charcoal dust or ash or a detergent on the affected plants.
- Apply plant extracts of chilli, neem, Tephrosia, Tithonia, Lantana and garlic that can reduce entry of army worms into the field.
- Handpick egg masses and caterpillars if populations are still low.
- Practice early planting or grow early maturing crop varieties.

BIOCONTROL

- *Bacillus thuringiensis* has consistently been shown to reduce the number of army worms on foliage. However, the natural strains of *B. thuringiensis* were found to be very potent, while the genetically modified strains performed better (Wang *et al.*, 2019).

CONTROL WITH PLANT PROTECTION PRODUCTS

- Use registered and approved pesticides within your country, which may include: malathion and lambda-cyhalothrin that have successfully reduced army worm abundance (Togola *et al.*, 2018).
- Neem extracts proved successful in reducing the population of army worms in Ghana (Shaiba *et al.*, 2019).

OTHER CONTROL METHODS

- Natural resistance has been developed for army worm management on maize but not yet for cassava.

H. TIGER MOTH (*PHOENICOPROCTA SANGUINEA*)

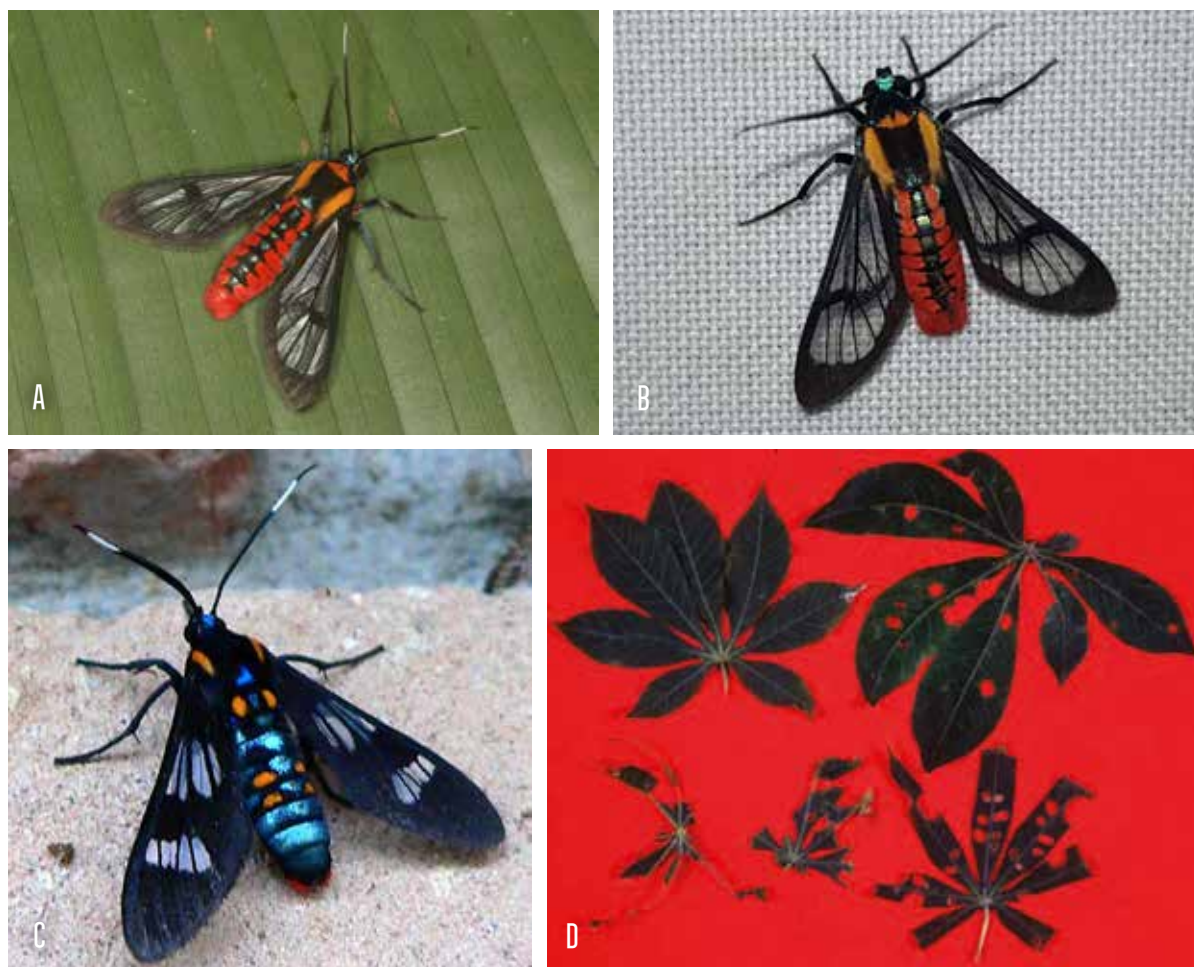


Figure 89 — Adult tiger moths and damage caused to cassava leaves

Source: iNaturalist, <https://stricollections.org/portal/collections/individual/index.php?occid=2082621>;
Smithsonian Tropical Research Institute, <https://stricollections.org/portal/taxa/index.php?taxon=156521>;
Discover Life, <https://www.discoverlife.org/mp/20q?search=Phoenicoprocta>;
file:///C:/Users/Owner/Downloads/Practical_Handbook%20(1).pdf

DESCRIPTION/IDENTIFICATION

PEST

Tiger moths, of the species *Phoenicoprocta sanguinea* belonging to the family Amatiidae, are small and eye-catching insects with a wingspan of 30 mm and a body approximately 12 mm long. Females have black forewings and smaller hindwings with transparent areas. The abdomen is coloured with metallic blue spots in the middle segments (Figure 89A & B). The bodies of males have a black background with blue, red and yellow metallic spots (Figure 89C). The forewings and hindwings are transparent. The males are prettier than the females, due to the presence of red tufts on the abdomen that are separated by central blue spots, and a yellow thorax. Males have black eyes and a blue head (Leraut, 2006; Bellotti et al., 2012).

SYMPTOMS

Young instars make round punctures in the leaves while feeding (Figure 89D). The older and bigger pests make irregular holes and leave a few leaf midribs intact.

DISPERSAL

Adult moths can fly from plant to plant and between fields, while the young instars can move with planting materials during transportation by humans.

DAMAGE

The young instars make round punctures in the leaves while feeding. In contrast, the older and bigger insects make irregular holes due to their eating habit, which leaves a few midribs intact. However, the damage caused is not economically significant (Bellotti *et al.*, 2012).

IMPACT ON YIELD AND QUALITY

The damage caused by tiger moths does not significantly impact yield, unless infestation occurs early during the plants' growth, when the punctures in the young cassava leaves slow growth and tuber formation.

GEOGRAPHICAL DISTRIBUTION

Tiger moths have only been reported on cassava in Senegal (Table 27).

Table 27 — Geographical distribution of tiger moth on cassava in the ACP countries

AFRICA	CARIBBEAN	PACIFIC
Senegal	Not reported	Not reported

QUARANTINE ORGANISM

Tiger moths are not widely distributed, so measures should be taken to control their introduction to new areas.

FAVOURABLE CONDITIONS

Tiger moths establish and thrive best in warm weather.

PEST LIFE CYCLE

Females lay eggs underneath the leaves in the upper third of the plants. The eggs are semi-spherical and creamy and measure about 1 mm in diameter. Eggs can be laid individually or in groups of 2–17, with females laying about 190 eggs in 14 days. The eggs take 4–5 days to hatch.

Larvae undergo five instar stages in 10–14 days. The instars change colours at each stage. First instars are yellowish, translucent, fawn, coffee-coloured and grey, becoming red by the fifth instar. The insect then falls to the ground, where it develops into a prepupal stage for 1 or 2 days before pupating fully in the soil or litter by making a cocoon. The pupal stage takes 12–16 days and pupae are coffee-coloured and 1.5–2.0 cm long, and 0.5–0.7 cm wide. On average, tiger moths take 41.2 days at 26 °C and 70% humidity to develop from egg to adult (Leraut, 2006; Bellotti *et al.*, 2012).

SURVEILLANCE/MONITORING

Not reported

CULTURAL CONTROL

Remove and burn any leaves with the tiger moth larvae and instars to reduce their populations.

- Handpicking egg masses and caterpillars can be tried when the populations are still low.

BIOCONTROL

- Use biological control with *Bacillus thuringiensis* kurstaki (Btk) a selective biological insecticide was reported to control the pest. *Bacillus thuringiensis* release a toxic protein in the digestive system of the insect, which kills it within 5 days. The caterpillar stops feeding soon after, and dies within five-day agents that would maintain it at moderate to low levels in the field (Gabrys, *et al.*, 2008; Bellotti *et al.*, 2012).

CONTROL WITH PLANT PROTECTION PRODUCTS

- *Phoenicoprocta sanguinea* infestation has grown in economic importance and may require the use of chemical pesticides such as Pyrethrins (0.01%) to reduce the population to manageable levels (Bellotti *et al.*, 2012).

OTHER CONTROL METHODS

- Natural resistance.

I. GALL MIDGE (*LATROPHOBIA BRASILIENSIS*)

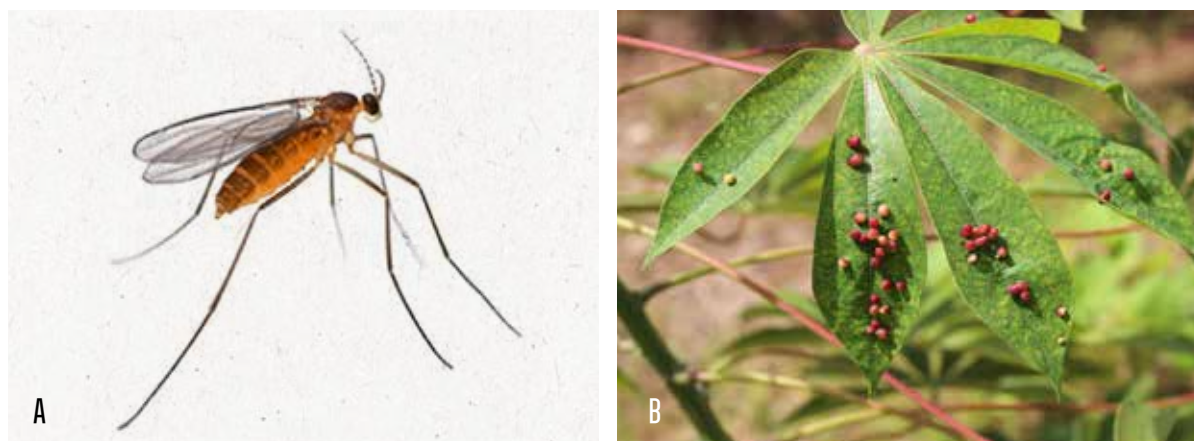


Figure 90 — (A) an adult gall midge and (B) the beads they produce deposited on cassava leaves

Source: National Agricultural Research Institute Guyana,

<https://myrmecos.wordpress.com/2008/09/24/busy-busy-busy/>,

<https://www.britannica.com/animal/gall-midge>

DESCRIPTION/IDENTIFICATION

PEST

Gall midges (*Latrophobia brasiliensis*) are characterised by beaded hairy antennae (Figure 90A) and few veins in the short-haired wings. They feed on plant sap in flowers/ flower buds, fruits, developing seeds and decaying material, although some are carnivores. They produce beads (Figure 90B) on the leaves, buds and stems. A large number also feed on fungi (and are sometimes beneficial to agriculture) (Homenauth and P-DeSouza, 2003).

SYMPTOMS

As a result of feeding by the larva (maggot) an oval chamber is formed around the maggot. The chamber gradually grows out as an elongated cylindrical gall (Alvarez *et al.*, 2012a).

DISPERSAL

It is a lightweight insect that normally flies low over crops. An early study showed that the insect could be dispersed from emergence sites by winds and thermal currents over distances up to 9 km (Alvarez *et al.*, 2012a).

DAMAGE

Under serious attack the roots weaken and leaves turn yellow (Alvarez *et al.*, 2012a).

IMPACT ON YIELD AND QUALITY

The pest does not cause major economic yield loss in cassava plants (Alvarez *et al.*, 2012a).

GEOGRAPHICAL DISTRIBUTION

Gall midges occur on cassava in Africa (Uganda and Tanzania) and the Caribbean countries (Table 28).

Table 28 — Geographical distribution of gall midges on cassava in the ACP countries

AFRICA	CARIBBEAN	PACIFIC
Uganda, Tanzania (Peter Sseruwagi, personal field observation)	Dominica, French Guiana, Grenada, Guyana, Saint Lucia, Saint Vincent and Grenadines, Suriname, Trinidad and Tobago, Venezuela	Not reported

QUARANTINE ORGANISM

Although the gall midge is not a serious economic pest on cassava, measures should be taken to avoid introducing it to areas where it does not currently occur.

FAVOURABLE CONDITIONS

Gall midge numbers decline significantly during the hot season. The insects thrive best at 20°C and high (96%) relative humidity (Gillespie *et al.*, 2000).

PEST LIFE CYCLE

The brightly coloured larvae live in leaves and flowers, where they cause tissue to swell (galls). Some live in galls produced by other insects. Pupation takes place in the gall or in the soil. Female adults deposit the eggs on top of cassava leaves and the larvae enter the leaf tissue, which causes the plant to develop the abnormal growths of galls, which are greenish-yellow to red, and narrower at the base with a curve. Larvae develop within 15–21 days, following which they merge as adults and fly to reproduce (Alvarez *et al.*, 2012a).

SURVEILLANCE/MONITORING

Look out for galls (balls) formed on top of cassava leaves and count them to assess their numbers, and the number of plants, followed by an assessment of the damage caused. Apply any control measures, including handpicking (if the number is low).

CULTURAL CONTROL

Use control strategies according to the cycle of the pest and crop stages (Homenauth and P-DeSouza, 2003).

- Where possible, plant in dry areas.
- Establish fields in open locations and space plants to allow adequate ventilation.
- Avoid late transplantation during first crop season.
- Destroy the weeds that serve as alternative hosts beneath and around plants.
- Remove all fallen leaves from fields, bury or burn.
- Colour traps can potentially be used for monitoring or mating disruption.

BIOCONTROL

- The biological control agents: platygasterid, eupelmid and pteromalid wasps can parasitize the larvae, phytoseiid mites feed on eggs and spiders feed on adults and can be used to control gall midges (Homenauth and P-DeSouza, 2003).
- Use of *Bacillus thuringiensis* var. israelensis (B.t.i.) was reported to effectively control midge larvae development.

CONTROL WITH PLANT PROTECTION PRODUCTS

- Potential insecticide, Deltamethrin.
- Control of midge larvae can also be achieved by using insect growth regulators (IGR) such as methoprene (application of these pesticides should be done in last resort and with appropriate risk mitigation measures).

OTHER CONTROL METHODS

- Use tolerant/resistant varieties.

7.4. BACTERIAL DISEASES

7.4.1. CASSAVA BACTERIAL BLIGHT

(*XANTHOMONAS AXONOPODIS* PV. *MANIHOTIS*)

SCIENTIFIC NAME

- Cassava bacterial blight (*Xanthomonas axonopodis* pv. *manihotis*)
- Cassava leaf spot/Bacterial angular leaf spot (*Xanthomonas campestris* pv. *cassavae*)
- Bacterial stem gall (*Agrobacterium tumefaciens* Biovar 1)
- Bacterial stem rot (*Erwinia carotovora* subsp. *carotovora*)
- Bacterial wilt (*Erwinia herbicola*)



Figure 91 — Bacterial blight symptoms on (A) a cassava leaf, (B) plants and (C) severely affected cassava crop with candle-stick appearance

Source: Pacific Pests, Pathogens & Weeds,

https://apps.lucidcentral.org/pppw_v10/text/web_full/entities/cassava_bacterial_blight_173.htm

DESCRIPTION/IDENTIFICATION

Cassava bacterial blight (CBB) is caused by the bacterium *Xanthomonas axonopodis* pv. *Manihotis* (Maraite, 1993; Maraite and Meyer, 1975; Mansfield *et al.*, 2012). The disease is expressed with abnormal leaf shedding and drying of leaves still attached to the stem. The leaves develop angular leaf spots and blighted areas. The green part of the stem has dark-brown to black streaks, with abnormal swellings of the bark and gummy exudates. Cutting the outer part of the bark reveals black stripes in the phloem tissue, while the xylem has browning and dark-green expanding spots (Persley, 1979).

SYMPTOMS

Small, angular, brown and water-soaked lesions between leaf veins on lower surfaces of leaves (Figure 91A); leaf blades turning brown as lesion expands; lesions may have a yellow halo; lesions coalesce to form large necrotic patches; defoliation occurs, with leaf stems remaining in horizontal positions as leaves drop; dieback of shoots (Figure 91B & C); brown gum may be present on stems, leaves and petioles.

Roots of highly susceptible cassava varieties develop discoloured tissue inside the stem, which is covered by dry and rotten spots (Boher *et al.*, 1995). CBB rarely affects cassava tubers. But when it does, especially in the susceptible varieties, it normally causes swollen roots, and dry, rotted spots (Lozano, 1986).

TRANSMISSION AND SPREAD

CBB spreads in infected but symptomless cassava cuttings and seed, by the aid of water splash (Persley, 1979), through infected tools, soil or wind, or through exchange of infected cassava cuttings (Maraite, 1993; Mansfield *et al.*, 2012).

DAMAGE

The disease causes wilting, dieback and stem tissue necrosis, and angular necrotic spotting of the leaves with a yellowing ring encircling the spots. The leaves' ability to carry out photosynthesis is impaired.

IMPACT ON YIELD AND QUALITY

CBB causes high economic losses of up to 100% in the susceptible varieties (Terry, 1977).

7.4.2. CASSAVA LEAF SPOT/BACTERIAL ANGULAR LEAF SPOT (*XANTHOMONAS CAMPESTRIS* PV. *CASSAVAE*)

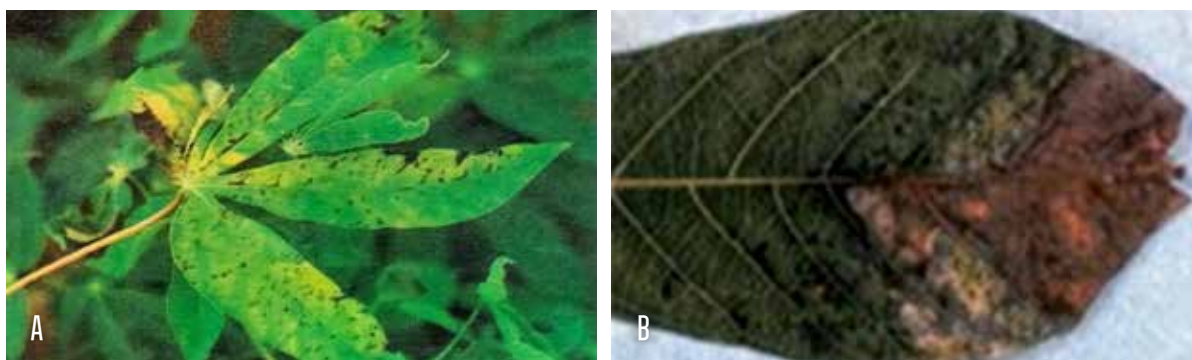


Figure 92 — Symptoms of (A & B) cassava leaf spots

Source: Lozano *et al.* (1981), Maraite (1993), <https://www.cabi.org/isc/datasheet/56927#toPictures>

DESCRIPTION/IDENTIFICATION

Cassava leaf spot is caused by the bacterium *Xanthomonas campestris* pv. *cassavae*. The disease is characterised by water-soaked, angular spots on leaves with gummy exudates that are restricted to the leaves (unlike those for CBB, which spread to stems and leaf stems).

SYMPTOMS

On the upper leaf surfaces the spots are brown with dark borders, sometimes surrounded by indistinct yellow margins (Figure 92A). On the underside, the spots are grey with less distinct borders (Figure 92B). Minor veins crossing the spots appear as black necrotic lines. The centres of the spots dry, crack and may fall out. Infected leaf tissues produce characteristic bright yellow exudates during periods of high humidity that look water soaked (Maraite, 1993; Maraite and Perreux, 1979).

TRANSMISSION AND SPREAD

Like CBB, cassava leaf spot spreads in infected, symptomless cassava cuttings and seed by the aid of water splash (Persley, 1979) and through infected tools, soil or wind, or by exchange of infected cassava cuttings (Maraite, 1993; Mansfield *et al.*, 2012).

DAMAGE

Leaves develop brown spots on upper leaf surfaces, which may cause the leaf to crack and fall off the plant in the severe cases. However, its effects are much less severe than those of cassava bacterial blight disease.

IMPACT ON YIELD AND QUALITY

The impact on yield and tuber quality is negligible, since the effect of the disease is much less severe than that due to CBB (Van den Mooter *et al.*, 1987).

7.4.3. BACTERIAL STEM GALL (*AGROBACTERIUM TUMEFACIENS* BIOVAR 1)



Figure 93 — Bacterial stem gall on cassava stems

Source: Ohio State University, <https://ohioline.osu.edu/factsheet/plpath-fru-19>

DESCRIPTION/IDENTIFICATION

Bacterial stem gall is caused by the bacterium *Agrobacterium tumefaciens* and is characterised by galls (swellings) on the stem nodes (Lozano and Bellotti, 1978).

SYMPTOMS

Symptoms appear on the lower part of the stem and other older plant parts and include galls (swellings) on the stem (Figure 93) and dieback (Lozano *et al.*, 1981).

TRANSMISSION AND SPREAD

Disease infection occurs through injuries arising from old leaves falling off the plant, which get infected on the ground by rainwater (Lozano *et al.*, 1981).

DAMAGE

Plants affected by bacterial stem gall have stunted growth and die back, especially when infection occurs earlier in their growth.

IMPACT ON YIELD AND QUALITY

Mild infection causes no yield loss to tubers, but severe infection involving stunting of plants may lead to reduced yield (size and number of tubers) (Hillocks and Wydra, 2002; Santana *et al.*, 2012; Alvarez *et al.*, 2012a; McCallum *et al.*, 2017).

7.4.4. BACTERIAL STEM ROT (*ERWINIA CAROTOVORA* SUBSP. *CAROTOVORA*) AND BACTERIAL WILT (*ERWINIA HERBICOLA*)



Figure 94 — Symptoms of bacterial stem rot appearing as (A) cankers on the stems and (B) wilting on leaves of cassava plants

Source: Lozano *et al.* (1981). <https://cgspace.cgiar.org/handle/10568/77965>, Reddy (2015) [https://www.google.ca/books/edition/Plant_Protection_in_Tropical_Root_and_Tu/Cc7WCQAAQBAJ?hl=en&gbpv=1&dq=Reddy,+P.+P.+\(2015\).+Plant+protection+in+tropical+root+and+tuber+crops+\(pp.+193-233\).+New+Delhi,+India::+Springer+India.&printsec=frontcover](https://www.google.ca/books/edition/Plant_Protection_in_Tropical_Root_and_Tu/Cc7WCQAAQBAJ?hl=en&gbpv=1&dq=Reddy,+P.+P.+(2015).+Plant+protection+in+tropical+root+and+tuber+crops+(pp.+193-233).+New+Delhi,+India::+Springer+India.&printsec=frontcover)

DESCRIPTION/IDENTIFICATION

Bacterial stem rot is caused by the bacterium *Erwinia carotovora* subsp. *carotovora*, while bacterial wilt is caused by *Erwinia herbicola* following stem damage by fruit fly (*Anastrepha* spp.) larvae. The diseases are characterised by soft stem rot, discoloration and cankers on the woody plant parts, followed by wilting in severe cases. Dry latex exudates are evident when the stem is pierced (Lozano and Bellotti, 1978; Lozano *et al.*, 1981).

SYMPTOMS

Cassava plants develop external cankers and internal rotting of stems (Figure 94A), necrosis of roots, wilting of young shoots or branches (Figure 94B), followed by dieback of shoots or in severe cases (Lozano and Bellotti, 1978; Lozano *et al.*, 1981).

TRANSMISSION AND SPREAD

The bacteria spreads by water splash and infected tools and planting materials (Lozano and Sequeira, 1974). Bacteria can also be transmitted in the seed (Elango and Lozano, 1980).

DAMAGE

The quality of planting materials is reduced by damage caused on the stems creating cankers, leading to poor germination, stunting and a few thick roots (Lozano *et al.*, 1981).

IMPACT ON YIELD AND QUALITY

In severely affected plants, the disease causes wilting, stunting and eventual death of plants, with 100% yield loss in the highly susceptible varieties (Hillocks and Wydra, 2002; Santana *et al.*, 2012; Alvarez *et al.*, 2012a; McCallum *et al.*, 2017).

GEOGRAPHICAL DISTRIBUTION

The bacterial diseases affecting cassava occur widely in sub-Saharan Africa, Central and South America, the Caribbean and the Pacific (Table 29) (Hillocks and Wydra, 2002; CABI/EPPO, 2016; CABI, 2020).

Table 29 — Geographical distribution of bacterial diseases affecting cassava in the ACP countries

BACTERIAL DISEASE	AFRICA	CARIBBEAN	PACIFIC
Cassava bacterial blight (CBB) (<i>Xanthomonas axonopodis</i> pv. <i>manihotis</i>)	Benin, Burkina Faso, Burundi, Cameroon, Central African Republic, Comoros, Democratic Republic of the Congo, Republic of the Congo, Côte d'Ivoire, Ghana, Kenya, Madagascar, Malawi, Mali, Mauritius, Mayotte, Niger, Nigeria, Réunion, Rwanda, South Africa, Sudan, Tanzania, Togo, Uganda	Barbados, Cuba, Dominican Republic, Mexico, Nicaragua, Panama, Trinidad and Tobago	Federated States of Micronesia, Fiji, Guam, Palau
Cassava leaf spot/ Bacterial Angular leaf spot (<i>Xanthomonas campestris</i> pv. <i>cassavae</i>)	Burundi, Democratic Republic of the Congo, Kenya, Malawi, Rwanda, Tanzania, Uganda	Colombia	Not reported
Bacterial stem gall (<i>Agrobacterium tumefaciens</i> Biovar 1)	Not reported	Not reported	Not reported
Bacterial stem rot (<i>Erwinia carotovora</i> subsp. <i>carotovora</i>) and Bacterial wilt (<i>Erwinia herbicola</i>)	Burundi, Democratic Republic of the Congo, Kenya, Malawi, Rwanda, Tanzania, Uganda	Latin America	Not reported

QUARANTINE ORGANISM

Although the overall risk from all five bacterial diseases: cassava bacterial blight, bacterial angular leaf spot, bacterial stem gall, bacterial stem rot and bacterial wilt is moderate, great care must be taken to screen cassava seeds and tissues before they are introduced to areas where bacterial diseases do not yet occur. Cassava planting material can be created using tissue culture in a laboratory, and therefore certified to be free from bacterial diseases. In addition, stem cuttings can be heat-treated and planted under strict quarantine observation to avoid spread of disease (Frison and Feliu, 1991). Upon arrival in the receiving country, the plantlets or planting materials should be grown in containment and kept under observation (FAO/IBPGR Technical Guidelines for the Safe Movement of Cassava Germplasm, <https://www.google.com.au/webhp?sourceid=chrome-instant&ion=1&espv=2&ie=UTF-8#q=international%20guidelines%20for%20cassava>).

FAVOURABLE CONDITIONS

Cassava bacterial diseases are more severe in wet conditions (with high humidity) and decrease during the dry season (Maraite and Meyer, 1975; Persley, 1976; 1978; Daniel and Boher, 1985) (Table 30). Bacterial diseases also attack wild cassava relatives such as *Manihot aipi*, *M. glaziovii* and *M. palmata*.

LIFE CYCLE

Table 30 — Life cycle of bacterial causal pathogens affecting cassava

BACTERIAL DISEASE	PATHOGEN LIFE CYCLE
Cassava bacterial blight (<i>Xanthomonas axonopodis</i> pv. <i>manihotis</i>), Bacterial angular leaf spot (<i>Xanthomonas campestris</i> pv. <i>cassavae</i>) and Bacterial stem gall (<i>Agrobacterium tumefaciens</i> Biovar 1)	Bacterial pathogens colonise surfaces of cassava leaves during high humidity, with the pathogen penetrating the plant (Daniel and Boher, 1985) through wounds and natural openings of cassava leaves such as the stomata (Kemp <i>et al.</i> , 2004). Bacteria are spread from one plant to another by raindrops and wind (Maraite, 1993). When the bacteria settle on the new cassava leaves, the disease cycle resumes. The pathogens can also be spread through cuttings and farm tools (Lozano and Sequeira, 1974). Bacteria can also be transmitted in the seed (Elango and Lozano, 1980).
Bacterial stem rot (<i>Erwinia carotovora</i> subsp. <i>carotovora</i>) and Bacterial wilt (<i>Erwinia herbicola</i>)	<i>Erwinia carotovora</i> subsp. <i>Carotovora</i> , <i>Pectobacterium carovora</i> and <i>Erwinia herbicola</i> seldom occur at epidemic levels. The pathogens overwinter on different plants and in the soil at low densities. They can be ingested by insects feeding on decayed vegetation and spread from the gut, where they infect plants through fresh pruning wounds and aging leaves. However, the pathogens do not infect healthy green leaves, although they can grow in the xylem vessels of the main stem.

SURVEILLANCE/MONITORING

Look for leaves that are drying and dying early. Check for the typical symptoms, such as angular spots on the leaves. Cut small pieces of the leaf from the edge of the spots and put them in a drop of water. Look for bacterial streaming, which should appear as white streaks in the water. On the stems, check for dark brown to black streaks on the green parts, and for the presence of a sticky liquid. Cut the stem, remove the bark and split it open to check for browning in the vascular tissues.

CULTURAL CONTROL

- Rotate cassava crops with non-hosts (Persley, 1978; Lozano, 1986).
- Plough crop debris into soil after harvest or remove and burn it (Fanou *et al.*, 2017).
- Prune infected parts (leaves and branches) from cassava plants.
- Propagate cuttings only from healthy mature cassava plants.
- Intercrop cassava with corn (maize) and melon.
- Practice clean weed management to remove possible alternative host plants.
- Delayed planting towards the end of the rainy season helps reduce spread (Zinsou *et al.*, 2004).
- In soils with low potassium, add fertiliser rich in potassium to reduce severity of CBB (Odurukwe and Arene, 1980).

BIOCONTROL

- Biological control has been studied using the bacteria *Bacillus cereus*, *Bacillus subtilis* and *Penicillium purpurescens* (Beattie and Williams, 1996).
- Foliar application of *Pseudomonas fluorescens* and *P. putida* can significantly reduce leaf infection by *X. axonopodis* pv. *manihotis* (Lozano, 1986). However, biological control has not yet gained wide practical acceptance.

CONTROL WITH PLANT PROTECTION PRODUCTS

- Tools should be regularly disinfected using a bactericide.

OTHER CONTROL METHODS

- Use of elite cassava varieties with proven resistance or tolerance to CBB (Banito *et al.*, 2008; Terry and Hahn, 1982; Zárate-Chaves *et al.*, 2021).

7.5. FUNGAL DISEASES

7.5.1. *CERCOSPORA* LEAF DISEASES

COMMON AND SCIENTIFIC NAMES

- A. Cassava brown leaf spot (*Passalora henningsii* *Cercosporidium henningsii*)
- B. Cassava leaf blight (*Cercospora vicosae*)
- C. White leaf spot (*Cercospora caribaea*)

A. CASSAVA BROWN LEAF SPOT (*CERCOSPORIDIUM HENNINGSII*)

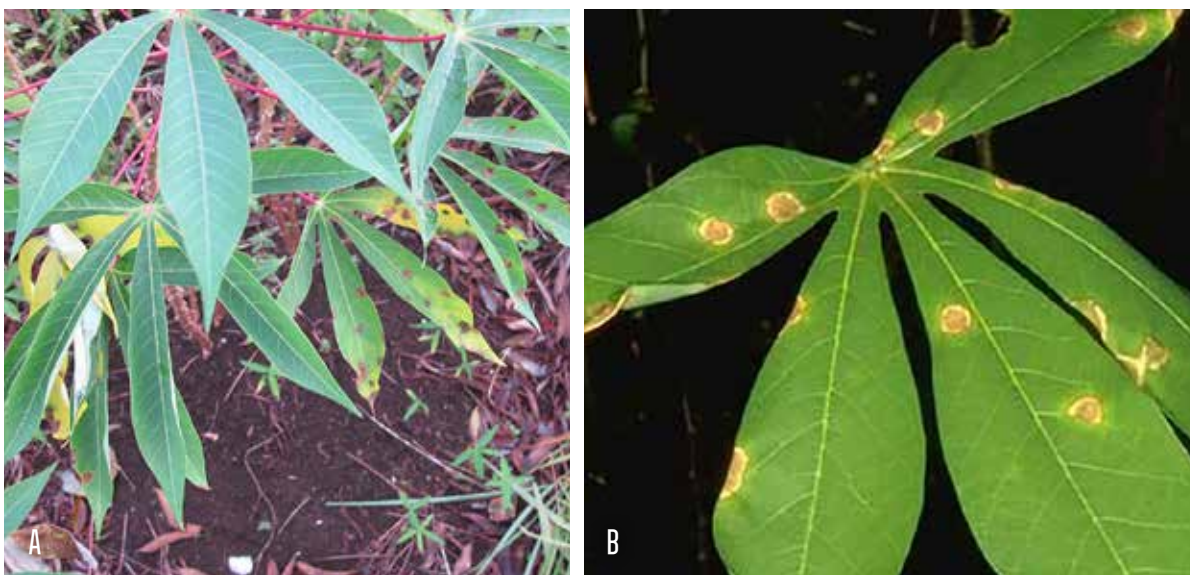


Figure 95 — Showing (A) Severe and (B) mild brown leaf spot symptoms on cassava leaves

Source: Pacific Pests, Pathogens and Weeds,

https://apps.lucidcentral.org/pppw_v10/pdf/web_full/cassava_brown_leaf_spot_095.pdf

DESCRIPTION/IDENTIFICATION

Cassava brown leaf spot is a disease of minor economic importance (Wydra and Msikita, 1998) that is caused by the fungus *Passalora henningsii*/*Cercosporidium henningsii*, which forms light brown spots with yellow margins on the older cassava leaves. The brown spots on the surface are circular with dark borders of 15 mm in diameter, which become angular and restricted by veins. On the underside of the leaves, the spots are grey with less distinct borders. Minor veins cross the spots as black necrotic lines. The middle of the leaf spots is dry, which leads to cracking and leaf fall. As the spots enlarge, the leaves turn yellow and fall off.

SYMPTOMS

Circular or irregular brown spots with darker margin between leaf veins on lower older (> 5 months old) leaves (pFigure 95A & B); centres of lesions may drop out giving leaves a shot hole appearance; if infection is severe, leaves may turn yellow, dry out and drop from the plant.

TRANSMISSION AND SPREAD

Movement of the fungal spores occurs by wind and water-splash, and long-distance spread occurs during distribution of planting material.

DAMAGE

The disease occurs on older, lower leaves at 5–6 months after planting, leading the leaves to fall prematurely.

IMPACT ON YIELD AND QUALITY

Losses in root yield of up to 30% have been reported from Africa (Théberge, 1985), 23–30% in South America (Takatsu *et al.*, 1990) and 17% in India. However, serious defoliation requiring disease control has not been reported in Pacific countries. The disease usually occurs late in the growth of the plants, and only on the older leaves.

B. CASSAVA LEAF BLIGHT (*CERCOSPORA VICOSAE*)



Figure 96 — Showing cassava leaf blight or leaf spot symptoms on (A) a cassava leaf and (B) leaves on a cassava plant
Source: Lertwit Sasipreyajun/Dreamstime, <https://www.dreamstime.com/cassava-leaf-blight-disease-bacteria-cassava-leaf-blight-disease-bacteria-plant-disease-image104127512>

DESCRIPTION/IDENTIFICATION

Cassava leaf blight, also known as diffuse leaf spot, is caused by the fungus *Cercospora vicosae* (Hillocks and Wydra, 2002).

SYMPTOMS

Large, diffuse white spots on upper surface of leaves; spots with grey centre on underside of leaves (Figure 96A & B) in humid weather (Lozano and Booth, 1976).

TRANSMISSION AND SPREAD

Spreads by water splash and infected tools; disease more severe in wet conditions; the most important method of spread is through exchange of infected cassava cuttings.

DAMAGE

Cassava leaf blight causes severe defoliation in the susceptible cultivars (Hillocks and Wydra, 2002).

IMPACT ON YIELD AND QUALITY

Since the disease occurs in older plants (> 6–9 months old), the yield losses are very low (Otim-Nape, 1980; Hillocks and Wydra, 2002).

C. WHITE LEAF SPOT (*CERCOSPORA CARIBAEA*)



Figure 97 — Symptoms of white leaf spot of cassava

Source: National Agricultural Research Institute Guyana,

http://www.newgmc.com/gmc_docs/brochures/pests%20%20diseases%20of%20cassava%20in%20guyana.pdf

DESCRIPTION/IDENTIFICATION

White leaf spot is caused by the fungus *Cercospora caribaea*.

SYMPTOMS

Leaves develop circular yellowing areas on the upper surface (Figure 97), which later develop small (1.5–2 mm) circular white lesions in the centre. These are visible on the underside of the leaf. The spots enlarge to 3–5 mm and form a purple border, retaining the yellow halo (Lozano *et al.*, 1981; Théberge, 1985).

TRANSMISSION AND SPREAD

White leaf spot is the most important fungal disease of cassava; spread by water splash and infected tools; the disease is more severe in wet conditions; particularly destructive in South America and Africa; most important method of spread is probably through exchange of infected plant cuttings.

DAMAGE

White leaf spot causes mild disease severity on the affected plants (Hillocks and Wydra, 2002).

IMPACT ON YIELD AND QUALITY

The impact of the disease on cassava plants is negligible (Hillocks and Wydra, 2002; Fanou *et al.*, 2018).

GEOGRAPHICAL DISTRIBUTION

All three *cercospora* leaf diseases occur widely in cassava (Table 31) growing areas in Sub-Saharan Africa, the Caribbean and the Pacific (Fanou *et al.*, 2018).

Table 31 — Geographical distribution of *cercospora* leaf diseases on cassava in the ACP countries

FUNGAL DISEASE/S	AFRICA	CARIBBEAN	PACIFIC
Cassava brown leaf spot, cassava leaf blight and white leaf spot	Angola, Benin, Burkina Faso, Chad, Ethiopia, Ghana, Ivory Coast, Kenya, Madagascar, Malawi, Mozambique, Nigeria, Sierra Leone, South Africa, Sudan, Tanzania, Uganda	Barbados, Belize, Costa Rica, Cuba, Dominican Republic, El Salvador, Guatemala, Haiti, Honduras, Mexico, Nicaragua, Panama, Puerto Rico, Trinidad and Tobago	American Samoa, Cook Islands, Fiji, French Polynesia, Federated States of Micronesia, New Caledonia, Niue, Palau, Papua New Guinea, Samoa, Solomon Islands, Tonga, Vanuatu

QUARANTINE ORGANISM

Generally, all three fungal diseases (cassava brown leaf spot, cassava leaf blight and white leaf spot) are of low economic importance, although they occur globally on cassava crops. Nevertheless, it is advisable that movement of cassava planting materials and leaves infected with fungal diseases are checked for symptoms before they are moved between regions and countries.

FAVOURABLE CONDITIONS

Fungal diseases are favoured by high temperature and humidity. The spread of the fungal spores occurs by wind and water-splash, while long-distance spread occurs during distribution of planting material. The fungal spores overwinter on the fallen leaves in cassava fields (Ayesu-Offei and Antwi-Boasiako, 1996).

PEST LIFE CYCLE

The fungal spores over winter on the older fallen leaves in cassava fields and re-infect new crops in subsequent growing seasons when the conditions are favourable (Ayesu-Offei and Antwi-Boasiako, 1996).

SURVEILLANCE/MONITORING

Fungal diseases of cassava tend to affect the older canopy/leaves of plants older than 4 months of age. Look for characteristic fungal disease symptoms on the older plants at least monthly to score for the number of plants with infections and symptom severity (damage levels).

CULTURAL CONTROL

Fungal diseases can be controlled effectively using cultural strategies that disrupt the life stages, as follows:

- Plant at wide spacing for reduced crop density to allow wind to dry the leaves as quickly as possible.
- Practice crop rotations with non-host plants every 3–5 years.
- Plant in the wet season so that plants reach the susceptible stage (6–8 months old) during the dry season.
- Avoid establishing new crops next to old, infected fields.
- After harvest, plough the crop debris into the soil or remove and burn it.
- Prune infected leaves and branches from the plant and either bury or burn them.
- Use cassava cuttings selected from healthy plants only while still in the field.
- Removed weeds in and around cassava crops to avoid disease spreading into the field.

BIOCONTROL

- Not reported

CONTROL WITH PLANT PROTECTION PRODUCTS

- Apply copper fungicides if the need arises.

OTHER CONTROL METHODS

- Plant resistant varieties of cassava if available (Lozano and Booth, 1976; Hahn *et al.*, 1980a; Boher and Verdier, 1994).

7.5.2. CASSAVA ANTHRACNOSE DISEASE

SCIENTIFIC NAME

— *Colletotrichum gloeosporioides* f. sp. *Manihotis*/*Glomerella cingulata*



Figure 98 — Showing symptoms of anthracnose disease (*Colletotrichum gloeosporioides*) damage on cassava (A) branches and (B) stems

Source: CABI Plantwise Knowledge Bank,

<https://www.plantwise.org/KnowledgeBank/pmdg/20167800251>

DESCRIPTION/IDENTIFICATION

PEST

Anthracnose disease is caused by a fungus known as both *Glomerella cingulata* and *Colletotrichum gloeosporioides* (the different names refer to the different forms – sexual and asexual – of the fungus).

SYMPTOMS

The disease causes wounds/cankers on stems and leaf stems (Figure 98A & B), leaves droop downwards and wilting leaves may eventually die (Moses *et al.*, 1996) and fall off the plant. The soft parts of the plant are twisted and distorted.

TRANSMISSION

The causal fungus is transmitted in cassava seed, with up to 40% incidence in some genotypes (Fokunang *et al.*, 1997). The fungus enters the necrotic tissue and infection develops during high relative humidity.

SPREAD

Disease emerges at the beginning of the wet season and worsens, and the spores are spread by wind. The coreid bug (*Pseudotheraptus devastans*), a sap-sucking insect that punctures the stem, aids infection by the fungus (Boher *et al.*, 1983).

DAMAGE

Anthracnose causes severe damage (cankers) on cassava stems, dieback and loss of leaves.

IMPACT ON YIELD AND QUALITY

The main effect of the disease is reducing the quality of stem cuttings as planting material (Théberge, 1985).

GEOGRAPHICAL DISTRIBUTION

Anthracnose diseases occur mostly on cassava in Africa (Table 32).

Table 32 — Geographical distribution of Anthracnose diseases on cassava in the ACP countries

AFRICA	CARIBBEAN	PACIFIC
Angola, Benin, Cameroon, Central African Republic, Democratic Republic of the Congo, Republic of the Congo, Côte d'Ivoire, Equatorial Guinea, Gabon, Gambia, Ghana, Guinea, Guinea-Bissau, Kenya, Liberia, Madagascar, Malawi, Mali, Mozambique, Niger, Nigeria, Rwanda, Senegal, Sierra Leone, South Africa, Sudan, Tanzania, Zanzibar Island, Togo, Uganda, Zambia, Zimbabwe	Not reported	Not reported

QUARANTINE ORGANISM

The disease does not cause large-scale economic damage to cassava. Control by quarantine can help to further reduce spread.

FAVOURABLE CONDITIONS

Disease emerges at the beginning of the wet season and worsens, and the spores spread by wind (Wydra and Msikita, 1998).

PEST LIFE CYCLE

The life cycle of the fungus (see section 'Spread', above) is repeated by propagating infected stems and other plant debris.

SURVEILLANCE/MONITORING

Monitor cassava fields weekly, from sprouting of the plants through to plant maturity. Check for symptoms such as wilting, dieback and cankers. As soon as the symptoms are detected, apply control measures to limit further spread of the disease (Wydra and Msikita, 1998).

CULTURAL CONTROL

Anthrachnose usually does not cause large-scale economic damage to cassava and control is usually not necessary, therefore cultural control can significantly reduce its effects and further spread.

- Avoid planting cassava cuttings with cankers/cuts as the fungus enters through wounds.
- Rogue or remove cassava plants infected with anthracnose disease.
- Remove crop debris and destroy it after harvesting cassava.
- Avoid growing cassava on the same field for multiple seasons. Practice crop rotation of cassava with grain crops such as maize, sorghum or millet.
- Late planting especially in the second rain season can reduce disease infections.

BIOCONTROL

N/A

CONTROL WITH PLANT PROTECTION PRODUCTS

- Treat cassava cuttings with a fungicide such as copper.

OTHER CONTROL METHODS

Other potential control methods include natural resistance, IPM, regulation and awareness creation.

- Use of resistant varieties.

7.5.3. CASSAVA ROOT ROT DISEASE

SCIENTIFIC NAME

- A. **Soft root rot/Phytophthora root rot** (*Phytophthora cryptogea*, *Phytophthora drechsleri*, *Phytophthora erythroseptica*, *Phytophthora nicotianae* var. *parasitica*) and **Pythium root rot** (*Pythium* spp., *Fusarium* spp.)
- B. **Dry root rot** (*Rigidoporus lignosus*, *Rosellinia necatrix*, *Lasidiplodia theobromae*, *Armillariella mellea*, *Sphaerotilbe repens*)
- C. **Dematophora root rot** (*Rosellinia* root rot) (*Dematophora necatrix* *Rosellinia necatrix*)
- D. **Diplodia root and stem rot** (*Diplodia manihoti*)
- E. **Black root and stem rot** (*Scytalidium* sp., *Hendersonula toruloidea*)

CASSAVA ROOT ROT DISEASE

A. SOFT ROOT ROT/ PHYTOPHTHORA ROOT ROT



Figure 99 — Showing *fusarium* rot-infected cassava roots
Source: PlantVillage, <https://plantvillage.psu.edu/topics/cassava-manioc/infos>

B. DRY ROOT ROT



Figure 100 — Showing cassava roots infected with *botryodiplodia* root rot disease
Source: PlantVillage, <https://plantvillage.psu.edu/topics/cassava-manioc/infos>

C. DEMATOPHORA ROOT ROT (ROSELLINIA ROOT ROT)



Figure 101 — Showing cassava roots infected with root rot disease
Source: PlantVillage, <https://plantvillage.psu.edu/topics/cassava-manioc/infos>

D. *DIPLODIA* ROOT AND STEM ROT

Photo not available

E. BLACK ROOT AND STEM ROT



Figure 102 — Showing black root and stem rot disease

Source: Brito *et al.*, 2017, <https://link.springer.com/article/10.1007/s10658-020-02024-7>

DESCRIPTION/IDENTIFICATION

Cassava root rots are diverse and include (Brito *et al.*, 2020):

- Soft root rot/*phytophthora* root rot, caused by *Phytophthora cryptogea*, *Phytophthora drechsleri*, *Phytophthora erythroseptica*, *Phytophthora nicotianae* var. *parasitica*;
- *Pythium* root rot, caused by *Pythium* spp., *Fusarium* spp.;
- Dry root rot, caused by *Rigidoporus lignosus*, *Rosellinia necatrix*, *Lasidiplodia theobromae*, *Armillariella mellea*, *Sphaerotilbe repens*;
- *Dematophora* root rot/*Rosellinia* root rot, caused by *Dematophora necatrix*, *Rosellinia necatrix*;
- *Diplodia* root and stem rot (*Diplodia manihoti*) and Black root and stem rot (*Scytalidium* spp., *Hendersonula toruloidea*).

The pathogens live in the soil and can affect the crop at any stage of growth (Alvarez *et al.*, 2012a).

SYMPTOMS

Root rot pathogens cause sudden wilting and severe soft or dry root rot (Figure 99-102). In soft rot, the infected roots exude a liquid of a repulsive odour, after which they decompose into the soil. Cassava leaves affected by soft root rot turn brown, wilt and are scorched. In severe cases, the leaves may drop off and the plant becomes dessicated and dies. The roots develop dieback and swelling of tubers with light brown, dark grey, blue, pink, white or black discoloration in the wet season. Cassava plants affected by dry root rot develop bulky roots that are slightly discoloured, produce liquid if pressed and have a characteristic odour of decay (Alvarez *et al.*, 2012a).

SPREAD

Cassava root rot pathogens spread in infected planting materials. In the field, the pathogens may spread with the aid of wind and rain, farm tools and irrigation water (Alvarez *et al.*, 2012a).

DAMAGE

Cassava root rot-affected plants develop dieback and swelling of tubers, discoloration and rotting with a foul odour (Alvarez *et al.*, 2012a).

IMPACT ON YIELD AND QUALITY

Cassava root rot diseases reduce the quality of cassava tubers through discoloration, rotting and development of a foul odour. The yield is reduced by 5–100%, especially in the susceptible varieties (Alvarez *et al.*, 2012a).

GEOGRAPHICAL DISTRIBUTION

Cassava root rot diseases occur worldwide in cassava producing areas (Hillocks and Wydra, 2002; Alvarez *et al.*, 2012a; Brito *et al.*, 2020).

QUARANTINE ORGANISM

Not reported.

FAVOURABLE CONDITIONS

Cassava root rot fungal diseases are favoured by wet conditions such as waterlogged and heavy, poorly drained low-nutrient soils (Alvarez *et al.*, 2012a).

PEST LIFE CYCLE

Several pathogens cause root rot diseases in cassava plants, including bacteria (El Karkouri *et al.*, 2010) and fungi (Cleary *et al.*, 2011; Harveson *et al.*, 2011). Usually, these pathogens stay in debris and soil for several years (Berg and McClaugherty, 2008). Infection depends on the availability of a suitable environment, such as high soil moisture, temperature and poor drainage (Dalman *et al.*, 2010; Abdel-Monaim and Abo-Elyousr, 2012). The pathogens (fungi and bacteria) enter the root or stems through wounds caused by farm tools, nematodes and other parasites. The spores in soil infect the plant, causing the symptoms of root rot (see above). The cycle recurs when infected cassava cuttings are planted or when the pathogen is transmitted through debris and infected water.

SURVEILLANCE/MONITORING

Regularly check cassava fields for the appearance of yellow mushrooms on the soil or white fruiting bodies at the bottom of cassava stems. Usually, these organisms are those that cause the rot. Look for symptoms of wilting leaves, defoliation, stem dieback and death of infected plants. In addition, consider digging up some symptomatic plants to check if the tubers are swollen and have soft brown rot internally. If the number of infected plants exceeds five, it is advisable to apply an IPM strategy to prevent further spread of the infection to the rest of the crop.

CULTURAL CONTROL

- Select cassava cuttings from healthy cassava plants.
- Use hot water to treat the cassava cuttings at a maximum temp of 49°C for 49 minutes.
- Plant cassava in loose textured and well-draining soils, using ridges where possible.
- Eradicate diseased plants with clear symptoms and destroy them.
- Remove and destroy all crop debris by burning.
- Sanitise all tools after use.
- Practice crop rotation with grasses whenever the disease has affected 3% of the plantation area.
- Keep soils drained for at least 6 months.

BIOCONTROL

- None reported.

CONTROL WITH PLANT PROTECTION PRODUCTS

- Dip cassava cuttings in metalaxyl.

OTHER CONTROL METHODS

- Plant root rot resistant cassava varieties if available.

7.5.4. CASSAVA TUBER ROT DISEASE

SCIENTIFIC NAME

- A. *Sclerotium/Tuber rot (southern blight)* (*Sclerotium rolfsii*, *Athelia rolfsii*)
- B. **Rust** (*Uromyces janiphae*)
- C. *Verticillium root and stem rot* (*Verticillium dahliae*)
- D. **Cassava ash** (*Oidium manihotis*)

CASSAVA TUBER ROT DISEASE

A. *SCLEROTIUM/TUBER ROT (SOUTHERN BLIGHT)*

Photo not available

B. RUST

Photo not available

C. *VERTICILLIUM* ROOT AND STEM ROT



Figure 103 — Showing symptoms of *verticillium* root and stem rot disease on cassava
Source: Ratana Kak/Dreamstime, <https://www.dreamstime.com/african-cassava-mosaic-disease-bacterial-blight-anthrax-bud-necrosis-brown-white-leaf-spot-diseases-root-rot-plant-image124306858>



Figure 104 — Showing symptoms of cassava ash on leaves:
Source: Alvarez *et al.*, 2012a

DESCRIPTION/IDENTIFICATION

Cassava tuber rots are diverse and include: *sclerotium/tuber* rot (southern blight), Cassava rust, *verticillium* root and stem rot and cassava ash disease.

- *Sclerotium/tuber* rot (southern blight): is caused by *Sclerotium rolfsii*, a common soil pathogen with a white mycelium of cottony appearance and numerous round sclerotia, which typically form in the affected plant host or cultures in the laboratory (Alvarez *et al.*, 2012a).
- *Rust* is caused by *Uromyces janiphae* pathogens (Schotman, 1989).
- *Verticillium* root and stem rot: is caused by a fungus *Verticillium dahlia*.
- Cassava ash disease: is caused by *Oidium manihotis* P. Henn., *Erysiphe manihotis*. The fungal mycelium is white producing numerous steam/roots on host plants. Conidiophores are upright and simple with the upper portion increasing in both length and width as conidia are formed (Alvarez *et al.*, 2012a).

SYMPTOMS

Generally, root and stem rot disease causes wilting, defoliation and death of the affected plant. Occasionally, yellowing of lower leaves and stunted growth of plants occurs (Subbarao, 2020).

- *Sclerotium/tuber* rot (southern blight): occurs in young cassava stems and mature roots and develops a whitish cotton-like mat at the base of the affected stems. The affected roots tend to rot.
- Rust disease symptoms appear as light to dark brown pustules on the leaf veins, petioles, or green branches. Mature pustules can be parasitized by *Darluca filum* fungus and may be surrounded by chlorotic halos. They cause deformation of affected plant parts (Alvarez *et al.*, 2012a).
- *Verticillium* root and stem rot: the roots and tubers begin to rot, followed by wilting of the stems (Figure 103) and eventual discoloration and falling off of the leaves and dieback of severely affected plants.
- Cassava ash disease is characterised by yellowish undefined spots (Figure 104), on the leaves with some necrosis (Alvarez *et al.*, 2012a). First, the disease symptoms appear as white mycelium (ash-like) on the surface of the leaves. This is followed by the infected leaves showing yellowish lesions, which are angular in shape with pale brown necrotic areas and of different sizes that are easily mistaken for damage by insects and mites.

SPREAD

All four pathogens that cause cassava tuber rot diseases can spread through infected planting material, crop debris, soil, contaminated farm tools and wind, especially in the dry season.

DAMAGE

Generally, tuber rot diseases cause wilting, defoliation, stunting and death of affected plants.

- *Sclerotium/tuber* rot due to *Sclerotium rolfsii* develops through wounds in the roots, although it is not always very damaging to young plants, but can occur with high incidence of root necrosis in a plant.
- Rust disease incidence and severity are low on cassava where they occur. Importance. Although recorded in Brazil and Colombia, this disease is considered to be of minor importance. It appears at the end of dry periods, sometimes causing a type of shoot proliferation in stem apices (Normanha, 1970).
- *Verticillium* root and stem rot causes the roots and tubers to rot, followed by wilting of the stems and eventual discoloration and falling off of the leaves and dieback of severely affected plants.
- Cassava ash commonly occurs in young stakes and mature roots, and develops plant parts with a cottony white mycelium mat (CIAT 1972; Ferdinando *et al.* 1968). Therefore, the disease is not known to cause significant damage to cassava (Alvarez *et al.*, 2012a).

IMPACT ON YIELD AND QUALITY

Sclerotium/tuber rot and rust pathogens cause root rot, which reduces the quality and quantity of cassava yields. *Verticillium* root and stem rot disease causes significant losses in many countries (Pegg and Brady, 2002; Inderbitzin and Subbarao, 2014). Despite widespread occurrence of cassava ash disease, in the dry season, it is of minor economic importance because it only attacks the lower mature leaves of cassava plants, which are less important for photosynthesis and hence may not necessarily affect plant growth and yield (Alvarez *et al.*, 2012a).

GEOGRAPHICAL DISTRIBUTION

Cassava tuber rot diseases occur on cassava in Africa and the Caribbean (Table 33) but have not been reported yet in the Pacific (Schotman, 1989; Hillocks and Wydra, 2002; Alvarez *et al.*, 2012a; Brito *et al.*, 2020).

Table 33 — Geographical distribution of tuber rot diseases on cassava in the ACP countries

CASSAVA TUBER ROT DISEASES	AFRICA	CARIBBEAN	PACIFIC
<i>Sclerotium/tuber</i> rot (southern blight), Rust, <i>Verticillium</i> root and stem rot and Cassava ash disease	Algeria, Democratic Republic of the Congo, Republic of the Congo, Egypt, Kenya, Madagascar, Malawi, Morocco, Mozambique, Nigeria, Rwanda, South Africa, Tanzania, Tunisia, Uganda, Zambia, Zimbabwe	Brazil, Colombia, Cuba, Dominican Republic, Jamaica, Mexico, Puerto Rico, Trinidad and Tobago	Not reported

QUARANTINE ORGANISM

Although most cassava tuber rot diseases are widespread, and therefore not considered quarantine risks, there are restrictions on entry of planting materials with the fungal and bacterial pathogens in countries such as Australia (Allen, 1995) where they would infect cotton and other commercial crops. The EPPO has listed *Verticillium dahliae* and *Verticillium alboatrum* as A2 quarantine pests together with infected planting materials (EPPO, 2007; 2014; Anon., 1993).

FAVOURABLE CONDITIONS

Sclerotium/tuber rot (southern blight) pathogens mostly occur in warm and humid soils (above 15 °C).

- Root rot diseases are often favoured by different climatic and environmental factors.
- Some rust species are favoured by moderate temperature and high rainfall, while other species are predominant during hot dry seasons.

For example, *Verticillium* root and stem rot increase under moderate to high temperatures, but not temperatures higher than 30°C, such as those in the lowland tropical areas, where the diseases are absent (Mace *et al.*, 1981). Cassava ash disease is prevalent in the dry season in warm areas (Alvarez *et al.*, 2012a).

- *Erysiphe manihotis* is the sexual stage of cassava ash and occurs during the dry season (Normanha 1970).

PEST LIFE CYCLE

The causal pathogens for cassava tuber rot diseases spread through soil or infected planting material, crop debris, contaminated farm tools and wind. The cycle of infection recurs when the plants are planted on the same plot or by use of contaminated tools or by wind during the dry season.

- *Sclerotium/tuber* rot (southern blight) pathogens overwinter as sclerotia, which are the primary inoculum (Agrios, 2005). In addition, the pathogens have a wide host range (>500 plant species), which retain the inoculum in the growing areas across different seasons.
- Like all obligate parasites, rust (*Uromyces* spp.) uses other hosts to complete their life cycle. After overwintering on the residual plant material, the spores germinate, are ejected by the help of water or air, and land on the leaf surface of a host plant where they germinate to grow further. They can complete the life cycle on a single plant or several plants (Voegelé *et al.*, 2009), including cassava.
- *Verticillium* pathogens, *Verticillium dahliae* survive as mycelia in dead tissue, soil, or infected plant material from where they affect the host. The pathogens enter the host plant through wounds or root tissue into the xylem, where the conidia spread throughout the plant. The reaction by the plant to block the infection ends up in limiting the flow of assimilates in the xylem, resulting in wilting (Brazee, 2015).
- White mycelium fungal spores of the causal organism for Cassava ash, *Erysiphe manihotis* grow on leaf surfaces of cassava plants, where they are either blown away by wind or swept off by rain water as ascospores and drop on other plants where they thrive. However, the spores can also be spread through planting materials with infected leaves. The spores are prone to water, which makes them explode and limit their further dispersal.

SURVEILLANCE/MONITORING

Look out for plants with the various symptoms described above for *Sclerotium* tuber rot (southern blight), rust, *Verticillium* root and stem rot and cassava ash disease as early as possible. If the numbers are low, you may rogue out the affected plants. Use the IPM strategies (below) to prevent further damage to the rest of the cassava crops.

CULTURAL CONTROL

- Practice crop rotation with cereal crops such as maize, rice, sorghum and millet or grasses by resting the plot. For example, rotating cassava with paddy rice or maize proved effective in reducing *V. dahliae* in the field due to flooding (Pullman and DeVay, 1981).
- Manipulation of soil fertility such as applying sufficient nitrogen and phosphorus was shown to reduce *Verticillium* wilt disease occurrence (Pennypacker, 1989; Davis *et al.*, 1994).
- Manipulation of soil moisture through irrigation proved to reduce the incidence of cassava tuber rot causal pathogens (Cappaert *et al.*, 1994).
- Soil fumigation or exposing the soil to sunlight reduces the viability of the fungal pathogens causing cassava tuber rot diseases in the soil (Xiao *et al.*, 1998).

BIOCONTROL

- The fungi: *Pythium oligandrum* (Al-Rawahi and Hancock, 1998), *Heteroconium chaetospora* (Narisawa *et al.*, 2000) and *Talaromyces flavus* (Nagtzaam *et al.*, 1998) were used as biological agents to control cassava tuber rot.
- Bacterial antagonists such as rhizobacteria may prevent the establishment of tuber rot causal pathogens (Sharma *et al.*, 1998).

CONTROL WITH PLANT PROTECTION PRODUCTS

- Spray cassava plants affected with cassava ash disease with sulphur-based pesticides (Ferdinando *et al.*, 1968).

OTHER CONTROL METHODS

- Use disease-resistant cassava varieties (CIAT, 1972).

7.5.5. CONCENTRIC RING LEAF SPOT

COMMON AND SCIENTIFIC NAME

- **Concentric ring leaf spot** (*Phyllosticta manihotae*, *Phyllosticta manihoticola*)



Figure 105 — Showing symptoms of concentric ring leaf spot disease with blight-brown spots on the upper surface of a cassava leaf

Source: Emile Frison/ResearchGate, https://www.researchgate.net/figure/Phomopsis-blight-brown-spots-with-concentric-rings-of-pycnidia-on-the-upper-surface-of_fig15_281633837

DESCRIPTION/IDENTIFICATION

PEST

Concentric ring leaf spot is caused by the fungus *Phoma* spp., which invades the leaves, followed by the leaf stems and the green parts of stems.

SYMPTOMS

Concentric ring leaf spot is characterised by large water-soaked spots (Figure 105), which appear as pale green and turn brown or black at the leaf tips, surrounded by unclear margins or along the midvein and secondary leaf veins. In early infections, the lesions appear as concentric rings on the upper leaf surface or stems, formed by the fungal fruiting bodies. The older lesions lack the rings and look like lesions due to *Cercospora vicosae*. On the lower side of the leaf, the lesions are dark brown and the veins have black thread-like filaments emerging from the spots (Álvarez *et al.*, 2012a). Under severe attack, the leaves fall off plants (Lozano and Booth, 1976).

TRANSMISSION AND SPREAD

The disease-causing pathogens (conidia) can be transmitted through water splashes during the rainy season. It can also be spread through planting infected cassava leaves and planting material obtained from the previous season's crops. The pathogens also thrive in alternative host plants such as eggplant/aubergine (*Solanum melongena*) (Frison and Feliu, 1991).

DAMAGE

Concentric ring leaf spot defoliates the plant and causes dieback, which in severe cases leads to death of affected plants (Álvarez *et al.*, 2012a).

IMPACT ON YIELD AND QUALITY

Under severe attack, the disease can cause high yield losses to cassava crops.

GEOGRAPHICAL DISTRIBUTION

Concentric ring leaf spot disease is widespread, especially in areas with high altitude, where it is cooler, and in the lowland areas during the wet season (Frison and Feliu, 1991).

QUARANTINE ORGANISM

Plant materials with concentric ring leaf spot disease and causal pathogens (*Phoma* spp.) are restricted from entry into the EU. Either the plant materials are treated with meristem-tip culture, which removes the causal pathogens, or only proven disease-free materials are allowed for export (Frison and Feliu, 1991).

FAVOURABLE CONDITIONS

Concentric ring leaf spot occurs in cultivated areas with temperatures below 22°C, especially during the rainy season (Álvarez *et al.*, 2012a).

PEST LIFE CYCLE

The fungus lives as spores in leaf litter or alternative hosts in cassava fields. Infection by the fungus occurs when the spores are blown by wind and settle on the plant leaves, where they penetrate through natural openings or wounds. Here the spores start to grow. The conidia disperse by the aid of wind or water splash fall on older leaves on the ground and the cycle repeats itself when the weather conditions are favourable.

SURVEILLANCE/MONITORING

Monitor cassava fields for the characteristic concentric ring leaf spot symptom (pale green, brown or black at the leaf tips), which is surrounded by unclear margins or along the midvein and secondary leaf veins. Count the number of infected plants to determine if and when to commence IPM measures.

CULTURAL CONTROL

Planting healthy cassava cuttings.

- Use appropriate sowing density (4,000 plants per acre or 10,000 plants per hectare).
- In the disease hot spot (high incidence) areas, plant cassava at the end of the rainy season.

BIOCONTROL

Not reported

CONTROL WITH PLANT PROTECTION PRODUCTS

- Azoxystrobin can be used against *Phyllostica* sp.

OTHER CONTROL METHODS

Not reported

7.5.6. SUPERELONGATION

SCIENTIFIC NAME

— Superelongation (*Sphaceloma manihoticola*, *Elsinoë brasiliensis*)



Figure 106 — Showing symptoms of superelongation of a cassava leaf and stem
Source: Semantic Scholar, <https://www.semanticscholar.org/paper/Chapter-3-Cassava-Superelongation-Disease-in-the-Alleyne/f8eecfb4defb70d79ef11017a444b5e038b41f53>

DESCRIPTION/IDENTIFICATION

PEST

Superelongation disease is caused by fungus *Sphaceloma manihoticola* and *Elsinoë brasiliensis*. It is characterised by exaggerated elongation of the stem internodes (Zeigler *et al.*, 1980).

SYMPTOMS

The characteristic symptom is exaggerated elongation of the stem internodes (Figure 106), which gives the disease its name. Other symptoms include distortion of the young leaves and cankers of different shapes and sizes on nerves underneath the leaves and stems. Affected leaves have white irregular spots on the blades, midribs, veins and leaf stems. The stems are thin and weak; the diseased plants are much taller than normal (Álvarez *et al.*, 2012b).

TRANSMISSION AND SPREAD

The disease can be transmitted in splashed rainwater and spread through infected cassava planting materials from the previous season's crops, crop debris or alternative plant hosts such as *Manihot* spp., *Euphorbia brasiliensis*, *Jatropha curcas* and *J. aconitifolia* var. *papaya* (Ziegler *et al.*, 1984). Shoot tips from residue plants are a major source of inoculum (spores), can survive for 6 months and are blown by wind.

DAMAGE

Superelongation disease causes necrosis of cassava leaves, which may result in severe defoliation, thinning and development of weak stems and eventually dieback (Lozano and Booth, 1976).

IMPACT ON YIELD AND QUALITY

Losses can exceed 80% of total production in young crops, but significant losses do not occur in crops that are more than 6 months old (Lozano and Booth, 1976; Álvarez *et al.*, 2012b).

GEOGRAPHICAL DISTRIBUTION

Superelongation disease was reported in Tanzania in Africa and several countries in the Caribbean (Table 34).

Table 34 — Geographical distribution of superelongation disease on cassava in the ACP countries

AFRICA	CARIBBEAN	PACIFIC
Tanzania	Barbados, Brazil, Colombia, Costa Rica, Cuba, Dominican Republic, Guatemala, Nicaragua, Panama, Thailand, Trinidad and Tobago, Venezuela	Not reported

QUARANTINE ORGANISM

Planting materials, including leaves, should be treated using meristem culture to eliminate the disease before introduction into another country.

FAVOURABLE CONDITIONS

Superelongation disease development and spread is favoured by wet conditions with annual mean temperatures of 20–28°C, relative humidity of 90–100% and rainfall of >1500 mm.

PEST LIFE CYCLE

Temperatures of 20–23°C and relative humidity of 90–100% increase the disease incidence. Minute spores, produced in the scabby lesions and spread by rain-splash and wind, germinate in free moisture and penetrate leaf surfaces directly.

SURVEILLANCE/MONITORING

Look for plants with typical symptoms of superelongation disease, including exaggerated elongation of stem internodes, distortion of young leaves and cankers of different shapes and sizes on nerves underneath the leaves, leaf stems and stems. The plants might also express white irregular spots on the blades, midribs, veins and leaf stems. The stems may be thin and weak, and the infected plants are much taller. Count the number of diseased plants to determine if and when to commence IPM measures.

CULTURAL CONTROL

- Plant healthy cassava cuttings.
- Practice crop rotation with cereal crops (maize, sorghum, millet or rice) or grasses.
- Plant cassava crops at the end of the rainy season.

BIOCONTROL

- Spraying with *Pseudomonas putida* successfully minimised the severity of damage caused by super elongation disease with increased yields (CIAT, 1985).

CONTROL WITH PLANT PROTECTION PRODUCTS

- Treating by immersing cassava cuttings for 5 minutes in copper-oxychloride solution at 3 to 5 g/L concentration (Álvarez *et al.*, 2012b) reduced the effect of the disease.
- Spray copper oxychloride at rates of 1 kg/ha on affected cassava plants.

OTHER CONTROL METHODS

- Use resistant varieties (Zeigler, 1982) such as the Colombian Agriculture and Livestock Institute (ICA)-Catumare or ICA-Cebucán in Colombia has proved successful in controlling the disease.

7.6. VIRAL DISEASES

7.6.1. CASSAVA MOSAIC DISEASE

COMMON AND SCIENTIFIC NAMES

Cassava mosaic *begomoviruses* (*Geminiviridae*: *Begomoviruses*)

- *African cassava mosaic virus*
- *East African cassava mosaic virus*
- *South African cassava mosaic virus*



Figure 107 — Showing severe leaf symptoms of cassava mosaic disease on:
(A) a leaf and (B) field and (C) constricted tubers. Source: Pacific Pests, Pathogens & Weeds,
https://apps.lucidcentral.org/pppw_v10/text/web_full/entities/cassava_brown_streak_disease_439.htm

DESCRIPTION/IDENTIFICATION

PEST

Cassava mosaic disease (CMD) is one of the two most economically important viral diseases limiting cassava production in sub-Saharan Africa. It is caused by six species of DNA viruses generally called cassava mosaic *begomoviruses* (CMBs), family *Geminiviridae*: genus *begomovirus*, including (Legg *et al.*, 2014):

- *African cassava mosaic virus* (ACMV)
- *East African cassava mosaic virus* (EACMV)
- *East African cassava mosaic Cameroon virus* (EACMCV)
- *East African cassava mosaic Malawi virus* (EACMMV)
- *East African cassava mosaic Zanzibar virus* (EACMZV)
- *South African cassava mosaic virus* (SACMV)

SYMPTOMS

Cassava mosaic disease (CMD) is typically expressed as yellowing mosaic patterns with distortion of the leaf surface, mottling, thinning of leaves (Figure 107A) and stunting of plants (Figure 107B) in severe infections (Sseruwagi *et al.*, 2004).

TRANSMISSION

The causal viruses are disseminated in infected cassava planting material obtained from the previous season's crops and transmitted by the whitefly (*Bemisia tabaci*) in sap during feeding from infected to healthy plants.

SPREAD

Key factors for spread of these viruses are cassava varieties, proximity of other fields or source of inoculum, crop density and virus strains present (Legg *et al.*, 2011). In addition, whiteflies can disperse CMBs both locally within and between cassava fields (Fargette *et al.*, 1990) and over a long distance by wind (Maruthi *et al.*, 2017).

DAMAGE

Compared to cassava from healthy cassava plants, CMD causes reduced tuber size, making them unmarketable but edible (Figure 107C).

IMPACT ON YIELD AND QUALITY

Cassava mosaic disease causes reduced plant growth, leading to yield loss of up to 100% in the severely affected plants (Legg *et al.*, 2014).

GEOGRAPHICAL DISTRIBUTION

Cassava mosaic disease only occurs on cassava in Africa (Table 35). It has also been reported in parts of Asia.

Table 35 — Geographical distribution of cassava mosaic disease on cassava in the ACP countries

AFRICA	CARIBBEAN	PACIFIC
Angola, Benin, Burkina Faso, Burundi, Cameroon, Central African Republic, Comoros, Democratic Republic of the Congo, Republic of the Congo, Côte d'Ivoire, Ghana, Kenya, Madagascar, Malawi, Mali, Mauritius, Mayotte, Niger, Nigeria, Réunion, Rwanda, South Africa, South Sudan, Tanzania, Togo, Uganda	Not reported	Not reported

QUARANTINE ORGANISM

The disease occurs widely in sub-Saharan Africa in east, west, central and southern Africa, wherever cassava is grown, in addition to having a wide host range. Hence, quarantine measures may only be instituted to restrict the movement of CMD diseased-planting materials from one region to another in case of an epidemic of a new strain or species of CMBs. However, as CMD spread is not known to occur in the tuberous roots, no restrictions have been imposed on trade in the cassava tubers in Africa or Europe.

FAVOURABLE CONDITIONS

The spread and symptoms of CMD are usually variable and irregular depending on plant age, cultivar (genotypes), environmental conditions (altitude, temperature and rainfall quantity) and virus species (Navangi *et al.*, 2020). High temperatures following the rainy season led to an increase in whitefly populations, with a resultant increase in the spread of CMD (Sseruwagi *et al.*, 2003). The direction and speed of the prevailing wind also influences the side of the cassava field that is infected and the rate of spread within and between the fields (MacFadyen *et al.*, 2018).

PEST LIFE CYCLE

Transmission of CMBs depends on the life cycle of whitefly (*B. tabaci*), which spans around 21 days, depending on the climate. Adult whiteflies feed for around 3 hours to acquire the viruses from diseased plants, after which, following which a latent period of at least 8 hours, around 10 minutes is needed to transmit the virus to healthy plants. Whiteflies can stay infective for about 7–9 days with no loss of the virus during moulting. Only about 10% of the virus-carrying adults were reported to transmit CMBs (Fargette *et al.*, 1990), which can be significant with abundant populations (Legg, 1999).

In newly planted fields, CMD infections appear in the first month after planting when whitefly begin to infest the crop. Once infected, cassava plants retain the virus throughout their growth, except for the CMD resistant varieties where reversion/recovery from the viral infection occurs.

SURVEILLANCE/MONITORING

Surveillance for CMD is guided by the objective and availability of resources. Monitoring should be designed around the life cycle of the disease, which is usually more evident on young (3–6 month) rather than old (>6 month) cassava crops. Monitoring for new infections should be carried out bi-weekly or monthly, as it takes approximately 14–21 days for viral symptoms to appear in field conditions (Sseruwagi *et al.*, 2004). In each field, plants should be assessed for presence or absence of CMD symptoms along two diagonal transects using an 'X' sampling design. The number of plants with CMD symptoms are counted and reported. Symptom severity is scored using a 1–5 scale, where 1 is no symptoms (visually healthy) and 5 represents severe damage with stunting (Hahn *et al.*, 1980b). The CMD-affected plants are identified either as cutting ('C') or whitefly ('W') borne infections, where the symptoms appear only on the lowest first formed or upper-most formed leaves, respectively (Sseruwagi *et al.*, 2004). The data can be collected on a paper form or advanced iForm app using a smart phone or a Tablet (Sseruwagi *et al.*, 2017).

CULTURAL CONTROL

Isolation of new cassava crops from older fields that may be infected with CMD.

- Use of disease-free planting material, confirmed by visual inspection of absence of viral disease symptoms from the source fields, or using novel virus-indexing molecular technology helps to prevent or delay virus infections in new crops and to

reduce damage with sufficient time to produce economic yield before the plants are overwhelmed by the disease, especially in disease-susceptible or tolerant cultivars.

- Roguing or removal of cassava plants infected with CMD in young crops (<3 months old), followed by replanting healthy planting materials.
- Practice crop hygiene at harvest by removing and burning crop debris/residues infected with CMBs/CMD that may serve as sources of inoculum for further spread of the viruses from the previous season.
- Clean weeding of cassava crops to remove potential alternative host plant species for either the whitefly vectors and/or cassava-infecting viruses within the immediate surrounding fields and bushes to new cassava crops.
- Intercropping cassava with maize at high density can lower whitefly populations (Fargette and Fauquet, 1988; 1990), which reduces the spread of CMD (Night *et al.*, 2011).
- Establish repellent crops such as marigold near or around cassava fields to repel whiteflies.
- Provide sufficient water for irrigation to cassava crops to avoid stress which encourages whitefly infestation that may transmit the viruses that cause CMD.
- Harvesting cassava crops early helps to avoid severe losses due to reduced pest and disease effects.

BIOCONTROL

- Use of natural enemies; parasitoid wasp species *Encarsia sophia* and *Eretmocerus mundus* proved to be more effective in controlling whiteflies on cassava crops, with parasitism rates of 34–58% of 4th instar whitefly nymphs (Legg, 1995). Reduction of whitefly populations in a cassava field, reduces the chances of spreading the viruses to more plants.

CONTROL WITH PLANT PROTECTION PRODUCTS

- No plant protection products have been reported to control CMD, but several can be used to control the whitefly vectors. See sections above on control of whiteflies using plant protection products, which may help reduce the whitefly vector populations that could spread CMD to other crops.

OTHER CONTROL METHODS

- Use of elite cassava varieties with proven resistance or tolerance to CMD (Thresh *et al.*, 2005; Thuy *et al.*, 2021).
- Hot water therapy (25–60°C) and two rounds of meristem tip culture of CMD-affected cassava planting materials reduced the causal viral loads by 100% (Nakabonge *et al.*, 2020).

7.6.2. CASSAVA BROWN STREAK DISEASE

COMMON AND SCIENTIFIC NAME

Cassava brown streak viruses (*Potyviridae: Ipomovirus*)

- *Cassava brown streak virus*
- *Uganda cassava brown streak virus*

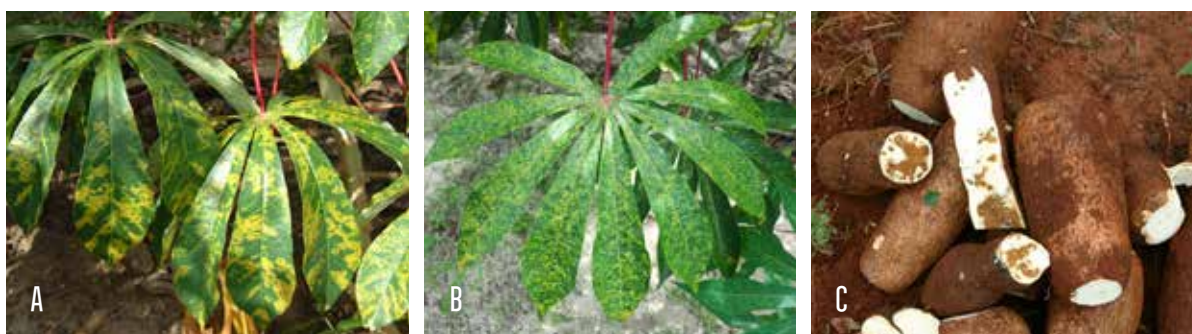


Figure 108 — Showing severe symptoms of cassava brown streak disease on (A & B) lower leaves of cassava plants and (C) necrosis – browning on the tubers

Source: PlantVillage, <https://plantvillage.psu.edu/topics/cassava-manioc/infos>; Pacific Pests, Pathogens & Weeds, https://apps.lucidcentral.org/pppw_v10/text/web_full/entities/cassava_brown_streak_disease_439.htm

DESCRIPTION/IDENTIFICATION

PEST

Cassava brown streak disease (CBSD) is caused by two distinct RNA viruses: cassava brown streak virus (CBSV) and Ugandan cassava brown streak virus (UCBSV), both of which belong to the genus *Ipomovirus* in the family *Potyviridae* (Mbanzibwa *et al.*, 2011).

SYMPTOMS

Cassava brown streak disease causes yellowing of older leaves, which appear yellowish and mottled with feathery-like patterns along the minor veins (Figure 108A & B). Streaks develop on the young green stems, while the tubers develop necrosis with brown colouration (Figure 108C) on the edible flesh (Rwegasira *et al.*, 2011) and constriction of tubers (Hillocks *et al.*, 2002; Legg and Hillocks, 2003). The symptoms vary depending on cultivar and the affected plant part. The leaves may show symptoms, while the roots appear healthy.

TRANSMISSION

Cassava brown streak viruses are disseminated in infected cassava planting material obtained from the previous season's crops and transmitted by the whitefly (*Bemisia tabaci*) in sap during feeding from infected to healthy plants (Maruthi *et al.*, 2017).

SPREAD

Key factors for spread of CBSD include cassava varieties, proximity of other fields or source of inoculum, crop density and virus strains present (Legg *et al.*, 2011). In addition, whiteflies can disperse CBSVs both locally and for short distances between cassava fields (Gwandu *et al.*, 2019). Long-distance spread is mainly through transportation of diseased planting materials (Maruthi *et al.*, 2017). Alternative hosts such as the Ceara rubber tree (*Manihot glaziovii*) also play an important role in retaining virus to propel disease spread.

DAMAGE

Compared to cassava from healthy cassava plants, CBSD causes the most damage on the tubers, which develop necrosis that renders it unfit for human consumption.

IMPACT ON YIELD

The disease is estimated to cause yield losses of 25–100%, depending on the susceptibility of cassava varieties in the high-pressure regions with retarded plant growth (Legg *et al.*, 2011). The disease causes browning of the edible part, resulting in reduced quality of food and leading to high risk of food insecurity in countries where cassava is a main staple food crop (Legg *et al.*, 2011). It was reported to be responsible for losses of at least US\$750 million annually in East and Southern Africa (Legg *et al.*, 2011).

GEOGRAPHICAL DISTRIBUTION

Cassava brown streak disease only occurs on cassava in Africa (Table 36).

Table 36 — Geographical distribution of cassava brown streak disease on cassava in the ACP countries

AFRICA	CARIBBEAN	PACIFIC
Burundi, Cameroon, Central Africa Republic, Democratic Republic of Congo, Kenya, Malawi, Mozambique, Rwanda, South Africa, South Sudan, Tanzania, Uganda, Zambia	Not reported	Not reported

QUARANTINE ORGANISM

Cassava brown streak disease is endemic in the eastern African Coastal lowland areas, where it was first reported in the 1930s in Tanzania. However, in the last decade the disease has spread inland into east, central and southern Africa, and threatens to spread to west Africa. Therefore, CBSD is considered a major quarantine disease in west Africa and all other cassava producing areas, where it is not yet found. Strict

measures have been put in place by phytosanitary departments at border posts to contain the entry of the disease into west Africa, the largest producer of cassava in Africa. Quarantine measures also control movement of CBSD-affected materials into Asia, the Caribbean, South and North America and the Pacific islands, where cassava is also produced (Legg and Hillocks, 2003).

FAVOURABLE CONDITIONS

The spread of CBSVs is favoured by high temperatures following the rainy season when whitefly populations are abundant (Boissot *et al.*, 2010; Sseruwagi *et al.*, 2003). The direction and speed of the prevailing wind also influenced the side of the cassava field that is infected and the rate of spread within and between the fields (MacFadyen *et al.*, 2018).

PEST LIFE CYCLE

Transmission of CBSVs depends on the life cycle of whitefly (*B. tabaci*), which spans around 21 days, depending on the climate (temperature). Adult whitefly feed for around 3 hours to acquire the viruses from diseased plants, after which, following a latent period of at least 8 hours, about 10 minutes is needed to transmit the virus to healthy plants. Whiteflies can stay infective for about 7–9 days with no loss of the virus during moulting. Only around 10% of the virus-carrying adults were reported to transmit the disease (Fargette *et al.*, 1990), which can be significant with abundant populations (Legg *et al.*, 2011).

SURVEILLANCE/MONITORING

Cassava brown streak disease should be monitored regularly, depending on the objective and resources available. Otherwise, it is recommended that cassava fields are surveyed and assessed for new infections bi-weekly or monthly, as it takes approximately 14–21 days for viral symptoms to appear in field conditions (Sseruwagi *et al.*, 2004). Monitoring can be carried out by sampling major cassava-growing areas for plants with typical CBSD symptoms, such as yellow blotches along the veins at midribs, brown streaks on the stems, and dry brown root rots, following along major transects (or motorable roads) traversing the sampling domain. Stops are made at regular intervals and a cassava field of crop ages 3–6 months after planting is assessed for presence or absence of CMD or CBSD. The incidence, symptom severity of infected leaves, stems or (tubers for CBSD) and adult whitefly abundance (Sseruwagi *et al.* 2004) are recorded. Data is either entered in a paper field data collection form or iForm app on a Tablet or smart phone. The leaf samples for detecting virus presence and identity are collected in sampling bags and stored on ice in a cool box, while adult whiteflies are collected using a pooter and stored in 70% ethanol until laboratory analysis. It is advisable to carry out whitefly surveys at peak population and CMD and CBSD in the following months.

CULTURAL CONTROL

- Isolation of new cassava crops from older fields that may be infected with CBSD.
- Use of disease-free planting material confirmed by visual inspection of absence of viral disease symptoms from the source fields, or by using novel virus-indexing molecular technology helps to prevent or delay virus infections in new crops and to reduce damage with sufficient time to produce economic yield before the plants are overwhelmed by the disease, especially in disease susceptible or tolerant cultivars.
- Rogue/remove cassava plants with CBSD symptoms and replant healthy planting materials.
- Practice crop hygiene at harvest by removing and burning crop debris/residues infested with CBSD that may serve as sources of inoculum for further spread of the causal viruses from the previous season.
- Clean weeding of cassava crops to remove potential alternative host plant species for either the whitefly vectors and/or cassava-infecting viruses within the immediate surrounding fields and bushes to new cassava crops.
- Harvest crop early to avoid severe losses due to necrosis of tubers.

BIOCONTROL

- Use of the natural enemies; parasitoid wasp species *Encarsia sophia* and *Eretmocerus mundus* have proved to be more effective in controlling whiteflies on cassava crops, with parasitism rates of 34–58% of 4th instar whitefly nymphs (Legg, 1995). Hence, the reduction of whitefly populations can help control the spread of CBSD.

CONTROL WITH PLANT PROTECTION PRODUCTS

- No plant protection products have been reported to control CBSD, but several can be used to control the whitefly vectors. See section on control of whiteflies using plant protection products, which may help reduce the whitefly vector populations that could spread CBSD to other crops.

OTHER CONTROL METHODS

- Screen plants for absence of CBSVs using molecular tools before harvesting the planting materials.
- Grow elite cassava varieties with proven resistance or tolerance to CBSD and whitefly.
- Hot water therapy (25–60°C) and two rounds of meristem tip culture of CBSD-infected cassava planting materials reduced the causal viral loads by 100% (Nakabonge *et al.*, 2020).

7.6.3. CASSAVA COMMON MOSAIC DISEASE

COMMON NAMES

- Cassava common mosaic virus
- Brazilian cassava common mosaic virus
- Cassava common mosaic *potexvirus*



Figure 109 — Showing severe leaf symptoms of cassava common mosaic disease

Source: Food and Agriculture Organization,

https://bioversityinternational.org/fileadmin/_migrated/uploads/tx_news/Cassava_349.pdf

[consulted on 5 January 2023]

DESCRIPTION/IDENTIFICATION

PEST

Cassava common mosaic disease (CCMD) is caused by a *potexvirus* called cassava common mosaic virus, also known as Brazilian cassava common mosaic virus or cassava common mosaic *potexvirus* (Calvert and Thresh, 2002).

SYMPTOMS

Disease-affected cassava leaves are distorted with yellow, white or light and dark green spots that look like blisters (Figure 109). The mosaic and chlorotic symptoms can be dark and with light green patches separated by the veins. In severe infection there is stunted plant growth. The leaf stalks develop a characteristic S-shape (Calvert and Thresh, 2002).

TRANSMISSION

There is no known vector of CCMV and the primary source of inoculum is infected planting material. Infected stem cuttings and garden tools can also carry the virus (Calvert and Thresh, 2002).

SPREAD

The disease spreads by planting CCMV-infected cassava planting materials (Calvert and Thresh, 2002) and on farm tools (Lozano and Nolt, 1989).

DAMAGE

Severely affected cassava plants are stunted.

IMPACT ON YIELD AND QUALITY

The disease is suspected to be able to cause economic impact if the symptoms are severe.

GEOGRAPHICAL DISTRIBUTION

Cassava common mosaic disease occurs in a few countries in Africa, the Caribbean and Pacific (Table 37).

Table 37 — Geographical distribution of cassava common mosaic disease on cassava in the ACP countries

AFRICA	CARIBBEAN	PACIFIC
Côte d'Ivoire	Argentina, Brazil, Colombia, Paraguay, Peru, Venezuela	Tuvalu

QUARANTINE ORGANISM

In the affected areas/countries, phytosanitary control measures should be imposed to minimise losses due to the disease. Plant materials should be checked for absence of the virus and if present, they must be treated using thermotherapy and meristem tip culture to avoid spread of the virus.

FAVOURABLE CONDITIONS

The disease is prevalent in cool semi-tropical areas in South America (Calvert and Thresh, 2002).

PEST LIFE CYCLE

There is no known insect vector of CCMV, so the main mode of transmission and spread is by infected planting materials (Calvert and Thresh, 2002).

SURVEILLANCE/MONITORING

Monitor plants with yellow, white or light and dark green spots that look like blisters and mosaic and chlorotic (yellowing) symptoms that are dark with light green patches separated by the veins and S-shaped stalks (Calvert and Thresh, 2002).

CULTURAL CONTROL

- Rogue out the CCMD-diseased plants and debris from the field and burn them.
- Disinfect farm tools such as cutting equipment used to harvest cassava stems.

BIOCONTROL

Not reported

CONTROL WITH PLANT PROTECTION PRODUCTS

Not applicable

OTHER CONTROL METHODS

- None reported

7.6.4. CASSAVA LEAF VEIN MOSAIC DISEASE

SCIENTIFIC NAME

- Cassava vein mosaic virus
- Cassava vein mosaic *caulimovirus*



Figure 110 — Showing severe leaf symptoms of cassava leaf vein mosaic disease

Source: Food and Agriculture Organization, [consulted on 5 January 2023]

https://bioversityinternational.org/fileadmin/_migrated/uploads/tx_news/Cassava_349.pdf

DESCRIPTION/IDENTIFICATION

PEST

Cassava leaf vein mosaic disease is caused by Cassava vein mosaic virus or Cassava vein mosaic *caulimovirus* (CsVMV/CVMV), *Caulimovirus* genus.

SYMPTOMS

Leaf symptoms occur in flushes with yellowing of veins on leaves appearing as a chevron pattern (Figure 110). The yellowed areas form a pattern of rings that join to create a circular spot. Leaves with severe symptoms often have deformed blades and downward-pointing leaves. Sometimes, symptoms disappear as their expression is influenced by climatic conditions. Leaves of infected plants become prematurely old and fall, reducing leaf area (Calvert *et al.*, 2012).

TRANSMISSION AND SPREAD

There is no known insect vector of CsVMV. The virus is mainly spread through infected planting materials (Calvert *et al.*, 2012).

DAMAGE

The disease does not affect plant vigour and most cassava varieties tolerate CsVMV/CVMV and produce stems of normal appearance that could be used as good planting material.

IMPACT ON YIELD AND QUALITY

The disease causes more losses when it occurs at the beginning of the cropping cycle.

GEOGRAPHICAL DISTRIBUTION

Cassava leaf vein mosaic disease has only been reported in Brazil in South America (Table 38).

Table 38 — Geographical distribution of cassava leaf vein mosaic disease on cassava in the ACP countries

AFRICA	CARIBBEAN/SOUTH AFRICA	PACIFIC
Not reported	Brazil	Not reported

QUARANTINE ORGANISM

Plant materials should be checked for signs of the virus and, if present, treated using thermotherapy and meristem-tip culture to avoid spread of the virus.

FAVOURABLE CONDITIONS

The disease is prevalent in cool semi-tropical areas in South America (Calvert and Thresh, 2002).

PEST LIFE CYCLE

Although the causative agents are *Caulimoviruses*, which are usually aphid-transmitted, the natural vector of CsVMV is unknown. However, the transmission and spread primarily occurs through movement of infected planting materials.

SURVEILLANCE/MONITORING

Monitor plants for leaf symptoms with yellowing of veins on leaves appearing as a chevron pattern or rings that join to create circular spots (Calvert *et al.*, 2012). Count the number of symptomatic plants in the field/area to determine if and when to deploy control measures.

CULTURAL CONTROL

Remove all CsVMV-infected plant debris with the symptoms from the field and burn them.

BIOCONTROL

Not reported

CONTROL WITH PLANT PROTECTION PRODUCTS

Not applicable.

OTHER CONTROL METHODS

None reported

7.6.5. OTHER VIRAL DISEASES/VIRUSES OF CASSAVA

SCIENTIFIC NAMES

- Cassava virus X (*Potexvirus*) (CsXV)
- Cassava Colombian symptomless virus (*Potexvirus*) (CCSpV)
- Cassava symptomless infections (Cassava American latent virus - CsALV, Cassava Ivorian bacilliform virus - CIBV)
- Cassava frogskin disease (Cassava frogskin Associated Virus - CsFSaV)



Figure 111 — Showing symptoms of cassava frogskin disease on cassava tuber
Source: CIAT, <http://gcp21.org/Bellagio2/Ceballos.pdf>

VIRAL DISEASE	DESCRIPTION / IDENTIFICATION / SYMPTOMS	TRANSMISSION AND SPREAD	DAMAGE / IMPACT
Cassava virus X (<i>Potexvirus</i>) (CsXV)	No symptoms on cassava	CsXV is transmitted through vegetative propagation. The vector is unknown (Frison and Feliu, 1991).	[Photo not available]
Cassava Colombian symptomless virus (<i>Potexvirus</i>) (CCSpV)	No known symptoms	CCSpV is transmitted through vegetative propagation. No vectors of CCSpV are reported (Frison and Feliu, 1991).	[Photo not available]

VIRAL DISEASE	DESCRIPTION / IDENTIFICATION / SYMPTOMS	TRANSMISSION AND SPREAD	DAMAGE / IMPACT
Cassava symptomless infections (Cassava American latent virus - CsALV, Cassava Ivorian bacilliform virus - CIBV)	No known symptoms for CIBV and CsALV on cassava	Similarly, the vector for CIBV is unknown, while for CsALV, the virus can be mechanically transmitted to cassava (Frison and Feliu, 1991).	[Photo not available]
Cassava frogskin disease (Cassava frogskin Associated Virus - CsFSaV) Also known as “lagarto-jacaré” or “caiman-lizard skin disease”	Symptoms vary according to temperature and genotype. Most infected plants do not present visible signs on the leaves and appear healthy and vigorous. The stems of infected plants thicken at the base. The root symptoms can be mild and show constrictions (Calvert <i>et al.</i> , 2012).	The virus is transmitted through grafts and by whitefly <i>Bemisia tuberculata</i> (Frison and Feliu, 1991) although at very low efficiency (Calvert <i>et al.</i> , 2012). The whitefly <i>Aleurotrachelus socialis</i> was associated with the transmission of cassava frogskin virus (Angel <i>et al.</i> , 1987).	The roots are thin and woody with a thick, cork-like peel with fibres. Economic losses occur due to lack of starch accumulation in the roots, leading to low yields or up to 90% yield loss (Calvert <i>et al.</i> , 2012).

GEOGRAPHICAL DISTRIBUTION

Occurrence of other viral diseases affecting cassava occur on in some countries in the Caribbean (Table 39).

Table 39 — Geographical distribution of other viral diseases affecting cassava in the ACP countries

VIRAL DISEASE	AFRICA	CARIBBEAN	PACIFIC
Cassava virus X (<i>Potexvirus</i>) (CsXV)	Not reported	Colombia	Not reported
Cassava Colombian symptomless virus (<i>Potexvirus</i>) (CCSpV)	Not reported	Colombia	Not reported
Cassava symptomless infections (Cassava American latent virus - CsALV, Cassava Ivorian bacilliform virus - CIBV)	Côte d'Ivoire	Brazil, Guyana	Not reported
Cassava frogskin disease (Cassava frogskin Associated Virus - CsFSaV)	Not reported	Brazil, Colombia, Costa Rica, Panama, Peru, Venezuela	Not reported

QUARANTINE ORGANISM

Restrictions are in place for cassava materials from countries infected with Cassava virus X, Cassava Colombian symptomless virus, Cassava symptomless infections virus, Cassava Ivorian bacilliform virus and Cassava frogskin disease. Materials may be imported only under strict conditions following testing with molecular tools and treatment by thermotherapy and meristem-tip culture to avoid spread of the viruses.

FAVOURABLE CONDITIONS

The viruses listed above require cool conditions to thrive, while dry or hot conditions reduce development of symptoms (Calvert *et al.*, 2012).

PEST LIFE CYCLE

Except for CsXV, CCSpV and CsALV have no known natural vectors, but are vegetatively transmitted. The disease cycle is maintained through repeated use of diseased planting materials. Cassava frogskin Associated Virus (CsFSaV) can be transmitted vegetatively and by whitefly *Bemisia tuberculata* (Frison and Feliu, 1991).

SURVEILLANCE/MONITORING

Look for the symptoms of each of these viral diseases and rogue and burn the plant materials if the incidence is more than 10% (Álvarez *et al.*, 2012).

CONTROL WITH PLANT PROTECTION PRODUCTS

Not reported

OTHER CONTROL METHODS

Not reported

7.7. PHYTOPLASMA DISEASES

7.7.1. CASSAVA ANTHOLYSIS

SCIENTIFIC NAME

Cassava Antholysis



Figure 112 — Showing cassava antholysis syndrome

Source: Emile Frison/ResearchGate, https://www.researchgate.net/figure/Cassava-antholysis-tepals-of-flowers-show-virescence-and-phyllody-leaf-like-structures_fig10_281633837

DESCRIPTION/IDENTIFICATION

PEST

Antholysis in cassava plants is caused by mycoplasma-like organisms (Jayasinghe *et al.*, 1984; Frison and Feliu, 1991). However, the disease does not have significant economic importance and is only seldom observed (Alvarez *et al.*, 2012a).

SYMPTOMS

Symptoms include the development of green pigment in parts of the plant that are not normally green, with pinkish-cream tepals; tissue overgrowth and phyllody, a condition where parts of the flower are replaced with leaves (Figure 112). This produces a syndrome known as antholysis. The centre of the flower elongates, while

the deformed flowers are sterile and prematurely abort. The malformed flowers can become necrotic. The flower head may change colour from pink to green. (Alvarez, *et al.*, 2012a). The affected plants lack symptoms in other organs (Jayasinghe *et al.*, 1984).

TRANSMISSION AND SPREAD

Transmitted by vegetative propagation (Frison and Feliu, 1991).

DAMAGE

Flower fertility is lost, resulting in non-functional flowers that abort prematurely (Alvarez, *et al.*, 2012a). This could limit plant-breeding efforts by cassava programmes or sexual seed production programmes for germplasm exchange projects.

IMPACT ON YIELD AND QUALITY

Flower fertility is lost, resulting in non-functional flowers that abort prematurely (Alvarez, *et al.*, 2012a).

GEOGRAPHICAL DISTRIBUTION

Cassava antholysis was only reported in a few countries in South America (Table 40) (Frison and Feliu, 1991).

Table 40 — Geographical distribution of cassava antholysis in the ACP countries

AFRICA	CARIBBEAN/SOUTH AMERICA	PACIFIC
Not reported	Brazil, Central America, Colombia, Venezuela	Not reported

QUARANTINE ORGANISM

Plant materials should be checked for absence of the virus and if present, they must be treated using thermotherapy and meristem-tip culture to avoid spread of the virus.

FAVOURABLE CONDITIONS

The viruses listed above require cool conditions to thrive, while dry or hot conditions reduce development of symptoms (Calvert *et al.*, 2012a).

PEST LIFE CYCLE

Planting infected materials is the only known means of propagating the disease.

SURVEILLANCE/MONITORING

Monitor fields for plants with pinkish-cream tepals, with elongated flowers and deformed necrotic flowers that abort prematurely. The petals may look like leaves ‘antholysis’ (Alvarez *et al.*, 2012a). Although the occurrence of plants with these symptoms may be low, it is important to remove them from the field to avoid spreading the disease further.

CULTURAL CONTROL

- Select planting materials from healthy plants (Alvarez *et al.*, 2012a).

BIOCONTROL

Not reported

CONTROL WITH PLANT PROTECTION PRODUCTS

Not reported

OTHER CONTROL METHODS

- Varietal resistance exists (Alvarez *et al.*, 2012a).

7.7.2. CASSAVA WITCHES' BROOM

SCIENTIFIC NAME

Candidatus Phytoplasma aurantifolia



Figure 113 — Showing symptoms of Witches' broom disease as (A & B) dwarfed growth of cassava shoots and (C) change in colour and oozing of broken stems
Source: CIAT, <https://www.flickr.com/photos/ciat/13505169343/in/photolist>

DESCRIPTION

PEST

Cassava Witches' broom (CWB) disease caused by *phytoplasma*, *Candidatus phytoplasma aurantifolia*, which lives within the phloem of cassava stems (Alvarez *et al.*, 2013).

SYMPTOMS

Plants are stunted (or dwarfed) with an excessive distortion of branches (Figure 113A); shoots have small leaves and short internodes (Figure 113B), but without distortion or chlorosis (Frison and Feliu, 1991). Affected cassava plants have weak shoots (Figure 113C), swollen lower stems and thin, fibrous tubers that have rough outer skins with deep cracks and low starch content (Alvarez, *et al.*, 2012a).

TRANSMISSION AND SPREAD

Leaf hoppers are suspected to be the most likely vectors of CWB disease although the mechanism of transmission is still under investigation. The leafhopper species can transmit the pathogens to several plant hosts (Weintraub and Beanland, 2006), including the sticky mallow (*Malvaviscus arborus*), croton (*Codiaeum variegatum*), Chinese hibiscus (*Hibiscus rosa-sinensis*) and passion fruit (*Passiflora edulis*), which may grow near or within cassava fields (Raj *et al.*, 2009). The disease is mainly spread by humans through transportation of CWB-diseased cassava materials into new uninfected areas.

DAMAGE

Cuttings from the CWB-affected shoots are weak and only a few shoots successfully grow from such cuttings.

IMPACT ON YIELD AND QUALITY

The disease will cause yield loss of 30–80% (Lozano *et al.*, 1981; Alvarez *et al.*, 2012b; 2013). The disease causes a general reduction in quantity and quality of cassava yield.

GEOGRAPHICAL DISTRIBUTION

Cassava witches broom occurs in Africa, the Caribbean and Pacific (Table 41) (Botti and Bertaccini, 2006; Tairo *et al.*, 2006; Arocha *et al.*, 2008; Hodgetts *et al.*, 2008; Arocha *et al.*, 2009; Davis and Tsatsia, 2009; Lee *et al.*, 2010).

Table 41 — Geographical distribution of cassava witches broom disease on cassava in the ACP countries

AFRICA	CARIBBEAN	PACIFIC
Ethiopia, South Africa, Sudan, Uganda	Brazil, Mexico, Venezuela, Peru	Australia (Western Australia), Cambodia, Central Vietnam, Fiji, New Caledonia, Philippines, Solomon Islands, Tonga, South Pacific Commission, Vanuatu, Wallis and Futuna, Western Thailand

QUARANTINE ORGANISM

Cassava witches' broom is not widely distributed globally. Therefore, it is advisable to control the movement of cassava planting materials from CWB-infected to new locations where the disease has not been detected. In addition, the movement of CWB-associated host plants and vectors must be restricted.

FAVOURABLE CONDITIONS

Moderate temperatures (13–20°C) favour disease establishment, and at higher temperatures the symptoms tend to disappear (Frison and Feliu, 1991).

PEST LIFE CYCLE

Although some vectors such as leaf hoppers are suspected to transmit the disease, the most likely means is by planting CWB-infected planting materials (Alvarez *et al.*, 2012b).

SURVEILLANCE/MONITORING

Monitor regularly for CWB symptoms on leaves, shoots, stems and tubers (where it is possible to sample the affected plants), and act on management as soon as possible before the disease spreads throughout the field. Take care not to confuse the symptoms (bunchy tops and leaf folds) with those by the cassava mealybug (*Phenacoccus manihoti*), which leave whitish woolly egg sacks and ants.

CULTURAL CONTROL

- Do not establish new cassava fields next to CWB-infected cassava.
- Plant disease-free planting materials.
- Remove and destroy CWB-affected plants.
- Remove all CWB-affected plant debris from field after harvest.
- Disinfect farm tools and equipment between harvesting cassava cuttings.
- Look out for and restrict movement of cassava relatives such as jatropha which could be infected with CWB.
- Remove weeds and shrubs within and in immediate surrounding of cassava fields that could act as alternate hosts and reservoirs for the disease.

BIOCONTROL CONTROL

- Not reported

CONTROL WITH PLANT PROTECTION PRODUCTS

- Not reported

OTHER CONTROL METHODS

- Use of elite cassava varieties with proven resistance or tolerance to CWB disease (Alvarez *et al.*, 2012a).
- Control leaf hoppers in the field that might be transmitting the disease.

7.8. POST-HARVEST PESTS AND PATHOGENS CAUSING ROOT ROT IN CASSAVA

7.8.1. PESTS OF DRIED CASSAVA OR CHIPS

COMMON AND SCIENTIFIC NAMES

- Red flour beetle (*Tribolium castaneum*)
- Lesser grain borer (*Rhizopertha dominica*)
- Other pests of dried cassava that are not presented include *Sitophilus* sp., *Tenebrionides mauritanicus*, *Trogoderma granarium*, *Araecerus fasciculatus*, *Dinoderus* sp., *Carpophilus* sp., and *Prostephanus truncatus*

A. RED FLOUR BEETLE (*TRIBOLIUM CASTANEUM*)

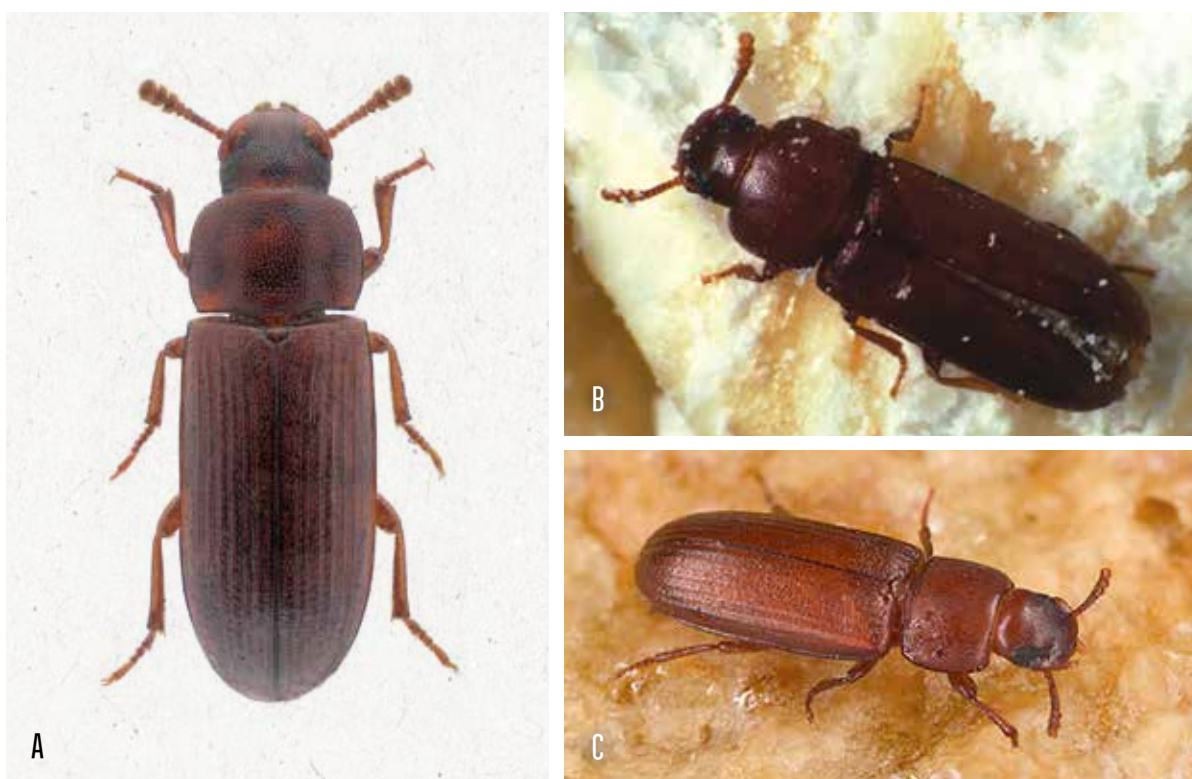


Figure 114 — Showing (A) an adult red flour beetle, (B & C) damage caused to stored cassava

Source: Udo Schmidt, <https://www.flickr.com/photos/coleoptera-us/32875109665/>;

Bayer Crop Science, <https://cropscience.bayer.co.uk/threats/pest-and-slugs/rust-red-flour-beetle/>;

Photo by Peggy Greb, <https://ars.usda.gov/oc/images/photos/nov05/d268-1/>

DESCRIPTION/IDENTIFICATION

PEST

Adult red flour beetles are small (3–4mm long) with a uniform rust, brown or black colour (Figure 114A). The head is pale brown with reddish-black eyes. The upper part of the thorax has tiny punctures and the wing cases are rough at the edges. The last segment of the abdomen has two upturned dark, pointed structures. The antennae are large at the tip and the last three segments are wider than the rest. The red flour beetle has a very broad host range and attacks a wide range of dried stored products (Brower *et al.*, 1991; Brown *et al.*, 2009).

SYMPTOMS

Beetles (adults and larvae) make tunnels in dry food products (Figure 114B). If infestation is severe, the food products develop greyish-yellow colouration due to mould (Figure 114C) and have a bad smell. Sometimes, adult beetles are visible on the surface of the dry food product such as tubers or chips.

DISPERSAL

Adult beetles can fly short distances or move on the ground in search of suitable materials to feed and lay eggs on. The beetles (larvae and adults) can also be transported in infested dry cassava tubers and chips (Romero *et al.*, 2010).

DAMAGE

Beetles (adults and larvae) cause extensive damage to stored dry cassava by making tunnels. The beetles excrete in the product, which destroys food quality.

IMPACT ON YIELD AND QUALITY

Under high infestation, the beetles excrete in the product, destroying food quality and encouraging growth of mould, which causes an unpleasant taste and a serious health risk to both humans and livestock.

GEOGRAPHICAL DISTRIBUTION

Red flour beetles affect cassava postharvest in many countries in Africa, the Caribbean and Pacific (Table 42).

Table 42 — Geographical distribution of red flour betteles on cassava in the ACP countries

AFRICA	CARIBBEAN	PACIFIC
Algeria, Angola, Benin, Botswana, Burkina Faso, Burundi, Cameroon, Central African Republic, Chad, The Democratic Republic of Congo, The Republic of Congo, Côte d'Ivoire, Djibouti, Egypt, Equatorial Guinea, Eswatini, Ethiopia, Gabon, Gambia, Ghana, Guinea, Guinea-Bissau, Kenya, Lesotho, Liberia, Libya, Madagascar, Malawi, Mali, Mauritania, Morocco, Mozambique, Namibia, Niger, Nigeria, Rwanda, Senegal, Seychelles, Sierra Leone, Somalia, South Africa, Sudan, Tanzania, Togo, Tunisia, Uganda, Western Sahara, Zambia, Zimbabwe	Barbados, Belize, Costa Rica, Cuba, Dominican Republic, El Salvador, Guatemala, Haiti, Honduras, Mexico, Nicaragua, Panama, Puerto Rico, Trinidad and Tobago	Australia, Fiji, New Caledonia, Papua New Guinea, Samoa, Tonga, Vanuatu

QUARANTINE ORGANISM

Tribolium castaneum is a prohibited organism in Europe under section 22(1) and susceptible food products may only be imported by permission. Some permits are only applicable to species required for research purposes in secure institutions.

FAVOURABLE CONDITIONS

The optimum conditions for the beetles are 35°C and 60–80% relative humidity, which support the development times for each stage, including the eggs, larvae and pupae.

PEST LIFE CYCLE

The red flour beetle life cycle undergoes four stages (Figure 115). The females lay 300–400 eggs over 5–8 months of their adult life. Under optimum conditions of 35°C and 60–80% relative humidity, the development time for each stage is about 3 days for eggs, 16 days for larvae and 5 days for pupae.

Monitoring for the pest in stored food should begin with careful inspection of the store. Examine the dry cassava tubers, chips, bags or wood with distinct and characteristic tunnels or dust at the entrance of the tunnels. In high numbers, the beetles make a rasping sound while feeding. Estimate the numbers and type by sampling the produce in the bags (if already packed) and the entire store to determine if and when to apply management strategies. If the population is low, hand picking the pest can be useful; however, high numbers require stronger measures to prevent damage to the stored food and spread to other stores.

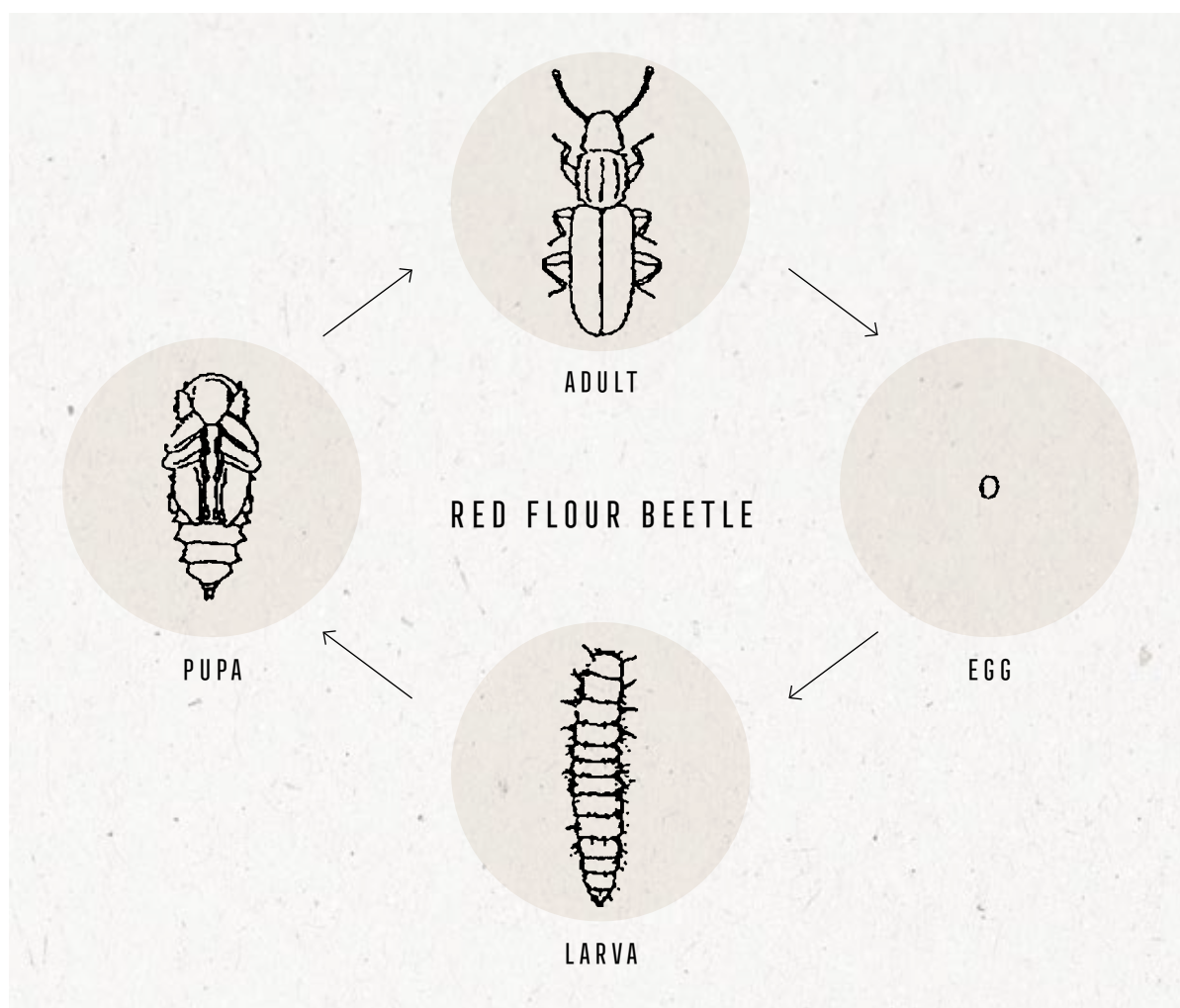


Figure 115 — The life cycle of the red flour beetle

Source: Food and Nutrition Library,

<http://www.nzdl.org/cgi-bin/library?e=d-00000-00---off-0fnl2%2E2--00-0----0-10-0---0---0direct-10---4-----0-1l--11-sk-50---20-preferences---00-0-1-00-0-0-11----0-0-&cl=CL3-.22&d=HASH01b1cf388877d23002e68620.11.8&gt=1>

CULTURAL CONTROL

- Maintain good store hygiene by cleaning the store at each harvest and burning infested produce and crop residues (Islam, 2017).
- Dip cassava in boiling water to force the pest out and kill it.
- Burn wood taken from storage facilities as it tends to harbour the beetles.
- Select produce that is not infested with the pest for storage.
- Remove the adult insects and larvae from dried cassava by sieving to reduce populations, although this is very labour-intensive.

BIOCONTROL

- Red flour beetles are affected by several parasites, parasitoids and predators that can be employed in their control (Brower *et al.*, 1991). However, the use of biological control has not yet been developed to a commercial level and remains largely for research purposes.

CONTROL WITH PLANT PROTECTION PRODUCTS

- Several pesticides, such as, pyrethroids and fumigants, have been used to control red flour beetle populations (Guru-Pirasanna-Pandi *et al.*, 2018; Ebadollahi and Taghinezhad, 2020). For instance, malathion was used successfully against the red flour beetle in Pakistan. However, the development of insecticide resistance has slowed down their use in many countries (Cato *et al.*, 2017).
- Fumigate the store to eliminate residual infestations. However, the red beetle has developed some level of resistance to phosphine (Cato *et al.*, 2017; Nayak *et al.*, 2017).

OTHER CONTROL METHODS

- Freezing for several days and heating for 24 hours can control the pest in stored food products.
- Use of low oxygen and carbon dioxide-enriched atmospheres can also control the pest in stored dry cassava.

B. LESSER GRAIN BORER (*RHIZOPERTHA DOMINICA*)



Figure 116 — Showing (A) Lesser grain borer and (B) damage caused to dried produce
Source: Purdue University, <https://extension.entm.purdue.edu/publications/E-238/E-238.html>

DESCRIPTION/IDENTIFICATION

PEST

Adult lesser grain borers (*Rhizopertha dominica*) are 2–3mm long and reddish-brown in colour with a slim cylindrical shape (Figure 116A). The wing cases that cover membranous hind wings have rows of coarse punctures, which are finer at sides and are covered with curved hairs. The front edge of the plate-like cover in the front segment of the thorax has a saw-toothed appearance. The head is hidden when viewed from above, while the antennae have a clear club-shape with three segments (Mason, 2018).

SYMPTOMS

Damaged dry cassava is filled with flour and has irregular shaped holes due to the feeding of the lesser grain borer. The produce also develops a sweet and slightly pungent odour (Mason, 2018).

DISPERSAL AND SPREAD

Adult lesser grain borers can fly in some months of the year but are not good fliers and are mainly carried by wind. The pest can also be moved in affected cassava chips by humans (Mason, 2018).

DAMAGE

Both adults and larvae cause significant economic damage to dried produce (Figure 116B), which loses weight and quality (Popoola *et al.*, 2017).

IMPACT ON YIELD AND QUALITY

Lesser grain borers can cause more than 50% loss of stored dried cassava chips if not checked. The affected cassava chips are of low quality for marketing and human consumption (Jood and Kapoor, 1992; Jood *et al.*, 1992).

GEOGRAPHICAL DISTRIBUTION

The lesser grain borer affects cassava postharvest in many countries in Africa, the Caribbean and some countries in the Pacific (Table 43).

Table 43 — Geographical distribution of red flour betteles on cassava in the ACP countries

AFRICA	CARIBBEAN	PACIFIC
Algeria, Chad, Egypt, Ghana, Guinea, Mali, Morocco, Namibia, Nigeria, Rwanda, Senegal, Somalia, South Africa, Sudan, Tanzania, Zimbabwe	Antigua and Barbuda, Argentina, Brazil, Chile, Costa Rica, Dominican Republic, El Salvador, Guadeloupe, Mexico, Nicaragua, Panama, Peru, Puerto Rico, Trinidad and Tobago, Trivelli	Australia, Fiji, New Zealand

QUARANTINE ORGANISM

Rhizopertha dominica is a prohibited organism in Europe under section 22(1) and susceptible food products may only be imported by permission. Some permits are only applicable to species required for research purposes in secure institutions.

FAVOURABLE CONDITIONS

The moisture content of the stored produce can influence egg laying and development rates, with lower (8%) moisture content inhibiting egg laying. Larvae develop more rapidly on whole dry produce than on flour.

PEST LIFE CYCLE

Lesser grain borer females lay 200–500 eggs in their lifetime. The eggs are white when laid and turn brown before hatching (Potter, 1935). The eggs are ovoid in shape, 0.6 mm in length and 0.2 mm in diameter, and are laid either singly or in clusters of up to about 20. The egg stage lasts as many as 32 days at 18.1 °C (65°F), but just 5 days at 36 °C (97°F).

Larvae undergo four larval instars in 27–31 days at 28 °C (82.5°F) and 46 days at 25 °C (77°F). The first two instars are not recurved, while the third and fourth instars have recurved heads and thoraxes towards the abdomen. Pupation takes 5–6 days at 28 °C (82.5°F) and 8 days at 25 °C (77°F). Adults start to feed 3–5 days after emerging from the pupal case and tunnelling out of dry produce. Egg laying begins about 15 days later for up to 4 months. Adults can live up to 240 days (Potter, 1935; Mason, 2018).

SURVEILLANCE/MONITORING

Monitoring for the pest in stored food should begin with careful inspection of the store. Examine the dry cassava tubers, chips, bags or wood with distinct and characteristic tunnels or dust at the entrance of the tunnels. If in high numbers, the beetles make a rasping sound while feeding. Estimate the numbers and type by sampling the produce in the bags (if already packed) and the entire store to determine if and when to apply management strategies. If the population is low, you may hand pick the pest, however, high numbers require stronger measures to reduce/prevent damage to the stored food and spread to other stores.

CULTURAL CONTROL

- Maintain good sanitation in the granaries and throw away infested food.
- Package produce in airtight structures, such as well-sealed barrels that are pressure-tested to confirm airtightness (Garcia-Lara *et al.*, 2013).
- Remove spilt produce from storage facilities to reduce the lesser grain borer populations.
- Remove adult insects from dry cassava by sieving to reduce populations, although this is very labour-intensive.
- Add ash and clay to the cassava chips to reduce the population of insects by causing the insects to die from desiccation.

BIOCONTROL

- There are a few parasites and predators that attack the lesser grain borer but their effectiveness in the field is uncertain. The fungus *Beauveria bassiana* can be used as a biological insecticide (Nboyine *et al.*, 2015).

CONTROL WITH PLANT PROTECTION PRODUCTS

- Indirect spraying in the cracks and crevices in the stores, bins and storage containers with pesticides such as malathion, pyrethrins and diatomaceous earth reduces the numbers of the pest, but care must be taken to the hazardous of these products and end consumption of the produce..

OTHER CONTROL METHODS

- Reducing temperatures below 20°C reduces the rate of population increase for the pest. This is because *R. dominica* cannot complete its life cycle below 20°C (Reed and Harner, 1998; Beckett and Morton, 2003).
- Heating stored cassava/chips to above 34°C for 24 hours reduces the rate at which the population of *R. dominica* increases (Beckett and Morton, 2003).
- Powdered sage (*Salvia officinalis* L.) or wormwood (*Artemisia absinthium* L.) leaves effectively reduced the number and delayed the development of *R. dominica* (Klys, 2004).

7.8.2. PATHOGENS OF DRIED CASSAVA/CHIPS

SCIENTIFIC NAME

- *Botryodiplodia theobromae*
- *Aspergillus flavus*
- *Fusarium solani*
- *Rhizopus* sp.



Figure 117 — Showing (A) severe symptoms of *Botryodiplodia theobromae* on cassava tubers and (B) dark discoloration with reduced quality

Source: PlantVillage, <https://plantvillage.psu.edu/topics/cassava-manioc/infos>



Figure 118 — Showing *Aspergillus flavus*-contaminated dried cassava in a market

Source: Food & Beverage Online, <https://www.21food.com/products/dried-cassava-chips-451556.html>

DESCRIPTION

A. *BOTRYODIPLODIA THEOBROMAE*

PEST

Botryodiplodia theobromae is a fungus that affects cassava root and tuber development.

SYMPTOMS

Leaves on affected plants appear brown, wilted and scorched (Figure 117). The affected leaves tend to fall off the plant and in extreme cases the plant dies. Upon examination of the roots and tubers, there is dieback and swelling, or discolouration to light brown, dark grey, blue or pink. The roots rot and are soft with a foul odour. During the wet season, affected plant bases develop whitish structures as a result of *Botryodiplodia fungi* infection.

TRANSMISSION AND SPREAD

Spread of root rot disease due to *Botryodiplodia theobromae* in cassava can occur in waterlogged and poorly draining soils, and through movement of the infected tubers by man (Onyeka *et al.*, 2005).

DAMAGE

Disease-affected roots and tubers dieback and swell, or are discoloured to light brown, dark grey, blue or pink. The roots rot become soft and produce a bad smell that greatly affects the quality of fresh cassava and the flour is unpalatable.

IMPACT ON YIELD AND QUALITY

Post-harvest, the disease affects both the fresh and processed tuber quality (Onyeka *et al.*, 2005).

B. *ASPERGILLUS FLAVUS*

PEST

Aspergillus flavus is a mycotoxigenic fungus that produces *B. aflatoxins* and a bright yellow-green conidial colour, when cultured on malt extract agar or Czapek yeast extract agar. *A. flavus* is a very important food-borne fungus that attacks mainly stored food products, like cassava and grains (maize, rice, sorghum, millet, ground nuts).

SYMPTOMS

Infection by *A. flavus* appears as bright yellow and green on the stored food (Figure 118) (Jeyaramraja *et al.*, 2018).

TRANSMISSION AND SPREAD

A. flavus fungus easily increases its population and can enter the stored cassava tubers through injuries caused by insects or nematodes (Jeyaramraja *et al.*, 2018).

DAMAGE

A. flavus leads to growth of moulds, which affects the quality of cassava, leading to loss of market value.

IMPACT ON YIELD AND QUALITY

A. flavus affects the crop's growth and yield, resulting in loss of market value. High doses of aflatoxins in food can lead to death in humans (especially children), while small doses may lead to immunologic or nutritional effects, or to liver cancer (Marroquín-Cardona *et al.*, 2014).

C. *FUSARIUM SOLANI*

PEST

Fusarium solani is a fungus that is of great economic importance in the agricultural and food industry. It is a phytopathogenic fungus that causes several crop diseases, including root and stem rots in the field and post-harvest during storage.

SYMPTOMS

Internal tissues of affected tubers are light brown to black due to fungal mycelium killing the cells.

TRANSMISSION AND SPREAD

Fungal pathogens can be spread by water, moisture or unsterilised processing tools (Kaaya and Eboku, 2010), where the mould grows on fermented cassava tubers during storage.

DAMAGE

F. Solani affects the quality of the tuber, which results in loss of market value.

IMPACT ON YIELD AND QUALITY

Fusarium spp., can cause 100% yield loss of agricultural produce due to deterioration of the product and mycotoxin, which affect food quality and safety (Kokkonen *et al.*, 2010; Goswami and Kistler, 2005).

D. RHIZOPUS SP.

PEST

Rhizopus sp., are the causal agents of *rhizopus* rot disease, including postharvest diseases in various agricultural foods (Chacha and Mamiro, 2019; Ono and Taniwaki, 2021). The fungal pathogens are characterized by branching mycelia with three types of hyphae: stolons, rhizoids and unbranching sporangiophores (instead of conidia).

SYMPTOMS

Rhizopus-affected produce has slightly darker colouration, with decaying parts. Fungal mycelium may be white and fluffy with shades of grey to black.

TRANSMISSION AND SPREAD

Rhizopus sp., fungus is prevalent in dried/stored cassava but also can infect the tubers in the field usually through injuries caused by insects or nematodes.

DAMAGE

Discolouration and rotting of cassava, including production of mycotoxins that make the food unsafe for human consumption, processing and marketing.

IMPACT ON YIELD AND QUALITY

Rhizopus sp. affects cassava's yield, and the fresh and dry tuber quality (Chacha and Mamiro, 2019; Ono and Taniwaki, 2021).

GEOGRAPHICAL DISTRIBUTION

Post-harvest tuber rot fungal pathogens such as *Botryodiplodia theobromae*, *A. Flavus*, *F. Solani* and *Rhizopus* sp. in cassava are prevalent in many countries where cassava is dried, stored and or processed, in Africa, the Caribbean and the Pacific. The level of contamination depends on the post-harvest management practices and food quality standards.

QUARANTINE ORGANISM

Cassava plant materials and products with fungal pathogens are restricted from entry into the EU, due to their ability to develop mycotoxins such as trichothecenes (T-2 and HT-2 toxins, diacetoxyscirpenol, deoxynivalenol and nivalenol), zearalenone and fumonisins (Marasas, 1994; FAO, 2001; Klich, 2003) that are harmful to humans and livestock if consumed (Christensen and Meronuck, 1986; Bennett *et al.*, 2007). To manage aflatoxin contamination during trade, measures have been instituted to regulate the level of the toxins in food by the EU (Gurtler and Keller, 2019).

FAVOURABLE CONDITIONS

Post-harvest root rot in cassava due to fungal pathogens is prevalent in many countries in Africa, especially under poor humid conditions both in the field and post-harvest during storage (Msikita *et al.*, 2005; Akinyele and Ikotun, 1989; Onyeka *et al.*, 2005). The growth of *Botryodiplodia theobromae*, *Fusarium solani* and *Rhizopus* sp. is favoured by waterlogged, poorly draining soils (Onyeka *et al.*, 2005). Post-harvest, tuber rot **fungi** develop when the moisture content of the cassava chips exceeds 14% at relative humidity of 70-90% (Bankole and Adebajo, 2003; Chacha and Mamiro, 2019). On the other hand, *Aspergillus flavus* thrives in media with a pH range of 1.7 to 11.2, although the optimum is a pH of 3–7 (Yoshinari *et al.*, 2010). But the growth of the fungus slows down at low pH (<3) and at higher pH (>7). Similarly, fungal growth reduces at temperatures as low as 10–12 °C or as high as 50–55 °C (Eshelli *et al.*, 2015).

PEST LIFE CYCLE

Various fungal pathogens (Bankole, 1994) overwinter in the field (soil, plants and debris) and stored produce (Mohana and Raveesha, 2007) and come alive when favourable conditions abound. The fungal pathogens enter the produce through wounds and where the spores establish and develop into moulds that affect the quality of produce (Essonon *et al.*, 2007), leading to loss of food and market value. The cycle repeats itself when the affected produce is not removed and destroyed or if the conditions recur.

SURVEILLANCE/MONITORING

Monitoring of post-harvest fungal infections should be carried out using standard protocols such as developed by Kaaya and Eboku (2010), where 1 kg of dried or processed cassava products were sampled per household for analysis in the laboratory.

CULTURAL CONTROL

- Plant cassava in well-draining soils to avoid infection.
- Remove and destroy the crop debris by burning.
- Harvesting cassava early prevents or reduces root rot development.
- Sanitise the tools after use.
- Exposing harvested or stored food crops to sunlight helps in detoxification of aflatoxin (Rushing and Selim, 2019).

BIOLOGICAL CONTROL

- Bacteria, yeasts, moulds and algae have shown potential to degrade aflatoxin during absorption and enzymatic decadence (Jard *et al.* 2011).
- A significant decrease in *A. flavus* growth was achieved in pre-harvest crops by inoculating with strains of the bacteria *Pseudomonas*, *Trichoderma*, *Ralstonia*, *Lactobacilli*, *Burkholderia* and *Bacillus* spp. (Akocak *et al.*, 2015; Yang *et al.*, 2017).
- Aflatoxin contamination was inhibited by use of non-toxigenic strains of *A. flavus* and other moulds (Udomkun *et al.*, 2017).

CONTROL WITH PLANT PROTECTION PRODUCTS

- Chemicals including acids (sodium bisulfite), alkalis (sodium hypochlorite, calcium hydroxide), oxidizing agents (halogens, potassium nitrate, and nitric acid) and aldehydes (e.g. glucose) were used to control aflatoxigenic fungal growth and aflatoxin production in stored produce (Carvajal and Castillo, 2009; Udomkun *et al.*, 2017).
- Aflatoxin growth can be impeded by adding liquid ammonia, urea or ammonium bicarbonate (Karlovsky *et al.*, 2016).
- Food preservatives such as propionic acid, crystal violet, p-amino benzoic acid, benzoic acid, boric acid and sodium acetate successfully prevented the growth of *A. flavus* and aflatoxin production (Aiko and Mehta, 2015).
- Azole fungicides were used to control fungal growth and aflatoxin production, such as tebuconazole (Mateo *et al.*, 2017).

OTHER CONTROL METHODS

- Grow resistant cassava varieties against root rot pathogens such as *Botryodiplodia theobromae* (Onyeka *et al.*, 2005).
- Steaming under pressure, dry roasting and other cooking methods were found to be effective in controlling or reducing aflatoxin contamination in many harvested food crops (Peng *et al.*, 2018).







8

HARVESTING

Cassava can attain full genetic yield potential of 80 t/ha if grown using improved varieties and good agricultural practices (International Institute of Tropical Agriculture, 1990). However, large yield gaps remain in most countries, especially in sub-Saharan Africa (Rosenthal, 2020). The yield gaps vary depending on the biotic and abiotic constraints, and crop management practices (Fermont *et al.*, 2009). According to Edoh *et al.* (2016) cassava yields varied from 20-30 t/ha and 25-70 t/ha for local and improved cassava varieties, respectively. However, much lower yields (5-15 t/ha) are mostly obtained under poor production practices in East Africa.

Harvesting cassava is the ultimate production activity any farmer looks for, as this is the period the farmers gets to benefit from their investment. In addition to the final tubers harvest, the leaves can be harvested throughout the season, and are consumed as vegetables (Latif and Müller, 2015). Like all commercial crops, it is very important to know when and how to harvest, and how to maintain the standards required by the market.

8.1. COMMERCIAL QUALITY

Mature market-ready cassava varies in standards, depending on customer specifications, end use and markets.

8.2. WHEN TO HARVEST CASSAVA TUBERS

Cassava can be harvested as soon as the roots attain an average weight of 2-4kg. In practice, cassava is mature and ready to harvest beginning 6–9 or 12–18 months after planting, depending on whether the variety is early or late maturing (Ekanayake *et al.*, 1997), respectively. Cassava may continue developing after this period, but roots will harden and become woody.

In high altitude areas where the weather is cold, cassava takes longer to reach maturity, with an average of 24 months before harvesting. Physical signs that cassava is mature enough to be harvested include: yellowing and dropping of the lower leaves and cracks developing around the plant, which could be an indication for increased size of the tubers.

A field of cassava may therefore be harvested over a period of several months, and staggering harvest offers a major advantage to adjust supply to the needs.



Figure 119 — Piecemeal harvesting of cassava

Source: <https://i0.wp.com/sustainable-secure-food-blog.com/wp-content/uploads/Untitled-design-2021-08-30T153759.998.png?fit=1200%2C675&ssl=1>

Harvesting cassava may also depend on the use: whether it is for home consumption, selling fresh tubers or processing into flour. Leaving cassava tubers longer in the ground (especially for short maturing varieties) may lead them to become woody and lose the characteristic cassava flavour (Lancaster and Coursey, 1984).

A plot can be harvested all at once if the crop is grown for commercial purposes and or processing. However, if for home consumption, harvesting may be carried out piecemeal (Figure 119) i.e., whenever the farmer feels like eating cassava, and this could be over the whole season until a new crop is mature to harvest.

If harvested too early, the yield may be low and of poor eating quality, while harvesting after 12 months and >24 months for early and late maturing varieties, respectively may lead to development of woody and inedible mid portions in the tubers (Amponsah *et al.*, 2018). Moreover, leaving cassava crops for too long in the field limits further use of the land and exposes the roots to soil borne pests and diseases.

8.3. HOW TO HARVEST CASSAVA TUBERS

8.3.1. METHODS OF CASSAVA HARVESTING

Cassava can be harvested using three methods, including: manual, semi-manual or mechanised harvesters (Amponsah *et al.*, 2017; 2018).

A. MANUAL HARVESTING

Manual harvesting is the traditional method practiced by most smallholder and semi-commercial farmers in the African, Caribbean and Pacific (ACP) countries. It can be carried out in several ways:

- First, one can use bare hands with or without tools such as a hoe, cutlass/panga, mattock or earth chisel. The plant is held by the stems at the base and uprooted out of the ground (Figure 120A & B), after which cassava tubers are carefully detached.
- Secondly, using tools such as a cutlass/panga (Figure 120C) or a hoe (Figure 120D), the soil is first dug away around the cassava plant, before it is uprooted out of the ground. This might involve first cutting off the upper parts of the stems with the leaves before harvesting the tubers.
- The other options involve using a mattock (Figure 120E) or an earth digging chisel (Figure 120F) to uproot the whole base with the tubers out of the soil. If the soil is hard, it is advisable to first dig away some soil to expose the tubers before pulling.

Challenges of manual harvesting: Manual harvesting can be challenging depending on the type of soil and weather conditions.

- For instance, muddy soil can stick on the tools or cassava tubers, making it difficult to clean and requiring much labour if more acreage is to be harvested (Kouakou *et al.*, 2016).
- It is estimated that manual harvesting needs at least 22-62-man days per hectare (Nweke *et al.*, 2002). Pulling cassava with bare hands alone needs 23-47-man h/ha, while using a hoe needs 42-51-man h/ha (Amponsah *et al.*, 2014). It is therefore advisable to harvest cassava when the soil is softer as during or after a rain shower when it is relatively dry. Also uprooting cassava is much easier in soils with relatively higher moisture content. Cutting off the upper part of the stem gives better grip on the plant to be harvested.
- In addition, manual harvesting leads to high loss of tubers due to damage by tools or breakage during pulling (Amponsah *et al.*, 2014). To reduce root damage, it is advisable to first carefully dig away the soil around the plant to expose the breadth and length of the tubers before pulling the whole root base out by hand or mattock/chisel and detaching the tubers.
- Manual harvesting increases the cost of production, especially where the crop is grown on large scale for fresh marketing or industrial processing.

To overcome the challenges associated with manual cassava harvesting, various attempts were made develop more efficient and appropriate methods, including semi-mechanical and mechanised techniques. However, earlier efforts aimed at mechanising cassava harvesting were challenged by lack of standardised agronomic cassava planting practices such as spacing, placement of cuttings during planting, variable terrain and scale of cassava cultivation.



Figure 120 — Showing various options for manual harvesting of cassava: (A & B) pulling with bare hands; (C) removing away soil around the plant using a panga/cutlass; (D) using a hoe before pulling; (E) cutting off the stems and using a mattock or (F) using a chisel to pull out cassava tubers

Source: <https://www.intechopen.com/media/chapter/57918/media/F1.png>;
<https://www.intechopen.com/chapters/57918>; <https://encrypted-tbn0.gstatic.com/images?q=tbn:ANd9GcS06n84HbHSjA5V8 TUwUGJDaucdaj65uAw&usqp=CAU>

B. SEMI-MANUAL HARVESTERS

CRI-harvester: The semi-manual harvester (Figure 121A & B) developed by the Crops Research Institute (CRI), Kumasi, Ghana (Amponsah *et al.*, 2018) works on the principle of a lever; “grip and lift”, which aids to reduce the effort by man while uprooting cassava tubers. The tool (Figure 121A) comprises of a metallic frame with a steel plate which is joined to an immobile ‘gripping jaw’. At the bottom is a ‘chisel tip’ which is used to dig into the soil to lift cassava tubers with the aid of the gripping jaw (Figure 121B).

The long handle helps to increase the force on the load, i.e., cassava tubers and soil around the plant during harvesting. According to Amponsah *et al.* (2017), the tool can easily be operated, and works best for cassava planted vertically. In addition, the CRI-harvester recorded the least tuber damage and breakages for cassava planted using the slanted placement method.

NCAM harvester: The semi-manual harvester (Figure 121C) developed by the National Centre for Agricultural Mechanisation (NCAM) in Nigeria, has a metallic frame that is attached to a ‘footboard’ and immobile ‘gripping jaws’. It is operated by a long handle that serves as a lever hinged to the frame that is designed for uprooting cassava tubers. To harvest, the gripping jaw is placed at the bottom of the cassava stem, and with one foot on the footboard the plant is lifted using the lever/handle (Figure 121D). Accordingly, both the CRI and NCAM harvesters were tested and proven to harvest up to 200 plants per man-hour (Amponsah *et al.*, 2018).

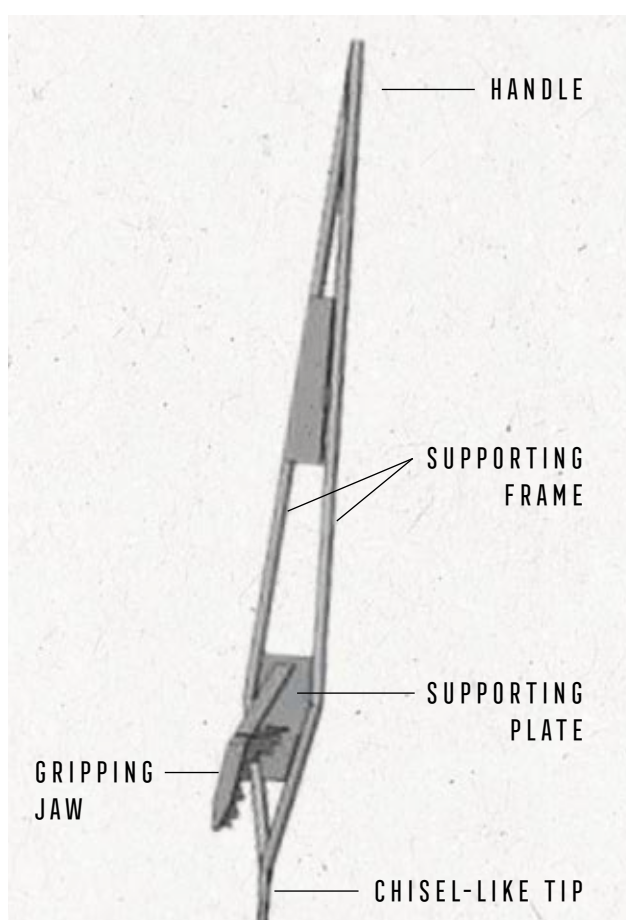




Figure 121 — Showing semi-manual cassava harvesters: (A) CRI-harvester by CSIR-Crops Research Institute (CRI), Ghana; (B) the CRI-harvester in use by a farmer; (C) a NCAM harvester by the National Centre for Agricultural Mechanisation (NCAM), Nigeria and (D) how it is operated during harvesting cassava in the field
Source: <https://www.researchgate.net/profile/Shadrack-Amponsah/publication/313667385/figure/fig2/AS:461433570893824@1487025744008/The-improved-manual-cassava-harvesting-tool.png>;
<https://www.intechopen.com/media/chapter/57918/media/F2.png>;
https://dierosoil.tripod.com/postharvest/Equipment/images1/harvester_01.jpg;
https://www.researchgate.net/publication/322567302/figure/fig4/AS:583974904664065@1516241875709/The-NCAM-harvester_W640.jpg

C. MECHANISED HARVESTERS

There are two types of mechanised harvesters, including: **semi-mechanised** and **fully mechanised** (Amponsah *et al.*, 2018).

Semi-mechanised harvesters: The NCAM-harvester (Figure 122A) and CLAYUCA-harvester (Figure 122B) are two models of semi-mechanised harvesters developed in Nigeria and Brazil, respectively. For both, only the cassava digging process is mechanised as seen in figure 122C, where the tractor mounted semi-harvester is digging out the cassava tubers, which are later collected and detached manually (Amponsah *et al.*, 2018).



Figure 122 — Showing tractor-operated semi-mechanised harvesters:
 (A) NCAM-harvester made in Nigeria; (B) CLAYUCA-harvester model P600 made in Brazil,
 and (C) a semi-mechanised cassava harvester mounted on a tractor

Source: <https://www.intechopen.com/media/chapter/57918/media/F7.png>;

<https://www.intechopen.com/media/chapter/57918/media/F6.png>;

<https://tractorexport.com/wp-content/uploads/2017/07/Cassava-harvester-on-6610.jpg>

Another semi-mechanised cassava harvester model 'TEK cassava harvester' (Figure 123A) was developed in Ghana. It has the following parts: digger, shakers with a slatted conical mouldboard, linkage points and a vertical support, improved to implement what is called the 'dig and pull' principle (Bobobee *et al.*, 1994).

The TEK harvester is mounted onto a tractor (Figure 123B) and digs the soil to bring the tubers to the surface by the aid of the inclined slatted conical mouldboard. Thereafter, the tubers are manually collected and detached (Figure 123C) from the

stalks (Amponsah *et al.*, 2018). However, sometimes a lot of soil is dug up, which may increase resistance and lead to high fuel consumption. The slatted conical mouldboard also serves to shake and sieve off the soil clods, which helps to clean the tubers, and increase the pace at which other harvesting processes (collecting, detachment and cleaning) are carried out.



Figure 123 — Showing (A) a fully mechanised TEK cassava harvester mounted on a tractor; (B) the TEK harvester in use in the field and (C) farmers manually collecting cassava tubers dug up by the TEK mechanical harvester

Source: https://www.tekcassavaharvester.com/wp-content/uploads/2018/05/DSC_0808-1024x681.jpg;
https://www.tekcassavaharvester.com/wp-content/uploads/2018/05/Binder1_008-1024x682.png;
<https://www.tekcassavaharvester.com/wp-content/uploads/2018/05/pib71.png>

Fully mechanised harvesters: There are various models of fully mechanised cassava harvesters that are equipped with implements for multiple harvesting processes such as digging cassava tubers onto the soil surface, detaching the tubers and transporting the harvested tubers from the field.

To enable the smooth and efficient operation of the mechanised cassava harvesters, the following must be done prior to harvesting cassava:

- i. Remove any obstacles that may damage the implement such as huge stones or rocks, tree stumps etc;
- ii. Remove weeds that may block the lifters;
- iii. Cut down cassava plant stalks to at least 30 cm to enable the harvesters to run properly;

According to Amponsah *et al.* (2018), mechanised harvesters operate best on cassava planted on ridges (Figure 124).



Figure 124 — Showing cassava planted on ridges,
which is the most suitable planting method for mechanised cassava harvesting
Source: <https://www.tekcassavaharvester.com/wp-content/uploads/2018/05/pib5.jpg>

The capacity of manual, semi-manual and fully mechanised harvesters was estimated as ‘man hours per hectare’ (man h/ha) at approximately: 22-51 man h/ha, 16-45 man h/ha and 1-4 man h/ha, respectively (Amponsah *et al.*, 2018). However, mechanised harvesters have the additional advantage of leaving the field ploughed after harvesting, which helps to save on fuel, time and cost for the next season.

A comparison of traditional and modern cassava production methods by Lansche *et al.* (2020) indicated that modern methods produced 3X more yield than traditional methods for similar plant populations and land area (Table 44).

Table 44 — A comparison of traditional and modern cassava production methods
(Example from Malaysia)

PARAMETER	TRADITIONAL	MODERN
Plowing (number of crossings)	manual (1)	mechanized (1)
Harrowing (number of crossings)	-	mechanized (2)
Mulching (number of crossings)	manual (1)	mechanized (3)
Planting, pcs/ha	26,000	26,000
Harvesting	manual	mechanized
Loading of crops	manual	mechanized
Fertilizing	manual	mechanized
Cattle manure, kg/ha	-	15'000
Manure application	-	mechanized
NPK (15/15/15), kg/ha	350	350
Cutting stems and leaves	manual	mechanized
Tuber yield, t/ha	5.0	17.8
Leaves yield, t/ha	0.6	2.04

Source: Lansche *et al.* (2020)

8.3.2. HYGIENE AND HANDLING

The EU market is very sensitive to food safety and therefore demands strict observance of standard guidelines for all operations along the cassava value chain. The standards for maintaining hygiene must be observed by all stakeholders, including: cassava farmers, field workers, tractor operators, transporters, packhouse employees and traders/exporters.

In addition, the containers used to carry fresh cassava tubers must also be cleaned of dirt, oils and any other contaminants. The workers should dress in appropriate field wear such as gum boots, overalls, and can use hand groves and head/hair covers such as headscarfs or hats. Dirty clothes and other gear should be cleaned often to prevent them accumulating bad odour and germs that might contaminate both the food and workers.



HARVESTING

KEY SUMMARY POINTS

Cassava harvesting should be carefully carried out at the right maturity time to maximise the yield and quality, while at the same time using the right tools and methods.

COMMERCIAL QUALITY

- Mature market-ready cassava varies in standards, depending on customer specifications, end use and markets.

WHEN TO HARVEST

- Cassava is mature and ready to harvest beginning 6–9 or 12–18 months after planting, for early - and late maturing varieties respectively.
- In high altitude areas with cold weather, cassava takes an average of 24 months to mature.
- Cassava crops may be harvested in phases (i.e. staggered harvesting).

HOW TO HARVEST CASSAVA ROOTS

- Manually, by use of bare hands and pulling or with the aid of tools such as a panga/cutlass, mattock or earth chisel.
- Using semi-manual cassava harvesters.
- Using semi-mechanised cassava harvesters mounted on tractors that only dig out cassava.
- By use of tractor operated mechanised harvesters for all processes.
 - To enable efficient operation of mechanised cassava harvesters: remove obstacles such as huge stones or rocks, tree stumps, weeds and cut down cassava plant stalks to 30 cm.

HYGIENE AND HANDLING

- Strictly observe market food safety standards and handling guidelines.







g

POSTHARVEST

Cassava is a highly perishable crop and begins to deteriorate as early as 1–3 days after harvest (Amponsah *et al.*, 2018). This is due to the fact that cassava tubers lack dormancy and do not play a role in propagation as they possess no bud primordia from which regrowth can occur (Onwueme, 1978, Cooke *et al.*, 1988, Wickam, 1988). The high perishability of cassava is a major challenge that limits storage, processing and marketing.

9.1. POSTHARVEST DETERIORATION

When the tubers are uprooted from the ground, the process of deterioration begins within 24 hours. The tubers soon develop a blue-black discolouration and brown streaking of the xylem tissue (Iyer *et al.*, 2010). This shortens the shelf life of the fresh tubers, resulting in wastage, poor quality products that are less palatable and attractive to the market and so causing high economic losses (Van Oirschot *et al.* 2000). Deterioration is worsened by presence of cuts or breakages, which serve as entry points for microorganisms that cause further deterioration (Reilly *et al.*, 2004). Buschmann *et al.* (2000) classified postharvest deterioration into three categories: mechanical damage, physiological and microbial deterioration.

- a. **Mechanical damage:** Mechanical damage is due to careless handling of cassava tubers during harvesting, transportation and processing (Iyer *et al.*, 2010). The damage created during harvesting can be due to rough pulling of the tubers from the ground or cuttings by farm tools and machinery. During transportation, mechanical damage may result from excess vibration or compression on rough roads to a fresh market or processing facility.
- b. **Physiological or primary deterioration** occurs as a result of mechanical injuries on the tubers (Fadeyibi, 2012). Mechanical injuries cause wounds which initiate a response involving biochemical and oxidative changes within 24 hours after harvesting (Reilly *et al.*, 2004). Deterioration sets in and is characterised by blue or brown discolouration that tends to increase at temperatures of 20–30 °C and low relative humidity of 65–80 % (Sánchez *et al.*, 2006).
- c. **Microbial or secondary deterioration:** is attributed to infection by fungi and/or bacteria and occurs in the flesh 5–7 days after harvest, resulting into wet and dry rotting. Physical damage arising from wounds, cuts and bruises during harvesting, handling operations and transportation are the entry points for microorganisms such as fungi and bacteria, which cause microbial deterioration or secondary deterioration (Falade and Akingbala, 2010a). Among the important microbial deterioration fungi are *Aspergillus niger*, *Aspergillus flavus*, *Aspergillus fumigatus*, *Penicillium citrinum* and *Rhizopus* spp., (Onyimonyi, 2002). The fungi cause dry rot under aerobic conditions, which results in discoloration, increased acidity, off-flavour and taste of the stored cassava tubers and chips (Gnonlonfin *et al.*, 2008). The bacteria include *Bacillus* spp.; which cause rapid development of acid in the cassava tubers and chips (Onyimonyi, 2002). Therefore, following primary deterioration through wounds and cuts, microbial deterioration of cassava tubers results in fermentation and softening of the root tissue after 4–5 days (Buschmann *et al.*, 2000; Reilly *et al.*, 2004).

Therefore, to manage postharvest deterioration, it is important to understand the cause and stage along the value chain (field handling, transportation, storage and processing) at which to impose intervention measures.

9.2. FIELD HANDLING

Harvested cassava should be handled carefully to avoid causing physical damage that can reduce both the shelf life and market quality of the tubers. Practice the following as a routine during field handling of harvested cassava.

- As soon as the tubers are harvested, they should be kept under a shade to avoid direct exposure to sunlight and to reduce field heat, which might cause blisters until transportation to the market or processing facility.
- If the tubers are to be sold in a fresh market (near or far), while detaching them off the stalk, leave a portion of 2-5cm to prevent rapid spread of decay into the root since rotting often begins from the neck.
- For long storage, select and retain only the tubers that lack any sign of damage. However, if the tubers get some cuts/wounds during field handling, keep them at 30–40 °C and 90–100 % relative humidity (RH) for 4 days (Fadeyibi, 2012) to delay the onset of physiological and microbial deterioration to prolong the shelf life and quality of the fresh tubers (Akingbala *et al.*, 2005).
- Avoid throwing or pushing the tubers into containers, which may also cause physical breakages and wounds that may result in quality deterioration through early decay or darkening of the cut surfaces (Plumbley and Rickard, 1991).
- Wash cassava tubers to remove the soil, especially if harvested on a rainy day. When washing, be gentle to avoid damaging the peels. The tubers could also be placed in a chlorinated water with 150 parts per million (ppm) of chlorine and water for 60 to 90 seconds after washing. Rinse the tubers in clean water and place them on trays.

9.3. TRANSPORT

For small volumes, most farmers carry the harvested cassava tubers using basins or gunny bags, either on their heads, back, wheel burrow or bicycle to storage yards, or for direct packaging for sale to fresh markets or processing facilities. Where large volumes are transported as for commercial farmers, the use of automated transport such as motor cycles (Figure 125A); pickups or lorries (Figure 125B) and tractors with carriage trailers is common. In either case, care must be taken to avoid bruising the tubers, or throwing them which will reduce their market quality. It is uncommon to use refrigerated trucks in transportation of cassava in most developing countries. In all cases take care to cushion the cassava tubers using materials such as dry grass or harvesting plastic crates to limit physical damage especially on a rough road.



Figure 125 — Showing transportation of cassava tubers using (A) motor cycles and (B) trucks

Source: https://media.nature.com/lw800/magazine-assets/d41586-019-00014-2/d41586-019-00014-2_16380922.jpg;
https://live.staticflickr.com/5552/14490361639_bb3913430f_b.jpg

9.4. STORAGE OF TUBERS

Proper storage of fresh cassava tubers guarantees the availability of quality and safe food even out of season. Generally, there are three methods of storing cassava, including: traditional, improved low-cost and modern high-cost storage methods.

A. TRADITIONAL STORAGE METHODS

Among the traditional methods are:

- Leaving cassava tubers in the ground until they are needed;
- Re-burying the tubers in trenches covered with plant material and soil (Figure 126);
- Piling the tubers in heaps and pouring water on them daily to keep them moist (Wickham, 1985);
- Smearing a thick coating of soft clay or mud on the tubers;
- Storing small quantities of cassava tubers in water can keep them in good condition for at least 7 days (Bancroft and Crentsil, 1995).
- Prunning cassava (in the field), which involves removing all the leaves, extended the freshness of cassava tubers in the field for 14 days (Nuwamanya *et al.*, 2019),

The above practices simply reduce moisture loss from the cassava tubers and can help prolong the storage life of the tubers by only few days. Unfortunately, keeping cassava tubers longer in the field is often challenged by the need for the land to grow new crops. Furthermore, the tubers develop fibres, decrease in starch content and instead increase in sugars and acidity, which affects the cooking and processing quality.

In addition, keeping the tubers longer in the soil also exposes them to infection by soil borne pathogens such as *Fusarium solani*, *Phaeolus manihotis* and/or damage by soil pests such as grubs, and animals such as rodents and wild pigs (Westby, 2002). However, cyanide levels were much reduced in the cassava tubers stored using the traditional practices (Akingbala *et al.*, 2005).



Figure 126 — Showing re-burying of cassava tubers in soil during short-term storage
Source: <https://www.genebanks.org/wp-content/uploads/2016/12/Hero-image-cassava.jpg>

B. IMPROVED LOW-COST STORAGE METHODS FOR FRESH CASSAVA

Improved low-cost storage methods are aimed at prolonging the shelf life of fresh cassava roots by at least 14 days (Crentsil *et al.*, 1995). This period may facilitate that the produce reaches the market in good quality. Improved low-cost storage practices include: storage in pits, field clamps and boxes with moist sawdust.

i. Field clamp

A field clamp is made of a layer of straw placed on a dry floor that is covered by a heap of 300-500 kg of roots/tubers, followed by a layer of straw and a final layer of soil (Figure 127). Ventilation is catered for at the bottom of the heap through holes. The method stores tubers in good condition for up to 8 weeks. However, it is very difficult to manage ventilation during seasonal variations of weather. Also, during the wet season, entry of rain water makes it difficult to keep the floor dry (Fadeyibi, 2012) and it is also very labour intensive (Westby, 2002).

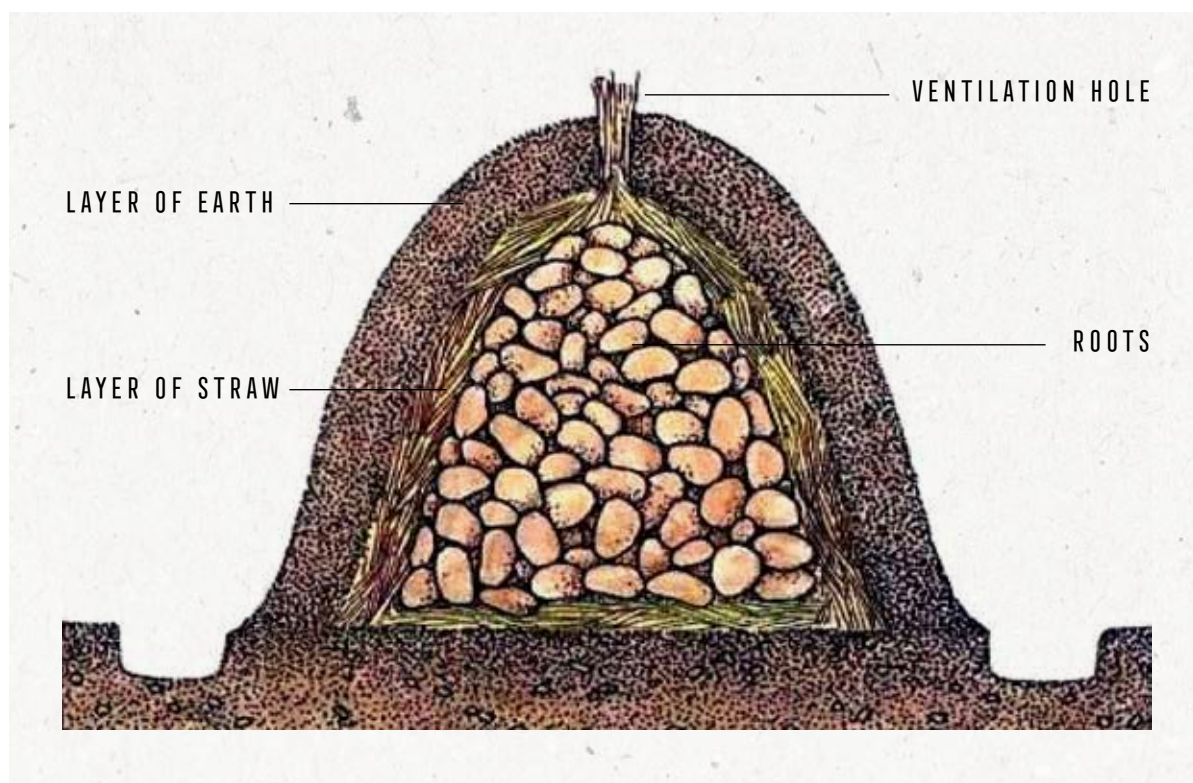


Figure 127 — Showing a cross-section of a field clamp structure for storage of fresh tubers

Source: <https://d3i71xaburhd42.cloudfront.net/cfd183c061e4576a494246a81eb01d36d5728569/3-Figure3-1.png>

ii. Storage in boxes lined with moist sawdust or wood shavings

Alternate layers of moist sawdust/or wood shavings and cassava roots are placed in a box, beginning and ending with a layer of sawdust (Figure 128A) (Fadeyibi, 2012). Cassava tubers stored well in the sawdust with relative humidity of 70-80% and temperature of 28-33 °C, with 50-75% of the tubers remained in acceptable quality after 4- 6 weeks (Olaleye *et al.*, 2013). The method involves adding water regularly

to keep the sawdust/wood shaving wet/cool (Figure 128B). However, the method is challenged by physiological deterioration and microbial decay, which occur if the saw dust is too dry or too wet, respectively (Babarinsa and Oluwalana, 2018). Also, exporters who used the method complained about the high cost of transport.



Figure 128 — Showing storage of tubers in boxes line with moist sawdust

Source: https://www.researchgate.net/profile/Olumuyiwa-Babarinsa/publication/324015173/figure/fig1/AS:619876200751106@1524801411999/Thermo-hygrometers-indicating-the-RH-and-Temperature_Q320.jpg;
https://www.researchgate.net/profile/Olumuyiwa-Babarinsa/publication/324015173/figure/fig3/AS:619876204949504@1524801412131/figure-fig3_Q320.jpg

C. IMPROVED HIGH-COST METHODS

i. Thatched roof pit

This method involves building a structure that has a thatched roof (Figure 129A) and digging a pit in the ground (Figure 129B) inside the structure. The roof helps to keep away rain water and the direct effect of sunlight. The sides are left open for ventilation and the method kept 65% of the cassava tubers in fresh condition for 120 days, while 35% of the tubers were lost due to rotting (Fiagan 1995).

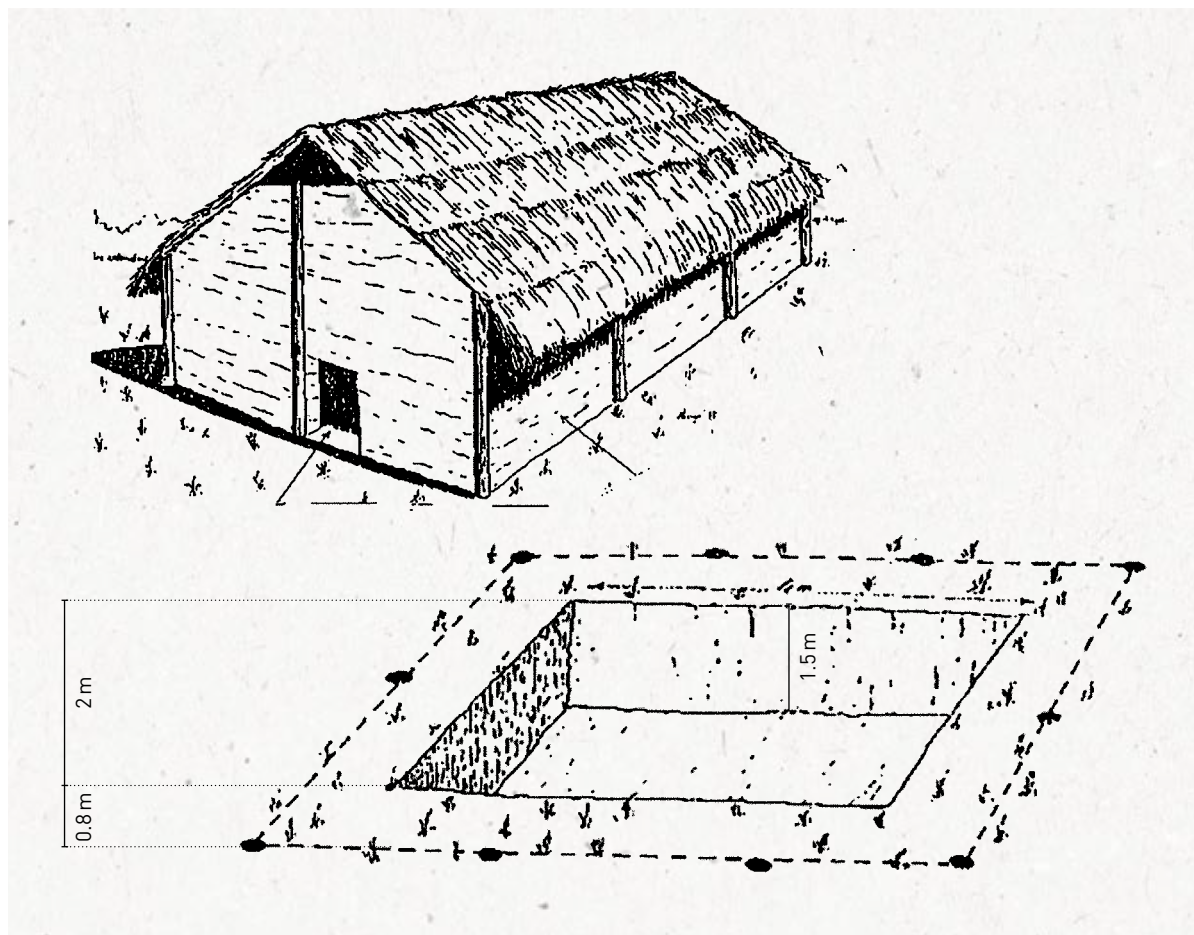


Figure 129 — Showing (A) a field clamp structure for storage of fresh cassava tubers and the (B) pit where the tubers are kept

Source: <https://www.fao.org/3/x5415e/x5415e08.gif>

ii. Storage in plastic bags or plastic film wraps

This method involves dipping cassava tubers in a 0.4% w/w solution of thiabendazole (fungicide) for 10 seconds and keeping them in plastic bags (Crentsil *et al.* 1995). Airtight plastic bags or plastic film wraps (Figure 130) kept the fresh tubers in good condition for 2-3 weeks. The method allowed the transportation of the tubers on rough roads without any loss of quality. Most of the thiabendazole remains in the thick peel of the tuber. For example, only about 1 mg/kg of residue was found in the

parenchymal tissue of cassava tested by the method, which was below the 5 mg/kg acceptable limit for potato (Cooke *et al.*, 1988a). The method is by far the best for commercial storage and transportation of fresh tubers (Fadeyibi, 2012). However, after 3 weeks that cassava still begins to deteriorate (Akingbala *et al.*, 2005).



Figure 130 — Showing storage of fresh cassava tubers in plastic bags or plastic film wraps
Source: <https://vinleaf.com/wp-content/uploads/2019/03/raw-cassava-20190326001.jpg>

iii. Frozen cassava storage

Refrigeration at low temperature of 0–5 °C and freeze drying can keep cassava tubers fresh (Figure 131A & B) for up to 2 weeks (Reilly *et al.*, 2004; Akingbala *et al.*, 2005; Fadeyibi, 2012). The method can be used at home, in businesses such as super markets or during long transportation for export. But the method is expensive since it requires constant supply of electricity (Kormawa *et al.*, 2002). Furthermore, the equipment can be costly and requires skilled technicians to maintain (Fadeyibi, 2012). The other disadvantage of the technology is that freeze drying can cause chilling injury, internal breakdown of cassava flesh, water/weight loss of 14 and 23 %, decay or browning disorder (Rickard and Coursey, 1981; Paull, 1999) and loss of eating quality (Brecht, 2003). To reduce on water/weight loss, the fresh tubers should be kept in polythene bags/wraps. However, the rate of deterioration is faster once the tubers are removed from the fridge (Lebot, 2009).



Figure 131 — Showing (A) frozen peeled fresh cassava and (B) how it is packaged in a plastic bag or film wrap
 Source: <https://mednutrition.files.wordpress.com/2012/06/dsc02309.jpg?w=1024;>
<https://mednutrition.wordpress.com/tag/cassava/>

iv. Waxing cassava with paraffin wax

Waxing cassava with paraffin wax (Figure 132) helps to extend its shelflife and food quality and has huge potential for adoption and commercialisation in the ACP countries. The method can keep cassava tubers fresh for 20 days (Atieno *et al.*, 2018), and if coupled with pruning, the tubers can keep for a further 8 days, i.e., making a total of 28 days (Nuwamanya *et al.*, 2019).



Figure 132 — Showing waxed fresh cassava in an export box
 Source: <https://mednutrition.files.wordpress.com/2012/06/57886580a62122328c69a137883d3ced.jpg>

In the flow chart (Figure 133) are the essential packhouse operations carried out during waxing cassava with paraffin. The unit operations are normally used to determine the types and quantities of tools and equipment necessary for constructing and equipping a packhouse.



Figure 133 — Showing the essential packhouse operations carried out during waxing cassava with paraffin wax
Source: Adopted from Matovu *et al.* (2016)

In addition, the site and facility should have the following:

1. Minimal risk of air pollution or contamination;
2. Protection from the sun and rain;
3. Reliable electricity supply;
4. Comfort and safety utilities for employees;
5. Clock-in areas and toilet facility.

9.5. PROCESSING CASSAVA

Processing cassava into different food forms such as chips, tapioca, flour or starch (Figure 134A-D) extends shelflife and maintains the quality better than the fresh root storage methods (Falade and Omojola, 2010b). In addition, processing helps to remove cyanide, a toxic compound that limits the consumption of bitter cassava varieties (Simonyan, 2014).



Figure 134 — Showing cassava processed in various products:
(A & B) chips; (C) tapioca and (D) flour or starch

Source: <https://nationaltoday.com/wp-content/uploads/2020/06/Tapioca-640x514.jpg>
https://img3.exportersindia.com/product_images/bc-full/dir_119/3557307/dry-cassava-chips-1475084771-2411928.jpeg
<http://fooddryingoven.com/wp-content/uploads/2019/05/dried-cassava-chips.jpg>
<https://www.curiouscuisiniere.com/wp-content/uploads/2019/02/Cassava-starch-Tapioca-starch.jpg>

Both traditional and modern technologies are used to process cassava into the various products above, and more. A brief description of some of the cassava processing methods and the equipment used is presented for each product as below.

A. CASSAVA CHIPS AND PELLETS

- Sort and weigh cassava tubers, followed by peeling and washing, either manually using hands or by use of automated peeling and washing machines (Figure 135A & B):



Figure 135 — Showing cassava (A) peeling & washing machine (all in one) and (B) peeling (alone)
Source: <https://www.juiceprocessline.com/wp-content/uploads/2021/02/cassava-washing-and-peeling-machine.jpg>;
<https://nationaltoday.com/wp-content/uploads/2020/06/Tapioca-640x514.jpg>

- The clean tubers are manually cut into pieces (chips/slices) of 2-5 cm (diameter) using sharp knives or a cutter machine (Figure 136);



Figure 136 — Showing a cutter for making cassava chips
Source: https://m.media-amazon.com/images/I/51LNiJNOGiL._AC_SX450_.jpg

- The chips are then spread out to dry in the open sun on raised platforms or in a drier machine to low moisture content of 11-13% (Mujaffar and Lalla, 2020); which enables them to store longer (Falade and Akingbala, 2010a; Adamade and Azogu, 2013). According to Usman and Idakkwo (2011), smaller sized chips/slices dry faster than bigger sizes, but it takes more time to cut smaller sizes by hand.
- The dried chips/slices can be packed in appropriate bags (jute bags or paper) for long storage or export (Adamade and Azogu, 2013).

B. TAPIOCA

- Using the flow chart for processing tapioca (Figure 137), follow step 1 to sort, weigh, peel and wash the cassava tubers;

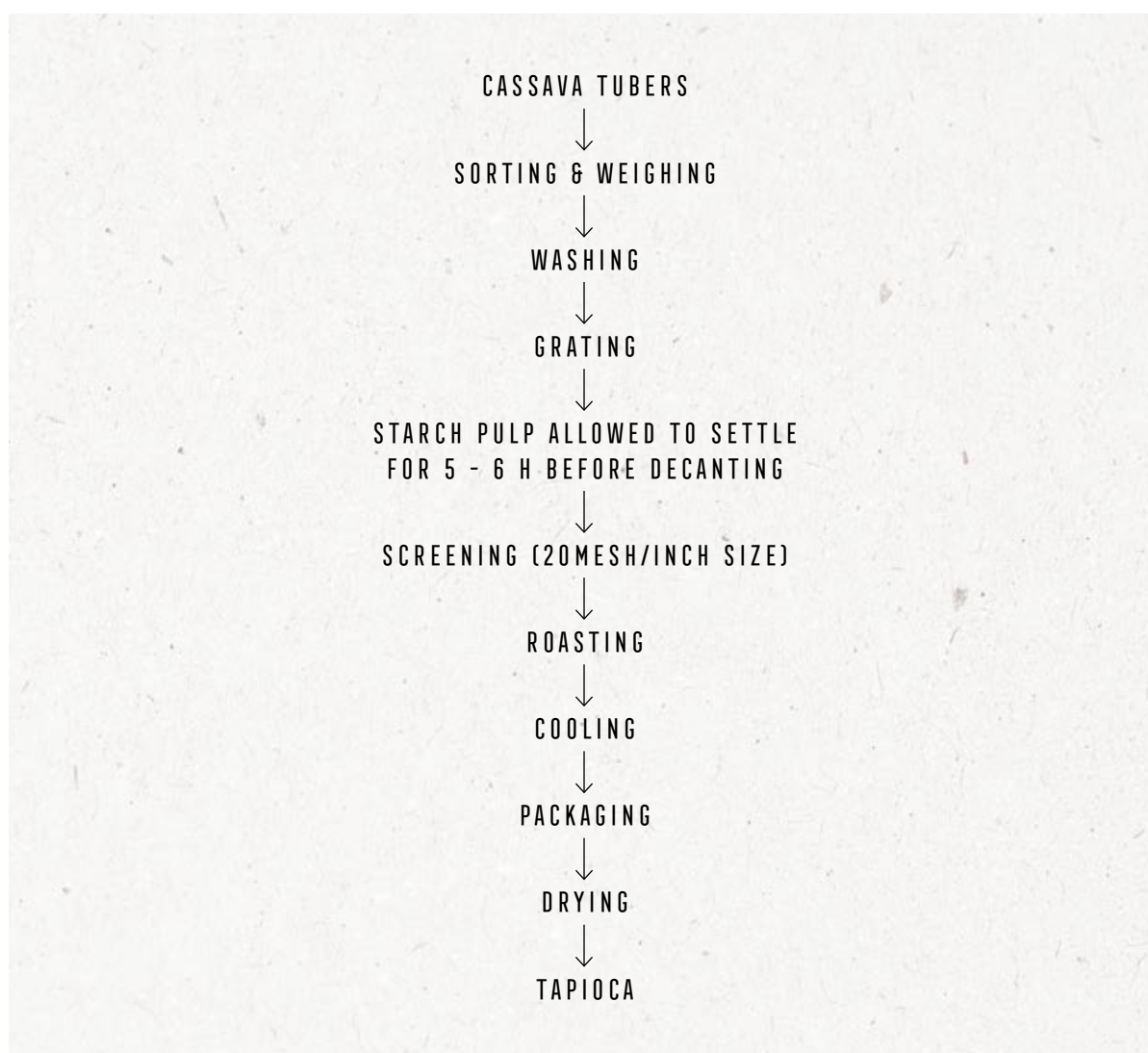


Figure 137 — Showing a tapioca processing flow chart

Source: <https://www.researchgate.net/profile/Monday-Otache/publication/336825378/figure/fig1/AS:818155026907137@1572074767714/Process-flow-chart-of-tapioca-gritfrom-cassava-tubers-2.png>

- Grate the washed cassava manually or by using the automated grater (Figure 138);



Figure 138 — Showing a grating machine used for grating cassava
Source: <https://sc04.alicdn.com/kf/HTB1rpKjKaSWBuNjSsrBq6y0mVXaa.jpg>

- Follow the rest of the steps in the tapioca processing flow chart (Figure 133), until the final product.

Tapioca is able to maintain some moisture content (8-12%) and a longer shelf life than the chips/slices (Falade and Akingbala, 2010a; Chukwu and Abdullahi, 2015).

C. CASSAVA STARCH

- Cassava starch is processed by first peeling and washing the tubers;
- The clean tubers are grated and sieved to remove the fibre to get a mash that is left to sediment and is decanted to collect the starch using traditional or automated methods (Figure 139);

Starch has a low gelatinization temperature and high water-binding capacity, which acts as a food stabilizer and a high viscosity which keep it from degrading quickly. However, the method of drying (Jekayinfa and Olajide, 2007) can affect the starch quality which may affect its marketability.

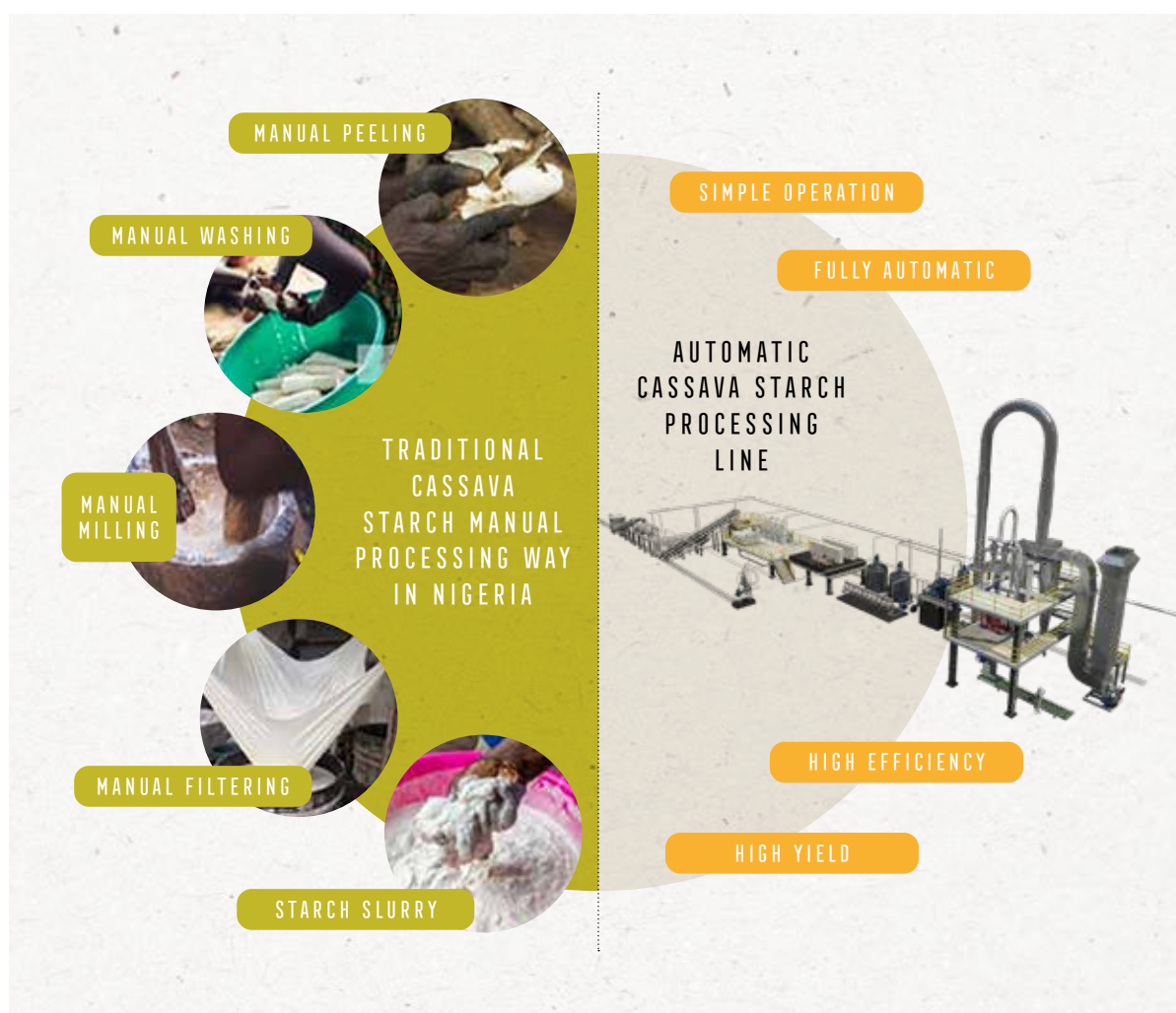


Figure 139 — Showing cassava starch processing using (A) traditional and (B) automated methods

Source: <https://www.cassavaprocessing.com/uploads/userup/20211202/20211202132125.jpg>

D. UNFERMENTED CASSAVA FLOUR

Unfermented cassava also referred to as **high quality cassava flour (HQCF)** is a white, smooth and odourless flour that is used as composite flour (Taiwo, 2006). It is processed using an improved processing technique that has been promoted for adoption by smallholder cassava farmers.

- The process involves grating using either manual graters or an automated cassava grater machine (Figure 138);
- The mash is dewatered using a screw or hydraulic press and pulverized;
- The result product is dried using solar or oven driers, and milled into flour before packaging.

All the the above processed products (chip/slices, tapioca, starch and flour) need proper packaging and storage to improve shelflife and safety (Uchechukwu-Agua *et al.*, 2015). A list of recommended packaging and storage materials for different cassava flour and garri products (Ogiehor and Ikenebomeh, 2006; Obadina *et al.*, 2009; Kulchan *et al.*, 2010; Ogugbue and Gloria 2011; Adejumo and Raji 2012; Uchechukwu-Agua *et al.*, 2015) is hereby presented in table 45.

Table 45 — Specifications recommended for packaging materials for storage of cassava products

PRODUCTS	PACKAGE MATERIALS	STORAGE CONDITIONS	DURATION (WEEKS)	REFERENCE	COMMENTS
Cassava flour-baked products	LDPE, OPP, plastic bag	30±1 °C	24	Shelf life stability in LDPE, OPP	Kulchan <i>et al.</i> (2010)
Garri	Gunny sack, HDPE, LDPE, plastic container	Ambient	9	Microbial stability in HDPE	Ogugbue and Gloria (2011)
Garri	LDPE, HDPE, plastic bucket and Hessian	30±1 °C	24	Proximate best in HDPE	Ogiehor and Ikenebomch (2006)
Fufu flour	Airtight dishes	28+2 °C; 33 °C, 76% RH	8	Flour was best at 33 % RH	Obadina <i>et al.</i> (2009)
Garri	Unpackaged, PP, PE, Hessian bag	16 and 36 °C	24	Best in PP and PE	Adejumo and Raji (2012)

Source: Uchechukwu-Agua *et al.* (2015)

E. FERMENTED CASSAVA

Cassava varieties are categorized as either “sweet” or “bitter” depending on the levels of anti-nutritional factors and toxins (cyanide) (Grace, 1977; Sundaresan *et al.*, 1987). The sweet and bitter varieties contain about 15-50 mg/kg and 15-400 mg/kg fresh weight of hydrogen cyanide, respectively (Famurewa and Emuekele, 2014). In the bitter varieties, the cyanide levels range from 75 to 1000 ppm and are influenced by variety, plant age, soil condition, fertilizer application and weather (Gomez and Valdivieso, 1983; Fomunyan *et al.*, 1984; Aregheore and Agunbiade, 1991). The high content of cyanogenic glucosides (CNG): linamarin (96%) and lotaustralin (4%), cyanohydrin and free hydrocyanic acid (HCN), make the bitter varieties poisonous (Cooke and Coursey, 1981; Aregheore & Agunbiade, 1991), and unfit for human and animal consumption (White *et al.*, 1998). Eating cassava products with high levels of cyanogens can cause acute intoxications (Mlingi *et al.*, 1992; Akintonwa *et al.*, 1994; Delange *et al.*, 1994; Ernesto *et al.*, 2002; Simeonova and Fishbein, 2004) with symptom such as vomiting, nausea, dizziness, stomach pains, weakness, headache, exaggerated goitre (Bourdoux, 1983), diarrhoea and inducement of paralysis leading to death in acute cases (Tylleskar *et al.*, 1992).

Sweet varieties are processed unfermented into products such as chips, pellets, Tapioca, starch and unfermented flour as described in sections 11.5 a-d. In contrast, the bitter varieties require more rigorous processing such as drying and fermentation. Fermentation is achieved by use of either lactic acid bacteria (*Lactobacillus coryneiformis*, *Lactobacillus delbruckii* and *Saccharomyces*) or yeast (baker's yeast) such as *Saccharomyces cerevisiae* (Oyewole and Odunfa, 1990; Akindahunsi *et al.*, 1999) at pH 6 and above (Hawashi *et al.*, 2019). The process of fermentation helps to reduce the toxic hydrocyanic acid (HCN) from the cyanogenic glucosides during hydrolysis of linamarin by linamarase enzyme (Conn, 1969) that occurs in cassava peels (Cardoso *et al.*, 2005; Famurewa and Emuekele, 2014). Furthermore, Chelule *et al.* (2010) and Nambisan (2011) found processing methods involving fermentation to be more effective in removal of linamarase and subsequent reduced residual cyanide than those without fermentation (Cooke and Maduagwu, 1978). The recommended safe level of cyanogens in cassava flour is 10 ppm or 10 mg HCN /kg according to the World Health Organisation (WHO) (FAO/WHO, 1991).

Several methods are used to reduce the cyanide content of bitter varieties, including: peeling, chipping, grating, soaking, boiling, fermenting and drying (Agbor-Egbe and Mbome, 2006; Obilie *et al.*, 2004). Traditional drying methods involve exposing cassava to direct radiation of the sun. Although this method is less costly, it is slow and encourages continuous fermentation and the development of mould, aflatoxicosis and mycotoxic infection that reduce the quality of cassava products. Modern methods of drying cassava are faster and can eradicate microbial infections leading to production of high-quality products with optimal cyanide levels (Famurewa and Emuekele, 2014).

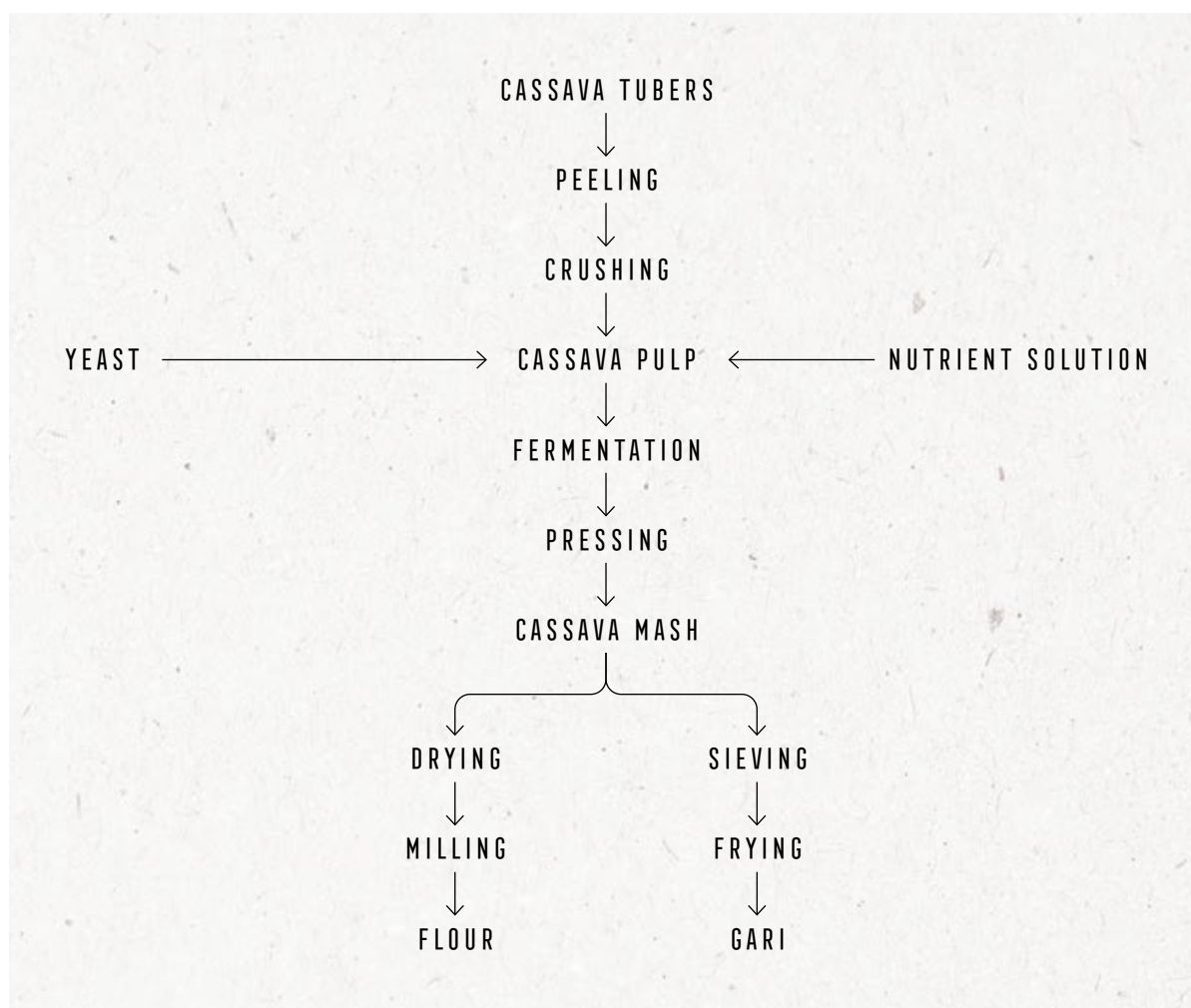


Figure 140 — Showing a production chart for cassava products (flour and gari) using solid-state fermentation (SSF) method
Source: <https://www.intechopen.com/media/chapter/67829/media/F1.png>

Among the most promising modern cassava fermentation methods is the “solid-state fermentation” or SSF method (Figure 140). The SSF methods can be used to degrade anti-nutritional factors and cyanide toxins and also enrich protein in cassava products such as flour, gari, starch and bread (Reade and Gregory, 1975; Vlavonou, 1988; Akindahunsi *et al.*, 1999). The SSF method is used to ferment products obtained from the roots/tubers, peels and leaves of cassava plants (Hawashi *et al.*, 2019). Therefore, in addition to reducing the toxic cyanogenic glycosides to a safe level, fermentation can increase proteins and reduce post-harvest losses for the various cassava food products (Caplice and Fitzgerald, 1999; Kostinek *et al.*, 2005; Achi and Akomas, 2006).



POSTHARVEST

KEY SUMMARY POINTS

CAUSE

- Postharvest cassava deterioration arises from poor handling of cassava tubers during and after harvest, which begins with mechanical damage such as cuts or bruises that allow pathogens to enter and cause physiological and microbial deterioration.

FIELD

- Handle harvested tubers with care by using harvesting baskets, and storing them under a shade until transportation to the market or processing plants;
- You may also keep small quantities of the tuber in water until transportation to the market or use at home;

TRANSPORT

- Be careful when loading cassava tubers on to the means of transport (bicycle, wheel burrow, motorcycle, truck etc.), not to cause physical damage;

STORAGE

- Store cassava tubers using traditional methods common in your area, or preferably, improved methods such as plastic bags, waxing or freezing that retain moisture to keep longer;
- You may also process the cassava into dry chips/slices, tapioca, flour or starch for longer shelf life and broader use in different products.

PROCESSING

- Cassava can be processed into various food forms such as chips, tapioca, starch, and unfermented and fermented flour to extend shelflife, food quality (including enriching proteins) and to reduce postharvest losses;
- Processing can be achieved by peeling, chipping, grating, soaking, boiling, drying and fermenting;
- Fermentation helps to reduce or remove cyanide (HCN), a toxic compound that limits the consumption of bitter cassava varieties
- Symptoms of HCN poisoning include vomiting, nausea, dizziness, stomach pains, weakness, headache, exaggerated goitre, diarrhoea, paralysis and death in acute cases.



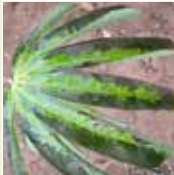




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APPENDIX

APPENDIX 1 - SUMMARY PEST DAMAGE AND CONTROL OPTIONS

INSECT PESTS				CONTROL OPTIONS (IPM)	STAGE OF CULTIVATION							
MOST DAMAGING GROWTH STAGE	PLANT GROWTH STAGE/ ORGAN DAMAGED	DAMAGE/IMPACT	FIELD SELECTION & PREPARATION		SELECTION OF PLANTING MATERIAL	PLANTING	CROP/PEST MANAGEMENT	TUBER DEVELOPMENT	HARVESTING OF STEMS & TUBERS	STORAGE OF STEMS & TUBERS	POST-HARVEST MANAGEMENT	
WHITEFLIES												
 Nymphs and adults	 Leaves	- Sooty mould causes reduced photosynthesis, plant growth and yield - Transmission of viruses that cause stunted growth and yield & tuber quality reduction. - Reduce quality of leaves.	Isolate new cassava crops from older fields to reduce whitefly entry.	x								
			Select disease-free planting materials using visual judgement or virus screening assays.		x							
			Use elite cassava varieties with proven resistance or tolerance to whitefly.		x							
			Grow thin-leaved cassava varieties for lower whitefly egg and nymphal instar densities.		x							
			Dip cassava stem cuttings in Deltamethrin or Cypermethrin to kill whitefly eggs, nymphs and adults before distribution to farmers.			x						
			Intercrop cassava at high density with cow peas or maize to reduce whitefly populations build up.			x						
			Establish repellent crops such as marigold near or around cassava fields to repel whiteflies.			x						
			Remove and burn crop debris and weeds (potential hosts) with whiteflies (eggs, nymphs and adults) and visible viral disease symptoms within and around young fields.				x					

INSECT PESTS				STAGE OF CULTIVATION									
MOST DAMAGING GROWTH STAGE	PLANT GROWTH STAGE/ ORGAN DAMAGED	DAMAGE/IMPACT	CONTROL OPTIONS (IPM)		FIELD SELECTION & PREPARATION	SELECTION OF PLANTING MATERIAL	PLANTING	CROP/PEST MANAGEMENT	TUBER DEVELOPMENT	HARVESTING OF STEMS & TUBERS	STORAGE OF STEMS & TUBERS	POST-HARVEST MANAGEMENT	
			Rogue/remove cassava plants infected with viral diseases in young crops (<3 months old), followed by replanting healthy planting materials.					x	x				
			Spray organic neem or azadirachtin on top and underneath of cassava leaves until completely wet to kill eggs, nymphs and/or repel adults of whitefly vectors.					x	x				
			Practice timely planting, method of planting, spacing, soil fertility (use of organic manure), weed and water management to ensure optimum plant growth and pest management.				x	x	x	x			
			Regularly monitor whitefly population occurrence and growth to guide deployment of control options.		x	x	x	x	x				
WHITEFLIES													
		- Sucking sap from cassava shoots. - Contaminating plant with toxic saliva leading to defoliation, stunting and drying of cassava plants. - Saliva encourages sooty mould growth which interferes with photosynthesis. - Reduce quality of leaves.	Use only healthy/pest free planting materials.			x		x					
			Avoid movement of plants/cuttings with mealybugs.			x							
arvae	Leaves, shoots and branches		Rotate fields to grow maize and legumes.					x					
			Dip cassava planting materials in Dimethoate at a rate of 10-20 ml in 14 litres of water during planting period to avoid spread of mealybugs.				x						
			Grow mealybug resistant cassava varieties.			x							
			Plant early at the start of the rainy season to enhance plant establishment and resisting mealybug attack.				x						

INSECT PESTS				STAGE OF CULTIVATION							
MOST DAMAGING GROWTH STAGE	PLANT GROWTH STAGE/ ORGAN DAMAGED	DAMAGE/IMPACT	CONTROL OPTIONS (IPM)	FIELD SELECTION & PREPARATION	SELECTION OF PLANTING MATERIAL	PLANTING	CROP/PEST MANAGEMENT	TUBER DEVELOPMENT	HARVESTING OF STEMS & TUBERS	STORAGE OF STEMS & TUBERS	POST-HARVEST MANAGEMENT
			Remove or prune infested plants, or plant parts (leaves, shoots) and burn or bury to kill the mealybugs.				x				
			Practice clean weeding of cassava crops to remove potential alternative host plant species for the mealybugs within and near cassava fields.				x				
			Apply manure or other fertilisers to reduce mealybug population, since the nutrition helps to produce more parasitoid wasps with higher fertility levels.				x				
			Mulch to enhance the antibiotic properties of cassava against mealybug infestation.				x				
			Organic Neem or Azadirachtin can be applied on cassava leaf surfaces and undersides until completely wet to kill eggs, nymphs and/or repel adults of whitefly vectors.				x				
			Immerse cassava cuttings in manipueira for 60 minutes to reduce infestation.		x						
			Cyanide at high levels acts as a phagostimulant for mealybugs, while rutin has an antibiotic effect on the pest.		x						
			Use of mulch and manure increased cassava resistance against mealybug infestation.				x				

INSECT PESTS				STAGE OF CULTIVATION							
MOST DAMAGING GROWTH STAGE	PLANT GROWTH STAGE/ ORGAN DAMAGED	DAMAGE/IMPACT	CONTROL OPTIONS (IPM)	FIELD SELECTION & PREPARATION	SELECTION OF PLANTING MATERIAL	PLANTING	CROP/PEST MANAGEMENT	TUBER DEVELOPMENT	HARVESTING OF STEMS & TUBERS	STORAGE OF STEMS & TUBERS	POST-HARVEST MANAGEMENT
MITES											
		• Loss of leaves and shoot tips. • Reduce quality of leaves. • Reduce number and size of tubers before 7 months. • Tuber yield loss of 50% in severe cases.	Spray plants with a strong jet of water to reduce build-up of mite populations. Apply insecticidal soap to plants. Plant cassava crops at the beginning of the rainy season for vigorous growth for plants to tolerate mite infestation. Intercrop with crops such as cowpea that may preferentially attract mites to itself and reduce damage to cassava plants. Clean weed cassava crops to remove potential alternative host plant species for CGM within and around the fields. Isolate new cassava crops from older fields that may be infested with CGM. Plant cassava cutting using the horizontal/burying method which kills off CGM on the stems as opposed to the vertical or inclined/slanting/tilted method that promotes early infestation upon sprouting for mites on the unburied parts of the stem cuttings. The predatory mite <i>Typhlodromalus aripo</i>, family Phytoseiidae has successfully controlled CGM in many regions of Africa. Organic Neem or Azadirachtin can be sprayed on cassava leaf surfaces (including the undersides of leaves) until completely wet to kill eggs, nymphs and/or repel adult mite infestation.				x				
Larvae and adults	Leaves, shoots and branches						x				

INSECT PESTS				STAGE OF CULTIVATION							
MOST DAMAGING GROWTH STAGE	PLANT GROWTH STAGE/ ORGAN DAMAGED	DAMAGE/IMPACT	CONTROL OPTIONS (IPM)	FIELD SELECTION & PREPARATION	SELECTION OF PLANTING MATERIAL	PLANTING	CROP/PEST MANAGEMENT	TUBER DEVELOPMENT	HARVESTING OF STEMS & TUBERS	STORAGE OF STEMS & TUBERS	POST-HARVEST MANAGEMENT
			Cassava stem cuttings may be dipped in Deltamethrin or Cypermethrin to kill whitefly eggs, nymphs and adults before distribution to farmers.	x							
			Plant mite resistant/tolerant cassava varieties.	x							
			Intercrop with crops such as cowpea may reduce damage; introductions of the predatory mite <i>Typhlodromalus aripo</i> .			x	x				
SCALES											
  Larvae	Leaves and branches, roots and tubers	- Death of plant due to early and heavy infestation - Reduction in number and quality of leaves - Reduction in number, size and quality of tubers (distortion of tuber shape)	Select scale-free planting material.		x						
			Dip cassava cuttings in Deltamethrin or Cypermethrin to kill the scales before distribution to farmers.		x						
			Cuttings can be treated with insecticidal bioproducts before planting to minimise the impact of the pest.		x						
			Grow scale resistant cassava varieties.			x					
			Early planting in dry season can delay infestation.	x	x	x	x				
			Isolate new cassava crops from older fields that may be infested with scales.	x							
			Remove and burn crop debris/residues infested with scales.	x	x						
			Clean weed cassava crops to remove potential alternative host plant species inside and around cassava fields.				x				
			Apply organic matter to soil to improve fertility.				x				

INSECT PESTS				STAGE OF CULTIVATION							
MOST DAMAGING GROWTH STAGE	PLANT GROWTH STAGE/ ORGAN DAMAGED	DAMAGE/IMPACT	CONTROL OPTIONS (IPM)	FIELD SELECTION & PREPARATION	SELECTION OF PLANTING MATERIAL	PLANTING	CROP/PEST MANAGEMENT	TUBER DEVELOPMENT	HARVESTING OF STEMS & TUBERS	STORAGE OF STEMS & TUBERS	POST-HARVEST MANAGEMENT
THRIPS											
 Adults	 Leaves	Reduction of leaf surfaces and number	Use thick layers of dried mulch on the soil to prevent larvae from easily entering the soil to pupate.				x				
			Remove cassava and other plant debris from the previous crop, improve drainage, and clean weed to reduce the pest population.				x				
 Adults	 Leaves	Reduction of leaf surfaces and number	Use overhead irrigation to wash the insects off the cassava leaves.				x				
			Use sticky traps with yellow and blue colours to attract the thrips, which eventually die.				x				
 Adults	 Leaves	Reduction of leaf surfaces and number	Provide sufficient water to enhance rapid plant growth and to reduce stresses that make plants more susceptible to thrips attack.				x				
			Deep ploughing, harrowing and solarisation or exposure to the sun help to kill pupae in the soil from previous season.	x							
 Adults	 Leaves	Reduction of leaf surfaces and number	Plant early to ensure proper establishment of cassava plants and prevent thrips attack.			x					
			Crop rotation also avoids successive planting of susceptible cassava varieties and reduces the occurrence of thrip infestation.				x				

INSECT PESTS				STAGE OF CULTIVATION							
MOST DAMAGING GROWTH STAGE	PLANT GROWTH STAGE/ ORGAN DAMAGED	DAMAGE/IMPACT	CONTROL OPTIONS (IPM)	FIELD SELECTION & PREPARATION	SELECTION OF PLANTING MATERIAL	PLANTING	CROP/PEST MANAGEMENT	TUBER DEVELOPMENT	HARVESTING OF STEMS & TUBERS	STORAGE OF STEMS & TUBERS	POST-HARVEST MANAGEMENT
			Intercropping with cowpea helped reduce the thrips populations.				x				
			Organic Neem or Azadirachtin can be sprayed on cassava leaf surfaces (including the undersides of leaves) until completely wet to kill eggs, nymphs and/or repel adults of thrips.				x				
			Cassava stem cuttings may be dipped in Deltamethrin or Cypermethrin to kill thrips eggs, nymphs and adults before distribution to farmers.		x						
			Use neem-based pesticides to control young nymphs by adding 0.1 to 0.5% of soft soap.				x				
			Reflective mulches deter thrips.	x		x	x				
VARIEGATED GRASSHOPPERS											
	Egg Larva and adult		- Chew leaves and debark stems. - Cause dieback of growing points and wilting of plants.	x			x				
	Leaves, shoots and branches		Hand pick grasshoppers. This is only feasible in small plots.				x				
			Use biopesticides such as <i>Metarhizium anisopliae</i> to control grasshopper population.				x				
			Spray chemical insecticide on first larval stages.				x				
			Apply pesticides on the borders of both the crop and the bushes prevent further entry into the field by the grasshoppers. This should be done at least 7-14 days, depending on the level of infestation.				x				

INSECT PESTS				STAGE OF CULTIVATION								
MOST DAMAGING GROWTH STAGE	PLANT GROWTH STAGE/ ORGAN DAMAGED	DAMAGE/IMPACT	CONTROL OPTIONS (IPM)	FIELD SELECTION & PREPARATION	SELECTION OF PLANTING MATERIAL	PLANTING	CROP/PEST MANAGEMENT	TUBER DEVELOPMENT	HARVESTING OF STEMS & TUBERS	STORAGE OF STEMS & TUBERS	POST-HARVEST MANAGEMENT	
TERMITES												
 Adults	 Stems, roots and tubers	Nodes and roots are destroyed causing poor plant growth, death and rotting.	Avoid fields infested with termites when selecting planting sites.	x								
			Plant cassava on ridges to avoid termite establishment.		x	x						
			Plant cassava cuttings during sufficient rains.	x								
			Uproot and destroy plants that fail to germinate to look out for termite infestation.					x				
			Collect driver ants and put them in areas affected by termites					x				
			Dig out termite colonies and remove the white ant hills/ termite mounds.					x				
			Put wood ash on the base of cassava plants.					x				
			Where possible sterilize the soil with hot water before planting if infestation is localized.	x								
			Remove all plant debris and burn to remove any suitable sites for termite colonisation.						x			
			Plant cassava crops during the rainy season.									
If mulch material is used in the field, avoid putting it too close to the cassava plant since it will attract termites.						x						
Irrigate the fields to keep soil moist in the dry season.							x					
Plant cassava cuttings at high density to cater for those that may off be due to termite damage.						x						

INSECT PESTS				STAGE OF CULTIVATION								
MOST DAMAGING GROWTH STAGE	PLANT GROWTH STAGE/ ORGAN DAMAGED	DAMAGE/IMPACT	CONTROL OPTIONS (IPM)	FIELD SELECTION & PREPARATION	SELECTION OF PLANTING MATERIAL	PLANTING	CROP/PEST MANAGEMENT	TUBER DEVELOPMENT	HARVESTING OF STEMS & TUBERS	STORAGE OF STEMS & TUBERS	POST-HARVEST MANAGEMENT	
			Use of driver ants significantly reduced termite infestation.				x					
			Expose the termites to predators such as ants, dragonflies, ground beetles, bats, birds, frogs.				x					
			Deploy <i>Beauveria bassiana</i> as biological control if available in the country.				x					
			Spray cassava plants with extract of neem leaves or fruits to keep away the termites.				x					
			Spray diazinon around the edges of the field to prevent termites from entering the crop and wherever there is an anthill.				x					
			Establish barrier plants such as basil (<i>Ocimum</i> spp.) to repel termites.				x				x	
WHITE GRUBS												
  arvae	 Roots	Feed on root systems causing poor plant growth and death in severe cases.	Check for white grub infestation by early ploughing in the growing season when the grubs are still closer to the surface.	x								
			If present, expose the white grubs (larvae, pupae and adults) to the sun and predators such as birds.	x	x							
			Practice crop rotation by inclusion of resistant or less susceptible deep-rooted legumes such as alfalfa, cowpea, and pigeon peas.			x	x					
			Use trap crops such as African marigold, sunflower, and castor or repellent plants such as chives, garlic, tansy, and catnip to prevent adult beetles from attacking the main crop.						x			

INSECT PESTS											
MOST DAMAGING GROWTH STAGE	PLANT GROWTH STAGE/ ORGAN DAMAGED	DAMAGE/IMPACT	CONTROL OPTIONS (IPM)	STAGE OF CULTIVATION							
				FIELD SELECTION & PREPARATION	SELECTION OF PLANTING MATERIAL	PLANTING	CROP/PEST MANAGEMENT	TUBER DEVELOPMENT	HARVESTING OF STEMS & TUBERS	STORAGE OF STEMS & TUBERS	POST-HARVEST MANAGEMENT
							x				
							x				
							x				
				x							
				x							
CATERPILLARS											
		Damage to leaves and complete defoliation of plants under heavy infestation	Pick and kill visible caterpillars in the field.				x				
Caterpillar	Leaves and shoots		Regularly check cassava fields for symptoms of damage.				x				
			Rotate cassava with maize to decrease the number of worms.			x					
			Use of a bio-rational pesticide such as <i>Bacillus thuringiensis</i> , which is environmentally friendly and effective against early instars of the pest.				x				
			Spray a contact insecticide (ex: cypermethrin).				x				

NEMATODES

OTHER ARTHROPOD PESTS												
MOST DAMAGING GROWTH STAGE	PLANT GROWTH STAGE/ ORGAN DAMAGED	DAMAGE/IMPACT	CONTROL OPTIONS (IPM)	STAGE OF CULTIVATION								
					FIELD SELECTION & PREPARATION	SELECTION OF PLANTING MATERIAL	PLANTING	CROP/PEST MANAGEMENT	TUBER DEVELOPMENT	HARVESTING OF STEMS & TUBERS	STORAGE OF STEMS & TUBERS	POST-HARVEST MANAGEMENT
LEAF-CUTTER ANTS												
		Cut leaves into portions and heavy infestation can completely defoliate plants.	Change land use such as interchanging grasses with shade crops.	x								
Adults	Leaves		Look for trails of the leaf cutter ants and locate their nest at night when they are most active.				x					
			Mark areas with established nests by use of painted sticks or cloth.				x					
			Set up barriers to prevent entry of leaf-cutting ants into the crop.				x					
			Set up formicide baits that contain an attractive substrate and an insecticide to trap leaf-cutting ants.				x					
CASSAVA SHOOTFLIES												
		Injure the terminal buds, leading to retarded plant shoot growth and development of lateral budding and branches.	Management is unwarranted due to the insignificant impact on cassava leaves and tubers, which are the economic components of cassava.									
Adults	Shoots											

OTHER ARTHROPOD PESTS				STAGE OF CULTIVATION							
MOST DAMAGING GROWTH STAGE	PLANT GROWTH STAGE/ ORGAN DAMAGED	DAMAGE/IMPACT	CONTROL OPTIONS (IPM)	FIELD SELECTION & PREPARATION	SELECTION OF PLANTING MATERIAL	PLANTING	CROP/PEST MANAGEMENT	TUBER DEVELOPMENT	HARVESTING OF STEMS & TUBERS	STORAGE OF STEMS & TUBERS	POST-HARVEST MANAGEMENT
FRUIT FLIES											
 Adults	 Stems	Puncture and make tunnels in the stems, forming brown corridors in the pith, which could result in stem rot.	Select healthy plants with white piths, indicating freedom from fruit fly infestation.		x						
			Change type of land use such as planting shade trees to create unfavourable conditions for the survival of the pest e under the shade of a tree plantation.	x	x						
			Clean up the fields of materials that serve to propagate the development of the pest.	x							
			Protect cassava plants by banding the stems with grease or polyisobutylene gel to avoid fruit flies settling and piercing the stems.		x						
			Use barriers and sugar baits (hydrolyzed maize) with pesticide to trap and kill of the fruit flies.			x	x				
			Use appropriate registered and approved pesticides for fruit fly management.				x				

OTHER ARTHROPOD PESTS												
MOST DAMAGING GROWTH STAGE	PLANT GROWTH STAGE/ ORGAN DAMAGED	DAMAGE/IMPACT	CONTROL OPTIONS (IPM)	STAGE OF CULTIVATION								
				FIELD SELECTION & PREPARATION	SELECTION OF PLANTING MATERIAL	PLANTING	CROP/PEST MANAGEMENT	TUBER DEVELOPMENT	HARVESTING OF STEMS & TUBERS	STORAGE OF STEMS & TUBERS	POST-HARVEST MANAGEMENT	
STEM BORERS												
	Adults	 Stems	Stems and branches may be reduced to sawdust. In the dry season the affected stems and branches lose their leaves and may die.	Remove and burn infested planting materials.				x				
				Plant healthy undamaged stakes.			x	x				
				Intercrop with maize to reduce stemborer populations until the maize is harvested.			x	x				
				Malathion applied manually with “polyspray” caused 100% larval mortality.				x				
			Create awareness on the biology, impact and management of stem borers.	x	x	x		x				
CASSAVA LACE-WING BUG												
	Adults	 Leaves	Damage to leaves is like that made by mites. Damaged or defoliated leaves reduce photosynthesis and hence growth and yield.	Good field sanitation should rid the field of weeds and plant residues from previous crops.	x							
				Practice integrated pest management.					x			

OTHER ARTHROPOD PESTS											
MOST DAMAGING GROWTH STAGE	PLANT GROWTH STAGE/ ORGAN DAMAGED	DAMAGE/IMPACT	CONTROL OPTIONS (IPM)	STAGE OF CULTIVATION							
				POST-HARVEST MANAGEMENT	STORAGE OF STEMS & TUBERS	HARVESTING OF STEMS & TUBERS	TUBER DEVELOPMENT	CROP/PEST MANAGEMENT	PLANTING	SELECTION OF PLANTING MATERIAL	FIELD SELECTION & PREPARATION
CASSAVA ROOT/BURROWER BUGS											
	Nymphs and adults		Tubers	Nymphs and adults make holes on cassava tubers, opening routes for entry of soil-borne pathogens such as the fungal species <i>Aspergillus</i>, <i>Diplodia</i>, <i>Fusarium</i>, <i>Phytophthora</i> and <i>Pythium</i>, and the alga <i>Genicularia</i> sp., which cause coffee-black lesions.	Control is difficult because of its polyphagous nature and adaptation to different soil environments.						

OTHER ARTHROPOD PESTS											
MOST DAMAGING GROWTH STAGE	PLANT GROWTH STAGE/ ORGAN DAMAGED	DAMAGE/IMPACT	CONTROL OPTIONS (IPM)	STAGE OF CULTIVATION							
				FIELD SELECTION & PREPARATION	SELECTION OF PLANTING MATERIAL	PLANTING	CROP/PEST MANAGEMENT	TUBER DEVELOPMENT	HARVESTING OF STEMS & TUBERS	STORAGE OF STEMS & TUBERS	POST-HARVEST MANAGEMENT
ARMY WORM											
		- Damage leaves/ foliage of crops. - Under high infestation, the larvae may burrow into the buds, which retards plant growth and affects yield.	Remove crop debris from a previously infested crop.	x							
			Clean weeding to remove potential host plants.					x			
Larvae	Leaves, buds and shoot		Put charcoal dust or ash or a detergent on the affected plants.				x				
			Use plant extracts such as chilli, neem, Tephrosia, Tithonia, Lantana and garlic that have proved successful in reducing the numbers on infested plants.				x				
			Handpicking egg masses and caterpillars has been tried out when the populations are still low.				x				
			Practice early planting or grow early maturing crop varieties.			x					
			Use registered and approved pesticides within your country, which may include, malathion, permethrin, and lambda-cyhalothrin that have successfully reduced army worm abundance				x				
			Rotate the use of chemical pesticides with different modes of action to prevent resistance development by the pest.			x					
			Observe strict regulation of planting materials from infested areas.		x						

OTHER ARTHROPOD PESTS												
MOST DAMAGING GROWTH STAGE	PLANT GROWTH STAGE/ ORGAN DAMAGED	DAMAGE/IMPACT	CONTROL OPTIONS (IPM)	STAGE OF CULTIVATION								
TIGER MOTH												
		- Young instars make round punctures in the leaves while feeding. - Adults make irregular holes on the leaves.	Remove and burn any leaves that are seen with the Tiger moth larvae and instars to reduce their populations.					x				
Young instars and adults	Leaves		Handpicking egg masses and caterpillars has been tried out when the populations are still low.					x				
GALL MIDGE												
		Under serious attack roots weaken and leaves turn yellow.	Where possible, plant in dry areas.	x		x						
Adults	Leaves		Establish fields in open locations and space plants to allow adequate ventilation.	x								
			Control weeds beneath and around plants.					x				
			Remove all fallen leaves from fields, bury or burn.					x				
			Colour traps present a potential tactic for monitoring or mating disruption.					x				
			Use tolerant/resistant varieties.		x	x						
		Avoid late transplantation during first crop season.					x					
		Destroy the weeds which serve as alternative hosts.					x				x	

Notes: 'x' = integrated pest management (IPM) interventions are indicated for the most appropriate cultivation and growth stage

BACTERIAL DISEASES				STAGE OF CULTIVATION								
CAUSAL PATHOGEN/S	PLANT GROWTH STAGE/ ORGAN DAMAGED	DAMAGE/IMPACT	CONTROL OPTIONS (IPM)	FIELD SELECTION & PREPARATION	SELECTION OF PLANTING MATERIAL	PLANTING	CROP/PEST MANAGEMENT	TUBER DEVELOPMENT	HARVESTING OF STEMS & TUBERS	STORAGE OF STEMS & TUBERS	POST-HARVEST MANAGEMENT	
CASSAVA BACTERIAL BLIGHT (CBB)												
<i>Xanthomonas axonopodis</i> pv. <i>manihotis</i>	  Leaves, stems and roots	• Wilting, dieback and vascular necrosis, and angular necrotic spotting of the leaves • Chlorotic ring encircling the spots. • The leaves' ability to carry out photosynthesis is impaired. • Possible death of plants. • Reduction of tuber numbers and size.	Rotate cassava crops with non-hosts			x						
			Plough crop debris into soil after harvest or remove and burn it.	x								
			Prune infected parts (leaves and branches) from cassava plants.				x					
			Propagate cuttings only from healthy mature cassava plants.		x							
			Intercrop cassava with corn (maize) and melon.				x					
			Practice clean weed management to remove possible alternative host plants.				x					
			Delayed planting towards the close of the rainy season helps reduce spread.				x					
			In soils with low potassium, add fertiliser rich in potassium to reduce severity of CBB.	x								
			Rotate cassava crops with non-hosts.				x					
			Tools should be regularly disinfected using a bactericide.				x	x				
			Use of elite cassava varieties with proven resistance or tolerance to CBB.				x					

BACTERIAL DISEASES				STAGE OF CULTIVATION							
CAUSAL PATHOGEN/S	PLANT GROWTH STAGE/ ORGAN DAMAGED	DAMAGE/IMPACT	CONTROL OPTIONS (IPM)	FIELD SELECTION & PREPARATION	SELECTION OF PLANTING MATERIAL	PLANTING	CROP/PEST MANAGEMENT	TUBER DEVELOPMENT	HARVESTING OF STEMS & TUBERS	STORAGE OF STEMS & TUBERS	POST-HARVEST MANAGEMENT
CASSAVA LEAF SPOT/BACTERIAL ANGULAR LEAF SPOT											
<i>Xanthomonas campestris</i> pv. <i>Cassava</i>	 Leaves	Leaves develop brown spots on upper leaf surfaces which may cause the leaves to crack and fall off the plant in severe cases.	Rotate cassava crops with non-hosts			x					
			Plough crop debris into soil after harvest or remove and burn it.	x							
			Prune infected parts (leaves and branches) from cassava plants.				x				
			Propagate cuttings only from healthy mature cassava plants.		x						
			Intercrop cassava with corn (maize) and melon.				x				
			Practice clean weed management to remove possible alternative host plants.				x				
			Delayed planting towards the close of the rainy season helps reduce spread.				x				
			In soils with low potassium, add fertiliser rich in potassium to reduce severity of CBB.	x							
			Rotate cassava crops with non-hosts.			x					
			Tools should be regularly disinfected using a bactericide.		x	x					
			Use of elite cassava varieties with proven resistance or tolerance to CBB.		x						

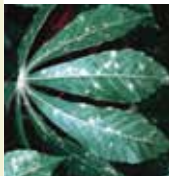
BACTERIAL DISEASES				STAGE OF CULTIVATION							
CAUSAL PATHOGEN/S	PLANT GROWTH STAGE/ ORGAN DAMAGED	DAMAGE/IMPACT	CONTROL OPTIONS (IPM)	FIELD SELECTION & PREPARATION	SELECTION OF PLANTING MATERIAL	PLANTING	CROP/PEST MANAGEMENT	TUBER DEVELOPMENT	HARVESTING OF STEMS & TUBERS	STORAGE OF STEMS & TUBERS	POST-HARVEST MANAGEMENT
BACTERIAL STEM GALL											
<i>Agrobacterium tumefaciens</i> Biovar 1	 Leaves and stems	- Affected plants develop galls (swellings) on stems. - Plants experience stunted growth and die back, especially when infection occurs earlier.	Rotate cassava crops with non-hosts			x					
			Plough crop debris into soil after harvest or remove and burn it.	x							
			Prune infected parts (leaves and branches) from cassava plants.				x				
			Propagate cuttings only from healthy mature cassava plants.		x						
			Intercrop cassava with corn (maize) and melon.				x				
			Practice clean weed management to remove possible alternative host plants.				x				
			Delayed planting towards the close of the rainy season helps reduce spread.				x				
			In soils with low potassium, add fertiliser rich in potassium to reduce severity of CBB.	x							
			Rotate cassava crops with non-hosts.			x					
			Tools should be regularly disinfected using a bactericide.		x	x					
			Use of elite cassava varieties with proven resistance or tolerance to CBB.		x						

BACTERIAL DISEASES												
CAUSAL PATHOGEN/S	PLANT GROWTH STAGE/ ORGAN DAMAGED	DAMAGE/IMPACT	CONTROL OPTIONS (IPM)	STAGE OF CULTIVATION								
				FIELD SELECTION & PREPARATION	SELECTION OF PLANTING MATERIAL	PLANTING	CROP/PEST MANAGEMENT	TUBER DEVELOPMENT	HARVESTING OF STEMS & TUBERS	STORAGE OF STEMS & TUBERS	POST-HARVEST MANAGEMENT	
BACTERIAL STEM ROT AND BACTERIAL WILT												
<i>Erwinia carotovora subsp. Carotovora</i> ; <i>Erwinia herbicola</i>	 Leaves and stems	- Leaves and shoots wilt, followed by die back. - Affected stems develop cankers (cracks), which reduces the quality of planting materials. - Severe infection leads to poor germination, stunting and yield loss.	Rotate cassava crops with non-hosts			x						
			Plough crop debris into soil after harvest or remove and burn it.	x								
			Prune infected parts (leaves and branches) from cassava plants.				x					
			Propagate cuttings only from healthy mature cassava plants.		x							
			Intercrop cassava with corn (maize) and melon.				x					
			Practice clean weed management to remove possible alternative host plants.				x					
			Delayed planting towards the close of the rainy season helps reduce spread.				x					
			In soils with low potassium, add fertiliser rich in potassium to reduce severity of CBB.	x								
Rotate cassava crops with non-hosts.					x							
Tools should be regularly disinfected using a bactericide.					x	x						
Use of elite cassava varieties with proven resistance or tolerance to CBB.					x							

Notes: 'x' = integrated pest management (IPM) interventions are indicated for the most appropriate cultivation and growth stage
 Above control measures apply to all bacterial diseases of cassava

FUNGAL DISEASES												
CAUSAL PATHOGEN/S	PLANT GROWTH STAGE/ ORGAN DAMAGED	DAMAGE/ IMPACT	CONTROL OPTIONS (IPM)	STAGE OF CULTIVATION								
				FIELD SELECTION & PREPARATION	SELECTION OF PLANTING MATERIAL	PLANTING	CROP/PEST MANAGEMENT	TUBER DEVELOPMENT	HARVESTING OF STEMS & TUBERS	STORAGE OF STEMS & TUBERS	POST-HARVEST MANAGEMENT	
CERCOSPORA LEAF SPOTS												
CASSAVA BROWN LEAF SPOT												
<i>Passalora henningsii</i> / <i>Cercosporidium henningsii</i>	 Leaves	Damage older, lower leaves at 5-6 months after planting, leading to premature loss of leaves.	Plant at wide spacing for reduced density to allow wind to dry the leaves as quickly as possible.			x						
			Practice crop rotations with non-host plants every 3-5 years.			x						
			Plant in the wet season such that plants reach the susceptible stage (6-8-months-old) during the dry season.									
			Avoid establishing new crops next to old, infected fields.	x								
			After harvest plough the crop debris into soil or remove and burn it.	x								
			Prune disease infected leaves and branches from the plant and either bury or burn them.				x					
			Use cassava cuttings selected from healthy plants only while still in the field.		x							
			Remove weeds in and around cassava crops to avoid disease spreading into the field.				x					
Plant cassava resistant varieties if available.												

FUNGAL DISEASES				STAGE OF CULTIVATION							
CAUSAL PATHOGEN/S	PLANT GROWTH STAGE/ ORGAN DAMAGED	DAMAGE/ IMPACT	CONTROL OPTIONS (IPM)	FIELD SELECTION & PREPARATION	SELECTION OF PLANTING MATERIAL	PLANTING	CROP/PEST MANAGEMENT	TUBER DEVELOPMENT	HARVESTING OF STEMS & TUBERS	STORAGE OF STEMS & TUBERS	POST-HARVEST MANAGEMENT
CASSAVA LEAF BLIGHT											
<i>Cercospora vicosae</i>	 Leaves	Severe defoliation in the susceptible cultivars.	Plant at wide spacing for reduced density to allow wind to dry the leaves as quickly as possible.			x					
			Practice crop rotations with non-host plants every 3-5 years.			x					
			Plant in the wet season such that plants reach the susceptible stage (6-8-months-old) during the dry season.			x					
			Avoid establishing new crops next to old, infected fields.	x							
			After harvest plough the crop debris into soil or remove and burn it.	x							
			Prune disease infected leaves and branches from the plant and either bury or burn them.				x				
			Use cassava cuttings selected from healthy plants only while still in the field.		x						
			Remove weeds in and around cassava crops to avoid disease spreading into the field.				x				
			Plant cassava resistant varieties if available.		x						

FUNGAL DISEASES												
CAUSAL PATHOGEN/S		PLANT GROWTH STAGE/ ORGAN DAMAGED	DAMAGE/ IMPACT	CONTROL OPTIONS (IPM)	STAGE OF CULTIVATION							
					FIELD SELECTION & PREPARATION	SELECTION OF PLANTING MATERIAL	PLANTING	CROP/PEST MANAGEMENT	TUBER DEVELOPMENT	HARVESTING OF STEMS & TUBERS	STORAGE OF STEMS & TUBERS	POST-HARVEST MANAGEMENT
WHITE LEAF SPOT												
Cercospora caribaea		 Leaves	Mild disease damage on the affected plants.	Plant at wide spacing for reduced density to allow wind to dry the leaves as quickly as possible.			x					
				Practice crop rotations with non-host plants every 3-5 years.			x					
				Plant in the wet season such that plants reach the susceptible stage (6-8-months-old) during the dry season.			x					
				Avoid establishing new crops next to old, infected fields.	x							
				After harvest plough the crop debris into soil or remove and burn it.	x							
				Prune disease infected leaves and branches from the plant and either bury or burn them.				x				
				Use cassava cuttings selected from healthy plants only while still in the field.		x						
				Remove weeds in and around cassava crops to avoid disease spreading into the field.				x				
				Plant cassava resistant varieties if available.		x						

FUNGAL DISEASES				STAGE OF CULTIVATION							
CAUSAL PATHOGEN/S	PLANT GROWTH STAGE/ ORGAN DAMAGED	DAMAGE/ IMPACT	CONTROL OPTIONS (IPM)	FIELD SELECTION & PREPARATION	SELECTION OF PLANTING MATERIAL	PLANTING	CROP/PEST MANAGEMENT	TUBER DEVELOPMENT	HARVESTING OF STEMS & TUBERS	STORAGE OF STEMS & TUBERS	POST-HARVEST MANAGEMENT
CASSAVA ANTHRACNOSE DISEASE											
<i>Colletotrichum gloeosporioides</i> <i>f. sp. Manihotis/</i> <i>Glomerella cingulata</i>	 leaves and stems	- Severe damage (cankers) on cassava stems and dieback, and loss of leaves. - Reduces the quality of stem cuttings. - Reduced number and size of leaves and tubers.	Avoid planting cassava cuttings with cankers/cuts since the disease affects through wounds.	x							
			Rogue or remove cassava plants infected with anthracnose disease.				x				
			Crop debris should be removed and destroyed after harvest.				x				
			Avoid growing cassava on the same field for more than 3 seasons. It is advisable that you rotate cassava with grains.			x	x				
			Late planting like in the second rainy season can reduce disease infections.			x					
			Treat cutting for planting materials with mancozeb and copper.		x						
			Use of resistant varieties.		x						

FUNGAL DISEASES												
CAUSAL PATHOGEN/S	PLANT GROWTH STAGE/ ORGAN DAMAGED	DAMAGE/ IMPACT	CONTROL OPTIONS (IPM)	STAGE OF CULTIVATION								
				FIELD SELECTION & PREPARATION	SELECTION OF PLANTING MATERIAL	PLANTING	CROP/PEST MANAGEMENT	TUBER DEVELOPMENT	HARVESTING OF STEMS & TUBERS	STORAGE OF STEMS & TUBERS	POST-HARVEST MANAGEMENT	
CONCENTRIC RING LEAF SPOT												
<i>Phyllosticta manihotae</i> , <i>Phyllosticta manihoticola</i>		• Leaf defoliation, dieback, and death of affected plants. • High yield loss in severe cases	Planting healthy cassava cuttings.		x							
	Leaves		Use appropriate sowing density.			x						
			In susceptible areas, plant at the end of the rainy season.			x						
			Azoxystrobin can be used against <i>Phyllosticta</i> sp.				x					
SUPER ELONGATION												
<i>Sphaceloma manihoticola</i> , <i>Elsinoë brasiliensis</i>		• Necrosis of cassava leaves, defoliation, thinning and development of weak stems and dieback. • Yield losses of 80% in young crops.	Use resistant varieties.		x							
	Plant healthy cassava cuttings.			x	x							
	Treat by immersion cassava cuttings for 5 minutes in copper-oxychloride solution at 3 to 5 g/L of centration, or spray the copper-oxychloride at rates of 1 kg/ha on cassava plants.			x	x							
	Leaves, petioles, shoots and stems		Practice crop rotation by inter changing cassava with grasses.			x	x					
			Plant cassava crops at the end of the rainy season.			x	x					

FUNGAL DISEASES												
CAUSAL PATHOGEN/S	PLANT GROWTH STAGE/ ORGAN DAMAGED	DAMAGE/ IMPACT	CONTROL OPTIONS (IPM)	STAGE OF CULTIVATION								
				FIELD SELECTION & PREPARATION	SELECTION OF PLANTING MATERIAL	PLANTING	CROP/PEST MANAGEMENT	TUBER DEVELOPMENT	HARVESTING OF STEMS & TUBERS	STORAGE OF STEMS & TUBERS	POST-HARVEST MANAGEMENT	
CASSAVA ROOT ROT DISEASES												
SOFT ROOT ROT/PHYTOPHTHORA ROOT ROT												
<i>Phytophthora cryptogea</i> , <i>Phytophthora drechsleri</i> , <i>Phytophthora erythroseptica</i> , <i>Phytophthora nicotianae</i> var. <i>parasitica</i>) & <i>Pythium</i> root rot (Pythium spp., <i>Fusarium</i> spp.)	 Roots and tubers	- Dieback and swelling of tubers. - Discoloration and rotting with a foul odour - Death of plant due to progressive decaying.	Select cassava cuttings from healthy cassava plants.		x							
			Use hot water to treat the cassava cuttings at maximum temp of 49 °C for 49 minutes.		x							
			Plant cassava in loose textured and well-draining soils using ridges where possible.			x						
			Eradicate diseased plants with clear symptoms and destroy them.				x					
			Remove and destroy all crop debris by burning.				x					
			Sanitize all tools after use.		x							
			Practice crop rotation with grasses whenever the disease has affected 3% of the plantation area.			x						
			Keep soils drained for at least 6 months.	x								
Dip cassava cuttings in metalaxyl.				x								

FUNGAL DISEASES											
CAUSAL PATHOGEN/S	PLANT GROWTH STAGE/ ORGAN DAMAGED	DAMAGE/ IMPACT	CONTROL OPTIONS (IPM)	STAGE OF CULTIVATION							
				FIELD SELECTION & PREPARATION	SELECTION OF PLANTING MATERIAL	PLANTING	CROP/PEST MANAGEMENT	TUBER DEVELOPMENT	HARVESTING OF STEMS & TUBERS	STORAGE OF STEMS & TUBERS	POST-HARVEST MANAGEMENT
DRY ROOT ROT											
<i>Rigidoporus lignosus</i> , <i>Rosellinia necatrix</i> , <i>Lasidioplodia theobromae</i> , <i>Armillariella mellea</i> , <i>Sphaerotilbe repens</i>		Deterioration of roots and tubers.	Select cassava cuttings from healthy cassava plants.		x						
			Use hot water to treat the cassava cuttings at maximum temp of 49 °C for 49 minutes.		x						
			Plant cassava in loose textured and well-draining soils using ridges where possible.			x					
			Eradicate diseased plants with clear symptoms and destroy them.				x				
			Remove and destroy all crop debris by burning.				x				
			Sanitize all tools after use.		x						
			Practice crop rotation with grasses whenever the disease has affected 3% of the plantation area.			x					
			Keep soils drained for at least 6 months.	x							
			Dip cassava cuttings in metalaxyl.	x							

FUNGAL DISEASES				STAGE OF CULTIVATION							
CAUSAL PATHOGEN/S	PLANT GROWTH STAGE/ ORGAN DAMAGED	DAMAGE/ IMPACT	CONTROL OPTIONS (IPM)	FIELD SELECTION & PREPARATION	SELECTION OF PLANTING MATERIAL	PLANTING	CROP/PEST MANAGEMENT	TUBER DEVELOPMENT	HARVESTING OF STEMS & TUBERS	STORAGE OF STEMS & TUBERS	POST-HARVEST MANAGEMENT
DEMATOPHORA ROOT ROT											
<i>Rosellinia</i> root rot, <i>Dematophora</i> necatrix <i>Rosellinia</i> necatrix		Yield loss of 5% to 100%, especially in the susceptible varieties.	Select cassava cuttings from healthy cassava plants.		x						
			Use hot water to treat the cassava cuttings at maximum temp of 49 °C for 49 minutes.		x						
			Plant cassava in loose textured and well-draining soils using ridges where possible.			x					
			Eradicate diseased plants with clear symptoms and destroy them.				x				
			Remove and destroy all crop debris by burning.				x				
			Sanitize all tools after use.		x						
			Practice crop rotation with grasses whenever the disease has affected 3% of the plantation area.			x					
			Keep soils drained for at least 6 months.	x							
			Dip cassava cuttings in metalaxyl.	x							

FUNGAL DISEASES												
CAUSAL PATHOGEN/S	PLANT GROWTH STAGE/ ORGAN DAMAGED	DAMAGE/ IMPACT	CONTROL OPTIONS (IPM)	STAGE OF CULTIVATION								
				FIELD SELECTION & PREPARATION	SELECTION OF PLANTING MATERIAL	PLANTING	CROP/PEST MANAGEMENT	TUBER DEVELOPMENT	HARVESTING OF STEMS & TUBERS	STORAGE OF STEMS & TUBERS	POST-HARVEST MANAGEMENT	
DIPLODIA ROOT AND STEM ROT												
Diplodia manihoti	[Photo not available]	yellowing, stippling, and wilting, and ultimately leaf drop. Reduced production of tubers.	Select cassava cuttings from healthy cassava plants.		x							
			Use hot water to treat the cassava cuttings at maximum temp of 49 °C for 49 minutes.		x							
			Plant cassava in loose textured and well-draining soils using ridges where possible.			x						
			Eradicate diseased plants with clear symptoms and destroy them.				x					
			Remove and destroy all crop debris by burning.				x					
			Sanitize all tools after use.		x							
			Practice crop rotation with grasses whenever the disease has affected 3% of the plantation area.			x						
			Keep soils drained for at least 6 months.	x								
		Dip cassava cuttings in metalaxyl.		x								

FUNGAL DISEASES											
CAUSAL PATHOGEN/S	PLANT GROWTH STAGE/ ORGAN DAMAGED	DAMAGE/ IMPACT	CONTROL OPTIONS (IPM)	STAGE OF CULTIVATION							
				FIELD SELECTION & PREPARATION							
				SELECTION OF PLANTING MATERIAL							
				PLANTING							
				CROP/PEST MANAGEMENT							
				TUBER DEVELOPMENT							
				HARVESTING OF STEMS & TUBERS							
				STORAGE OF STEMS & TUBERS							
				POST-HARVEST MANAGEMENT							
BLACK ROOT AND STEM ROT											
Scytalidium sp., Hendersonula toruloidea		deterioration of roots and stems.	Select cassava cuttings from healthy cassava plants.								
			Use hot water to treat the cassava cuttings at maximum temp of 49 °C for 49 minutes.								
			Plant cassava in loose textured and well-draining soils using ridges where possible.								
			Eradicate diseased plants with clear symptoms and destroy them.						x		
			Remove and destroy all crop debris by burning.							x	
			Sanitize all tools after use.								
			Practice crop rotation with grasses whenever the disease has affected 3% of the plantation area.								
			Keep soils drained for at least 6 months.								
			Dip cassava cuttings in metalaxyl.								

FUNGAL DISEASES												
CAUSAL PATHOGEN/S	PLANT GROWTH STAGE/ ORGAN DAMAGED	DAMAGE/ IMPACT	CONTROL OPTIONS (IPM)	STAGE OF CULTIVATION								
				FIELD SELECTION & PREPARATION	SELECTION OF PLANTING MATERIAL	PLANTING	CROP/PEST MANAGEMENT	TUBER DEVELOPMENT	HARVESTING OF STEMS & TUBERS	STORAGE OF STEMS & TUBERS	POST-HARVEST MANAGEMENT	
CASSAVA TUBER ROT DISEASE												
SCLEROTIUM/TUBER ROT (SOUTHERN BLIGHT)												
	Roots and tubers.	• Root rot	Rotate cassava with paddy rice or maize.			x						
			Manipulate soil fertility such availing sufficient nitrogen and phosphorus.				x					
			Soil fumigation or solarization.	x								
RUST												
	Leaves and stem.	• Die back	Rotate cassava with paddy rice or maize.			x						
			Manipulate soil fertility such availing sufficient nitrogen and phosphorus.				x					
			Soil fumigation or solarization.	x								
VERTICILLIUM ROOT AND STEM ROT												
	Stem/plant base	• Wilting, defoliation, stunting and death of affected plants. • Significant yield losses in susceptible varieties.	Rotate cassava with paddy rice or maize.			x						
			Manipulate soil fertility such availing sufficient nitrogen and phosphorus.				x					
			Soil fumigation or solarization.	x								

FUNGAL DISEASES													
CAUSAL PATHOGEN/S	PLANT GROWTH STAGE/ ORGAN DAMAGED	DAMAGE/ IMPACT	CONTROL OPTIONS (IPM)	STAGE OF CULTIVATION									
				FIELD SELECTION & PREPARATION	SELECTION OF PLANTING MATERIAL	PLANTING	CROP/PEST MANAGEMENT	TUBER DEVELOPMENT	HARVESTING OF STEMS & TUBERS	STORAGE OF STEMS & TUBERS	POST-HARVEST MANAGEMENT		
CASSAVA ASH													
	Leaves	- Mature leaves develop white mycelium (ash-like) appearance on surface. - Afterwards the leaves turn chlorotic with yellowish lesions.	Rotate cassava with paddy rice or maize. Manipulate soil fertility such as availing sufficient nitrogen and phosphorus. Soil fumigation or solarization.			x							
							x						

Notes: 'x' = integrated pest management (IPM) interventions are indicated for the most appropriate cultivation and growth stage

VIRAL DISEASES												
CAUSAL PATHOGEN/S	PLANT GROWTH STAGE/ ORGAN DAMAGED	DAMAGE/ IMPACT	CONTROL OPTIONS (IPM)	STAGE OF CULTIVATION								
				FIELD SELECTION & PREPARATION	SELECTION OF PLANTING MATERIAL	PLANTING	CROP/PEST MANAGEMENT	TUBER DEVELOPMENT	HARVESTING OF STEMS & TUBERS	STORAGE OF STEMS & TUBERS	POST-HARVEST MANAGEMENT	
CASSAVA MOSAIC DISEASE												
African cassava mosaic virus (ACMV), East African cassava mosaic virus (EACMV) & South African cassava mosaic virus (SACMV)	 Leaves, stems and tubers	• Reduced number, size and quality of leaves, stems and tubers. • Root/tuber constriction. • Yield loss of up to 100% in the severely affected plants.	Isolate new cassava crops from older fields to reduce whitefly entry.	x								
			Select disease-free planting materials using visual judgement or virus screening assays.		x							
			Use elite cassava varieties with proven resistance or tolerance to whitefly.		x							
			Grow thin-leaved cassava varieties for lower whitefly egg and nymphal instar densities.		x							
			Dip cassava stem cuttings in Deltamethrin (2.5%) or Cypermethrin (150g/L) to kill whitefly eggs, nymphs and adults before distribution to farmers.		x							
			Hot water therapy (25°C -60°C) and two rounds of meristem tip culture (MTC) of virus-infected cassava planting materials reduced by 100% the causal viral loads.		x							
Intercrop cassava at high density with cow peas or maize to reduce whitefly populations build up.						x						
Establish repellent crops such as marigold near or around cassava fields to repel whiteflies.							x					
Remove and burn crop debris and weeds (potential hosts) with whiteflies (eggs, nymphs and adults) and visible viral disease symptoms within and around young fields.							x					
Rogue/remove cassava plants infected with viral diseases in young crops (<3 months old), followed by replanting healthy planting materials.							x					
Spray organic neem or jojoba oil (a.i., azadirachtin) on top and underneath of cassava leaves until completely wet to kill eggs, nymphs and/or repel adults of whitefly vectors.							x					
Select disease-free planting materials using visual judgement or virus screening assays.					x							
Use elite cassava varieties with proven resistance or tolerance to whitefly.					x							

VIRAL DISEASES				STAGE OF CULTIVATION							
CAUSAL PATHOGEN/S	PLANT GROWTH STAGE/ ORGAN DAMAGED	DAMAGE/ IMPACT	CONTROL OPTIONS (IPM)	FIELD SELECTION & PREPARATION	SELECTION OF PLANTING MATERIAL	PLANTING	CROP/PEST MANAGEMENT	TUBER DEVELOPMENT	HARVESTING OF STEMS & TUBERS	STORAGE OF STEMS & TUBERS	POST-HARVEST MANAGEMENT
CASSAVA BROWN STREAK DISEASE											
Cassava brown streak virus (CBSV) & Uganda cassava brown streak virus (UCBSV).		- Reduced number, size and quality of leaves, stems and tubers. - Affected tubers develop necrosis, i.e., brown colouration which renders it unfit for human consumption. - Yield loss of 25% to 100% depending on the susceptibility of cassava varieties.	Isolate new cassava crops from older fields to reduce whitefly entry.	x							
			Select disease-free planting materials using visual judgement or virus screening assays.		x						
			Use elite cassava varieties with proven resistance or tolerance to whitefly.		x						
			Grow thin-leaved cassava varieties for lower whitefly egg and nymphal instar densities.		x						
			Dip cassava stem cuttings in Deltamethrin (2.5%) or Cypermethrin (150g/L) to kill whitefly eggs, nymphs and adults before distribution to farmers.		x						
			Hot water therapy (25°C-60°C) and two rounds of meristem tip culture (MTC) of virus-infected cassava planting materials reduced by 100% the causal viral loads.		x						
			Intercrop cassava at high density with cow peas or maize to reduce whitefly populations build up.			x					
			Establish repellent crops such as marigold near or around cassava fields to repel whiteflies.				x				
			Remove and burn crop debris and weeds (potential hosts) with whiteflies (eggs, nymphs and adults) and visible viral disease symptoms within and around young fields.				x				
			Rogue/remove cassava plants infected with viral diseases in young crops (<3 months old), followed by replanting healthy planting materials.				x				
			Spray organic neem or jojoba oil (a.i., azadirachtin) on top and underneath of cassava leaves until completely wet to kill eggs, nymphs and/or repel adults of whitefly vectors.				x				
			Select disease-free planting materials using visual judgement or virus screening assays.		x						
			Use elite cassava varieties with proven resistance or tolerance to whitefly.		x						

VIRAL DISEASES											
CAUSAL PATHOGEN/S	PLANT GROWTH STAGE/ ORGAN DAMAGED	DAMAGE/IMPACT	CONTROL OPTIONS (IPM)	STAGE OF CULTIVATION							
				FIELD SELECTION & PREPARATION	SELECTION OF PLANTING MATERIAL	PLANTING	CROP/PEST MANAGEMENT	TUBER DEVELOPMENT	HARVESTING OF STEMS & TUBERS	STORAGE OF STEMS & TUBERS	POST-HARVEST MANAGEMENT
CASSAVA COMMON MOSAIC DISEASE											
Cassava common mosaic virus (CCMV), Brazilian cassava common mosaic virus or Cassava common mosaic potexvirus	 Leaves, stems and tubers	- Reduced number and size of leaves, stems and tubers. - Severely affected cassava plants are stunted.	Rogue out the diseased plants and debris from the field and burn them.				x				
			Disinfect farm tools such as cutting materials used to harvest cassava stems.			x					
CASSAVA LEAF VEIN MOSAIC DISEASE											
Cassava vein mosaic virus (CsVMV/ CVMV), Cassava vein mosaic caulimovirus	 Leaves, stems and tubers	- Reduced number and size of leaves, stems and tubers. - Higher losses occur when infection comes early in the cropping season.	Remove all infected plant debris with the symptoms from the field and burn them.	x	x	x					x
OTHER VIRAL DISEASES/VIRUSES											
Cassava virus X (Potexvirus) (CsXV) Cassava Colombian symptomless virus (Potexvirus) (CCSpV) Cassava symptomless infections (Cassava American latent virus - CsAIV, Cassava Ivorian bacilliform virus - CIBV) Cassava frogskin disease (Cassava frogskin Associated Virus - CsFSaV)	Leaves, stems and tubers	- Reduced number and size of leaves, stems and tubers - Roots are thin and woody with a thick, cork-like peel with fibres. - Economic losses occur due to lack of starch accumulation in the roots, leading to low yields or 90% yield loss.	Remove all infected plant debris with the symptoms from the field and burn them.	x			x				

Notes: 'x' = integrated pest management (IPM) interventions are indicated for the most appropriate cultivation and growth stage

PHYTOPLASMA DISEASES											
CAUSAL PATHOGEN/S	PLANT GROWTH STAGE/ ORGAN DAMAGED	DAMAGE/IMPACT	CONTROL OPTIONS (IPM)	STAGE OF CULTIVATION							
				FIELD SELECTION & PREPARATION	SELECTION OF PLANTING MATERIAL	PLANTING	CROP/PEST MANAGEMENT	TUBER DEVELOPMENT	HARVESTING OF STEMS & TUBERS	STORAGE OF STEMS & TUBERS	POST-HARVEST MANAGEMENT
CASSAVA ANTHOLYSIS											
Phytoplasma	 Leaves	- Flower fertility is lost, resulting in non-functional flowers that abort prematurely. - The disease does not have significant economic importance.	Select planting materials from healthy plants.	x							
CASSAVA WITCHES' BROOM											
Candidatus Phytoplasma aurantifolia	 Leaves	- Stunted (or dwarfed) plants excessive distortion of branches. - Shoots have small leaves and short internodes. - Affected plants have weak shoots, swollen lower stems and thin, fibrous tubers that have rough outer skins with deep cracks and low starch content. - Cuttings from the CWB-affected shoots are weak and only a few shoots successfully grow. - Can cause yield loss of 30 to 80 percent, with reduced quality.	Plant disease-free cassava planting materials. Remove and destroy disease-affected cassava plants. Disinfect farm tools and equipment between harvesting the cassava cuttings. Look out for and restrict movement of cassava relatives such as jatropha which could be infected with cassava witches' broom. Do not establish new cassava fields next to those infected with the disease. Remove weeds and shrubs within and in immediate surrounding of cassava fields that could act as alternate hosts and reservoirs for the disease. Use resistant or tolerant to CWB disease. Control leaf hoppers in the field that might be transmitting the disease.	x	x				x		x
	 Shoot			x			x				

Notes: 'x' = integrated pest management (IPM) interventions are indicated for the most appropriate cultivation and growth stage

POST-HARVEST PESTS OF DRIED CASSAVA/CHIPS													
MOST DAMAGING GROWTH STAGE	PLANT GROWTH STAGE/ ORGAN DAMAGED	DAMAGE/IMPACT	CONTROL OPTIONS (IPM)	STAGE OF CULTIVATION									
				FIELD SELECTION & PREPARATION	SELECTION OF PLANTING MATERIAL	PLANTING	CROP/PEST MANAGEMENT	TUBER DEVELOPMENT	HARVESTING OF STEMS & TUBERS	STORAGE OF STEMS & TUBERS	POST-HARVEST MANAGEMENT		
RED FLOUR BEETLE (<i>TRIBOLIUM CASTANEUM</i>)													
		Beetles (adults and larvae) make tunnels in dried tubers and excrete in the product which destroys food quality.	Check cassava chips for presence or absence of pest/s.							x	x	x	
			Check warehouses or storage areas before building up stocks.								x	x	x
			Clean the store at each harvest to remove and burn infested produce and crop residues.										x
			Treat tubers or sacs before placing in storage.										
			Freezing cassava chips for several days and heating (above 34°C) for 24 hours can control the pest in stored food products.									x	x
larva and adult	Tubers (dried tubers and chips)		Use of low oxygen and carbon dioxide-enriched atmospheres can also control the pest in stored dry cassava.								x	x	
			Add ash and clay to the cassava chips to reduce the population.								x	x	
			Powdered leaves of <i>Salvia officinalis</i> L. and <i>Artemisia absinthium</i> L. can reduce or delay insect development.								x	x	

POST-HARVEST PESTS OF DRIED CASSAVA/CHIPS											
MOST DAMAGING GROWTH STAGE	PLANT GROWTH STAGE/ ORGAN DAMAGED	DAMAGE/IMPACT	CONTROL OPTIONS (IPM)	STAGE OF CULTIVATION							
				FIELD SELECTION & PREPARATION	SELECTION OF PLANTING MATERIAL	PLANTING	CROP/PEST MANAGEMENT	TUBER DEVELOPMENT	HARVESTING OF STEMS & TUBERS	STORAGE OF STEMS & TUBERS	
				POST-HARVEST MANAGEMENT							
LESSER GRAIN BORER (<i>RHIZOPERTHA DOMINICA</i>)											
		Damage dried produce by making tunnels, which losses weight and quality.	Check cassava chips for presence or absence of pest/s. Check warehouses or storage areas before building up stocks. Clean the store at each harvest to remove and burn infested produce and crop residues. Treat tubers or sacs before placing in storage. Freezing cassava chips for several days and heating (above 34°C) for 24 hours can control the pest in stored food products. Use of low oxygen and carbon dioxide-enriched atmospheres can also control the pest in stored dry cassava. Add ash and clay to the cassava chips to reduce the population. Powdered leaves of <i>Salvia officinalis</i> L. and <i>Artemisia absinthium</i> L. can reduce or delay insect development.						x	x	
									x	x	x

Notes: 'x' = integrated pest management (IPM) interventions are indicated for the most appropriate cultivation and growth stage

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The COLEAD Crop Protection Database allows you to search by crop and active substance to obtain information on the Good Agricultural Practices (GAPs) that ensure compliance with current European Union (EU) and Codex Alimentarius maximum residue limits (MRLs).

https://resources.colead.link/en/vue-substance-active-culture?field_culture_tid=All&field_substance_active_tid=329







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