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Quality declared planting material

Protocols and standards for vegetatively propagated crops

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Cover photo: Cassava stakes with adequate length and width and 5–7 nodes. Ceballos, CIAT. 2006.

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FAO PLANT PRODUCTION AND PROTECTION PAPER 195

Quality declared planting material

Protocols and standards for vegetatively propagated crops

Expert consultation
Lima, 27-29 November 2007

Coordinated by
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Preface

Ensuring that farmers have timely access to seed and planting material of good quality is one of the most important elements of successful agricultural production and development. This issue was presented as a major component of the *High-Level Conference on World Food Security: The Challenges of Climate Change and Bioenergy* held in June 2008 at FAO Headquarters in Rome. Conference participants recognized that increased access by farmers to appropriate locally adapted seeds is a key element in support of agricultural production in the context of high food prices and climate change.

Despite this reality, seed and planting material available to small-scale farmers in many developing countries is often of insufficient quality, which undermines potential yield and performance of crop production. With the support of FAO, the challenge of raising the quality of the seed produced locally and used by small-scale farmers has been addressed in a range of programmes and initiatives in many countries.

In 1993, the FAO Plant Production and Protection Division (AGP) further contributed to these efforts by initiating an expert consultation that produced technical guidelines on standards and procedures for quality seeds – known as the quality declared seed (QDS) system. QDS, as a quality assurance scheme for seed production, is less demanding than full quality control systems and, thus, can be more easily implemented in situations where resources are limited. It is now used and consulted worldwide and has proven particularly useful as a source of practical information on seed standards for a range of crop species propagated by true botanical seed. A revision of the QDS system aimed at expanding the crop coverage and better adapting to changing circumstances and needs was published by FAO in 2006.

However, crop species propagated by diverse vegetative structures such as setts, stem cuttings, tubers, suckers, corms and others have not been included in QDS, even though some of these species are of major importance for agricultural production and food security. With the exception of potato and *Musa* species, vegetatively propagated crops have received very little attention in formal seed quality regulatory systems. Many of these crops, such as yam, cassava and sweetpotato, belong to tropical or subtropical agricultural systems and are staple foods in many developing countries. Although some are considered minor crops at the global level, they contribute significantly to the food security of rural populations in specific countries and regions. Now, there are expanded possibilities for improving and developing vegetatively reproduced crops, thanks to the availability and dissemination of advanced technologies, in particular plant

micropropagation and production of disease-free materials. In addition, as the whole world could see, the 2008 International Year of the Potato (IYP) provided a timely opportunity to advance the development of global instruments for improved potato production.

For all these reasons, FAO, in collaboration with the International Potato Centre and a team of international experts, has developed and prepared a set of protocols and standards for the production of quality planting material of the most important vegetatively reproduced crops. Presented in this publication, they offer a practical and useful tool for seed producers and technicians at the community level and also for national seed services and the agricultural research community. We believe that a better quality of the materials used for planting will contribute significantly to improved agricultural production and productivity, and therefore to food security in many parts of the world. This publication will be of particular value to developing countries in tropical and subtropical zones where vegetatively propagated crops have the potential to make a real contribution to fighting hunger and poverty, driving economic development and sustaining rural livelihoods.

Shivaji Pandey
Director
Plant Production and Protection Division

List of acronyms and abbreviations

AbaMV	Abacá mosaic virus
APLV	Andean potato latent virus
AVA	Arracacha virus A
AVB-O	Arracacha virus B
BanMMV	Banana mild mosaic virus
BBrMV	Banana bract mosaic virus
BBTV	Banana bunchy top virus
BSV	Banana streak viruses
CBB	Cassava bacterial blight
CBDV	Colocasia bobone disease virus
CBSD	Cassava Brown Streak Diseas
CFU	Colony forming unit
CIAT	International Centre for Tropical Agriculture
CIP	International Potato Center
CIRAD	*Centre de coopération internationale en recherche agronomique pour le développement*
CMD	Cassava mosaic disease
CMV	Cucumber mosaic virus
DsMV	Dasheen mosaic virus
ELISA	Enzyme-linked immunosorbent assay
EPRV	Endogenous pararetrovirus
FYM	Farmyard manure
FSD	Frog-skin disease
GYSV	Garlic yellow streak virus
IDM	Integrated disease management
IITA	International Institute of Tropical Agriculture
K	Potassium
KoMV	Konjac mosaic virus
LYSV	Leek yellow stripe virus
masl	Metres above sea level
MS	Murashige and Skoog salt mixture
N	Nitrogren

NARS	National agricultural research system
NASH	Nucleic acid spot hybridization
NPK	Nitrogen-phosphorus-potassium
OYDV	Onion yellow dwarf virus
PapMV-U	Papaya mosaic virus
PBRSV	Potato black ring spot virus
PGRRI	Plant Genetic Resources Research Institute of Ghana
PIBS	*Plants Issus de Bourgeons Secondaires*
PLRV	Potato leaf roll virus
PVT	Potato virus T
QDPM	Quality declared planting material
QDS	Quality declared seed
RKN	Root knot nematodes
RRD	Root rot disease
SED	Super-elongation disease
SoMv	Sowbane mosaic virus
SPCFV	Sweetpotato chlorotic flecks virus
SPCSV	Sweetpotato chlorotic stunt virus
SPFMV	Sweetpotato feathery mottle virus
SPLCV	Sweetpotato leaf curl virus
SPLV	Sweetpotato latent virus
SPMMV	Sweetpotato mild mottle virus
SPV	Severe Sweetpotato virus disease
SPVG	Sweetpotato virus G
SSR	Simple sequence repeat markets
TaBV	Taro bacilliform badnavirus
TaRV	Taro reovirus
TaVCV	Taro vein chlorosis virus
TropMV	Tropaeolum mosaic virus
TLB	Taro leaf blight
USA	United States of America
UMMV	Ulluco mild mosaic virus
UMV	Ulluco mosaic virus
UVC	Ulluco virus C

Introduction

FAO has always recognized the potential contribution of traditional, local crops in achieving global food security. However, in the context of soaring food prices, the focus became even more acute on the need to make use of crops other than the major commercial crops. In turn, this raised awareness that most of these traditional crops had little or no recognized science-based systems for production of their planting materials.

In 1993, FAO published its first *Quality Declared Seed System (QDS) Manual – Technical guidelines on standards and procedures* based on the results of an expert meeting. It was updated in 2003. In all, new information was gathered and made available with regards to better field and seed protocols and standards for the quality seed production of 92 crop species reproduced by means of true seeds.

However, the 2003 meeting participants also recognized the dearth of knowledge and procedures for quality standards of non-commercial planting materials, especially for locally important traditional crops. Therefore, it recommended, among other things that FAO should hold an expert consultation to consider a quality assurance scheme for vegetatively reproduced crops. In addition, the Fourteenth Triennial Symposium of the International Society for Tropical Root Crops, held November 2006 in Thiruvananthapuram, India, also identified the "production of quality planting materials to overcome the degeneration due to diseases and pathogen accumulation" as a key emergent theme.

As a result, FAO's Plant Production and Protection Division (AGP) embarked on the preparation of protocols and standards for the world's most important vegetatively reproduced food crops. FAO proposed to collaborate with the International Potato Centre (CIP) in order to debate the main points and discuss draft proposals prepared by selected experts working in related fields all over the world.

FAO and CIP organized an Expert Consultation at CIP headquarters in Lima, Peru, from 27 to 29 November 2007 with the participation of 12 international experts and several national experts. This activity took cognizance of the great contribution of the potato to food security, recognizing its importance as a vegetatively propagated crop. Therefore, the expert consultation added potato crop to the list, in spite of the fact that its propagation method had been perfected by the private sector, as a contribution of CIP to the International Year of the Potato 2008.

The meeting discussed and approved experts' papers on the protocols for safe multiplication of disease-free materials of vegetatively reproduced crops of several species of global importance. The goal was to establish protocols that would improve the quality and availability of planting materials, particularly for the small farmer. The crops described varied greatly in their reproductive means and therefore on the types of processes needed to obtain quality materials. However, in general terms, the technical papers focused on quality and sanitary aspects of the production of planting materials.

The participants agreed upon the common principles and structure of the protocols as well as standards for quality developed planting material (QDPM). The working groups revised the technical protocols which are presented in this publication as the main output of the Expert Consultation. The Expert Consultation also recommended:

- capacity building for the implementation of QDPM in the field and promotion of its use in community-based seed multiplication activities;
- developing a monitoring system to observe the operation of QDPM in the field and integrate results and lessons learned;
- promoting governmental responsibility in the implementation of QDPM and in promoting the use of clean planting materials.

Description of quality declared planting material

PLANTING MATERIAL OF VEGETATIVELY PROPAGATED CROPS

Vegetatively propagated crops have a fundamental role in improving food security and human nutrition. They also are used in production of starch and biofuels and have many other uses within the agro-industrial chain. In addition, they can substitute for other types of crops in cases of economic need, such as in the case of high food prices.

Reproducing the planting materials of these crops presents complex problems and many logistical issues for their extensive use. This is particularly an issue for smallholder farmers because of:

- absence of formal seed systems (except potato);
- lack of knowledge of phytosanitary measures and quarantine issues related to safe movement of germplasm, plants and planting material across national borders;
- lack of consistent supplies of good quality planting material;
- variable demand for clean planting material;
- bulkiness and perishability of planting materials;
- use of traditional varietal mixtures, including local varieties.

OBJECTIVE AND PRINCIPLES OF QDPM

The QDPM process has been developed to guide the production of clean, disease-free planting material of vegetatively reproduced crops. Its overall goal is to raise the physiological and phytosanitary quality of the plant reproductive materials available to smallholders, and as a consequence, to increase agricultural production and productivity. It is meant to be implemented primarily by seed producers at community level or field extension workers.

In order to have quick and easy access to these materials, these practical QDPM protocols and standards have been designed to allow easy monitoring and verification of the production and distribution process. At the same time, they complement and are amenable to formal seed quality control systems, but also are cognizant of national and local conditions to ensure they are appropriate and achievable for target users. They also link activities initiated by experimental

stations and researchers of national and international institutions to seed multiplication activities of smallholders.

The source materials can come from *in vitro* cultivation or several other means used to obtain disease-free planting material but in all cases, QDPM must conform to national and international arrangements on phytosanitary issues.

QDPM sees very clear roles for the public and the private sectors, with the public sector responsible for maintaining germplasm, introducing and breeding new materials, clearing materials and handling virus indexing while the private sector is responsible for mass multiplication and distribution.

COMPATIBILITY WITH NATIONAL SEED REGULATIONS

Any QDPM scheme for plant multiplication must comply with prevailing national seed regulations. Thus, persons wishing to start such a scheme should first check its compatibility with local regulations. Most national seed regulations state that only registered seed varieties can be multiplied and may include a requirement to register producers. For this reason, QDPM-derived seed cannot be labelled as "certified" seed.

QDPM is a particular case of QDS. The reproductive means and, therefore, the production practices of planting materials are substantially different from those applied to the crops reproduced by means of true seeds.

LABELLING

Correct labelling is an important element in the responsible and successful distribution of the planting material to be distributed. The information that should be provided on labels varies according to crops but, in general, all should contain the following (using the example of yams):

- batch number
- weight of the batch
- number of seed yams or setts in batch
- length of seed tubers
- variety name or code
- name of seed yam grower
- farm location where seed tuber was produced
- date seed tuber was harvested
- name of inspector or code
- quality standard brand/logo, as appropriate.

The local authority, whether government or a seed grower cooperative, would specify the penalty for any false labelling, assuming there is approval of this principle in local culture. Records of off-types should be kept as proof of error. The following identifies some specific aspects of labelling.

Xanthosoma

For Xanthosoma, labelling is important to avoid mixing materials with different specifications. The stocks should be labeled as follows:

- variety
- origin
- category
- weight (corms or cormels)
- height (*in vitro* plant or plantlets)
- harvest date

Colocasia

- Labelling is essential for *Colocasia* and should include:
- batch number
- weight and/or number per batch
- variety name and/or code
- seed grower's name and location
- date harvested
- inspector code
- quality standard brand/logo, as appropriate.

Sweetpotato

Each container of seed tubers or vines should be appropriately tagged to identify the phase of production. If the container or bundle of vines is not tagged, the planting material cannot be distributed as obtained under QDS scheme. Label information includes:

- variety name/code of planting material
- grower's name and farm location
- harvest date
- batch number and weight, length and number of seed tubers or vines per batch
- inspector name/code
- quality standard brand/logo, as appropriate
- All labelled seed tubers or vines should follow quality standards.

Banana suckers

For suckers commercialized locally, labelling is infrequent. Nevertheless, the following information should be requested by persons acquiring suckers:

- location of field, name of grower, contact information for grower;
- cultivar, age of plantation, primary management practices (inputs used, routine practices applied) for field from which suckers were extracted;
- summary of presence of pests and diseases in plantation and frequency of inspection;
- description of practices used in extraction and treatment of suckers, including storage location after paring.

Tissue culture plants

For tissue culture plants, the following information should be requested:

- name, address and contact information of laboratory where *in vitro* plants were produced;
- name and address of the agent commercializing the plants;
- cultivar, special clone information and location of mother plants from which shoot tips were extracted, If mother plants are maintained in screen house, location of original mother plants;
- number of plantlets produced and sub-cultures used per shoot tip in routine tissue culture multiplication;
- virus indexing protocols followed and results, including for example, banana bunchy top virus (BBTV), cucumber mosaic virus (CMV), banana streak virus (BSV), banana bract mosaic virus (BBrMV) and banana mild mosaic virus (BanMMV);
- name, address and contact information for laboratory conducting virus indexing;
- other disease screening conducted, protocols and results;
- name, address and contact information for laboratory conducting other disease screening;
- name and address of official plant sanitary control organization certifying plants.

STRUCTURE

The protocols follow a common pattern as much as possible *vis-à-vis* the existing variability among these crops and according to their different characteristics.

1. Introduction
    - Scientific name, origin, distribution
    - Mode or modes of propagation commonly used by small farmers
    - Reproduction rate.

Main seed-borne diseases and pests including brief life cycle, identification, detection, natural spread, field symptoms, alternate hosts, control methods and/or any other element useful to characterize the diseases/pests.

2. Protocol for the production of planting materials by small-scale producers at field level from clean material sources*:*
   - Field facilities and equipment
   - Source of material, including positive selection
   - Field requirements
   - Field inspection
   - Agronomic practices such as isolation, rotation, and negative selection
   - Harvesting and handling
   - Post-harvest treatments
   - Storage and transport
   - Quality standards for the supplied product
3. Size and weight:
   - Table of tolerances (%) for common economically important seed-borne pests and diseases, both at field and at storage
   - Labelling requirements (list)
   - Capacity (%) to sprout and develop a normal plant (when possible)
   - Varietal purity (%).
4. Outline of multiplication programme.

Andean tubers

Carlos Arbizu
International Potato Center (CIP), Lima, Peru

Oca	***Oxalis tuberosa* Molina**	***Oxalidaceae***
Ulluco	***Ullucus tuberosus* Caldas**	***Bassellaceae***
Mashua	***Tropaeolum tuberosum* Ruiz and Pavon**	***Tropaeolaceae***

The tubers of oca, ulluco and mashua are used by about 50 million consumers from the Andes of Venezuela to northwest Argentina. They are grown at altitudes ranging from 2 800 to 4 000 masl, with more intensive cultivation from central Peru to central Bolivia. Oca is also consumed by about 2 million people in Mexico and New Zealand.

Oca, ulluco and mashua are perennials. Their upper parts senesce at the end of the growing season but their tubers persist and grow the following season. They are planted in well-drained soils, preferably with high organic matter content and are ready for harvesting after 7–8 months. When there is an excess of production, oca tubers are traditionally dehydrated into *kaya,* and ulluco tubers into *lingle*, which can be stored several years.

"Oca" is derived from the Quechua words "ok'a", "occa" and "uqa". It has been identified as a cultivated species of *Oxalis tuberosa* with 64 chromosomes. More than 80 species of its wild relatives have been identified in the Andes with ploidy levels ranging from diploid to hexaploid. The basic chromosome number of oca is x=8.

"Ulluco" is derived from the Quechua word "*ulluku".* The species *Ullucus tuberosus* comprises two subspecies including the cultivated ullucos with a basic chromosome number of x=12 and triploid wild forms of *U. tuberosus aborigineus*.

"Mashua" is derived from the Quechua word "maswa" or "mashwa". *Tropaeolum tuberosum* is a tuber-forming species of *Tropaeolum*, regarded here as an Andean tuber crop.

PEST AND PATHOGEN CONSTRAINTS OF ANDEAN TUBERS

Oca is severely damaged by the weevil *Adioristidius tuberculatus.* Cultural and biological pest management have been undertaken to reduce weevil damage in the Andes. Oca may also be infected by several pathogens: *Fusarium oxysporum*,

F. roseum, *Rhizoctonia solani*, *Mucor piriformis*, and *Rhizopus* spp (Ames, 1997). Well-drained soils and selection of sound tubers have the best results for prevention of tuber rot.

Ulluco cultivation can be affected by the weevil *Amathynetoides nitidiventris*, which damages tubers as well as other plants including carrot, broad beans and maize. Other constraints for ulluco cultivation are 15 fungal diseases, one bacterial disease (Ames, 1997) and four virus diseases. The virus diseases are described below (Chuquillanqui, *pers. com.).*

VIRUS DISEASES

Ulluco viruses

The ulluco viruses include the following (Fuentes and Chuquillanqui, 2004).

Potato leaf roll virus (PLRV), found in Peru, Bolivia, Argentina and Colombia, is transmitted by the green peach aphid *Myzus persicae* from ulluco into potato and *vice versa*. Unlike symptoms in potato plants, it is not expressed in the ulluco plants. Ulluco yield can be down to 30 percent for secondary infection.

Potato virus T (PVT), found in potatoes, mashua, and oca in Peru, Bolivia and Argentina, is mechanically transmitted through machinery and plant contact. Although the virus is transmitted by planting material, infected plants do not show symptoms.

Andean potato latent virus (APLV), found in Peru, Bolivia, Ecuador and Colombia, is mechanically transmitted by machinery and pests. One insect vector, *Epitrix* spp., has symptoms that have not been shown in ulluco plants.

Ulluco virus C (UVC), found in Peru, Bolivia, Ecuador, Argentina and Colombia, is transmitted by contact between plants, but ulluco does not show symptoms. However, UVC in combination with UMV results in severe mosaic. Secondary infection can reduce yields by up to 27 percent.

Arracacha virus A (AVA), which infects ulluco in Peru, and the starchy root crop arracacha (*Arracacia xanthorrhiza*) in Central America, is mechanically transmitted, most probably by a nematode although contact transmission between plants has not been demonstrated. Symptoms are not usually seen, although in association with other viruses, changes in leaf shape and mosaic may be observed.

The Papaya mosaic virus (PapMV-U), found in Peru, Bolivia, Ecuador, Argentina and Colombia, is usually transmitted by contact between plants. The ulluco plants remain symptomless, but in combination with UMV, the plants show mosaic. Secondary infection can reduce ulluco yield by about 30 percent. The disease can also infect oca, mashua and papaya plants.

Ulluco mosaic virus (UMV) is transmitted by the green peach aphid *Myzus persicae*. Infected plants show mild mosaic to chlorotic speckling and changes in leaf shape. Yield can be reduced by up to about 30 percent. The disease has been found in Peru, Bolivia, Ecuador, Argentina, and Colombia.

Ulluco mild mosaic virus (UMMV) is mechanically transmitted by contact between plants or during field operations. Infected plants show speckled symptoms. It has been found in Peru, Bolivia, Ecuador, Argentina, and Colombia.

Oca viruses

Four viruses have been identified in oca crops: potato black ring spot virus (PBRSV), arracacha virus B (AVB-O), PapMV-U and PVT (Chuquillanqui, *pers. comm*). The latter two also have been found in ulluco, their main features are described above.

PBRSV, found in Peru, is mechanically transmitted through nematodes. Although infected potato plants can show necrotic ring spots, there is not a clear reference about symptoms in oca plants.

AVB-O, found in Peru, Bolivia and other South American countries where potato, oca and arracacha are grown, is also transmitted by nematodes, and probably by planting materials. Infected plants are symptomless.

Mashua viruses

Four viruses infect the mashua crop: PapMV and PVT (described in Ulluco above) and sowbane mosaic virus (SoMV) and a complex of isolates of tropaeolum mosaic virus (TropMV) (M-6, M-8) (Chuquillanqui, *pers. comm.).*

SoMV, found in South America, is transmitted by leaf miners, beetles and aphids. It also infects several cultivated plants including potato, spinach, *Vitis* sp. and *Prunus domestica*.

TropMV has several strains that require further clarification. They are transmitted by aphids. Its geographical distribution is not clear.

Control of virus diseases

The main strategy in preventing virus infection is using healthy tuber material, roguing infected crop plants, removing weed species that are alternative hosts, and spraying to control aphids and other vectors such as *Epitrix* spp.

PLANTING MATERIAL

Seed tubers are usually used for planting oca, ulluco and mashua. Tubers are usually produced in the field at the same growing season as the ware crops. Agronomic management of planting material for oca, ulluco and mashua is similar.

Producing planting material in plots after 6–8 years of fallowing at 3 500–3 800 masl will largely ensure good quality material that has less than 5 percent virus infection and is nematode and weevil free. Seed tubers should originate either from a positive selection process or basic seed production. They should maintain the variety type in terms of tuber shape and surface colour. Good quality tuber seed should show a clean and shiny tuber surface. Oca and ulluco should weigh 20–30 g and mashua should weigh 30–40 g.

Andean farmers have traditionally used 3–5 small tubers (5–10 g each) per planting station for oca, ulluco and mashua, as opposed to the 20–40 g recommended above. As the small tubers are set at the late (senescent) stage of plant development, virus particles probably would not have transported to the last-developed tubers making some of the small tubers virus free (Chuquillanqui, *pers. comm*.). For this reason, the cultivated oca, ulluco and mashua did not become extinct during their 3 000–5 000 years of continuous cultivation.

Improving the traditional system of planting material by positive selection

Farmers in the Andes usually renew their tuber seed every 3–5 years, believing that their seeds are "lazy". For this reason, there is a continuous tuber seed flow within and between communities, mainly at harvest and planting. Furthermore, in traditional Andean systems, farmers usually consider the rotational system and fallowing the key factors in reducing pests and diseases. In addition, women select the best quality tuber seed from the stores. They select the normal tuber shapes of the varieties with clean shiny tuber surfaces, understanding that the planting material is viral- and bacterial-disease free which, to a certain extent, is positive selection. This system has met most of the annual tuber seed requirements of oca, ulluco and mashua of the Andean rural families.

The positive selection criterion for producing good quality seed of ulluco according to Garay (1995) and Lopez (2004) is a process whereby crop materials are planted in the field. Before the flowering stage, vigorous young plants with deep green or deep green yellowish colour foliage and large/normal leaves without symptoms of virus diseases are identified, labelled and selected for harvesting separately. The tubers of each selected plant must be seen as healthy and conform to the shape and colour of the cultivar. In this way, a stock of clones is obtained. In the following growing season, the seed tubers of each clone are multiplied by planting them in separated rows of about 10 plants per clone. In the event that any plant of a clone shows any virus, bacterial or fungal disease or any undesirable character, all the plants of the clone are eliminated (negative selection). Conversely, if all the plants grow vigorously and disease free, the plants of the clone are maintained and harvested separately (positive selection). This process is repeated in the following growing season so that eventually there will be a stock of clean seed. This strategy can be followed easily in farming communities, thus merging

ancestral traditional knowledge and modern technologies to increase oca, ulluco and mashua productivity at a low cost.

RAPID MULTIPLICATION TECHNIQUE

When seed tubers are scarce, rapid multiplication techniques through positive selection can be used to meet planting needs. The vegetative parts used include tuber sprouts, juvenile cuttings and lateral shoot cuttings (Bryant *et al.,* 1981; López, 2004).

Sprout cuttings

These require exposing healthy tubers to diffuse light at ordinary room temperature for promotion of vigorous sprouting. When the sprouts are about 3–8 cm long, they are detached from the tubers by an anticlockwise movement to minimize damage and planted in a disinfected sand bed in the greenhouse. Once rooted, they are transplanted in the field and the usual cultural practices are followed until harvesting. Almost all sprout cuttings will survive. They can be harvested from mother tubers three times, about every 3–4 weeks. No rooting solution is needed. A plant originated from a sprout could yield 600–800 g of tubers. Normal plants developed from sprout cuttings also can be considered mother plants for juvenile cuttings or lateral shoot cuttings. This technique can be used for all three crop species. Sprout cuttings also have been used successfully for control of weevil in oca.

Juvenile cuttings

These start with plants arising from small tubers (10–30 g), sprout cuttings or *in vitro* plantlets. The plants should be 10–15 cm high, with 5–6 leaves per stem. Each stem cutting is further subdivided into smaller nodal cuttings (less than one mm in diameter) with axillary buds, after which they are rooted and sprouted in a sand bed. It is not necessary to use a rooting hormone. Once the cuttings have rooted and sprouted (2–3 weeks), they are transplanted in the field or used as new mother plants to increase the multiplication ratio. Tuber yield of a plant harvested from a juvenile cutting ranges from 0.5 to 1.0 kg per plant. The mother plant can provide further stem cuttings.

Lateral shoot cuttings

Pathogen-free tubers (30–40 g) are planted in pots or beds to root and sprout. Mother plants also can be raised from top shoot cuttings, juvenile cuttings or *in vitro* tuberlets. Once the mother plants are about 20 cm high, top shoot cuttings are collected, rooted and sprouted to raise more mother plants and increase the multiplication rate. The removal of the plant's top promotes the development of many lateral axillary buds, resulting in lateral shoot cuttings 6–12 cm long in about three weeks. This process can be repeated 3–4 times, meaning that a mother plant can yield up to 100 cuttings. The application of nitrogen fertilizer will stimulate growth of lateral shoots to stimulate cutting production. A plant derived from a lateral shoot cutting can yield 0.5–1.0 kg of tubers.

FIELD OR GREENHOUSE MANAGEMENT

Facilities and equipment

Laboratory facilities should contain equipment for thermotherapy, tissue culture and virus detection by either ELISA or NASH. These are needed to eliminate pathogens from clones of oca, ulluco, and mashua. Greenhouses for virus indexing and rapid multiplication techniques to obtain pre-basic and basic seed material are also required.

Sanitary management

Once plants emerge in the field, they should be maintained free of *Epitrix* sp. to prevent virus transmission. Also the field should be weed free to prevent aphids, beetles and weevils from infesting the crops. Frequent inspections should be made in the field and the greenhouse to check that there are no outbreaks of any pest or disease.

Harvesting

Tuber surfaces of oca, mashua and ulluco are very delicate. Therefore, harvesting methods and handling should ensure minimal damage to the tubers to prevent rapid deterioration during storage and transportation.

Storage

Careful selection of tubers free of mechanical and insect damage is a key to keeping loses at a minimum. They should be stored in boxes of about 50 kg or less with eucalyptus leaves at the base and at the top and kept in well-ventilated rooms where temperatures usually range from 8 to 12°C (Tupac,1999).

Disease control

Tuber seed should be selected from disease- and pest-free plants. Diseased plants must be removed from the field or greenhouse as soon as they are detected. All tubers showing atypical shape or damage in the store should be eliminated. The greenhouse, store and all associated structures should be kept clean and free of dead or decaying plants and tubers. Regular surveillance for occurrence of mites and tuber moths in the greenhouse should be part of the routine, and necessary control measures should be taken to prevent and/or stop their development. Tuber seed stores should be disinfected with sodium hypochloride before use to control bacteria and fungi.

Bananas, plantains and other species of *Musaceae*

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Musa acuminata Colla and *Musa balbisiana* Colla *Musaceae*

TAXONOMY, ORIGIN, DISTRIBUTION

Groups
Musa AA, AAA, and AAAA[1]

Groups
Musa BB, BBB

Interspecific groups
Musa AB, AAB, ABB, AAAB, AABB and ABBB
More than 20 subgroups

Origin
South Asia, Southeast Asia and the Pacific with secondary diversification zones in West and Central Africa for the plantain subgroup, and in the East African Highlands for the Lujugira banana subgroup.

Distribution
Throughout low and mid-altitudes of the humid, wet-dry and dry tropics and subtropics (approximately 120 countries).

PROPAGATION METHODS COMMONLY USED
Five methods are commonly employed to obtain planting material for the establishment of new planting material of banana and plantain:

- suckers extracted from banana and plantain fields which are in production,
- suckers reproduced in field sucker multiplication plots,

[1] AA=banana diploid; AAA=banana triploid; AAAA=banana tetraploid

- plants from micro-corms grown out in a nurseries,
- plants originating from secondary buds (PIBS[2]), produced in a humidity chamber, seedbeds and grown in nurseries,
- tissue culture plants grown in two-phase nurseries.

MAIN DISEASES AND PESTS TRANSMITTED BY PLANTING MATERIAL

Bacterial diseases

- moko disease (*Ralstonia solanacearum* Smith, phylotype II)
- xanthomonas wilt (*Xanthomonas vasicola* pv. *musacearum*)

Viral diseases

- banana bunchy top virus (BBTV)
- cucumber mosaic virus (CMV)
- banana streak viruses (BSV)
- banana bract mosaic virus (BBrMV)
- banana mild mosaic virus (BanMMV)
- abacá mosaic virus (AbaMV)

Fungal diseases

- fusarium wilt or Panama disease *Fusarium oxysporum* f.sp. *cubense*, often referred to as Foc

Nematodes

- burrowing nematode *Radopholus similis* Cobb
- root-lesion nematodes *Pratylenchus coffeae* (Zimmerman) Filipjev and Schuurmans Stekhoven and *Pratylenchus goodeyi* Sher and Allen

Insects

- black weevil *Cosmopolites sordidus* Germar

PRIMARY CONSIDERATIONS OF PLANTING MATERIAL QUALITY

The quality of a batch of planting material has three components that should be taken into account by the buyer, the seller and the quality controllers.

- **Disease.** The material for planting or any associated rooting medium should not be a source of disease or insect pests. If planting material is brought from

[2] From French acronym of Plants Issus de Bourgeons Secondaires

another country, region or continent, it can introduce a new insect pest or disease with devastating effects for producers. Viruses are often spread in this way. Diseases such as black leaf streak disease also can be introduced in this way, although once this disease is present in a region, planting material is no longer an important path for its local dissemination. If planting material of local origin is highly infected with commonly occurring diseases and insect pests, the grower will also experience lower yields and a shorter plantation life. Planting material prepared by different methods has different risks of pest and disease transmission.

- **Variety**. The batch of planting material should contain only the desired variety. In addition, the material should originate from plants with superior production, resistance and quality traits. Acquisition of improved planting material is an opportunity to upgrade the production potential of the variety. Certain high rate multiplication techniques can be used to multiply carefully selected plants with elite characters.
- **Size and uniformity**. The batch of planting material should have the size and uniformity appropriate to the objectives and resources of the grower. In cases targeting a small marketing window, the grower may want highly uniform planting material. If the planting is for home consumption or local sale, less uniform planting material may be preferable to spread the first harvest over a longer time period.

DISEASES AND INSECT PESTS

Diseases and insect pests are an important element of the quality of planting material for two reasons. First, certain diseases and insect pests are not yet present in all *Musa* growing regions. Extreme vigilance is therefore essential to prevent the spread of these phytosanitary problems to new regions, especially through planting material or other means. These quarantine diseases include moko disease, xanthomonas wilt, fusarium wilt, tropical race 4, banana bunchy top disease and banana bract mosaic disease. Planting material and other plant parts contaminated with sigatoka disease or black leaf streak disease also can potentially spread the diseases to non-infected areas.

Second, diseases caused by bacteria, fungi, virus, nematodes and insect pests infect plantain and banana planting materials reduce bunch size, stand density and stand productive life. Ideally, both the planting material and the field where the new plantation is to be established should be free of these diseases and pests. In practical terms, it only may be possible to minimize these problems in the planting material without achieving completely clean seed. Appropriate practices for the quality of planting material should be decided upon according to the phytosanitary problems present and the multiplication method to be used.

PROTOCOL FOR THE PRODUCTION OF PLANTING MATERIAL

Common methods for multiplication of planting material

Five methods are common for obtaining planting material for the establishment of new plantings of banana and plantain. Each method has specific requirements in terms of facilities and equipment, generates planting material at a characteristic rate and has particular risks of pest and disease contamination. The methods range from a few suckers extracted from backyard gardens, to small seedbeds with a few hundred seedlings distributed at the local level, to a manufacturing unit for producing several million *in vitro* plants per year. The five techniques are described below. Good practices for different stages of plant multiplication are described in later sections.

SUCKERS EXTRACTED FROM BANANA AND PLANTAIN FIELDS IN PRODUCTION

Fields in production have numerous types of planting material. Sword and maiden suckers are generally considered the most reliable and productive planting material for direct extraction from production fields. The suckers are 0.5–1.0 m long, with a cone-shaped growing stem and small narrowing to the most expanded leaves. However, any shape or type of sucker or the main corm can be used, either intact or cut into pieces, to plant a new plantation, although harvest intervals may be lengthened by less satisfactory material. After extraction with hand tools, suckers or corm pieces must be subjected to diverse practices (described in Table 1) to minimize the transfer of pests and diseases, and then planted directly into a new field. Depending on the variety, each mat in a plantation may yield 1–3 suitable suckers. Over-extraction or careless extraction of suckers may result in weakened plant support and stem toppling.

Suckers reproduced in multiplication plot

Suckers or corm pieces are used to plant a high density stand. When the plants reach the stage of flower differentiation – well before flower emergence – decapitation or false decapitation is used to stop further flower development.

TABLE 1
Key banana plant multiplication steps

Steps	Suckers selected from production field	Suckers grown in a sucker multiplication plot	Microcorms	PIBS	Tissue culture
Sucker selection	X	X	X	X	X
Sucker preparation	X	X	X	X	X
Field selection		X			
Field management		X			
High humidity chamber				X	
Tissue culture laboratory					X
Weaning nursery					X
Hardening nursery			X	X	X

This action stimulates the emergence of 10–20 suckers per stem. These suckers are then extracted and prepared to avoid pest and disease multiplication and transfer.

Microcorms

Small, cone-shaped suckers from 200–300 g, called "peepers", are extracted from a production field or a sucker nursery, treated and then planted into a nursery for 6–8 weeks, until plants reach an appropriate size for transplanting.

PIBS

Sword corms (minimum 12–25 cm diameter or 150–400 g) or pieces of larger corms, peeled and stripped completely of leaf sheathes, are placed in wet sawdust in a humidity chamber made of plastic sheeting. The destruction of the main growing point of the sucker releases the axillary buds at the base of each leaf sheath for sprouting. The resulting shoots are carefully excised and transferred to nursery bags, under similar conditions to microcorms, until the plants are ready for transplanting. A single sucker can produce 15–60 shoots.

Tissue culture

Under controlled laboratory conditions, small corms are pared down and disinfected prior to the extraction of the shoot tips. Each shoot tip can be used to produce up to 1 000 *in vitro* plants, which are weaned at high humidity and relatively low light and then transferred to a nursery to be grown on before transplanting into the field.

GOOD MULTIPLICATION PRACTICES FOR EACH TECHNIQUE

Good multiplication practices must be employed in the five methods mentioned to produce plants of superior production potential with a minimum risk of pests and disease. These practices can be categorized by key steps common for two or more techniques as shown in Table 1.

Sucker selection

1. Select and mark plants of the desired variety with normal or below average height, a stout trunk and firm roots should be. Plants should be free of undesirable variations of the varietal characteristics. Selection should be made between flowering time and harvest to mark plants with above average bunch size.
2. When suckers are selected, whether to be used as planting material or as starting material in horticultural or tissue culture multiplication techniques, document their origins (country, village, farmer) and identify and describe the plot they come from. If the suckers are used in tissue culture, the source of shoot tips should be specified as monoclonal (originating from a single mother plant) or polyclonal (originating from more than one mother plant).

3. The plot must be well managed and selected suckers should be healthy. Good suckers are cone shaped and do not develop broad leaves until they are more than 1 m high. However, preferred sucker size depends on the technique to be used. Sword suckers are generally considered preferable to water suckers or corm pieces, but suckers of all sizes and even corm pieces can be used as planting material, provided they are free of quarantine diseases and relatively free of other pest and diseases. If the sucker is to be virus indexed, selected suckers should have at least one large new leaf.
4. For suckers or any derived planting material destined for international transportation, especially from tissue culture, the following quarantine diseases should be absent from country of origin.
   - Moko disease due to *Ralstonia solanacearum* Smith, phylotype II
   - Xanthomonas wilt caused by *Xanthomonas vasicola* pv. *musacearum*
   - Tropical Race 4 *Fusarium oxysporum* var. *cubense*
   - BBTV
   - BBrMV.

For suckers or other derived planting material destined for local or within-country sale or exchange, the above diseases should be completely absent from the field and all surrounding fields. The farther these diseases are from the source of the planting material, the lower the risk of contamination of the seed material.

Other pests and diseases, especially nematodes, weevils and other viral and bacterial diseases, should be absent or have a low prevalence in the field from which planting material is extracted. This should be confirmed by regular field inspections.

If the sucker is to be used in tissue culture, virus index leaf samples from the mother plant and from all the extracted suckers. Virus indexing also can be used for PIBS to ensure virus-free material for a well-planned and executed multiplication programme.

Sucker preparation

1. To prevent the spread of diseases from one sucker or corm piece to another, disinfect paring tools in a 5 percent sodium hypochlorite solution or a 20 percent iodine solution after each sucker has been propagated.
2. Suckers removed from the mother plant must be pared in the field before being transported, by removing all the roots and the outer surface of the corm until it is uniformly creamy white. Any suspicious part of a different colour should be removed. If darkened galleries, dead or discoloured areas or other damage make up one-fourth to one-third of the sucker, the sucker should be discarded.

3. Cross section the pseudostem 10–15 cm above the corm to identify any off-coloured rings, liquid or brownish spots. Suckers or corms showing these symptoms should be eliminated.
4. Once sucker paring is complete, transport the suckers immediately to a site at least 1 km from any banana fields to limit the risk of weevils reaching them to lay new eggs.
5. Depending on the type of multiplication techniques used, submit the sucker to the following practices.
   - If suckers are to be planted directly or put into a multiplication plot, they can be treated by immersion in hot water (30 seconds in boiling water or 20 minutes in water at 50°C) to kill weevil eggs and nematodes. Paring may not be necessary prior to this hot water treatment.
   - For PIBS, suckers which have been pared need to be further prepared before being placed in the high humidity chamber. The leaf sheathes should be carefully stripped away one by one, to expose axillary bud nodes at the base of each leaf. Suckers are cut in the form of an × across the stem section to destroy the main growing point. Suckers can also be treated with a up-to-date fungicide and dried in the shade for one day before planting (Plate 1).
   - Suckers for production of starting material for tissue culture should be maintained in an area free of banana plants or in an insect-proof enclosure. Once planting material has been verified as virus free, it can be planted in large pots in a screen exclusion house to ensure that there is no contact with virus- bearing vectors. Such material can serve as a regular source of small corms for new shoot tips (Plate 2).

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Plate 1
PIBS sucker prepared with ×.

LESCOT, CIRAD. 2006.

Plate 2
PIBS sucker with numerous shoots.

FIELD SELECTION AND MANAGEMENT OF SUCKER MULTIPLICATION PLOTS

1. For a sucker multiplication plot, free the field of nematodes specific to bananas or plantains. It should not have been planted with banana or

plantain for at least a single cycle and preferably for a minimum of three years. No bananas should be growing in neighbouring fields to avoid contamination from water runoff or through human traffic or implements.

2. Select a site with deep soil of medium texture. It should be well drained with adequate rainfall or irrigation to ensure continuous plant growth. Plantation densities can be up to 10 000 plants/ha, three to four times the density of a field managed for fruit production.
3. Management of irrigation, nutrition, weeding, and pest and disease control must be more meticulous in sucker multiplication plots than in fields for fruit production. Throughout the crop cycle, off-types should be eliminated. If any quarantine diseases such as bacterial disease, fusarium wilt or virus appear, the entire field should be quarantined and the suckers should not be sold or distributed.
4. Four to five months after planting, the apical meristem is eliminated or stopped by decapitation, false decapitation or folding of the pseudostem. Plants also can be ridged to increase the number of suckers and accelerate their growth and development.
5. When suckers reach the desired size, prepare them using the procedures described above or as microcorms.

HIGH HUMIDITY CHAMBER FOR PIBS

Multiplication techniques using a high humidity chamber include PIBS and corm fractioning such as sett and minisett multiplication techniques. The sanitary quality of the substrate and the conditions of humidity, light, temperature and drainage in the chamber are critical to ensuring quality material production. The high yield of plants from a single sucker makes this a useful technique for local multiplication of superior or new clones. When viruses are present, initial testing of foundation suckers is necessary. The following practices contribute to high quality PIBS.

1. Fill the sprouting bed within the chamber with clean, non-toxic sawdust free of soil or any other plant residue.
2. Locate the chamber at least 1 km from any banana plantation to avoid any accidental contamination through runoff or other transmission method.
3. Moisten substrates with enough clean water (not contaminated with banana pathogens) to maintain a high humidity within the chamber. Too much water will encourage the growth of bacteria and fungi.
4. Maintain the temperature between 25 and 40°C (up to 50°C at the hottest time of day). Shading of the chamber is recommended as the chamber should not be in direct sunlight.
5. Excise only vigorous, healthy shoots with normal leaf characteristics for planting in nursery bags. Off-type suckers or suckers from undesired varieties should be eliminated from the chamber. Suckers not producing shoots or producing very few shoots also should be eliminated and replaced.

LABORATORY TECHNIQUES FOR TISSUE CULTURE MULTIPLICATION

Tissue culture production is a specialized operation. It requires experience and strict procedures to minimize risks from contamination, somaclonal variation and virus activation and, at the same time, to keep production costs within acceptable limits. The cost of in vitro plants is the primary factor that limits wider spread use of this method.

The presence of BSV endogenous pararetrovirus (EPRV) in the genome of interspecific triploid (AAB) and tetraploid (AAAB) varieties results in severe limitations on the use of tissue culture multiplication. The tissue culture process itself is suspected of activating BSV EPRVs in virus-free shoot tips, especially in plantain species. *In vitro* techniques are not recommended for the multiplication of the AAB varieties, especially plantains, and AAAB varieties. These varieties can be multiplied from local mother plants for distribution to farmers within the same region. However, tissue culture plants of these cultivars should not be exchanged between countries. Other varieties, especially those with the *Musa acuminata* genome, present no risk of BSV EPRV activation during tissue culture.

For high quality tissue culture plants, the laboratory should be equipped with the necessary equipment to complete the following three phases. Specific protocols and procedures should be followed. The traceability of the shoot tip should be maintained throughout the *in vitro* multiplication process.

Phase 1: Introduction of virus-free and bacteria-free bud material under aseptic conditions

All introduced suckers and their mother plants should be tested for viruses and bacteria, a procedure that takes 1–2 months. Before extracting the shoot tips, the plant fragments should be disinfected to eliminate surface contaminants. Once the material has been disinfected, all further work should be done in a laminar flow chamber. The shoot tips should be 1.5 cm x 1.5 cm x 1.0 cm and put on a sterile culture medium in sterilized flasks.

Phase 2: Multiplication of buds or shoots

The shoot tips that survive Phase 1 give rise to buds or shoots. At regular intervals, these new shoots should be transferred to a new sterile medium. To reduce the incidence of off-types (somaclonal variants), production from a single shoot tip should be limited to 1 000 plantlets. The number of subcultures should not exceed 10.

Phase 3: Regeneration and rooting of shoots or buds

The shoots obtained during Phase 2 should be transferred to a regeneration medium consisting of Murashige and Skoog (MS) salts, sugar (and eventually

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Plate 3
In vitro *plants ready for planting.*

activated charcoal) to support root growth.

For phases 2 and 3, the recommended temperature range is 20°C to 35°C. Artificial lighting should be provided from cool white fluorescent tubes for 12 to 16 hours each day. The laboratory should adhere to set procedures to maintain the sterility of the tissue culture laboratory including no footwear from outside, and required wearing of cap and gown.

At this point, the plantlets can be commercialized either within the country or exported if they can be guaranteed free of pathogens. To limit the risk of contamination and maintain the quality of the plantlets, they should be transported in sterile conditions with no exterior contact. This recommendation should be compulsory when the tissue culture plants are exported to another country. Rooted plants in non-sterile soil or other media should not be moved between countries (Plate 3).

Accurate records are important for all batches of tissue culture plants to minimize labelling errors and phytosanitary risks, and to facilitate internal and external controls and traceability by official organizations and buyers. The documentation provided with each shipment of plants should include the following information.

- ✓ Name, address and contact information of laboratory where *in vitro* plants were produced.
- ✓ Name and address of the company offering the plants for sale.
- ✓ Variety, special clonal information and location of mother plants from which shoot tips were extracted. If mother plants are maintained in a screen house, the location of the original mother plants.
- ✓ Number of plantlets produced and subcultures used per shoot tip in routine tissue culture multiplication.
- ✓ Maximum percentage of off-types guaranteed (commonly proposed upper limit 3 percent).
- ✓ Virus indexing protocols followed and results (BBTV, CMV, BSV, BBrMV, BanMMV).
- ✓ Name, address and contact information of laboratory conducting virus indexing.
- ✓ All other disease screening conducted, protocols and results.

- ✓ Name, address and contact information of laboratory conducting other disease screening.
- ✓ Name and address of official plant sanitary control organization certifying the plants.
- ✓ Instructions for storage conditions prior to weaning.
- ✓ Weaning instructions clearly specified.

The importing country may quarantine the material to make observations for signs of diseases within eight weeks, during which time the plantlets should be maintained in quarantine nurseries isolated from all other banana or plantain plantations. Any plants or symptoms associated with quarantine diseases (BBTV, BBrMV, TR4 *fusarium* wilt, *ralstonia* and x*anthomonas* bacterial wilts) should be subjected to further testing. If tests are positive, the entire shipment of tissue culture plants should be destroyed.

WEANING NURSERY

The practices below are necessary only for *in vitro* plantlets. By the end of four weeks, plantlets will have rooted and have three to four green leaves.

1. A weaning nursery is an enclosed area like a greenhouse. Move the new plants from the laboratory to the weaning nursery as quickly as possible to avoid stressing the plants. In transit, avoid long periods in darkness or direct sunlight and temperatures below 18°C or above 30°C.
2. Before transplanting, remove old substrate (e.g. agar) from the plantlets and rinse them in clean water before dipping them in a broad spectrum up-to-date fungicide solution.
3. Transplant plantlets into containers of 10–30 cm^3 filled with clean substrate the quality of which is guaranteed and verifiable, such as commercial peat soil or potting soil with high water holding capacities. Crop and plant residues (e.g. rice husk, sifted coir fibre, sawdust), alone or mixed with other residues and well composted, are prime raw materials to prepare substrate and can be used either fresh or composted. Sterilize all raw materials for substrate mixtures before use.
4. Water with clean water. If the presence of nematodes is suspected, the water can be filtered with a 5 µ filter.
5. Facilitate drainage during watering and improve sanitary conditions by placing the plantlets on tables or benches.
6. For the first week, use plastic sheeting to create a compact chamber around young plantlets to maintain a high relative humidity. This accelerates leaf development and reduces plant stress.
7. Recently transplanted plantlets are very sensitive to changes in climatic conditions (temperature, relative humidity and especially light). The success of the weaning phase (survival rate and quality of plant material) depends

on their gradual acclimatization to the less humid and brighter conditions in the environment they are going to.

8. Eliminate all off-type plants as soon as they are identified.

HARDENING NURSERY

The three types of planting material to be processed in hardening nurseries are tissue culture plants that come from a weaning nursery, PIBS shoots produced in a high humidity chamber and microcorms. This phase is intended to bring the plantlets to the stage where they are ready to be planted in the field. Properly hardened plants are ready for transplanting when the last fully emerged leaf measures 20–30 cm in length. No off-type plants should be present. The plants should be of one variety only.

1. Put planting material into individual bags (perforated polyethylene bags for drainage) or pots with a capacity of 0.8 to 3 litres filled with a clean, good quality substrate (as described for the weaning phase). The volume of the bag or pot depends on the time the plants will spend in the nursery, which should be at least 3–4 weeks.
2. Shade the nursery, including the side walls, to maintain a uniform 50 percent light level, at least during the first week, especially for the tissue culture and PIBS plants. Shade should be reduced gradually and finally eliminated before the end of this phase to create field conditions. Excessive shade will cause elongated, spindly plants that will suffer from transplanting shock.
3. The nursery should be well drained so that excess water drains rapidly when plants are watered.
4. Take measures to avoid specified pests and pathogens with potential to cause infection in the nursery, such as:
   - nematodes: use clean water, filtered if necessary, and clean substrate in bags or pots, and inspect roots regularly for possible presence of pathogenic nematodes,
   - virus: remove any weeds inside the nursery and for 10 m around the perimeter of the nursery, use repellents or screen for exclusion of insects, including ants; use broad spectrum insecticides if necessary (the plantlets are very attractive to certain aphids, which increase the risk of infection of cucumber mosaic virus),
   - bacterial diseases: avoid zones with known sources of moko disease and *xanthomonas* wilt; water may need to be sterilized or brought from bacterial wilt-free sources
5. As the plants grow, they should be spaced out to avoid leaf overlap. They also can be graded to create more uniform lots which will be ready for transplanting at the same time.

TABLE 2
Risk of transmission of pests and diseases by multiplication method with 100 percent use of good practices

Pest/disease	Suckers selected from field in production	Suckers grown in a multiplication plot	Microcorms	PIBS	Tissue culture
	0 = zero risk; 1 = low risk; 2 = moderate risk; 3 = high risk				
Bacterial diseases*	2	1.5	1	2	0.5
	(3)	*(2.5)*	*(2)*	*(2)*	*(1)*
BBTV*	2	1,5	1	2	0
	(3)	*(2.5)*	*(2)*	*(2)*	*(3)*
BSV	1	1	1	2	2
	(2)	*(2.5)*	*(2)*	*(2)*	*(3)* (plantain)
Other viruses	2	1,5	1	2	0.5
	(3)	*(2.5)*	*(2)*	*(2)*	*(0.5)*
Foc*	2	1.5	1	2	0.5
	(3)	*(2.5)*	*(2)*	*(2)*	*(1)*
Nematodes	1	1	0	0	0
	(3)	*(2)*	*(2)*	*(2)*	*(2)*
Weevils	1	1	0	0	0
	(3)	*(2)*	*(2)*	*(0)*	*(0)*

* If the pest or disease is not present in the region or country, the risk is substantially lower
Note: bracketed figures indicate results with limited use of good multiplication practices

6. Stunted plantlets and off-types can be detected when plants are being re-spaced or sized. The most common types of somaclonal variants include dwarfism, gigantism, "massada" mosaic-like, variegation, chlorosis or necrotic leaf patches and droopy leaves. All off-type plants or plants lacking vigour should also be eliminated. The proportion of somaclonal variants from plants coming from the same tissue culture laboratory should not exceed 5 percent. If more than 5 percent are off-type, the whole batch should be destroyed.
7. Schedule regular application of fertilizers, adapted to the local conditions. Increase application as the plants grow.

QUALITY STANDARDS FOR PLANTING MATERIAL

Risks of pest and disease infestation

The multiplication practices described above are designed to minimize the risks of transmitting pests or diseases through the planting material produced (Table 2). It is therefore essential to follow each step and specification closely (Plate 4).

Plate 4
Root nematode damage. Bioversity. 2006

Quality standards for planting materials

The quality standards outlined below provide guidance to buyers who are placing an order or receiving a batch of suckers for direct planting or plants produced in a nursery. If the batch exceeds the proposed tolerance limits, then it should be rejected. Once the batch has been acquired, suckers or plants with defective qualities still can be eliminated to improve the quality and uniformity of the resulting plantations. This selection can occur when the plants are being loaded for transport, moved to the field or transplanted.

Size and weight

Suckers and corm pieces for direct planting

- Many different sizes and types of planting material can be used satisfactorily to establish a plantation. Smaller suckers or portions of suckers sprout slowly and have a higher rate of failure compared to larger suckers. Suckers or corm pieces should measure at least 12 cm in diameter. The upper limit on sucker size is set by practical concerns of transport and logistics.

Plants produced in nursery

- Last fully emerged leaf should be at least, but not greater than, 20–30 cm in length, with leaves progressively larger in size from oldest to youngest.

  Tolerance of plants not reaching size criteria: 1 percent.

Summary tables of standards

TABLE 3
Suckers/corms for direct planting

Size of suckers and corms	At least 12 cm in diameter	
Suckers/corms health	Removal of roots/peeling to reduce contamination with pests/diseases	Tolerance of corms with partial or total roots: 3 % of corms
	Creamy-white corms resulting from elimination of insects galleries, nematodes, bacterial/fungal diseases	Tolerance: 2 % corms with more that 1/3 corm removed or not totally creamy-white colour
Pseudostems	Cross section without off-coloured rings or liquid or brownish spots	Tolerance of cross sections with off-coloured rings: 0%

TABLE 4
Plants produced in the nursery

Plant size	Last leaf 20-cm length, oldest leaves larger than young ones	Tolerance of plants not reaching size criteria: 1 %
	Height of plant not to exceed two times height of pot	Tolerance of plants not reaching size criteria: 5 %
Off types	Dwarfism, gigantism, mosaic-like, variegated, chlorotic/necrotic leaf patches, droopy leaves	Tolerance off types: 1 %
Container	Damage/loss of substrate	Tolerance: 2 % plants

- The height of plant should not exceed two times the height of the bag or pot.

  Tolerance of plants not reaching size criteria: 5 percent.

Other quality planting material standards applicable to a batch of seed or plants

Suckers for direct planting (Table 3)

- Removal of roots and peeling to reduce risk of contamination by weevil eggs and larvae, nematodes, bacteria and fungi.

  Tolerance of corms with partial or total roots: 3 percent of the corms.
- Creamy-white colour of the corm which results from the elimination of weevil larvae galleries, nematodes and others bacterial or fungus diseases; at least two-thirds of the corm should remain after paring (if not, discard).

  Tolerance of 2 percent of corms with more than one-third corm removed during paring or not totally creamy-white colour.
- Pseudostem cross-section with no off-coloured rings, liquid or brownish spots (bacterial or fungal symptoms).

  Tolerance of pseudostem cross-sections with off-coloured rings: 0 percent.

Plants produced in nursery (Table 4)

- Plants with off-type characteristics – dwarfism, gigantism, mosaic-like, variegated, chlorotic or necrotic leaf patches, droopy leaves.

  Tolerance of off-types: 1 percent.
- Condition of container: damage to container or loss of substrate.

  Tolerance: 2 percent plants.

EXAMPLE OF MULTIPLICATION PROGRAMMES WITH AND WITHOUT QUARANTINE DISEASES

The major challenge for the production of clean, high quality planting material is choosing the appropriate technique for the local pest and disease problems and then planning the production process for timely planting. This is especially important in rainfed plantations where planting can be completed during only a few months in the year.

Alternative programmes to produce 50 000 plants when quarantine diseases are present

The use of locally produced suckers for direct fields planting, sucker multiplication plots, microcorms or PIBS brings with it a very high risk of multiplication of the quarantine diseases that may be present. The only options available depend on *in vitro* multiplication with clean shoot tips thoroughly indexed as free of virus. The initial emphasis of these multiplication programmes should be on disease-free material but, over a period of 5–10 years, the selection process should also

include the identification of superior clones with a high and uniform production potential.

Option 1, in Table 5, is more applicable where *in vitro* plants are inexpensive and the re-infection rate is high. This approach is used in areas where there is a threat from Foc or where BBTV pressure is very high. Under such conditions, the use of sucker multiplication plots represents a high risk of re-infection before PIBS can be implemented. Option 2 may be applicable where the risk of re-infection is lower and where tissue culture plants are more expensive.

Alternative programmes when major quarantine diseases are absent

In regions where there are no quarantine diseases present, there are numerous options to produce clean planting material. The major challenge in such regions is developing superior clones with a high and uniform production potential. The use of *in vitro* multiplication is not illustrated among the options in Tables 6, 7 and 8, but may be very effective once superior clones have been identified.

TABLE 5
Options for planting material multiplication where quarantine diseases are present

Option 1. *In vitro* plants			Option 2. *In vitro* plants, sucker multiplication plot, PIBS		
Steps	**Time (months)**	**Factors in multiplication**	**Steps**	**Time (months)**	**Factors in multiplication**
Selection, indexing, cleaning of 55 virus- and disease-free shoot tips of desired variety	1–12	Small losses due to shoot tip survival and multiplication	Selection, indexing, cleaning of 2 virus and disease-free shoot tips of desired variety	1–6	Small losses due to shoot tip survival and multiplication
Production of 53 000 *in vitro* plants	6	1 shoot tip yields 1 000 *in vitro* plants	Production of 210 *in vitro* plants	6	1 shoot tip yields 1 000 *in vitro* plants
Hardening and weaning nursery to produce 50 000 plants	6	Loss of 3% off-types, damaged containers, plants not surviving transplant	Hardening and weaning nursery to produce 205 plants	6	Elimination of 3% off-types and damaged containers
			Protected sucker multiplication plot to produce 2 000 suckers	8	1 plant yields 10 suckers
			High humidity chamber with 2 000 suckers	6 months	1 sucker yields 25 PIBS
			Weaning nursery with 50 000 plants	6 months	Small loss of damaged containers and plants not surviving transplant

TABLE 6
Planting material multiplication from fields in production (quarantine diseases absent)

Option 3. Suckers from plantation for direct planting			Option 4. Microcorms from plantation into weaning nursery		
Steps	**Time (months)**	**Factors in multiplication**	**Steps**	**Time (months)**	**Factors in multiplication**
15–20 ha field (1 000 plants/ha) planted for production from which suckers extracted	10	1 plant yields 2 to 5 suckers	15–20 ha field for production from which **microcorms** are extracted	8	1 plant yields 2 to 5 suckers
50 000 suckers pared and treated for planting	0.5	Small losses of suckers not sprouting	**Microcorms** pared, treated and grown out in nursery	2	Very small losses of plants not surviving transplant

TABLE 7
Planting material multiplication from sucker multiplication plots (quarantine diseases absent)

Option 5. Sucker multiplication plot			Option 6. Microcorm multiplication plot, microcorm nursery		
Steps	**Time (months)**	**Factors in multiplication**	**Steps**	**Time (months)**	**Factors in multiplication**
2 ha field planted for production from which suckers extracted	10	1 plant yields 2 to 5 suckers	2 ha field planted for production from which suckers extracted	8	1 plant yields 2 to 5 suckers
1 ha sucker multiplication plot (5 000 plants/ha) from suckers pared and treated	10	1 plant yields 10 suckers	1 ha microcorm multiplication plot (5 000 plants/ha) from suckers pared and treated	8	1 plant yields 10 microcorms
			Microcorms pared, treated and grown out in nursery	2	Very small losses of plants not surviving transplant

TABLE 8
Planting material multiplication with PIBS (quarantine diseases absent)

Option 7. PIBS from suckers from a field in production			Option 8. PIBS from a sucker multiplication plot		
Steps	**Time (months)**	**Factors in multiplication**	**Steps**	**Time (months)**	**Factors in multiplication**
1 ha field planted for production from which suckers extracted	10	1 plant yields 2 to 5 suckers	100 plants planted for production from which suckers extracted	10	1 plant yields 2 to 5 suckers
2 100 suckers into high humidity chamber	6	1 sucker yields 25 PIBS	250 suckers in multiplication plot from suckers pared and treated	8	1 plants yields 10 corms
Weaning nursery with 50 000 plants	6	Small loss of damaged containers and plants not surviving transplanting	2 100 suckers into high humidity chamber	6	1 sucker yields 25 PIBS
			Weaning nursery with 50 000 plants	6	Small loss of damaged containers and plants not surviving transplanting

Cassava

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Manihot esculenta Crantz
Euphorbiaceae

Cassava (*Manihot esculenta* Crantz), known as mandioca or yuca in different regions of South America, is a perennial native of tropical America. Its probable center of origin is Northeastern and Central Brazil (Allen, 2001; Olsen and Schaal, 2001) with a second center of diversity and domestication site in Central America (Nassar, 1978).

Cassava can be propagated from either stem cuttings or botanical seed, but the former is the main practice. The root is not a reproductive organ. Plant height can vary from 1–4 m. Its growth habit influences the amount of planting material that a mother plant can produce. Erect, non-branching types produce larger amounts of cuttings (up to 30) which facilitates the harvest, storage and transport of stems (Ceballos and de la Cruz, 2002). The reproductive rate of high branching types can be as low as 1:3. A plant grown from stem cuttings (stakes) can produce as many primary stems as there are viable buds on the cutting. However, in some varieties with strong apical dominance, only one stem develops (Alves, 2002).

The number of commercial stakes obtained from a single mother plant typically ranges from 7 to 10. Propagation rate depends upon variety, climate, management, age of planting material and soil conditions.

PESTS AND DISEASES
Main diseases

Cassava stems are attacked by various pathogens that induce internal or external rot and/or cortical or epidermal cankers (Lozano *et al.,* 1977). Other pathogens – viruses, mycoplasms cassava bacterial blight – invade the woody stem tissue systematically without leaving any visible symptoms.

Systemic pathogens include vascular (viruses, bacteria and/or phytoplasma) and cortical or epidermal (different fungi) causal agents that invade the host systematically without leaving any visible signs in the mature portion of the stem. For this reason, a high percentage of the plants coming from diseased cuttings are infected, and these plants may constitute the source of primary inoculum in the new plantation. This is the means through which systemic pathogens are disseminated.

TABLE 9
List of main cassava diseases

Code	Common name	Agent
CBB	Cassava bacterial blight	*Xanthomonas axonopodis* pv. *manihotis*
CBSD	Cassava brown streak disease	Virus
CMD	Cassava mosaic disease	Virus
	Foliar diseases	*Cercospora* spp., Cercosporidium *spp.,* Colletotrichum *spp., Phaeromularia* spp.
FSD	Frog-skin disease	Suspect a virus and phytoplasm
	Root rots	*Armillaea* spp., *Diplodia manihotis, Fusarium* spp., *Phytophtora* spp., *Scletorium* spp.
SED	Super-elongation	Induced by *Sphaceloma manihoticola*

The most important diseases (Table 9) propagated by infected planting materials are:

- cassava mosaic disease (CMD) and cassava brown streak disease (CBSD) are both present in Africa and CMD is found in India and Sri Lanka as well,
- cassava bacterial blight (CBB, *Xanthomonas axonopodis* pv. *manihotis*) is found in Asia, Africa and Latin America and the Caribbean,
- super-elongation disease (SED), a fungal disease induced by *Sphaceloma manihoticola* (Teleomorph: *Elsinoe brasiliensis*) that is widespread in the Americas,
- several species of *Phytophthora*-induced root rot and infected stems, which are the most important dissemination mechanisms,
- *Diplodia manihotis* fungus, which causes root and stem rots in Africa and Latin America and the Caribbean,
- frog skin disease (FSD), which is of unknown etiology although suspects are a virus and a phytoplasm.

Other diseases which are not as acute as those mentioned above – although in certain regions the assessed importance of diseases may be different – include:

- other foliar diseases affecting cassava productivity in tropical lowlands with high rainfall belonging to genera *Cercospora, Cercosporidium, Phaeoramularia* or *Colletotrichum* (Jennings and Iglesias, 2002),
- *Phoma* species which cause leaf and stem lesions in the tropical highlands,
- root rots that are induced by different species of *Sclerotium, Armillaea* and *Fusarium*.

Main pests transmitted by the propagation material

Stems are attacked by insects and mites that are localized on the epidermis or within the stem (Lozano *et al*., 1977). Different species of mites feed on cassava leaves. When they migrate, they are found on the stem surfaces of the infected plants

where they attack the germinating buds. Infected cuttings are the most important vehicle for the mite *Mononychellus tanajoa*, scale insects (*Aonidomytilus albus, Saissetia miranda*) and mealy bugs (*Phenacoccus herreni* and *P. manihoti*). The eggs and larvae of other insects such as thrips (*Frankliniella williamsi, Corynothrips stenopterus* and *Caliothrips masculinus*) and lace bug (*Vatiga* spp) adhere to the surface of stems and spread via infested cuttings.

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Plate 5
Galleries made by stem borers

The insects most commonly found within the stem are the stem borers from different species of Coleoptera (*Coelosternus* sp. and *Lagochirus* sp.), Lepidoptera (particularly *Chilomima* spp) and Hymenoptera fruit flies (*Anastrepha* spp.) (Plate 5). The surface or subterranean cutworms that feed on the stem (*Agrotis ipsilon* and *Prodenia eridania*) are often carried inadvertently from one place to another. The galleries that they make in the stem facilitate access of micro-organisms that cause stem rot, but also provide an easy way to identify infested planting material.

PROTOCOL FOR THE PRODUCTION OF PLANTING MATERIAL

Greenhouses and laboratory facilities

Rapid multiplication methods have been developed and range from the use of microstakes to tissue culture techniques. One-node microstakes have been used successfully for rapid multiplication schemes. For these conditions, irrigation would be a requirement. The size of the stake to be used depends on:

- the moisture in the soil at planting time – adequate rains or access to irrigation would allow the use of shorter stakes,
- storage period of the planting material – the shorter the storage period the shorter the stakes can be,
- varietal characteristics – some clones have better sprouting capacity than others, and
- overall physiological and nutritional quality of the planting material.

Alternatively, two-node microstakes can be grown at high density in moist chambers where they sprout. The resulting shoots (15–20 cm long) are harvested after three weeks, their lower sections are immersed in water to produce roots, and then they are transferred to soil.

The facilities needed for an efficient hardening of these small plants are not complex. A cool or fresh environment under shade, where extreme temperatures

can be avoided and adequate humidity provided, should be available. Screen houses are ideal. The plantlets can be transplanted to the field when they are two-months old.

Tissue culture approaches such as the use of meristems and somatic embryogenesis have been used for cassava rapid multiplication (Fregene *et al.,* 2002). These two protocols produce small plants grown *in vitro*, which require a hardening process (Segovia *et al.,* 2002). The critical period for the hardening process, after the plants are taken from *in vitro* conditions, lasts one week. Hardening (using the facilities as described above) starts with the transfer of the small plant from the *in vitro* condition to a container (plastic bags or seedling trays) with a mixture of soil and sand that has ideally been sterilized at 100°C. A high moisture condition is required for the first week after transplanting. Different alternatives have been proposed ranging from the use of plastic disposable coffee cups with small holes at the base and placed inverted over the plant so that it is completely covered, to the use of moist chambers (Fregene *et al.*, 2002; Segovia *et al.*, 2002). After the first week, the plants are gradually exposed to conditions with lower humidity and higher temperature and in two months can be transplanted to the field.

Field management requirements

The process of production of cassava planting material in the field is the same as for root production. It is recommended to keep an area of the production field as a source of new planting material for the next cycle. This area (about 10 percent of the total area) is specially managed following the same criteria as for nurseries designed with that purpose.

Specific multiplication nurseries are planted when a new variety is identified, or when clean planting material is produced from meristem culture of an old variety. In this case, the primary product is the planting material (stakes) rather than the roots. This would be particularly relevant for planting material that has been analyzed as clean of the viral diseases present in Africa or frog skin disease in the Americas. When planting material is confirmed to be disease free, it is important to avoid **allowing** the new, clean crop to have contact with insect disease vectors, such as white flies, in order to prevent their re-infection. Although the use of insecticides may be considered, it is not fully effective. White flies are prevalent in lowland environments and are seldom present at over 1 800 masl. Crop rotations are important in the case of fields that have been affected by root rots because the inoculum would remain in the soil and a new crop would very likely be infected.

The production of planting material needs to be properly managed to avoid lack or excess of water from any source, prevent the attacks by pests and diseases and provide adequate soil fertility. The ultimate objective is to have cassava plants (10–18 months old) that have stems with optimum sanitary and physiological

conditions, well grown and irrigated. Adequate soil fertility is important because it maximizes a quick sprouting of the next generation, with vigorous and healthy plants and a uniform plant stand.

Monitoring nurseries

Production of planting material begins with material that is free from pests and diseases. Before planting, the producer will verify the absence of contaminants and volunteers from previous seasons, as well as the availability of irrigation facilities and drainage in the field. Inspections should take place during the development of the crop: 1 month after planting (MAP) to monitor crop establishment, and then at least every other month until harvest (typically 10–12 MAP).

Under certain conditions, such as when cassava is grown at a high altitude (>1500 masl), with short rainy periods or cold winters (in latitudes > 20°), planting material is harvested from older plants (18 months). In this case, the monitoring visits can be spread.

During the inspections, the whole nursery should be screened for potential sanitary problems. Plants attacked by diseases or pests should be eliminated. Proper availability of nutrients and water should be guaranteed. Weed control should be done carefully, in particular in the first three months of the crop. The varietal purity of the nursery can be checked at 3–5 MAP. The most distinctive descriptors of cassava (colour and length of the petiole, shape of leaf lobules, presence of pubescence in the shoot, colour of the stem) allow for an easy identification of off-type plants that can then be eliminated. For some diseases, such as CBSD or frog skin disease, it is necessary to inspect the roots since they may offer the only source of symptoms that allow for the identification of infected plants (Calvert and Thresh, 2002).

It is recommended to make an official inspection of the plant multiplication nursery at 5–7 MAP. Up to 1 percent of off-type plants can be accepted. For CBB and SED, up to 2 percent of plants with symptoms is acceptable. Depending on disease pressure and the variety being multiplied, CMD and CBSV acceptable levels at that time can range from 0 to 5 percent but this tolerance level should be agreed with the authorizing agency.

Harvesting planting material

Any part of the cassava stem can be used for propagation purposes. However, the thickness of the stem used for cuttings should not be less than one-half the diameter of the thickest part of the stem of the particular variety being used.

Cuttings from green stems (slightly lignified) will germinate, but they are susceptible to attack by pathogens and insects and tend to dehydrate rapidly. Cuttings from stems older than 18 months are too lignified, contain small amounts

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Plate 6
Cross section of a cassava stem showing the relationship between diameter of the pith and total diameter and the latex exudation.

of food reserves, and have reduced viability, delayed and slow sprouting, and/or poor vigor. It is recommended that planting material be taken from stems ranging from 8–18 months of age. The younger the plant, the more lignified the part of the stem selected for the cutting should be. One practical way of knowing whether a stem is sufficiently mature is to determine the relationship between the diameter of the pith and the stem cutting in a transversal cut. If the diameter of the pith is equal to or less than 50 percent of the diameter of the stem, it is sufficiently mature to be used for propagation (Plate 6).

Since cassava has no physiological maturity, it is preferable to maintain the planting materials in the nursery fields rather than harvest them too early and store for 2–3 months. The young branches are cut and discarded and the main stems, offering the quality standards described above, are cut and tied together in bunches of about 50 stems. On average, each stem yields 5–7 stakes. However, depending on age and varietal characteristics, stems can yield 3–12 stakes. There is no dormancy period and stakes can be planted immediately after harvest, when even thin (green) stems could sprout and produce a vigorous plant. Each bunch is identified with a plastic tag with the name of the variety, date and location of harvest clearly written using permanent ink markers or graphite pencils.

At harvest, stems are screened for evidence of insect damage, particularly stem borers. If relevant for the region, roots should be checked for FSD or CBSD.

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Plate 7
Storage of selected stocks to be used for multiplication.

Storage

Stems (about 1–2 m long) can be stored as they have been harvested from the field. Otherwise they can be cut to the proper size for planting (about 20 cm long). To prevent dehydration during storage, it is recommended that the stems be cut into planting stakes just prior to planting. Bunches of long stems should be placed vertically on the ground, in shade (Plate 7) and with the apical portion of the stem up. Sometimes, farmers cover the stem bunches with foliage remaining from the crop to further reduce dehydration of the

stems. The storage area should be shaded and offer high but not excessive relative humidity (about 80 percent) and moderate temperatures (20–30 °C).

Chemical treatment

It is recommended to spray or submerge stems in a protective solution including an insecticide and a up-to-date fungicide (usually copper-based). The solution could also be used on planting material just prior to planting. Stems to be stored for a long time should be treated twice with the same solution: immediately after harvest of the stem and just before planting. It is very important that operators wear protective gloves, aprons, glasses and masks.

Varietal differences

There is genetic variation on sprouting capacity of cassava stakes. Differences are accentuated when the cuttings are stored: the longer the period of storage, the greater the variation. The sprouting capacity of different varieties can be compared after a short storage period of 15 days and thereafter every two weeks for up to three months.

Mechanical damage

The epidermis and buds of cuttings can be bruised or damaged by friction and machete wounds during their preparation, transportation, storage and planting. Each wound is a new potential site of entry for micro-organisms that may cause rots. All precautions should be taken to avoid rough handling when stems are cut and transported. The cuts should be made with a well-sharpened machete or with circular saw, in which case the stem should be held with both hands while it is being cut. Due regard must be given to health and safety measures for the staff working in these operations.

QUALITY STANDARDS FOR THE PLANTING MATERIAL (TABLE 10)

Age of the planting material

Stems should not be stored longer than three months under optimal conditions, preferably less than one month.

Number of nodes per cutting

Each stem node has an axillary bud. Theoretically, one plant can be obtained from each node. It has been found that cuttings with 1–3 nodes have low percentages

TABLE 10
Summary table of standards

Stem age	Less than 3 months storage after cutting
Stem length	18–25 cm
Number of nodes per cutting	5–7
Pith thickness	Up to 50 % of total diameter of the stem
Tolerance to pests and diseases	Not more than 5 %

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Plate 8
Cassava stakes with adequate length and width and 5–7 nodes.

of sprouting in the field. Longer stakes, with 8–10 buds, have a better chance of conserving their potential viability, but require more planting material per unit area. Ideal stakes should, therefore, have from 5–7 nodes and be about 20 cm long (Plate 8).

Pith thickness

The pith diameter should be, as a general rule, 50 percent or less of the total diameter of the stem. When stems are cut into planting stakes, the exudation of latex from the cortex is an indication of good stem condition.

Visual inspection of stems/stakes

A visual inspection of the stems can detect physical damage during storage and/or transport as well as presence of symptoms or signs of diseases and pests. Good quality planting material should have less than 5 percent of stems showing these types of problems.

Sprouting capacity

Quick and vigorous sprouting is the ultimate objective of any scheme to produce cassava planting material. Therefore, a sample of stakes can be tested for sprouting capacity by placing them in plastic bags with soil and adding water to the bag. Sprouting percentage and vigor can be evaluated within 10 days.

Cocoyam

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Xanthosoma sagittifolium (L) Schott
Araceae, subfamily *Aroidea*

GEOGRAPHIC ORIGIN AND DISTRIBUTION

Xanthosoma sagittifolium (cocoyam) originated in the American tropics. It is cultivated in tropical and subtropical zones, between latitudes 30 degrees north and 15 degrees south. It is a globally important root and tuber crop, ranked sixth in planted area and production after cassava, potato, Sweetpotato, yam and taro. The main areas of distribution of the crop include the Caribbean (Cuba, Dominican Republic, Puerto Rico, West Indies), Central and South America; USA (Florida, Hawaii), West Africa (Cameroon, Ghana, Nigeria, Togo), and tropical Asia (Indonesia, Malaysia, the South Pacific Islands).

COMMON NAMES

Gualuza, macal, malanga, malangay, okumo, otoe, quiquisque, quiscamote tiquisque, uncucha, yautía in Spanish. *Mangareto, mangarais, mangarito, taioba* in Portuguese. *Chou Caraïbe* in French. Cocoyam, tannia, taniera in English.

REPRODUCTIVE MEANS

Conventional propagation includes cutting portions from the central corm, cormels or small cormels. These undergo a dormancy period of approximately five weeks, during which sprouting does not occur (Wilson, 1984). The removal of the apical dominance accelerates sprouting of the lateral buds by destroying the apical bud.

PESTS AND DISEASES (TABLE 11)

Dasheen mosaic virus

Dasheen mosaic virus (DsMV) is the most important viral pathogen of cultivated aroids worldwide (Chen *et al*., 2001). It was first reported by Zettler *et al*. (1970) and first identified in Central America in Costa Rica by Ramirez (1985). DsMV is classified as a Potyvirus, family Potyviridae, consisting of a flexuous filamentous particle (less than 700 nm) containing a positive-sense strand RNA genome. The visible symptoms on the plants include leaf distortion, vein chlorosis, mosaic feathering along the veins (Zettler *et al*., 1989) and, with a severe attack, stunted plants.

TABLE 11
Other pests and diseases

Disease or pest	Causal agents	Affected area	Symptoms	Preventive treatments
Bacterial soft rot	*Pseudomonas solanacearum*	Leaves and tuber	Leaf chlorosis, wilting and tuber rot	Use of disease-free planting material. Crop rotation. Use of well-drained soils. Disinfection of tools for cutting the planting material. Roguing.
Tuber soft rot	*Erwinia carotovora* pv. *atroseptica*	Tubers	Tuber soft rot	As above for bacterial soft rot.
Bacterial leaf spot	*Xanthomonas campestris*	Leaves	Early symptoms start close to the leaf margin on the underside of the leaf as small water-soaked spots. In later stages, the leaf spots become brown, necrotic and coalesce, resulting in large irregular necrotic areas with a bright yellow border	Use of disease-free planting material. Overhead irrigation and overcrowding must be avoided. Infected leaves should be removed.
Leaf blight	*Phytophthora* spp.	Leaves	Brown leaf spots with circular shape	Eliminate alternative hosts before planting. Discard infected leaves
Dry rot	*Fusarium oxysporum*	Leaves	Dry rot with cottony appearance	Use of disease-free planting material. Crop rotation with grasses.
Root knot nematode	*Meloidogyne* spp.	Roots	Crown galls	Crop rotation with legumes, e.g. *Mucura pruriens.*
Root lesion nematode	*Pratylenchus* spp.	Roots	Root lesions	As above for root knot nematode.

DsMV is transmitted exclusively by aphids (Brunt *et al.*, 1996) and can spread very rapidly in the field (Pernezny *et al.*, 1993). Although DsMV is not lethal, it retards growth and reduces yield.

Wild species of *Xanthosoma* are sources of the virus. Other genera which are natural hosts include:

- *Aglaonema, Alocasia, Amorphophallus, Arisaema, Cyrtosperma*. Symptoms: mosaic.
- *Cryptocoryne, Dieffenbachia, Philodendron, Richardia, Zantedeschia* spp. Symptoms: mosaic and malformation of the leaves.
- *Colocasia esculenta*. Symptoms: mosaic, chlorosis and dwarfism (Seller *et al.*, 1970).

Control of DsMV depends on:

- use of disease-free planting material;
- chemical control of aphids;

- isolated and non-contaminated planting areas;
- efficient control of alternative host plants.

Root rot disease (RRD)

Root rot disease is the most devastating disease in *Xanthosoma* (Tambong *et al*., 1998) and can cause total yield losses (Saborido *et al*., 2004).

The symptoms are stunting, yellowing of the foliage, and reduction or total loss of the root system. The causal agents are *Rhizoctonia* spp.(Giacometti and Leon, 1994), *Sclerotium rolfsii* (Bejarano-Mendoza *et al*., 1998), *Fusarium* spp. (Saborido *et al*., 2004) and *Pythium myriotylum*, which has been reported as the main causal agent (Tambong *et al*., 1999).

The disease is spread through planting material and infected soil (Nzietchueng, 1984) and the pathogen persists in the soil for many years.

Control of RRD depends on:

- use of disease-free planting material;
- chemical control;
- planting at wide spacing, on high mounds and regulation of the time of planting;
- planting on ridges, also crop rotation;
- use of organic fertilizers.

PROTOCOLS FOR PRODUCTION OF PLANTING MATERIALS

Losses caused by pathogens completely justify a production program of materials free from pathogens. In the case of DsMV, these losses have been always higher than 25 percent and in the case of RRT have reached 90 percent of the crop.

Another important justification is the effect of physiological rejuvenation (Dottin and Perez Ponce, 1998), which could increase the yield by more than 30 percent, including plants infected with virus. In Nicaragua, Reyes *et al*. (2005) reported an 86 percent increase in yields through the effect of rejuvenation.

LABORATORY

Xanthosoma micropropagation is straight forward. Most culture media compositions are well defined, as are the techniques. In addition, virus-free material can be obtained. DsMV is a potyvirus and can be eliminated only via meristem culture. A 70–100 percent virus-free material can be obtained using this method. Other pathogens, including those responsible for RRD are also excluded.

PÉREZ PONCE. 2004.

Plate 9
Micropropagation of Xanthosoma *via Temporary Immersion Systems.*
A. *Systems in production.* **B.** Xanthosoma *explants after multiplication phase.*

Laboratory requirements

A building of approximately 120 m², equipped with a stereoscopic microscope, four laminar flow cabinets and ancillary equipment are needed for the laboratory. The production capacity of a laboratory of this type is between 200 000 and 500 000 plants free of pathogens per year. This production can reach 1 million per year with good efficiency and advanced micropropagation technologies. The personnel required are two specialists (one plant biotechnologist and one plant pathologist), four laminar flow operators and two laboratory technicians.

In order to improve the production efficiency, sunlight can be employed in the growing rooms. Introducing temporary immersion systems' micropropagation technologies can increase the production twofold (Plate 9).

GREENHOUSE

For production of 500 000 *in vitro* plants per year and a rotation of five times per year, the capacity of the greenhouse should have the following:

- total area of 400 square meters;
- effective area of 300 square meters;
- stands or benches for 2 700 trays of 38 holes (stations) each;
- capacity in the number of *in vitro* plants: 100 000;
- cladding (greenhouse covering) of thick plastic for the roof with aphid-proof mesh on the sides, and fan ventilation;
- double door to avoid entry of insects and vectors, footwear disinfection and changes of laboratory coats.

AGRONOMIC PRACTICES INCLUDING ROTATIONS

The planting of *in vitro* plants or seedlings should be done in isolated areas where no species of *Xanthosoma* or *Colocasia* have been planted. For example, areas previously planted with sugarcane have been very efficient for the planting of potato seeds, because of the non-existence of host plants, and minimal aphid populations. In these cases, re-infection has been almost zero, and hence the rotation with sugarcane and other crops that require large areas is recommended.

The main agronomic considerations and practices are the following:

- grow the crop in a range of pH of 5.5 to 6.5 and temperatures between 20 to 35°C;

- temperatures lower than 18°C slow leaf growth while temperatures higher than 35°C increase the foliage, but limit the corms' normal formation;
- plant in isolated areas, preferably areas of sugarcane;
- do not exceed planting density of 10 000 plants per ha;
- make mounts at planting distances of 1.2m x 0.8 m;
- ensure there is good drainage if the area is subject to heavy rains;
- put yellow traps to capture and monitor the aphids;
- specialist should apply insecticides after evaluation of aphid population and virus incidence;
- always plant *in vitro* plants in a new area;
- rotate corms, cormels and plantlets every two years with other crops of the family of gramineae (poaceaes), preferably sugarcane, maize or sorghum.

Other

- Clean tools are essential, use a very sharp knife disinfected with 0.01% sodium hypochlorite each time a new corm is sectioned.
- In the greenhouse, soil should never be used as a substrate because it is a contaminating agent.
- Commercial substrates of inorganic base free from micro-organisms are recommended.
- When there is no possibility to use these substrates, then some substitutes may also be used, such as sugarcane baggage, rice pads or other similar material.
- A substrate with 60 percent baggage well decomposed and 40 percent rice pads has been used for *in vitro* plants of *Colocasia* and *Xanthosoma* with very good results without contamination problems.

FAST MULTIPLICATION

Corms and cormels from *in vitro* plants developed in isolated areas should be used for fast multiplication, and should include the parameters from the basic category as shown in Table 3.

Steps to follow:

1. Select corms and cormels without pest and diseases symptoms, and no mechanical damage.
2. Section material in segments between 15–25 grams.
3. Disinfect with up-to-date fungicides, bactericides and up-to-date nematicides which are allowed and available in the region.
4. Propagate plants either in trays or sand beds.

- Tray system: Use trays with 100-150ml indentations. A soilless substrate is recommended. After 40–50 days, plants should be 12–15 cm tall and ready to take to the field. This is the most recommended option.
- Sand bed system: Induce budding of corms and cormels in a sand bed then transfer 15x10cm plastic bags filled with substrate (avoiding soil mixtures). After 50–60 days, plants should be 20–25 cm high and ready to go to the field.

QUALITY STANDARDS FOR THE PLANTING MATERIAL

Crop monitoring

- To ensure the genetic, sanitary and physiological quality of the materials produced, very vigorous monitoring is required, via the following field inspections:
- inspect field 45 days before planting to verify that the area is isolated from other plantations of *Xanthosoma* and Colocasia, and to take soil samples if there are relevant pathogens;
- inspect the plantation with the objective of evaluating the quality of soil preparation, drainage and proposed planting distances;
- make a second inspection 45 days after planting to evaluate the percentage of survival of planted material, their development, and the presence of pests and diseases;
- make 4–6 more inspections with same objectives, until harvest;
- make negative selections (roguing) to eliminate plants with virus symptoms, one month after planting in the case of *in vitro* plants and plantlets and two months in the case of corms or cormels; eliminate all plants with symptoms of disease, as well as the atypical plants (atypical plants could appear *in vitro* plants as a result of somaclonal variation, and in *Xanthosoma* this should not exceed 1 percent).

FIELD INSPECTION METHODS, SAMPLING RECOMMENDATIONS (TABLE 12) AND TOLERANCES

For most the important diseases, DsMV and RRD, *in vitro* plants should remain at a minimal percentage of infection, if the isolation and negative selection (rouging) requirements are followed correctly. Each program should establish permissible values of infection, depending on local conditions.

TABLE 12
Sampling recommendations

Area (ha)	Number of samples	Number of plants per sample
1	5	100
1–2	6	100
2–3	7	100
3–4	8	100
4–5	9	100

In the case of plantlets, the difference of infection with the *in vitro* plants should not exceed 2 to 5 percent, because under the production conditions, re-infection is

very quick. Hence only planting material of the highest phytosanitary quality should be taken to the field.

The seeds obtained from *in vitro* plants should have 0 percent corms or cormels affected by fungi or bacteria, and a maximum of 3 percent with mechanical damage, insect damage, dehydration or sprouting. In the case of seeds obtained from plantlets, 0 percent should be affected with fungal and/or bacterial diseases. Up to 5 percent is permitted from other damage.

In the case of genetic purity, somaclonal variations should be eliminated by negative selection and at harvest. The genetic purity should be 99 percent for the *in vitro* plants and 98 percent for the plantlets.

Harvest

Xanthosoma is a perennial crop, but for practical purposes, it is harvested after 9–12 months. The growth and developmental cycle can be divided into three main periods.

- Period 1: During the first two months, growth is slow. This period starts with sprouting and ends when the cormels emerge.
- Period 2: The second period is characterized by a rapid increase in shoot growth until 6–7 MAP. It is during this period that the plants achieve their maximum leaf area, pseudostem diameter and height.
- Period 3: During the third period the leaves starts to wilt and the total dry weight of the plant above ground decreases until harvest. This is the moment of major remobilization of photo-assimilates from leaves to the corm and cormels. The senescence of the plant, at the end of this period (approximately 9–10 MAP), is used by farmers as a harvest index.

Harvesting, whether manual or mechanized, should be done when the soil is at medium moisture content so that the damage to the corms is minimal. The corms should not be exposed to the sun for more than two hours. They are then transferred to a shady area for their classification as primary, secondary or tertiary corms. Corms affected by disease, mechanical damage or any other downgrading features are discarded.

Storage

A stock of seeds (corms and cormels) can be comprised of primary, secondary or tertiary corms and should not be mixed in the same stock. The number of corms or cormels per stock should be: primary corms: 1 000, secondary corms: 5 000, and tertiary corms: 5 000.

The storage conditions should be as follows:

- roof and floor constructed of concrete or other impermeable material;

TABLE 13
Summary table of standards

Pests, diseases, and other criteria	Phase 1 (in vitro plants)	Phase 2 (plantlets or *in vitro* plants)	Phase 3 - QDPM (corms, cormels or plantlets)
Genetic purity %	99	99	98
DsMV %	0	3	5
RRD %	0	1	3
Pseudomonas solanacearum %	0	1	2
Erwinia carotovora %	0	0	2
Xanthosomas campestris %	0	0	3
Phytophora spp. %	0	5	5
Fusarium oxysporum %	0	5	5
Meloidogyne spp. %	0	1	2
Pratylenchus spp. %	0	1	2
Weight or height	12–15 cm	12–15 cm (in trays) 20–25 cm (in plastic bags)	70–130 g
Mechanical damage %	0	2	3
Budding %	99	99	98

- no solid walls are required because it is recommended that the plant material is fully ventilated, but away from direct sunlight;
- temperature should between 20–23 ºC with 60 percent relative humidity;
- storage of the seeds should not exceed 45 days;
- stock of seeds should be inspected every 15 days, all those with symptoms of diseases, pests or post-harvest damage and infections should be eliminated (Table 13).

MULTIPLICATION PROGRAMME

Estimated reproductive rate

The multiplication index of *Xanthosoma* is 6–8 plants in a cycle of 10–12 months by the traditional system. The results shown in Table 14 can be obtained using meristem culture and *in vitro* micropropagation.

The programme outlined in Table 15 should be adapted to local conditions, making it possible to include the following alternatives:

- **Alternative 1. Use of *in vitro* plants directly in commercial production.** This alternative can be applied when the laboratory and greenhouse have

TABLE 14
Estimated reproductive rate

Initial material	Duration (months)	Material obtained	Number of units
Meristem	6–8	*In vitro* plant	1 000–10 000
In vitro plant	10–12	Seeds/corms	15–20
In vitro plant	12–14	Plantlets	40–50

TABLE 15
Summary of the multiplication programmeme using micropropagation

Site	Objectives	Time (months)	Multiplication rate	Material produced
Laboratory	Introduction and multiplication	6	1 : 100	*In vitro* plant
Greenhouse	Adaptation of *in vitro* plants	2	1 : 1	Adapted *in vitro* plant (Phase 1)
Isolated area	Field multiplication	10	1 : 1	Basic corms and cormel seed (Phase 2)
Greenhouse	Multiplication of corms and cormels	2	1 : 40	Basic plantlets (Phase 3 – QDPM)
Entire multiplication programme		20	1 : 4 000	
Field	Commercial production	10	1 : 15	Corms and cormels, ware

a high production and quality level. It has the advantage of planting completely clean material with a high increase in yield in a short period of time. Field planting can start eight months from start of the programme. The major disadvantages are the high costs of *in vitro* plants. Evaluations of this alternative are being conducted in Dominican Republic, where more than 100 000 *in vitro* plants of *Colocasia* are being planted in isolated production areas.

- **Alternative 2. Use of corms and cormel sections from *in vitro* plants in commercial production.** Besides increasing the growth period of the *in vitro* plants in the field and the risk of introducing infections if techniques are not done correctly, it can increase the yield 2–4 fold, yielding 15–20 seeds per *in vitro* plant.
- **Alternative 3. Use the corms and cormels of *in vitro* plants to produce plantlets.** The advantage over Alternative 2 is that it doubles the minimum number of plants, hence 40 or more plantlets can be obtained from each *in vitro* plant. It is also safer to have a plantlet with a well-developed root system than a section of corm or cormel. A greater greenhouse capacity is needed for this alternative.

Garlic

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Allium sativum L.
Alliaceae

Garlic (*Allium sativum* L.) is a monocotyledonous species that originated in Central Asia. Initially, this species was classified in the *Liliaceae* family, but recent taxonomic research has identified garlic as *Alliaceae* (Hanelt, 1990). The crop has a worldwide distribution, mostly in mild climates.

Garlic produces bulbs and has a floral-scape stem. The inflorescences seldom set seed but often develop bulbils on the top of the inflorescences (Purseglove, 1975). Garlic bulbs are divided into cloves that together with the bulbils are the propagules of the crop.

Garlic is classed as:

a. Asian or violet for subtropics;
b. pink with low chilling requirements but long photo-periods;
c. white with medium to high chilling requirements and long photo-periods;
d. purple with high chilling requirements and long photoperiod (Burba, 1991).

Garlic varieties also can be grouped as either "hard neck" or "soft neck" The hard neck garlic varieties show a solid, tall scape emerging from the center of the bulb with flowers that often abort and with bulbils at the top of the scape. Hard neck cultivars yield 4–16 cloves per bulb but are difficult to separate due to the hardness of the scape. The soft neck cultivars seldom show a scape and yield 10–40 cloves, but of a smaller size than hard cultivars. Most commercial cultivars are soft neck because of their ease of cultivation, machine planting and long shelf life. However, the propagation rate of garlic depends on genotype and crop husbandry.

The most important pathogens and pests affecting garlic are the following.

VIRUSES

Leek yellow stripe virus (LYSV), garlic yellow streak virus (GYSV) and onion yellow dwarf virus (OYDV) are the main garlic viruses that belong to the Potyvirus Group (Bos, 1982; Walkey *et al.*, 1987). Garlic mosaic is caused by one or more

Plate 10
A. Garlic mosaic. B. Thrips (Thrips tabaci). *C. Mites* (Rhizoglyphus ***spp.***). *D. Mildew.*

of the above viruses. Its main symptoms are chlorotic mosaics especially in young leaves (Plate 10 A). These viruses can be transmitted via cloves and aphids. Tissue culture-derived plants from meristems are grown in clean, isolated fields to minimize the spread of these viruses.

Thrips

Thrips tabaci and *Frankliniella occidentalis* are the main insect pests of garlic. They infest the crop in its early development stages and cause severe damage during bulb formation and bulking (Plate 10 B).

Mites

Rhizoglyphus spp. infest bulbs or cloves, making them flaccid and affecting their shooting ability (Plate 10 C). The mites take approximately 14 days to develop from egg to adult stage and can live up to 121 days. Mites tolerate high temperatures but cannot survive temperatures below 11°C. Mechanical damage should be avoided because it facilitates the entry of *Rhizoglyphus* spp.

Fungi

Onion white rot, *Sclerotium cepivorum*, one of the major garlic diseases, is spreading in the main producing areas worldwide (Plate 10 E). Leaves of infected plants show chlorotic and wilting bulbs. The fungus starts developing in the base and sclerotia, a latent form of the fungus, grows on the surface at a later stage of its development. Sclerotia can survive up to 20 years, even after the plant dies, and can spread via water, equipment, machinery or propagules. Disease develops most easily in low temperatures (15–20°C) and soil humidity (15 percent). Maintaining pathogen-free or clean planting materials and sclerotia-free soil, and cleaning equipment with formaldehyde before moving to another field are recommended for controlling this disease (Delgadillo-Sánchez, 2000).

Downy mildew, purple blotch, basal rot, blue mold rot and rust are fungal diseases of garlic caused by *Peronospora destructor, Alternaria porri, Fusarium oxysporum, Penicillium* spp. and *Puccinia allii*, respectively (Plate 10 D, Plate 10 F). Some of these, such as blue mold rot or basal rot, appear or remain until harvest and even during storage.

Nematodes

Ditylenchus dipsaci (root knot eelworm) symptoms are shortening and thickening of leaves with yellow or brown spots (Plate 10 G). The bulb "neck" becomes flaccid until the bulb rots, galls may be seen in the roots. The use in crop rotation of non-host plants such as carrots and lettuces can reduce the nematode population density in the soil. Treating bulbs with hot water and use of approved safe up-to-date nematicides provide a means of control.

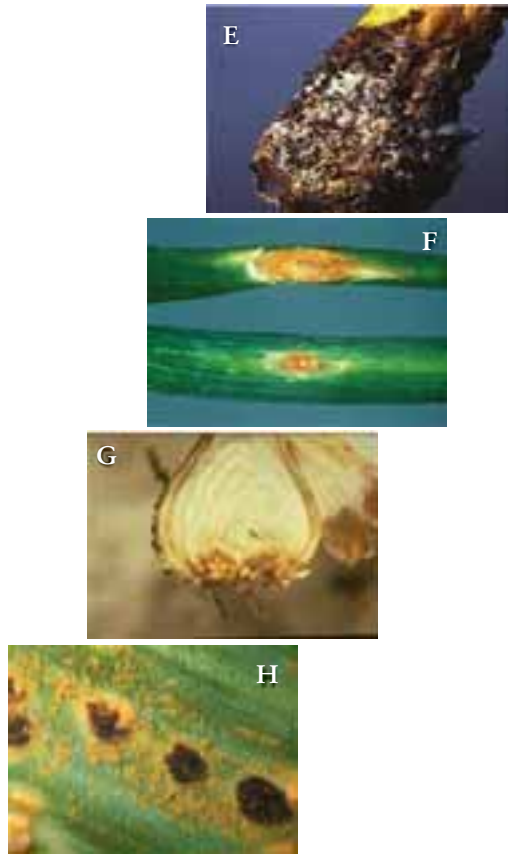


Plate 10
E. Onion white rot. F. Purple blotch. G. Root knot. H. Rust (Puccinia allii).

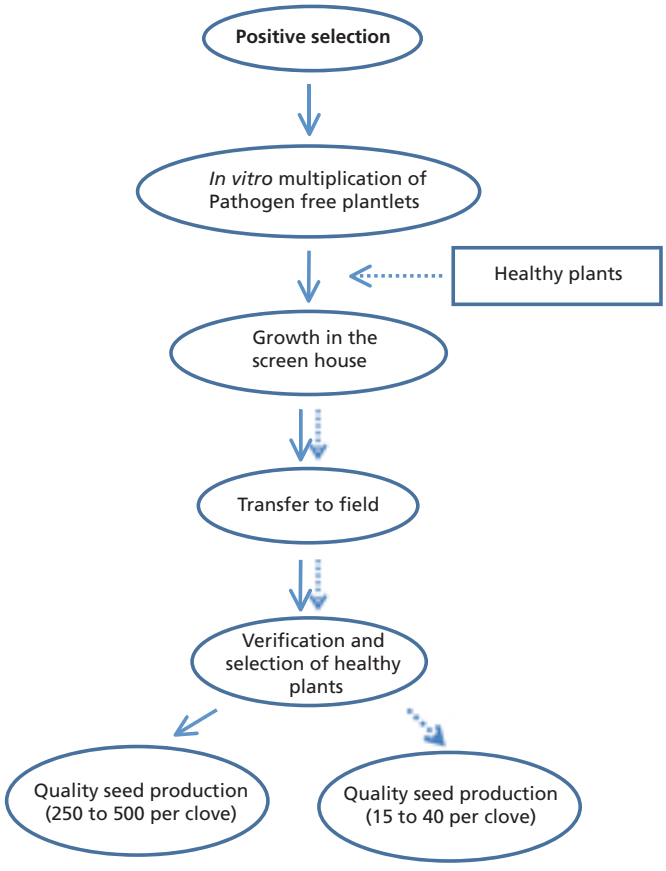
PROTOCOL FOR PRODUCING GARLIC PLANTING MATERIAL

Source material

The source material should be from pathogen-free mother plants after strict selection or from meristem tip culture-derived plantlets. Meristem tip culture allows regeneration of virus-free garlic plants (Walkey *et al.*, 1987; Chovelon *et al.*, 1990). Recent regeneration protocols use meristems from inflorescences or roots because they are virus free (Appiano and D'Agostino, 1983) and are available in larger amounts (30 or more per clove) which allows a high multiplication rate. This regeneration from meristems of roots and inflorescences was successful with garlic cultivars (Haque *et al.*, 1997; Robledo-Paz *et al.*, 2000; Xu *et al.*, 2001; Martín-Urdíroz *et al.*, 2004). The technique involves dissecting meristems from a healthy plant and growing them in a synthetic medium with micro- and macronutrients, vitamins and growth regulators, which allow shoot formation or adventitious embryos. The shoots are induced to form the root system for a whole plant, whereas the embryos are grown in an environment where they can germinate and develop plants (Figure 1).

Recent research results have shown successful transfer of host plant genes resistant to pests and pathogens in garlic through a transgenic approach (Kondo *et al.*, 2000; Robledo-Paz *et al.*, 2004; Eady *et al.*, 2005), and in compliance with national regulations. There are protocols for regeneration of transgenic plants that show host plant resistance to the insect *Spodoptera exigua* (Zheng *et al.*, 2004) or fungi (Robledo-Paz, 2008, pers. comm.).

FIGURE 1
Multiplication scheme for planting materials



The healthy material to be used as pathogen free propagules should be identified through laboratory tests because some pathogens are symptomless in infected plants. The tests will depend on the pathogens or pests known in each country. The bulbs to be used as propagules should not have been stored in cooling or freezing systems. The cloves should be separated from the bulbs 6 to 10 days before planting to avoid dehydration and loss of vigor. Physical damage should also be avoided to reduce the incidence of pathogens. In cases where nematodes and fungi have been observed earlier in the field, propagules may be treated with up-to-date fungicides and nematicides before planting.

There is a correlation between the size of the clove propagules and the harvest of the respective bulbs. Hence the selection and planting of large, sound cloves is recommended. The cloves should be planted in a vertical position.

FIELD FACILITIES AND EQUIPMENT

A planter for mechanical sowing is needed, along with an insect-proof screen house for acclimatizing tissue culture-derived plants before transplanting in the field.

Conventional propagation of cloves or bulbils also can be done in a screen house to ensure high plant health standards.

Field requirements

Selected fields should not have grown onion or garlic crops for at least three years.

Garlic grows in any cool soil with good drainage. Ideally it prefers flat, non-saline, stone-free conditions with pH 6–6.5. Before sowing, the soil's fertility should be determined. Garlic has shallow roots, therefore soil tillage will allow satisfactory seedling emergence and growth.

Field inspections

Crop monitoring should be undertaken at least twice during the cropping season. The first inspection should be when adult plants show their morphology and health status. Each 10 to 15 rows should be sampled by walking through the field and inspecting to ensure that there are no volunteers, infected plants or off-types. The second inspection should be when the leaves start wilting and before bulb lifting. Ten plants from four sites in the field should be taken for thrip assessment – this must be done weekly when there are more than 20 individual thrips per plant. For monitoring white rot, soil samples should be taken at 20 cm depth for each 50 m^2. For detecting *Ditylenchus dipsaci*, the field should be divided into 2–8 ha blocks with samples from 15 to 20 cm depth.

CROP HUSBANDRY

Appropriate field selection, crop rotation and vigorous and pathogen-free propagules ensure a high-yielding crop. The following crop husbandry practices are needed to achieve success.

- **Hand planting** should be in double rows 25 cm apart with 90 cm between each double row.
- **Mechanized planting** is in double rows 30 cm apart with 1 m between them. Cloves are sown 5 to 6 cm deep and 6 to 11 cm apart. The planting can be in rows or beds varying in width that allow planting of up to six rows.
- **Bed width** depends on the field size, the equipment for managing the crop and the irrigation system.
- **Fertilizer application** recommendations in normal soils range from 120 to 300 kg/ha nitrogen (N), 120 to 240 kg/ha phosphorus (P), and up to 185 kg/ha potassium (K), as needed. About 25 percent of the N and 100 percent of

both P and K are applied at planting and the remainder when the plants are 8 to 9 cm high.

- **Weeding** is an important operation and should be done in the early stages of crop development and every 20 days thereafter to avoid yield and quality loss and to control the incidence of pathogens and pests.
- **Irrigation** should be provided every 15 to 20 days in soils with average texture and between 20 to 25 days in clay soils.
- **Scapes** should be removed as soon as they appear to accelerate maturity and avoid yield loss.
- **Tissue culture-derived plants** need acclimatization and growing in the screen house for at least eight weeks before field planting; thereafter they are managed as those from cloves.

HARVEST

Harvesting starts when the lower leaves turn brown and the leaves covering the bulbs dry. Inspecting some of the bulbs before harvest is recommended to ensure that they are of appropriate size and ready for their harvest. At harvest, bulbs are lifted by cutting their roots to about 3–5 cm with a cutter adapted to a tractor or to the plough.

Post-harvest management

Immature bulbs are taken out of the ground by hand or placed at the top of the row and covered with the vines and top soil, or are moved to shade with good ventilation to complete their maturity. The curing process takes one or two weeks, after which roots and foliage are cut, leaving 2 to 5 cm of stem. The curing could also be under hot air at 27°C and 60 to 75 percent RH for approximately 48 hours. After the curing process, damaged bulbs showing any defects or unhealthy symptoms are eliminated, the healthy bulbs are graded by size and packed in mesh bags or corrugated carton boxes according to buyers' requirements. The bulbs can be immediately put on sale or kept in storage.

STORAGE AND TRANSPORT

After putting the bulbs in the containers, they are stored in well-ventilated rooms at 1–2°C and 60–70 percent RH. The shelf life in this environment could be from 90 to 210 days. Any garlic bulbs stored above 70 percent RH may be infected by fungi and show disease. Cloves may start shooting when stored at 4°C, which means their storage at this temperature should be for short periods. Stored garlic bulbs should be monitored frequently to detect any pathogens, pests or changes in the store's temperature. The vehicle for transporting the garlic bulbs should have a good aeration system.

TABLE 16
Summary table of standards

Size of bulbs (minimum diameter)	2.5 cm
Weight of bulbs (minimum)	25 g
Varietal purity (true-to-type, minimum)	99.8 %
Tolerances for pests and diseases	
Ditylenchus dipsaci, OYDV, *Thrips tabaci, Peronospora destructor, Sclerotium cepivorum, Puccinia allii* and *Botrytis porri*.	0.0 %

QUALITY STANDARDS FOR PLANTING MATERIALS (TABLE 16)

Size and weight

Bulbs are grouped according to their diameter:

a. Class Extra or Superior have a minimum diameter of 4.5 cm,

b. Class I includes those of good quality,

c. Class II includes those with a minimum diameter of 3 cm.

A maximum of 3 percent of bulbs below the minimum diameter still can be accepted for all classes. The bulbs to be used as propagules for the next planting season should not be less than 2.5 cm diameter and 25 g weight.

TOLERANCE TO ECONOMICALLY IMPORTANT PESTS AND PATHOGENS IN THE FIELD AND STORAGE

There should be "zero-tolerance" for quarantine pests because garlic propagules must be grown in pathogen-free areas. Which pests are considered "quarantine pests" depends on the country of origin. The tolerance for other pests and pathogens depends on the damage they may cause to the crop in each of the places where garlic is grown or imported. *In vitro* propagules can move to another country if an international plant health certificate is included in the shipment to show the pathogen-free status of the material. The pests and pathogens for which there should be zero tolerance include *Ditylenchus dipsaci*, OYDV, *Thrips tabaci*, *Peronospora destructor*, *Sclerotium cepivorum*, *Puccinia allii* and *Botrytis porri*.

TRUE-TO-TYPE VARIETIES

The percentage of bulbs that are not true-to-type should not exceed 0.1 percent in the first phase of the production programme and 0.2 percent for QDPM propagules.

Hausa potato

Elizabeth Acheampong
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Solenostemon rotundifolius (Poir) J.K. Morton
Lamiaceae

TAXONOMY

Solenostemon rotundifolius (Poir) J.K. Morton, commonly known as Hausa potato or frafra potato is a tropical root crop. *S. rotundifolius* belongs to the family Lamiaceae, subfamily Nepetoideae, tribe Ocimeae (GRIN, 2008). Its synonyms include: *Coleus dysentericus* Baker, *Coleus rotundifolius* Chev and Perrot, *Plectranthus rotundifolius* Spreng, and *Plectranthus tuberosus* Blume.

Solenostemon rotundifolius (Poir) J.K Morton is also known by common names. In English, this includes Hausa potato, frafra potato, coleus potato, country potato, Sudan potato, Madagascar potato and Chinese potato. In French, it is known as *pomme de terre de Madagascar* or *pomme de terre du Sudan*. Other local names: *innala, ratala* (Sri Lanka); *koorka* (India); *tumuku*, (Ghana, Nigeria); *piaha* (Ghana); *saluga* (Nigeria); *kembili, manding-bambara* (Mali); *ketang* (Indonesia); *patata de los Hausas* (Spanish); *vatke* (Ethiopia); *coleus* (Portuguese, Brazil); *Hausa kartoffel* (Germany) (Burkill, 1995; NRI, 1987; GRIN, 2008).

DISTRIBUTION

Hausa potato, a crop of the dry savanna, grows from Senegal to Nigeria and generally in the rest of tropical Africa, Sri Lanka and South East Asia. It is believed to have originated in Africa and from there it was introduced to India, Indonesia, Malaysia and the Philippines (Zeven and de Wet, 1982; GRIN, 2008). It has been widely grown in drier parts of tropical Africa, especially in Western Sudan, Ubangi and the Congo basin, but its cultivation has now given way to plants of the New World such as cassava (*Manihot esculenta*), sweetpotato (*Ipomoea batatas*), groundnut (*Arachis hypogea*) and others (Busson, 1965, cited by Burkill, 1995).

CENTRES OF CULTIVATION

The centres of cultivation of Hausa potato are northern Ghana, specifically the Northern, Upper East and Upper West Regions, and Kita, Mali (Burkill, 1995). Elsewhere, it is an occasional crop of secondary importance. Even though the tuber is nutritional, palatable and has the capacity for yielding on soils of low fertility (Burkill, 1995), Hausa potato is a famine food and is usually harvested

ACHEAMPONG. 2007.

Plate 11
Harvest from three different hausa potato accessions.

and preserved for use during the long dry season. It has been reported that Hausa potato is sometimes used in the treatment of dysentery and certain eye disorders (Burkill, 1995).

The tubers resemble the Irish potato but are much smaller (Plate 11). They are of various sizes and shapes and may be cylindrical, round or elliptical. The large size tubers are about 6.5 cm long by 1.8 cm diameter.

The Hausa potato has an aromatic sweetish flavour and is considered a delicacy. A Hausa potato germplasm collection held at the Plant Genetic Resources Research Institute (PGRRI) of Ghana recognizes four main varieties based on skin colour – white, black, brown and red. Flesh colour was white in all skin types. However, according to the literature, reddish yellow, dark brown and light grey flesh colours also have been reported (Burkill, 1995).

Surveys conducted by Tetteh (1993) and Abapoll (1997) listed reasons for the crop's low production. They identified the labour intensity of the crop, lack of planting materials, small tuber size, low yield, and spoilage under storage, pests and diseases.

BOTANY

Hausa potato is an erect, semi-succulent annual herb. It is bushy from the base, up to 30 cm tall, prostrate or ascending, and has a succulent stem and somewhat thick leaves (Plate 12).

ACHEAMPONG. 2007.

Plate 12
Photograph of 24-day-old hausa potato plant derived from sprouted seed tuber.

It has small flowers, which are blue, pinkish white or pale violet in a distal inflorescence. The flowers are hermaphroditic, produced on an elongated terminal raceme. Small tubers are produced in clusters at the base of the stem.

PROPAGATION AND REPRODUCTION RATE

Hausa potato is propagated vegetatively. Small tubers are usually selected for planting as it is widely believed that larger tubers tend to rot in the soil (Plate 13).

The crop also can be propagated by stem cuttings. However, farmers use seed tubers rather than stem cuttings. Hausa potato also can be propagated by tissue culture (Acheampong and Asante, 1998).

ACHEAMPONG, 2007.

Plate 13
A – selected seed tubers for planting;
B – tubers for food.

CULTIVATION CONDITIONS

The plant requires high rainfall, evenly distributed with low night temperatures. Irrigation is feasible in the dry season (Burkill, 1995). It also requires well-drained sandy loams as heavy clay soils are not suitable. The plant cannot withstand water logging and, thus, is usually grown on broad ridges and mounds (NRI, 2008; PGRRI, pers. comm.)

In northern Ghana, the Hausa potato matures in three months. The crop is planted during the heavy rains in July to August and harvested before the end of the rains in October or November. All 23 accessions held in the PGRRI field gene bank are from northern Ghana and mature within three months, However current information available in the literature and on the Internet reports that the crop matures in five to six months, although it can be up to eight months in India (Burkill, 1995; NRI, 1987). The tubers are formed in clusters at the base of the aerial stems, but stem tillering will produce more tubers where they make contact with the soil.

The Hausa potato is relatively free from pests and diseases in the field. However, it has been reported that *Pycnarmon cribata, Phostria piasusalis* and a leaf folder, *Hymenia curvalis*, have an important presence in India.

PROTOCOL FOR THE PRODUCTION OF PLANTING MATERIALS OF HAUSA POTATO

Hausa potato can be propagated by three types of planting material. The most common is propagation by sprouted tubers but stem cuttings or tissue culture plantlets can also be used. The tubers for planting are usually saved from the previous season's crop.

Source of material

Ideally, the source of planting material should be virus-tested tissue culture plants. However, since the protocol for virus indexing of Hausa potato may not be available, mother plants could be sourced from tissue culture plants or plants identified through positive selection. Large healthy tubers without bruises should be selected and moved to a screen house where the tubers can sprout and the plants can acclimate. Plants raised in screen houses can be explants for tissue culture.

The tissue culture plantlets should be acclimatized in the screen house and used to raise plants, which can then be multiplied by stem cuttings in the nursery. Tubers derived from such plants are used for planting although, more research is needed to verify the efficiency of this practice. Alternatively, large tubers selected for planting could be surface sterilized with sodium hypochlorite to reduce the risk of rotting in the field.

FIELD FACILITIES AND EQUIPMENT

A screen house is essential to acclimatize the tissue culture plants for transfer to the field. A screen house also can be used for producing plants from cuttings.

Field requirements

To ensure plants are disease free, mother plants should be cultivated in a disease- and pest-free environment. A one-year fallow period with at least one tilling can reduce soil pathogens. Hausa potato grows well on well-drained sandy loams. The plant should be grown on raised beds to avoid water logging as contact with too much water tends to cause rotting.

Field inspection

The field and nurseries should be regularly inspected to ensure that there is no outbreak of pests and diseases that would reduce the quality of the planting material. The crop should be weeded twice before the plants cover the bed. However, there should be regular inspection for roguing off-types, diseased plants and weeds.

AGRONOMIC PRACTICES

The crop should be grown on broad ridges about 90 cm wide or on mounds. The seed tubers should be spaced at 25–30 cm intervals. The bed should be maintained free of weeds after planting to reduce the incidence of pests and diseases. Tissue culture plantlets should be fully acclimatized in the screen house for about six weeks before they are transferred to the field. By this stage, they can be planted at the same spacing as the seed tubers.

The soil should be sandy loam with ridges that drain easily; wet but not waterlogged as this would cause the plants to rot; and tilled just before planting so the tuber have adequate moisture at planting. The beds should be fertilized, as Hausa potato needs well fertilized soil. However, under traditional planting where imported fertilizers are beyond the reach of the farmer, the beds are manured with cow dung. After the ridges are well manured, another layer of soil is put on top of the manure in such a way that only the tuber is covered with soil, and the sprouted shoot is out of the soil.

Under ideal conditions, fertilizers are applied to the nursery beds. It has been reported that before planting, NPK fertilizer could be applied at the rate of 125 kg/ha. In Ghana, where the crop has to be rotated with other crops such as

groundnuts and cereals to maintain acceptable yields, it is believed that nematodes may be one of the reasons for the low yields.

Moist soils are preferable and ideally irrigation should be applied. The main reason for delaying planting until August is the irregularity of the rains before this period, but with irrigation, it would be possible to plant seed tubers from April to May. The option of planting stem cuttings of improved material directly on the field in April and May under irrigation needs to be further investigated.

HARVESTING

In Ghana, Hausa potato matures in three months. The tubers are ready for harvesting when the leaves begin to wither and should be harvested as soon as the crop matures. However, harvesting of crop produced on river banks can be delayed for a month or more if the soil is not too dry. The tubers are small and usually range between 1 and 20 g. The crop is normally harvested by hoe and then sorted by hand to avoid bruising.

Post-harvest treatments

Traditionally, seed tubers are selected at harvest. Large tubers are used for food, and small tubers are preserved for planting the following season. This negative selection should be avoided. Seed tubers should be selected from the best. Tubers for planting should be healthy and have satisfactory colour.

At the centre of intensive cultivation in the Upper East Region of Ghana, seed tubers are mixed with dry millet husks, put in earthenware pots and kept in a room for the six months until the next planting season. Sometimes the pot of tubers is kept in a tree. The crop is sometimes spread out on the cool floor of a room or kept on dry leaves under the shade of a tree.

The tubers of Hausa potato can only be stored for two months after which they start to sprout. Once the tubers have sprouted, they are not palatable. Tubers selected for food during the lean season are parboiled and sun dried for use throughout the long dry season, which lasts for six months.

STORAGE AND TRANSPORT

Tubers selected for planting should be stored in cool, dry and well-ventilated area. They can be kept in open baskets or in earthenware pots in a room away from direct sunlight. The basket of tubers should be elevated to avoid contact with ants and other pests. The seed tubers should not get wet during the prolonged storage period otherwise they would rot.

As the seed tubers would be sprouted by the time of planting, extreme care has to be taken not to break the sprouted shoot during transportation. If it is broken, the tuber will not develop another shoot and cannot be planted. In addition, it

PGRRI. BUNSO, GHANA.

Plate 14
Variation in colour, shape and size of five different hausa potato accessions.

would not be palatable, causing the total loss of the production. Ideally, seed tubers should be transported within two months of harvesting before they sprout.

QUALITY STANDARDS FOR THE PLANTING MATERIALS

Size and weight

Optimally, a seed tuber should not be less than 14 g. However, the largest tubers of some accessions are about 3 g. The mean weight of the largest tubers from five PGRRI accessions ranged from 2.52 to 28.2 g while the mean weight of the small tubers (traditional planting material) from the five accessions ranged from 0.9 g to 1.8 g (Plate 14).

Tolerance/risks

Even though there are few known Hausa potato pests and pathogens in Africa, precautions should be taken. One report indicates several diseases affect the crop in India. Because of the risk of inadvertent transfer of pathogens, Hausa potato should be transferred between countries only as virus-tested tissue culture plants. Within countries, tubers should be surface sterilized with sodium hypochlorite to avoid transfer of pathogens from one area to the other.

Varietal purity

At least 98 percent of the Hausa potato plants must correspond or be consistent with the characteristics of the respective parent.

Germination

Acceptable sprouting of tubers should be 95–99 percent (Table 17).

TABLE 17
Summary table of varietal and germination standards

Tuber size (minimum)	14 g
Varietal purity (minimum)	98 %
Germination/tuber sprouting (minimum)	95 %

Konjac

Sivasubramanian Edison
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Amorphophallus konjac K. Koch
Araceae

SCIENTIFIC NAME, ORIGIN AND DISTRIBUTION

Konjac (*Amorphophallus konjac* K. Koch, syn. *A. rivieri*; Japanese: *konnyaku*; Chinese: pinyin: jǔruò), is also known as konjak, konjaku, devil's tongue, voodoo lily, snake palm or elephant yam (this name is also used for *A. paeoniifolius*). It belongs to the Araceae family and is native of the area from warm subtropical to tropical East Asia, Japan and China south to Indonesia. It is an important commercial crop in China, Indonesia, Japan and elsewhere in sub-tropical Asia. Its cultivation is so extensive that its wild origins are obscure. It is currently found in western Yunnan (China) at an altitude up to 1 200 meters, which indicates its cold tolerance. Some literature still refers to this plant as *A. rivieri*, *A. rivieri* var. *konjac* or *A. rivieri konjac*. Konjac, as with most of the *Amorphophallus* species, produces a single leaf up to 1.3 m across, bipinnate and divided into numerous leaflets. The flowers are produced on a **spathe** enclosed by a dark purple **spadix** up to 55 cm long.

Konjac was cultivated in China 1 500 years ago and spread to Japan and other countries, historically for food (flour for noodles and a jelly called "konjaku") but increasingly as a health food for processing and the pharmaceutical industries. Konjac tubers have the unique feature of containing high levels of *glucomannan*, which is the best known natural edible water-soluble fibre. Its high water soluble fiber content combined with low energy value, low vitamin content and low nutritional value make it an ideal food supplement to help regulate body functions resulting from poor diets that induce obesity.

Konjac has great export value. Unlike other *Amorphophallus* spp, *A. konjac* requires a cooler climate and is generally grown altitudes between 500–1 000 m. China, a major producer, has a long history of planting konjac, and Chinese farmers have developed a traditional production system. China's main konjac growing areas are in the Yunnan-Guizhou Plateau and the mountainous areas of Yunnan, Sichuan, Guizhou, Hubei, Guangxi and Shaanxi Provinces in the Yangtze River basin. Also, Fuyuan County in Qujing City of Yunnan Province is one of the major production areas in China.

EDISON. 2006.

Plate 15
Making minisetts of konjac.

COMMONLY USED MODES OF PROPAGATION

The most common method of konjac propagation is by corms or cormels. Flowering and seed setting occurs in *A. konjac*, but true seeds cannot be used for commercial cultivation because of genetic variability. The normal vegetative multiplication rate is 1:4 (Plate 15).

MAIN SEED-BORNE DISEASES AND PESTS

The pests and diseases of Konjac are common for most of the *Amorphophallus* spp. although the incidence, severity and strains vary from place to place. Among the field diseases, mosaic is the most important from a quarantine point of view as it is transmitted through planting material and insect vectors; thus the inocula build-up over a period can pose a serious threat to cultivation. Continuous monocropping also can contribute to build up the disease.

Mosaic-infected plants are generally dwarfed and chlorotic in appearance and exhibit mosaic with mottling which is more pronounced in young leaves. The leaflets become narrow and symptoms of leaf distortion, such as leaf strapping, rat tailing, shoe stringing, puckering and upward curling of leaf lamina, are prominent in severely infected plants. Konjac mosaic virus (KoMV) and Dasheen mosaic viruses (DsMV) are known to infect Konjac plants (Shimoyama *et al.*, 1992). DsMV is transmitted through the infected corms of *A. konjac* and by cotton aphids (*Aphis gossypii*).

Out of 17 species in five families (*Aizoaeceae, Araceae, Chenopodiaceae, Leguminosae* and *Solanaceae*), KoMV has been detected in naturally infected plants in only three Araceae species: *A. konjac, A. oncophyllus* and *A. virosus*. Although *Arisaema serratum* and *Colocasia esculenta* belong to Araceae, they do not host KoMV.

Collar rot, also referred to as southern blight and caused by *Sclerotium rolfsii*, is another important field disease, which becomes more serious if drainage is not efficient. Under hot and humid weather, foliar diseases, especially leaf blight and leaf rot, affect the leaflets of the plants. The leaf blight is caused by *Phytophthora colocasiae* while leaf rot is caused by *Corynespora cassicola*. In addition, bacterial leaf blight caused by *Pseudomonas pseudoalcaligens* subsp. *konjaci* and soft rot caused by *Erwinia carotovora* subsp. *carotovora* are serious diseases transmitted via planting material.

Apart from field diseases, konjac tubers suffer from serious post-harvest rotting due to various pathogens. Mechanical injury during harvesting predisposes the tubers to rotting caused by fungi and bacteria. Over 12 fungal and bacterial pathogens are reported to cause tuber rot. The root knot nematode, *Meloidogyne incognita*, causes dry rot of the tubers, punctures the roots and predisposes the roots and tubers to subsequent fungal infections, and may also reduce sprouting. Although *A. konjac* has no serious insect pest problem in the field, mealybug can cause considerable damage during storage.

PROTOCOLS FOR PRODUCING QUALITY PLANTING MATERIAL

Infrastructure

Facilities

A well-established tissue culture laboratory with an adequate number of greenhouses is the primary requirement. The nucleus seed material should be produced by government organizations or registered seed producers.

Equipment

Standard equipment, glassware, chemicals and miscellaneous items are required for the above facility.

PROCESS

Source of planting material

Apical meristem tissues from the sprouts should be obtained from seed tubers collected from disease-free field crops of known genetic origin. Each country should register and release the konjac varieties after molecular characterization. The plantlets obtained from the calluses should be grown in separate bottles to produce the minitubers, or else transplanted in insect-proof greenhouses after hardening off. It is possible to obtain tubers of approximately 50 g in the greenhouse.

These tubers (nucleus seed) should be inspected, through routine virus indexing, in random lots for the absence of viruses. Such tubers should be multiplied in the farmers' fields of seed villages. The villages should be selected in such a way that they are located near the main konjac growing areas. The planting material thus produced should be properly cured, treated with up-to-date fungicide, packed properly and labelled before transport (Plate 16).

EDISON. 2006.

Plate 16
Planting konjac minisetts.

FIELD REQUIREMENT

Seed production plots should not have grown konjac for at least three years and

should be free of volunteer konjac material. The registered seed growers should grow the seed crop in minimum 0.5 ha plots. The soil type should be a fertile sandy loam or sandy clay loam with good drainage. The soil should not have more than ten colony forming units (CFUs) of soft rot (*Erwinia carotovora*) and southern blight (*Sclerotium rolfsii*) pathogens. A certificate of soil analysis for the pathogens and also for the nutrient status of the soil should be obtained before accreditation for seed production.

Field inspection

Seed certification officers should periodically check the crop at different stages of crop growth. The planting material produced would be approximately 200–300 g, which should be the ideal size for commercial cultivation. In practice, farmers sell corms weighing 200 g and above to factories, as they prefer to use smaller cormels as planting material. However, if they used 200–300 g corms as planting material, their net income would certainly be higher. The planting material produced in the registered seed growers' fields should be properly cured, soil and roots removed, treated with up-to-date fungicide, properly packed, labelled and certified before transport.

Officially designated inspectors should visit the seed crop at least three times during the cropping season – at seven days after planting, one month after planting and finally one month before estimated harvest. Before entering the field, the inspector should confirm with the seed grower the exact location of the seed field, the variety, which it is said to be, and the previous cropping of the field. Fields of more than 5 ha should be divided into areas of 5 ha maximum each and inspected separately.

AGRONOMIC PRACTICES

Planting pits must be prepared to an appropriate size to hold the seed corm size of 200–300 g. The pit must be filled with well-rotted farmyard manure (FYM) or other organic manure at the rate of 12.5 tonnes per ha and then covered with soil. A fertilizer application of 27:20:23 kg per ha of NPK is recommended. The ideal spacing is 90 cm x 90 cm with 8 000 to 9 000 plants/ha. Adequate care must be taken to control any observed foliar diseases with appropriate chemical control measures, especially leaf blight, leaf rot and bacterial leaf blight. These practices will contribute to high tuber yields.

EDISON. 2006.

Plate 17
Konjac in store.

HARVESTING, CURING, POST-HARVEST TREATMENTS AND PACKING

The seed crop should be harvested only after complete drying of the plants.

Irrigation should be withdrawn at least one month before harvest. In the event of rain, harvesting should be done after the soil has completely dried. The harvested seed tubers should be cured for seven days in a ventilated, shaded and dry place (Plate 17). The tubers should be properly cleaned, treated with up-to-date fungicide and packed in layers in cardboard boxes of 10 kg capacity. Inert material should be placed between the tuber layers to avoid damage during transport. The cardboard boxes should have ventilation holes in their sides.

Uniform-sized whole tubers ranging between 200–300 g should be selected as seed tubers.

The seed tubers produced at the primary centre and in the farmers' fields should be randomly indexed for viruses and other pathogens. The seed certification agency should have the planting material evaluated in an official seed-testing laboratory.

SUMMARY OF QUALITY STANDARDS FOR THE PLANTING MATERIAL (TABLE 18)

TABLE 18
Summary table of standards

Weight of propagules	200–300 g
Varietal purity	98 %
Germination	99 %
Maximum tolerances for pests and diseases	
Mosaic disease (in field)	1 %
Collar rot (in field)	5 %
Other pests and diseases (in storage)	5 %

Potato

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Solanum tuberosum L.
Solanaceae

GEOGRAPHIC ORIGIN AND DISTRIBUTION

The potato (*Solanum tuberosum* L) originated in the highlands of South America. The world's fourth most important food crop after rice, wheat and maize, the potato is cultivated in 157 countries in the tropical, subtropical and temperate zones of the world. Eight species are cultivated. The most common is *S. tuberosum*, a tetraploid (2n=4x=48) which produces tubers under long-day conditions and is cultivated worldwide. The other seven species are found mainly in the Andes and produce tubers under short-day conditions. These include the tetraploid subspecies *S. tuberosum andigena*, the diploid (2n=2x=24) species, *S. phureja*, *S. stenotomum*, *S.goniocalyx*, *S. ajanhuiri*, the triploid (2n=3x=36) species *S. chaucha* and *S. juzepzuckii,* and the pentaploid (2n=5x=60) *S. curtilobum*. Approximately 187 wild *Solanum* species, which are closely related to the potato, are distributed from the United States of America (USA) to the south of South America.

REPRODUCTIVE MEANS

Potato is traditionally grown from tubers, but it also can be grown from other vegetative organs such as stems or sprouts, and also from true seed.

The tuber is an underground modified swollen stem that serves as a storage and reproductive organ. It develops from the tip of the stolon, which is an elongated underground stem, 35– 50 days after plant emergence. A potato plant has a multiplication rate ranging from 1:10 to 1:15. The tuber has buds, also called "eyes", arranged in a spiral manner, from which sprouts and shoots develop. After harvest, the tuber undergoes a dormancy period of two to three months, during which development is temporarily suspended. Some species, such as *S. phureja* and *S. chaucha,* have a short or no dormancy requirement. Tuber dormancy can be broken by an application of various treatments including the exposure of tubers to changing high and low temperatures or chemical treatments including gibberellic acid. Tubers of 40–60 g are commonly used as seed potatoes, larger tubers are cut into two to four pieces containing at least one eye. Several techniques using other reproductive parts of the plants have been developed to increase the propagation rate.

Stem cuttings consisting of at least one node are commonly used as a rapid multiplication technique. The stem has nodes, each with three buds in the axils of leaves. The buds may grow out to form lateral stems, stolons or inflorescences, aerial tubers or a new plant. Stem cuttings are harvested when the mother plant is 20–30 cm tall. A mother plant can yield 15 to 20 single nodal stem cuttings. A root-inducing hormone can be applied to the cutting to improve rooting and establishment.

Microtubers or *in vitro* tubers of 1–3 g can be produced from an *in vitro* plant. Adoption of microtubers as a seed propagule has been uneven globally and there is a lack of consensus on optimal production practices for microtubers. Microtubers require care and are not suitable for field planting, but they can be used as planting material to produce minitubers or in combination with other techniques for rapid multiplication in a greenhouse.

PHYSICAL AND PHYSIOLOGICAL FACTORS AFFECTING QUALITY OF PLANTING MATERIAL

Physiological age of the tuber

There are three physiological ages of tubers, *viz*. apical dominance, multiple sprouting and multiple branching. Following the end of tuber dormancy, the tuber develops a single sprout at the rose end, which is opposite the stolon end of the tuber. As the tuber ages, apical dominance is broken and other buds start sprouting. This tuber with multiple sprouts is ready for planting. With further tuber aging, the sprouts develop multiple branches and the tuber may dehydrate significantly and, in fact, planting a senile tuber with multiple branches will result in failure of plant emergence or a weak plant with many stems and lower tuber yield. Plants of very senile tubers produce few potatoes.

Management of mother plants for rapid multiplication of cuttings

The physiological age of the mother plant and cuttings is an important factor in the production of cuttings. They should always be kept juvenile. Old mother plants that are in the process of tuber formation often produce physiologically old cuttings that will develop into small plants. These small plants often have a very early tuber formation and produce few small tubers. A mother plant derived from a tuber or from old cuttings will form compound leaves. It is preferable to harvest cuttings at 35–40 days after planting the mother plants – when the mother plant has not yet produced tubers. Tuberization may be delayed by extending the photoperiod to 16 hours.

A juvenile mother plant derived from an *in vitro* plant has simple leaves and, as a source of apical stem cuttings, is preferable to a physiologically older mother plant that has compound leaves. Apical stem cuttings from juvenile mother plants will develop into vigorous plants and yield a high number of tubers, whereas those derived from older mother plants may develop into small plants and yield a lower

TABLE 19
Identification, detection, natural spread, field symptoms, alternate hosts, control methods and/or any other element useful to characterize the diseases/pests

Disease/pest	Causal agent	Affected area	Disease symptoms	Preventive treatments
Severe mosaic	*Potato virus Y*	Leaves and tuber	Variable according to strain and variety. Mild to severe mosaics on leaves including leaf distortion. Necrosis of veins, leaves may shrivel and drop (primary symptom). Stunting. Tubers may show cracking or surface rings and internal necrosis in the case of infection of sensitive varieties with PVY (including N and NTN) and other necrotic strains.	Planting clean seed, isolation and hygiene. Use of resistant varieties. Insecticides are of little value in control of PVY due to very rapid transmission by migrating aphid vectors.
Leaf roll	*Potato leaf roll virus*	Leaves and tuber	Rolling of lower leaves which may feel "crispy". Purplish colour of young leaflets in primary infection. Net necrosis in tubers of some varieties.	Planting clean seed, isolation, hygiene and use of insecticides (aphicides). Use of resistant varieties.
Mild mosaic	*Potato virus X*	Leaves	Mild mosaic pattern of light and dark green on leaves. No leaf distortion. Can cause severe symptoms in mixed infections with other viruses.	Planting of clean seed and use of resistant varieties.
Black Leg	*Erwinia carotovora* pv. *atroseptica*	Stems and tubers	Non-emergence or stunted pale green or yellow foliage, rolled upper leaves, black stems and foliage easily pulled out. Tuber soft rot (black) extending from heel end.	Planting of clean seed.
Potato cyst nematodes	*Globodera. rostochiensis* and *G. pallida*	Whole plant	Stunted weak plants with a tendency to wilt. Small bead-like cysts (just visible to naked eye) attached to roots and tubers.	Crop rotation and the use of resistant varieties, if available.
Powdery scab	*Spongospora subterranea*	Tubers	Tubers erupt with ragged-edged scabs more circular than common scabs.	Crop rotation. Planting of clean seed and use of resistant varieties.
Stem canker and black scurf	*Rhizoctonia solani*	Stems and tubers	Brown to black particles on tuber skin. Stem bases with brown cankers which can circle stem causing leaf rolling and wilting.	Crop rotation. Planting of clean seed and use of resistant varieties.
Bacterial wilt	*Ralstonia solanacearum*	Tubers	Vascular rings of tubers go brown, leading to rotting of whole tuber.	Planting of clean seed, avoidance of irrigation with contaminated water. Conformance to local minimum rotation requirements

tuber number due to precocious tuber formation. Continuous cutting of apical shoots and an extended photoperiod of 16 hours will ensure mother plants stay juvenile with simple leaves. The main seed-borne diseases and pests including their life cycles are outlined in Table 19.

PRACTICES TO PRODUCE GOOD QUALITY PLANTING MATERIAL

Field selection/crop rotation

For seed production, it is preferable to select a field that has not been planted with any Solanaceous crop for at least three years. Ideally, choose fields that are isolated from other potato (ware) crops and sites within traditional (elevated) seed production areas. Find out if the field has any history of potato diseases,

particularly bacterial wilt. This information will be useful to take the necessary measures to prevent or control soil-borne pathogens. The national potato programme or other specialists in an area may be able to arrange soil testing for bacterial wilt before planting.

PLANTING MATERIAL

Always use healthy, preferably certified, seed as planting material. This helps minimize disease risks. It is a good idea to identify the source of potential planting material a year or more in advance and to visit the seed field, if possible, to assess the health and management of that crop. Always ask questions about the seed source, including the number of field generations, and ask for the results of any testing or inspections that have been made. National authorities may be able to provide the results of any testing or inspection.

AGRONOMIC PRACTICES

Weeding and fertilizer applications, and chemical applications for pest control operations should be implemented as recommended by competent and trained staff. Inspect the crop regularly for the presence of colonizing aphids that transmit viruses by examining the undersides of leaves. Avoid mixing varieties, and rogue all off-types and volunteers. Potato volunteers are a source of many diseases and must be controlled in seed production sites. Seed field entry should be restricted to the minimum.

HARVESTING

Tubers should be harvested when they are physiologically mature, which is when the peel (skin) is well set. Immature tubers are prone to peeling during harvest and storage operations, which puts it at risk of disease infection. Tubers can be induced to mature by cutting, pulling or killing the haulms 10–20 days before harvest. Early killing of haulms also can be used for pest control (particularly aphid borne viruses) and controlling seed size. Avoid harvesting the seed potatoes when the soil is wet or during rainy days, as the tubers will carry soil and be at risk of disease infection. If tubers are harvested wet, they should be dried before storage. The drying should be done away from direct sunlight and heat.

STORAGE

Cure the seed potatoes for two weeks to suberize the outer layer of potato peel and thicken the periderm. This reduces infection and water loss. Before storage, seed potatoes should be sorted to eliminate damaged, diseased and off-type tubers. Repeated seed examination and sorting is recommended during storage. In general, seed tubers must be stored in a protected facility, avoiding direct exposure to sunlight and extreme changes in temperature and humidity. Seed potato tubers can be stored in cold rooms (5–10°C) with humidity control to extend storage life and reduce dehydration. Another option is to use diffused-light storage, in particular in tropical and subtropical regions. Diffused light storage reduces

sprouting and weight loss of the tubers during storage, helps tubers turn green which increases resistance to pests, and allows farmers to store the seed for longer periods and thus increases yields. Storing tubers in the dark results in long weak sprouts and low tuber yield.

SEED MULTIPLICATION PROGRAMMES

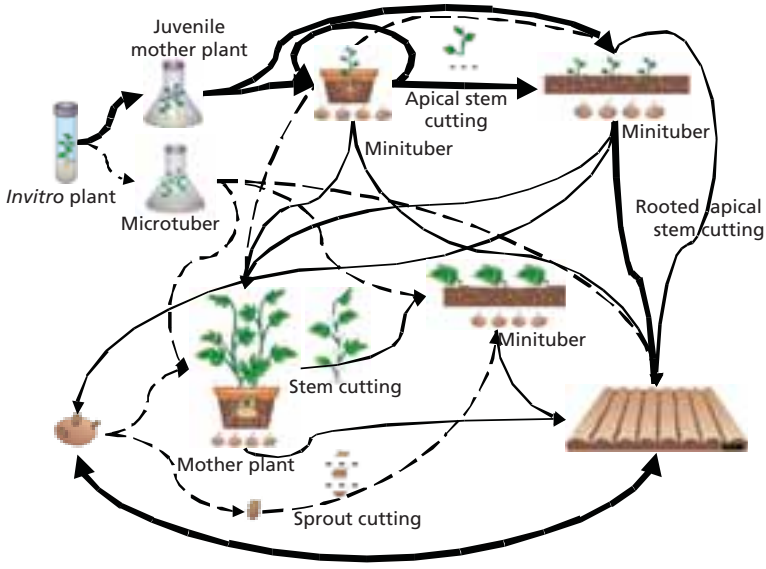
Unlike many clonally propagated crops, seed potato legislation exists in many countries. Hence, the QDPM scheme for potato should consider local legislation. Certified or formal potato seed programmes started as early as the 1900s in Germany and 1914 in the USA.

Sophisticated seed organizations and regulations exist in many countries, which usually have one mandated agency with the legal base to manage and coordinate all phases of the programme. These agencies are typically responsible for organizing seed producers, and formulating and enforcing regulations, field and storage inspections and labeling. Certified seed is often classified in categories such as "prebasic" (highest health status), "basic" and "certified" following each cycle of field multiplication. Seed is entered into the appropriate class and inspected by competent persons according to criteria specified for that class. Failure to meet the specified standards results in rejection or the possibility of re-entering the seed at a lower class if available.

Certified seed programmes are not fully standardized among countries. However, some regions are initiating efforts to harmonize seed categories, the number of field generations within each category or class, and tolerance levels. In developing countries, farmers commonly use their own saved seed and often less than 5 percent of seed is certified. Many agencies promote training farmers to recognize common seed-borne diseases and to practice positive selection of healthy mother plants and/or negative selection with the removal or roguing of infected plants. Inevitably seed stocks decline in vigor as diseases accumulate over time (degeneration). Replanting with certified or QDPM-derived seed may represent a sound investment for the potato producer depending on prevailing economics. As potato producers periodically lose their seed as a result of natural disasters or civil unrest, agencies participating in restoration of seed supplies should consider sourcing seed of an appropriate quality standard where possible (Figure 2).

In vitro plants are used as a source of juvenile mother plants to produce apical stem cuttings or are directly planted into a nursery or in soilless media such as in aeroponics, to produce minitubers. In turn, minitubers are planted in-field to produce tuber seed. Tuber seed is traditionally planted in-field but also can be used as source of sprout cutting or mother plant to produce stem cuttings and minitubers (Plate 18). A combination of propagation techniques may increase seed multiplication rate substantially (Table 20). Thicker lines and broken lines show, respectively, common and least common pathways.

FIGURE 2
Vegetative propagation of potato and seed production



CIP. 2006.

Plate 18
Sprout cutting from tubers and apical stem cuttings with simple leaves from juvenile mother plant.

STANDARDS FOR CONVENTIONAL FIELD MULTIPLICATION OF POTATO

Inspection methods including varietal purity (Table 21 and 22)

Timing and number of inspections

Two field inspections and one post-harvest inspection are recommended. The first should be made when the variety to be inspected has developed its full characteristics, normally before flowering. This is typically the point when plants are touching within the row but just before they meet across the row. The inspection should be made early in the morning or under overcast conditions to facilitate recognition of virus symptoms. The second inspection should be made at least two weeks later, usually when flowering has finished. A third tuber inspection should be performed post-harvest. Crops that are likely to be irrigated should be inspected at the optimum growth stage. If possible, inspection should be done by trained personnel from a seed agency, extension agency, national programme, local government, or suitable community or grower group representatives. If self certified, then this should be indicated on the label.

ROTATION

The minimum requirement for rotation is four years without potatoes on the site.

TABLE 20
Rate of multiplication for potato propagation techniques

	Source	Product	Harvest time	Estimated mean multiplication rate	Comments
Field	Tuber seed	Tuber	90 days	10:1	
	In vitro plant/ node/ apical stem/sprout cutting	Tuber	90 days	8:1	Management of i*n vitro* plant /cuttings to avoid precocious tuberization and low tuber number yield.
Greenhouse	40-day-old plant	Stem node cutting	40 days	15:1	Avoid harvesting old cuttings.
	Mother plant derived from *in vitro* plant or cutting	Apical stem cutting	Every 10 to 15 days	25:1	Maintain mother plants as juvenile. Use foliar fertilizer. Extend photoperiod.
	Tuber	Sprout cutting	14 days	25:1	
	In vitro plant/ node/ apical stem/sprout cutting	Minituber	90 days	10:1	Requires careful management of i*n vitro* plant/cuttings to avoid precocious tuberization and low tuber number yield. High density planting (50–100 plants/m²) to produce 350–900 minituber/m².
	In vitro plants for aeroponic culture	Minituber	90 days	30:1 to 80:1	High-density planting (16–67 plants/m²) to produce 1 200–2 000 mini-tubers > 1.5g/m².
Laboratory (*in vitro*)	*In vitro* plant	*In vitro* plant	30 days	5:1 to 10:1	*In vitro* plants can be continuously multiplied.
	In vitro plant	Microtuber	45-60 days	1:1	High density of *in vitro* plants may yield a high number of microtuber/m²

TABLE 21
Tolerances (field inspection)

Disease or defect	Tolerance
Incorrect variety	1 %
Leaf roll (virus)	5 %
Severe mosaic (virus)	5 %
Total severe virus (leaf roll + severe mosaic)	**10 %**
Mild mosaic (virus)	10 %
Total virus	**10 %**
Blackleg	2 %
*Bacterial wilt**	nil

* If a plant with bacterial wilt is found, the diseased plant should be clearly indicated; do not rogue the diseased plant to avoid spreading the disease in the field; avoid entering the field; harvest the healthy plants first; do not harvest the eight plants immediately adjacent to the diseased plant along the row and adjacent two rows. Sort out the seed for external brown rot symptoms at harvest, before storage, two or three times during storage at one month intervals, and again when preparing the seed before planting. If the situation is more serious and there is nothing to rogue because the tubers are in a state of putrification due to the disease, the field should be abandoned or should go through rotations that reduce bacterial wilt, avoiding solanaceous crops as rotation choices.

MINIMUM ISOLATION REQUIREMENTS

Seed crops to be inspected against the QDPM grade should be isolated from other potato crops or other solanaceous crops by a minimum of 50 m. Inspectors should

also note the presence of ground keepers and weeds. If the potato haulm cannot be properly inspected because of the presence of weeds, then the crop should not be granted QDPM status.

GROWING SEASON INSPECTIONS

Inspection

Inspections for QDPM should include examination of 10 counts of 100 plants each for crops of 2 ha or less. The inspector should make counts while walking diagonally across the field. An additional 10 counts of 100 plants each should be examined for every additional 2 ha of crop. A post-harvest tuber inspection should also be performed (Table 22).

Aphids

Inspectors should examine the undersides of leaves for aphids, note any presence and estimate the degree of infestation. Infestations should be recorded as slight (one or two aphids on a few plants), moderate (a few aphids on most plants) or severe (many aphids on some plants). Inspectors should recommend early haulm destruction for crops with moderate or severe infestations.

Roguing (negative selection)

Growers may rogue any obviously infected plants for which there will be no maximum tolerance prior to inspection. Inspectors may also rogue plants

TABLE 22
Tuber inspection tolerances (post-harvest)

Disease, pest or defect	Individual tolerances	Group tolerance
*Wart disease (*Synchytrium endobioticum*)*	Nil	Nil
Nematodes		
Bacterial wilt		
Ringrot		
Potato spindle tuber virus		
Colorado beetle		
Blight *(Phytophthora infestans)*	1 %	5 %
Blackleg *(Erwinia carotovora* subsp. *atroseptica* or *Erwinia chrysanthemi* or both)		
Watery wound rot *(Pythium ultimum)*		
Pink rot *(Phytophthora erythroseptica)*		
Dry rot *(Fusarium* spp.*)*		
Gangrene *(Phoma* spp.*)*		
Powdery scab *(Spongospora subterranea)*	5 %	8 %
Black scurf (*Rhizoctonia solani*)		
Common scab *(Streptomyces* spp*)*		
Tuber necrosis caused by strains of PVY	0.5 %	0.5 %
Soil	2 %	2 %
Off-type – Tuber seed from field	1 %	1 %
Off-type – Minituber and microtuber	nil	nil

while inspecting the crop but should count any infected plants against the tolerances shown.

Haulm destruction

In general, the haulm should be destroyed 3–4 weeks after the second inspection. This could be reduced to two or even one week if aphids are present and the tubers have bulked to sufficient size.

Tuber size

Seed size may influence the tuber yield of a potato crop. Plant emergence, vigor, growth and tuber yield are related to seed size. In general, larger seed sizes result in higher total yield than smaller sizes. However, the benefits of using larger sized seed diminish as the size of seed increases above 70 g. The optimum seed size depends on factors such as the availability and cost of seed, plant density and produce market price. Overall, seed size of 40–70 g will provide optimum returns. Seeds smaller than 40 g are less productive than larger seeds. Seeds larger than 80 g increase seed costs.

CIP. 2006.

Plate 19
Sprout cuttings from tubers.

CIP. 2006.

Plate 20
Minituber production in aeroponic system.

Microtubers and minitubers are smaller than conventional tuber seed but their high health quality may compensate for the lower tuber yield expected from smaller seed size. Microtuber size often ranges from 0 to 1 g; microtubers of at least 0.5 g can be planted superficially at 3 cm depth in the field. Minitubers, whose sizes often range from 0.5 g to 30 g, can be sorted into two sizes to facilitate planting and uniform management by seed size class. The smaller size of minitubers ranging 0.5– 5 g can be planted superficially at 3 cm depth, while the minitubers of 5– 35 g can be planted deeper (Plate 19 and 20).

Bags

Seed potatoes should be stored and transported in new and unused bags.

Sweetpotato

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Ipomoea batatas (L.) *Convolvulaceae*

ORIGIN

Sweetpotato, *Ipomoea batatas* (L.) Lam., was domesticated in or near South America around 8 000–6 000 BC. Colombia, Ecuador, Guatemala and northern Peru have the greatest diversity in sweetpotato germplasm. Secondary centres of genetic variability are Papua New Guinea, the Philippines and parts of Africa.

MODES OF PROPAGATION

The sweetpotato can reproduce asexually by:

i. storage roots that subsequently sprout to give new plants; and
ii. vine cuttings that form roots at the nodes, producing daughter plants.

The sweetpotato also can reproduce sexually by seed, but this is used only for breeding purposes. Sweetpotato is a perennial but is cultivated as an annual for vines and storage roots. It has a photoperiod of 11.5-hour day length or less to promote flowering, although with a 13.5-hour day, flowering ceases but the storage root yield is not affected. Short days with low light intensity promote root development.

The sweetpotato's estimated reproductive rate using vine cuttings is 1:15 to 1:20. At optimum conditions, one tissue culture plantlet can produce 64 000 cuttings, which is enough to plant an 800 m^2 field plot in one year.

PESTS AND DISEASES

A wide range of pathogenic organisms attack the sweetpotato plant. Although most are widespread, the damage levels they incur are variable. These organisms include viruses, fungi and bacteria, and damage caused by nematodes.

Globally, at least 20 viruses are known to infect sweetpotato individually or as mixed infections. Sweetpotato feathery mottle virus (SPFMV) is the most common. In mixed infections, sweetpotato chlorotic stunt virus (SPCSV) and SPFMV can

MARTIN, 2006.

Plate 21
Root knot nematode.

be associated with the severe sweetpotato virus disease (SPV), the most important disease of sweetpotato in Africa. Other viruses include: sweetpotato mild mottle virus (SPMMV), sweetpotato latent virus (SPLV), sweetpotato chlorotic flecks virus (SPCFV), sweetpotato virus G (SPVG) and sweetpotato leaf curl virus (SPLCV). Whiteflies and aphids act as vectors for some viruses.

Bacterial diseases can be economically damaging and include bacterial stem and root rot (*Dickeya dadantii*) in some parts of the world. Bacterial wilt (*Pseudomonas solanacearum*) is important in southern China and soil rot (*Streptomyces ipomoea*) is important in parts of the United States of America and Japan. Control measures such as good crop hygiene and resistant varieties are common recommendations.

Root-knot nematodes (RKNs – *Meloidogyne* spp.) occur worldwide (Plate 21). The extensive RKNs and their interactions with pathogenic fungi and bacteria in plant disease complexes rank RKNs among the major pests. Nematode attacks of sweetpotato cause stunting, yellow foliage, abnormal flower production, round- to spindle-shaped swellings (galls), necrotic root systems and low yields. More than 50 species of RKNs have been described, but *Meloidogyne incognita*, *M. javanica*, *M. arenaria* and *M. hapla* account for more than 95 percent globally.

Worldwide, there are at least 270 species of insects and 17 species of mites that feed on sweetpotatoes. Insect pests are categorized as defoliators, virus transmitters, stem borers and root feeders. Sweetpotato weevils, *Cylas* spp., (Plate 22) are the main pests. Worldwide there are three main economically important sweetpotato weevils: *Cylas formicarius* occurs globally, while *C. puncticollis* and *C. brunneus* are the main species in Africa. The West Indian sweetpotato weevil, *Euscepes postfasciatus*, occurs in Central and South America, the Caribbean and the Pacific Islands.

The most damaging stage of weevils is the larval stage. The larvae mainly attack stems and underground parts, but may also feed on leaves. Adult weevils oviposit at the base of vines and in exposed roots, while the larvae tunnel through storage roots causing major economic losses. The damage caused by larvae and adults also stimulates the production of terpene phytoalexins, which make the storage roots toxic for human consumption. The weevil population and its damage are most prevalent during dry seasons, probably because drought increases soil cracking, thus exposing roots to weevils.

PROTOCOL FOR PRODUCTION OF PLANTING MATERIAL

Facilities and equipment

Established protocols, including those specific for a region or country or those developed by the International Potato Center (CIP), are followed to clean up sweetpotato material derived from various sources including field, screen house or tissue culture. The protocols produce virus-indexed plantlets for local, regional and international use (Figures 1 and 2). Important facilities include well-equipped greenhouses or screen houses, tissue culture laboratories, virus-detection equipment and indicator plants for virus indexing. Basic equipment for tissue culture includes: autoclave, laminar flow cabinet, pH meter, sensitive balances, refrigerators and heaters. A growth room for *in vitro* plantlets can be constructed locally. The size of fields for multiplication and increase of stocks of clean plants depends on the demand for clean planting materials and the capacity of the country, organization or agent to meet the demand.

SMIT. CIP, 2006.

Plate 22
Sweetpotato weevil.

AGRONOMIC PRACTICES

In the tropics, sweetpotato is propagated from vine cuttings, but in temperate regions it may also be grown from rooted sprouts (slips) pulled from bedded storage roots. Apical cuttings 30–45 cm long are planted by inserting them into the soil at an angle. In some parts of East Africa, cuttings may be wilted or left in the shade for a few days. In India, the central portion of a cutting is buried in the soil, leaving a node exposed at each end.

Sprouts are obtained by planting small- or medium-sized roots close together in nursery beds. The resulting spouts are removed from the storage roots when 22–30 cm long and planted in the field. Cuttings and sprouts are planted on mounds, ridges or on the flat if the soil is deep and well drained. Mounds are used extensively in the tropics, especially where the water table is high. Mounds up to 60 cm high, and 90–120 cm apart are planted with three or more cuttings. Ridging is suitable for mechanical preparation of the ground. Ridges are about 45 cm high and 90–120 cm apart, with cuttings planted at 30 cm intervals.

Sweetpotato is often the lead crop in a rotation cycle. The exception is in very fertile soils where planting sweetpotato at the start of the rotation should be avoided, as excessive vegetative growth occurs at the expense of storage root formation.

SEED CROP MONITORING

A sweetpotato crop for planting material (seed) production, when well established with good vine growth, should be carefully inspected by an experienced breeder,

seed inspector or other trained personnel to detect off-type plants within a variety. In addition, all plantings undergo inspection for varietal purity by the appropriate authority during the growing season.

INSPECTION METHODS

Field inspections are conducted before and during harvest to identify high-yielding hills and desirable shape, and to detect off-type plants, variety mixtures, serious diseases and pests. Inspection results allow positive selection of roots or vines to serve as the breeder seed for planting the next season's crop. Field inspections are conducted to coincide with the time when diseases are most conspicuous, such as a month after planting when SPVD may be clearly identified. A representative inspection of a large field includes 1 percent of the field taken randomly in four different places. For smaller fields, a higher percentage can be inspected.

HARVESTING

At harvest, roots are dug out of the soil, with each hill handled and graded separately. Only those hills that have a high yield of well-shaped roots and are free of any defects are selected. Only disease-free vines are cut to serve as breeder seed.

STORAGE

Post-harvest handling includes curing of seed roots and proper sanitation, which requires removal of all old sweetpotatoes and fumigation of the storage house before storage of new roots. Dust and debris from the grading and packing area must not come in contact with seed roots or vines. Vines must be stored in well-ventilated, shaded places before planting. All storage roots and vines for seed must be transported in net bags or well-aerated containers to avoid excess heat damage due to respiration and close packing (Table 23).

TABLE 23
Summary table of standards

Vine length	25 cm
Tolerance for other varieties (varietal purity)	2 %
Tolerances for pests and diseases	
Black rot	0.5 %
Root knot nematodes (RKNs)	1 %
Scurf	0 %
Wireworms	10 %
Wilt	0.5 %
SSR-Pox1	10 %
Mosaic and stunting virus	1 %
Leaf curl (SPLCV)	5 %
Other virus (e.g. purpling of old leaves, chlorotic spots, vein clearing)	5 %
Storage rot	None
Sweetpotato weevil	None

MULTIPLICATION PROGRAMME PROTOCOL

Breeder seed is the highest quality category of all varieties officially released in a country and, thus, is produced and maintained by a sweetpotato breeder. The breeder seed is carefully maintained until the next multiplication cycle when it is repeated. Guidelines for production of foundation seed, registered and certified seed are generally the same. The guidelines relate to land requirements, inspections and standards for fields, seeds and plants (Table 24).

TABLE 24
Maximum tolerances for disease, insect damage, and internal quality standards for Foundation, Registered, Certified and QDS categories of Sweetpotato

Standards	Foundation (Generation 1)	Registered (Generation 2)	Certified (Generation 3)	QDS[2] (Generation 4)
			%	%
Black rot	None	None	0.10	0.50
Root-knot nematodes	None	0.20 %	0.50	1.00
Scurf	None	None	0.10	0.50
Wireworms	1.00 %	2.00 %	5.00	10.00
Wilt	None	None	0.10	0.50
SSR-Pox[1]	None	5.00 %	5.00	10.00
Sweetpotato viruses				
Mosaic and stunting	None	None	None	1
Leaf curl	None	None	None	5
Other (e.g. purpling of old leaves, chlorotic spots, vein clearing)	None	None	None	5
Other varieties (varietal purity)	None	None	None	2
Storage rot	None	None	None	None
Sweetpotato weevil	None	None	None	None

[1] Seed with pox will be labelled *Sterptomyces* soil rot (pox) below 5 percent. For foundation, registered and certified seed, there should be no physiological disorder, variety mix-up or rotten tubers.
[2] Quality declared seed

Sweetpotatoes grown for certification are handled in much the same way as the commercial crop with the following exceptions:

- plants showing any mutations and symptoms are discarded;
- 4-year rotation is followed;
- only vine cuttings may be used for production of foundation seed;
- fields to be certified must have at least one field inspection by the relevant official during the growing season (Figure 4).

MATERIALS FOR RAPID MULTIPLICATION

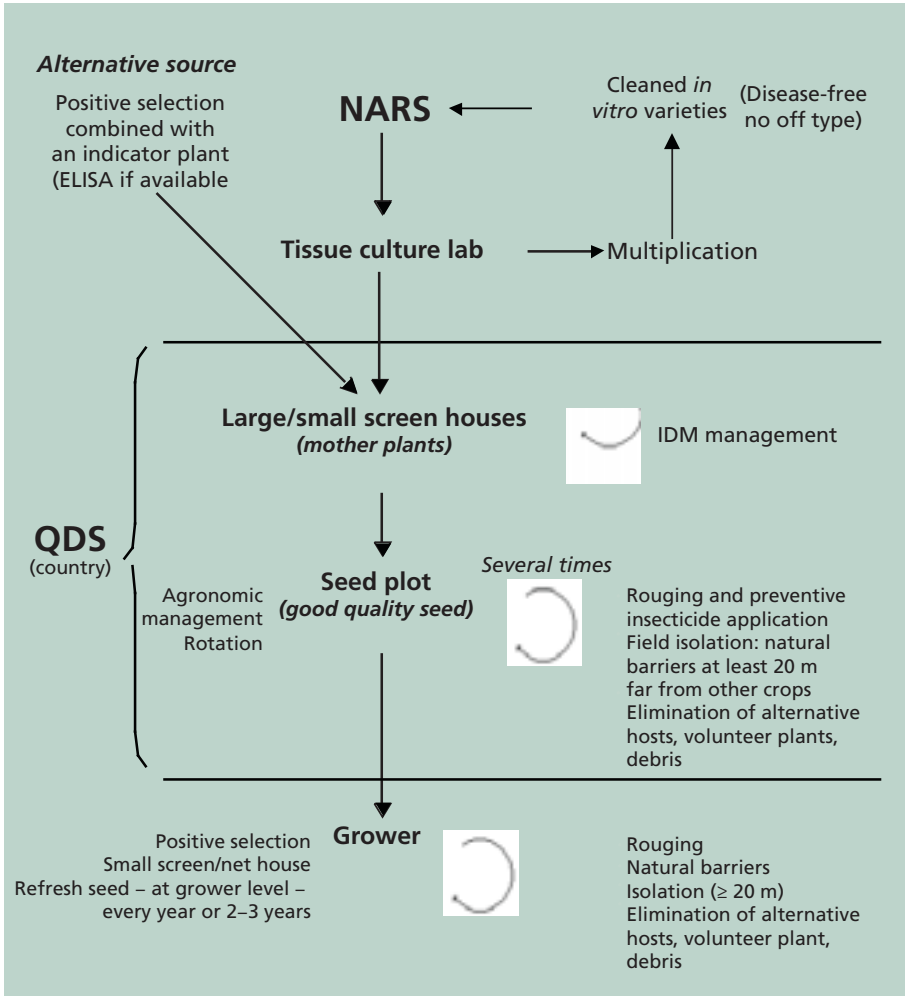
Fertilizer

NPK 17-17-17 at the rate of 42 g/m^2 is applied after planting. Urea is applied at the rate of 13 g/m^2 after each harvest of cuttings, followed by light watering. Manure at 2.5 kg/m^2 is applied as farmyard manure before planting. The manure should be well decomposed.

Insecticides

Where the presence of weevil infestation is proven, cuttings may be dipped in a systemic insecticidal solution for several minutes before planting, in accordance with national legislation. This is done to ensure the elimination of all stages of the weevil and provide some protection for the young plants. To control aphids, white

FIGURE 3
Outline of sweetpotato multiplication programme

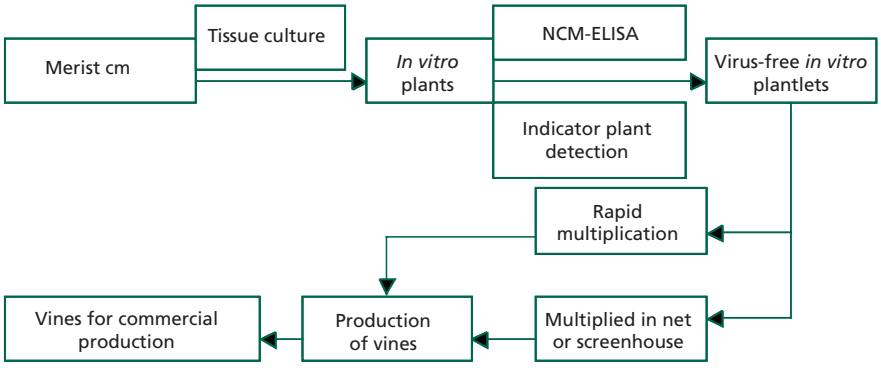


flies and mites, acaricides or available alternative marketed up-to-date pesticides may be applied, following the recommended dosage and method of application.

Up-to-date fungicides

Systemic up-to-date fungicides may be applied following the recommended dosage, only when disease symptoms appear.

FIGURE 4
Protocol for virus free sweetpotato planting material production



Varieties
Appropriate varieties for rapid multiplication need to be identified.

Cuttings
Three node cuttings are taken from vines whose leaves were previously removed. Apical or top cuttings are planted separately.

Preparation of nursery beds
The beds are 10 m long, 1.2 m wide and 20 cm high. Fertilizer (17-17-17), manure (2.5 kg/m^2) and insecticide are applied and mixed thoroughly with the soil before planting.

PREPARATION OF PLANTING MATERIAL
Cuttings are taken from vigorous mother plants after about three months. Leaves are removed from the vines. Three node cuttings are prepared from all parts of the vine. Apical cuttings with three nodes are planted separately. When risk of weevil infestation is proven, the cuttings could be dipped in a solution of a systemic insecticide before planting.

Planting
Density: 50 cuttings/m^2 (0.2 m between rows × 0.10 m intra-row). The cuttings are planted upright with two nodes below the soil surface.

CULTURAL PRACTICES
Irrigate two to three times a day, early morning and late afternoon with a hose pipe or watering can.

Weed periodically to maintain weed free beds.

Rogue all diseased plants as soon as identified.

Shade the nursery beds where there is excessive sunlight and heat, using mats or other locally available materials. Remove the mats when the first leaves develop. Avoid keeping the mats on for more than two weeks to prevent etiolation.

Cut (harvest) vines with apical cuttings (25 cm long) 5 cm above the soil level, leaving some nodes on the stems to enable further production of cuttings from the auxiliary buds. The procedure of cutting above the soil surface has a 98 percent chance of selecting weevil-free plants.

Data to be collected in rapid-multiplication beds includes:

- percentage of sprouting or establishment (two weeks after planting);
- harvesting (cutting) dates of apical cuttings;
- number of apical cuttings harvested;
- percentage of rooting success (survival in the open field); and
- reaction of cuttings in the open field related to the yield.

Taro

Mary Taylor
Secretariat of the Pacific Community, Suva, Fiji

Colocasia esculenta (L.) Schott
Araceae

Taro, *Colocasia esculenta* (L.) Schott is a root crop of the monocotyledonous family, Araceae (Matthews, 1995). It is a widely distributed food crop, locally important in many parts of the humid tropics and subtropics, but also cultivated in temperate zones. Until relatively recently, the centre of origin and domestication for *Colocasia* was accepted as Southeast Asia (Plucknett, 1976), with Papua New Guinea as a major centre of diversity. However, the understanding now is that most cultivars found throughout the Pacific were domesticated from wild sources in Melanesia (Plucknett *et al.,* 1970; Kuruvilla and Singh, 1981). With domestication also occurring in Southeast Asia and separation of land masses in Sundra and Sahull, two gene pools developed (Matthews 2003; Lebot *et al.*, 2005a).

Two botanical varieties of taro are recognized: *C. esculenta* var. *esculenta* commonly known as dasheen, and *C. esculenta* var. *antiquorum* (Plate 23), commonly known as eddoe (Purseglove, 1972). The dasheen varieties, with large central corms, suckers or stolons are dominant in the Pacific, whereas the eddoe type with a relatively small central corm and a large number of smaller cormels is found mainly in Asia. In recent years, this separation of the cultivated taros into two varieties has been challenged (Hay, 1998a) and the literature today often refers to large corm (dasheen type) and small corm (eddoe type) or intermediate types.

TAYLOR, 2006

Plate 23
Tubers of the small-corm (eddoe type) often referred to as Colocasia esculenta *var.* antiquorum.

METHODS OF PROPAGATION

Taro is a vegetatively propagated crop. Plants are monoecious, with flowering and seed setting common in wild and naturalized taros. However, information regarding seed dispersal and germination is limited (Matthews, 1997). Taro can be induced to produce seeds by spraying with gibberellic acid, a practice used in breeding programmes. For propagation, farmers commonly use headsetts (tops) or suckers with the dasheen type and cormels with the

TAYLOR, 2006

Plate 24
Taro leaf blight caused by Phytophtora colocasiae.

eddoe type. Some taros produce runners or stolons that can also be used as planting material. Headsetts or large suckers establish quickly, resulting in vigorous plants. The proliferation rate for both dasheen (suckers) and eddoe types (cormels) is genotype dependent, but can be influenced by agronomic practices. Whole corms can be used for increasing planting material by cut corm or minisett production.

PESTS AND PATHOGENS

Taro is attacked by a wide range of insect pests, the most serious of which is the taro beetle, *Papuana* spp. (Theunis and Aloali'i, 1999). The adult beetles feed on the corms, causing significant damage. Above ground symptoms vary with the age of the plants. Taro is also attacked by fungi, bacteria, nematodes and viruses, some of which cause serious diseases (Jackson, 1980). Heavy infestations of *Tarophagus proserpina* (taro planthopper) can cause plants to wilt and, exceptionally, to die, but most importantly they transmit viruses, namely Colocasia bobone disease virus (CBDV) and possibly taro vein chlorosis virus (TaVCV). Taro leaf blight (TLB), caused by *Phythophthora colocasiae,* is widespread throughout Asia and the Pacific (Plate 24). Cultural control is labour intensive and chemical control is difficult and costly. The use of tolerant/resistant varieties is the only sustainable approach to managing TLB.

Corm soft rot, caused by *Pythium* spp., can have a significant impact on the plant depending on its age and whether the taro is in wetland or dryland cultivation. Healthy planting material, stored for 4–5 days and then inspected for rot, is the best form of control.

Several viruses are known to infect taro. The most common is Dasheen mosaic virus (DsMV), which is easily transmitted by aphids (Plate 25). It can be eliminated by meristem culture but symptoms appear rapidly on field planting. Taro bacilliform badnavirus (TaBV), although widespread, has little impact on growth. However it is thought to be involved in the serious alomae virus complex. CBDV, restricted to Papua New Guinea and Solomon Islands, is reported to be associated with the two most devastating viral diseases of taro, alomae and bobone. Plants recover from bobone infection, but not alomae, which is a lethal disease, restricted to Papua New Guinea and the Solomon Islands. Destruction of infected plants is crucial, as the disease spreads rapidly and destroys entire plantings. Infection with both CBDV and TaBV has been considered the cause of alomae, but some plants can be infected with both viruses and not develop alomae (Revill *et al.*, 2005). With TaVCV, leaves show a distinct vein chlorosis more pronounced than the vein chlorosis sometimes associated with TaBV. By contrast to infection with CBDV

(which is also a rhabdovirus), galls are not present on the leaf blades and petioles, and plants are generally not stunted. Taro reovirus (TaRV) has only been detected together with other viruses, meaning that it is possible TaRV is not a serious pathogen of taro (Revill *et al.*, 2005).

TAYLOR, 2006.

Plate 25
Taro plant infected with dasheen mosaic potyvirus (DsMV).

Removal and subsequent burning or burial of infected plants is strongly recommended for all viruses, as plants serve as a source of infection. Guidelines recommend that all transfers of taro should be as sterile, virus tested plantlets growing in a tissue culture medium (Zettler *et al.*, 1989). Further information on taro pests and diseases can be obtained from TaroPest, an illustrated guide to pests and diseases of taro in the South Pacific (Carmichael *et al.*, 2008).

PROTOCOL FOR THE PRODUCTION OF PLANTING MATERIAL

There are essentially five types of taro planting material: headsetts/tops or suckers, cormels, corm pieces or minisetts, runners or stolons, and tissue culture plantlets. Planting material is generally produced in the field at the same time as the crop, which means best practices used for taro production will also be of benefit to the planting material. The protocole for reproduction of taro planting material is illustrated in Figure 5.

SOURCE OF MATERIAL

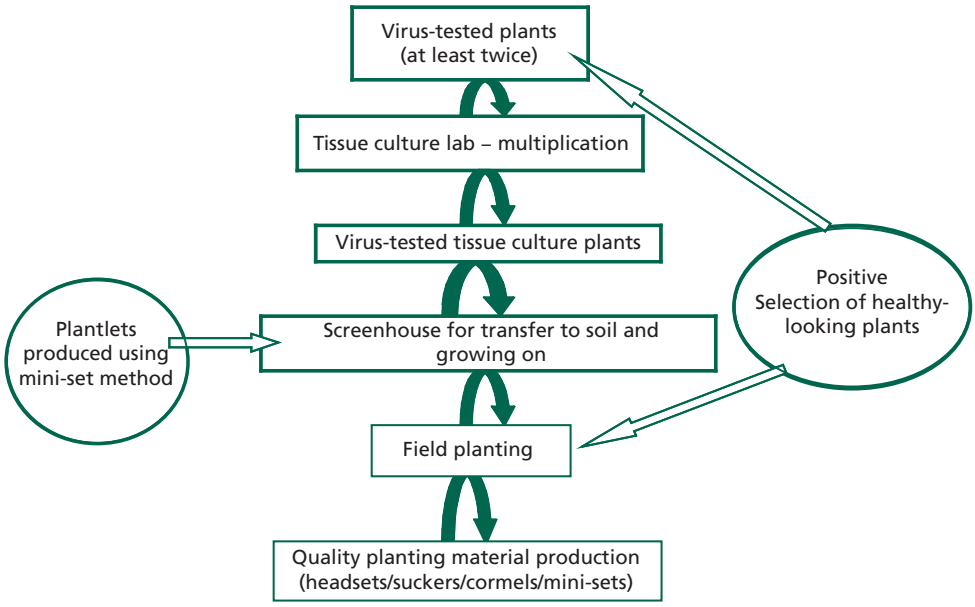
The source of planting material (mother plants) should either be virus-tested tissue culture plants (Plate 26) or plants identified through positive selection as healthy, disease-free, pest-free mother plants. Virus-tested tissue culture plants would be preferable as some taro viruses are symptomless, which could affect the efficiency of positive selection. If virus-tested tissue culture plants are used, documentation should be provided that details which viruses were tested for, the frequency of testing and which methods were used.

TAYLOR, 2006

Plate 26
Virus tested tissue culture taro plants as source of planting material.

Five viruses have been recorded for taro, but their presence can be country specific. Country records should be examined to

FIGURE 5
Outline of a multiplication programme



determine what viruses are important. It is advisable to sample all stock material and test it for viruses at least twice, before large numbers of plants proliferate. These plants can then be grown on to produce headsetts, suckers and/or minisetts from the corm, or cormels. Each minisett should weigh 20–50 g and possess at least one axillary bud. They should be dusted with wood ash or an up-to-date fungicide mix to reduce rotting, after which they are planted in a nursery bed or screen house (as with tissue culture plants) until the sprouted plants are ready for field planting.

FACILITIES AND EQUIPMENT

A screen house is essential for acclimatizing tissue culture plants before transfer to the field. A screen house is also necessary for growing on small suckers or cormels, and for managing the minisett method. Cutting tools, such as knives, must be kept clean to prevent the spread of pests and diseases. The knife can be dipped in diluted bleach solution between each cut (Plate 27).

FIELD REQUIREMENTS

The full benefits of using disease-free mother plants can only be achieved if the plants are cultivated in a disease- and pest-free environment. At least three

months fallow with two tillings is advised for reducing taro disease pathogens, although a year is preferable. Taro will grow on a wide range of soil types from heavy clay loams to light volcanic soils. However, quality and yield will improve with friable, fertile soil that has high water capacity and is rich in organic matter. A slightly acid soil (pH range 5.5 to 6.5) with moderate clay content is preferred. Maintaining available calcium levels at recommended concentrations will prevent the development of *Pythium* corm rot. Ideally, plant calcium levels should be monitored by leaf analysis throughout the growth of the crop.

TAYLOR, 2006.

Plate 27
Preparing suckers of the dasheen type referred to as Colocasia esculenta *var.* esculenta *for planting.*

Field inspection

Fields and nurseries should be regularly inspected to ensure no outbreak of any pest or disease that would impact on the quality of the planting material. Diseased plants must be eliminated from the field and nursery. Inspection should occur shortly after planting (within two weeks) and then at three months and just prior to estimated harvest (at six months). The number of plants to be inspected will be influenced by the size of the field or nursery.

The screen house nursery should be kept clean and free of decaying or dead plant material. Red spider mite can be a problem and, therefore, a regular spraying of miticide may be necessary.

AGRONOMIC PRACTICES

The taro field should be maintained free of weeds after planting, to reduce the incidence of pests and diseases. Tissue culture plants can only be transferred to the field after 6–8 weeks in the screen house, when they have been fully acclimatized. Once ready for transfer to the field, they can be planted at the same spacing used for headsetts and suckers, generally1.0 m by 1.0-1.5 m. Nitrogen and potassium applications should begin after a minimum of two leaves have formed and should be made in several small applications during the first two-thirds of the plant cycle. Ideally, soil and leaf tests should be conducted to determine the rates required, with N and K targets generally in the range of 55–90 kg/ha. Permanently moist soils are preferable. Ideally, irrigation should be applied just after planting and one week later. Subsequent irrigation may be given at 12–15 days intervals, depending on the moisture retention capacity of the soil. About 9–12 irrigation applications will be required for the crop, although the irrigation should be stopped 3–4 weeks before harvest. In the case of a rainfed crop, if there is prolonged drought, supplementary irrigation is required, especially for the eddoe type.

HARVESTING

Planting material can be grouped according to size at the harvesting stage, as there may be a variation in the final size. Taro corms can remain in the ground after suckers are harvested, and will provide 5–10 more suckers over time. Hilling the soil against the side of the taro, together with the light application of fertilizer is important. Visibly damaged and diseased material should be properly discarded at harvesting.

Post-harvest treatments

After harvest, suckers/headsetts are trimmed, thoroughly washed in clean water and dipped in disinfectant such as 0.5 percent sodium hypochlorite (totally submerged for a timed minute), then removed, drained and allowed to dry in a cool, clean area. The headsetts are held for 4–5 days to allow the wound periderm to form over cut surfaces and to allow culling of any diseased tops (CTAHR, 1997). Similarly cormels are left in a cool, dry place for 1–2 days.

The "holding" area should be cool, clean, dry, well ventilated and out of direct sunlight. The planting material also should be elevated during this time to avoid contact with ants or other pests. Cormels can be kept in sand spread over the floor to avoid rotting.

STORAGE AND TRANSPORT

Before planting, headsetts/suckers are held for 4–5 days, and cormels are held for 1–2 days in a well-ventilated area. Longer storage is not ideal. For transport, there is no one prescribed system but precautions should be taken: to avoid physical injury, bruising, microbial contamination and deterioration; to provide adequate ventilation for respiration and exchange of gases; and to provide protection from the sun.

QUALITY STANDARDS FOR THE PLANTING MATERIAL

Size and weight

The optimum size of a headsett or sucker should be not less than 5 cm diameter at the base of the petiole. Headsetts are prepared by cutting the leaf stalk 1–2 cm below the top of the mother corm, removing all dead leaves and outer petiole bases, and finally trimming a small amount of the plant to make it neater, with the final length of the headset about 15–20 cm. Suckers should be treated in the same way as headsetts. For cormels, 30–50 g is the preferred size.

Tolerances/risks

Transfer of taro from one country to another should only be as virus-tested tissue culture plants. However within countries, the extent to which a pest and/or disease is present is location specific. For example, in Fiji, taro beetle is a problem on one island but not on the other. Therefore, planting material for the island that is free of beetle should not originate from the island where taro beetle exists.

Several taro pests and diseases have the ability to damage the marketability of the crop significantly or destroy it completely. Therefore, the supplied product should be produced in areas free of these pests and diseases (zero tolerance). In the case of the viruses, mother plants should have been sourced from virus-tested plants certified as virus free. The pests and diseases for which there should be zero tolerance are:

- taro beetle
- TLB
- CBDV
- alomae
- pythium.

Among the pests and diseases described above, some are known to be widespread globally, others have just been found in the Pacific, but could exist elsewhere. However, the level of tolerance acceptable for these pests and diseases would depend on the extent to which they are present in the country.

Varietal purity

At least 98 percent of the taro plants must conform to the characteristics of the respective parent.

Germination

For headsetts and cormels, the germination percentage of 99 percent and 95 percent respectively should be the minimum standard (Table 25).

TABLE 25
Summary table of standards

Standard	Headsett/sucker	Cormel
Size	5 cm diameter, at the base of the petiole	30–50 g
Varietal purity (minimum)		98 %
Germination (minimum)	99 %	95 %
Tolerances for pests and diseases: taro beetle, TLB, CBDV, alomae, pythium	0 %	

Yam

Malachy Akoroda
University of Ibadan, Ibadan, Nigeria

Dioscorea spp.
Dioscoreaceae

The main species of the genus *Dioscorea* commonly used for human food include: *D. rotundata* Poir., *D. cayenensis* Lam., *D. alata* L., *D. bulbifera* L., *D. esculenta* (Lour) Burk., *D. dumetorum* (Kunth) Pax., *D. trifida* L., *D. japonica* Thunb., *D. hispida* Dennst., and *D. oposita* Thunb.

However, only two species, *D. rotundata* (white yam) and *D. alata* (water yam or Lisbon yam) account for over four-fifths of food yams consumed worldwide in terms of tonnage as well as spread over the major consumption areas. In West Africa, which produces about 95 percent of the world output of edible yams, both species are utilized, but *D. rotundata* is predominant and revered. In some countries, *D. alata* dominates at 60 percent, as in Côte d'Ivoire (Okonkwo *et al.*, 2004).

GEOGRAPHIC ORIGIN AND DISTRIBUTION

Yams are mainly pan-tropic in distribution. Africa, which has 83 percent of the cultivated areas, has the major producing countries, especially Benin, Côte d'Ivoire, Ghana, Guinea, Nigeria and Togo. In 2005, global yam production was 49.3 million tonnes from 4.5 million ha, 96 percent of which was in tropical Africa. Nigeria alone accounts for about 70 percent of the world production. It is the second most important root/tuber crop in Africa, with production reaching just under one-third the level of cassava. The other main yam producing areas are in South America, mostly Brazil and Colombia; the Caribbean islands of Cuba, Haiti and Jamaica; the Philippines and Japan in Southeast Asia; and Portugal which is the only European country that produces yams. The gross fresh tuber yields overall average 10.85 t/ha.

REPRODUCTIVE MEANS

Yams reproduce sexually from true botanical seed. Vegetative reproduction is by aerial tubers and underground tubers. Although vines can be used to reproduce the plant, that method is mainly by researchers. The following protocol outlines the use of tubers as planting material for small farmers who grow most of their crop for home food and only secondarily for sale.

DISEASES AND PESTS

The main seed-borne pathogens and pests of seed tubers are soft rots, dry rots, nematodes and beetles. Virus symptoms may be observed on leaves and vines after planting healthy tubers. Spread of most of these pathogens and pests is through vegetative planting material, either seed tubers or cut tuber setts.

Virus

Occurs as mosaic disease on distorted/deformed linear/shoestring vein clearing, chlorotic leaves and on vines as mottling and stunting. It is spread by aphids *Aphis citricola* and by infected setts.

Scorching

Observed as chlorosis, it can be due to either virus or fungi that are spread when sap inoculation infects healthy setts during cutting.

Tuber rot in storage

Caused by bacteria or fungi with symptoms of soft or dry rot with an unpleasant smell. Infected tubers must be removed. Wounding or bruising in the field or during transportation aids the rots, which are spread by rain, insects and wind.

Nematode damage

Starts in the field where cracked skin and cavities in a tuber with dead tissue are obvious symptoms. Galling of yam roots are clear signs of the nematodes *Scutellonema bradys*, *Pratylenchus* sp. and *Meloidogyne* sp. Nematodes persist in the soil and may be controlled by rotation, use of clean setts and by dressing setts with an up-to-date nematicide.

Insect pests

Beetles of several species affect tubers in the field and storage. They bore holes in the tubers and are controlled by applying insecticides recommended in the country for setts at planting. The leaf-eating beetle *Crioceris livida* has brown to black adults, their larvae feed on lamina causing die-back and defoliation with localized damage, especially after the rains start. Females lay eggs on the underside of lamina, which give rise to soft larvae that are covered by slimy frothy secretions. They are washed off by heavy rain and pupate in the soil, completing their life cycle in a month.

The following points are given as the protocol for field agronomists, extension staff and small farmers on the production of quality planting materials.

SEED YAM TUBER MULTIPLICATION PROTOCOL

- Select mother tubers for quality and appropriate size.
- Cut selected tubers into small setts, each with a portion of brown skin or peel.

- Practice good field care and maintenance to ensure satisfactory growth and yield.
- Set optimum weight, dependent on the target seed tuber weight required at harvest.
- Store good seed tubers in optimum conditions to minimize losses in weight and their regenerative potential.

The expected seed yam should be 150–400 g with a preference for setts of approximately 200 g. The differential response of individual varieties makes the optimum seed weight difficult to prescribe. In practice, the distribution of resulting seed yams is normally distributed but often skewed in favour of smaller weights. These seed yam weights are recommended especially where the target is to obtain ware tubers of about 2 kg or more.

REQUIREMENTS FOR FIELD PRODUCTION AND STORAGE BARNS

Setting up field production and preparing storage barns requires:

- land for field production;
- nursery tools including hoes, cutlass, knives, watering can and gloves;
- source of nursery water such as well, river or spring;
- chemicals (to be used to treat cut surfaces of setts);
- secure nursery;
- barn to store setts and mother tubers;
- stationery and suitable writing materials for records;
- mother tubers to start the production;
- bags, baskets and plastic containers for packaging;
- nets for immersing setts;
- tables on which to cut setts.

GREENHOUSES AND LABORATORY FACILITIES

Farmers do not need greenhouses. Only baskets or polythene bags are required in the preparation of the minisetts from tubers before they are transplanted to the field.

However, the production of seed yam from imported tissue culture plantlets is an option. It is assumed that the yam tissue culture plantlets will have to be acclimatized before being planted in buckets of soil in nurseries or into the field.

The following 16 steps are recommended for producing seed yam from imported tissue culture plantlets.

1. Assemble two buckets, scissors, marker pen, labels, plastic bags, masking tape, hand sprayer, clean water, NPK fertilizer, rope, small flat wooden

stick, washing bottle, and jiffy pellets or cocopeat sterilized at 121°C for an hour, left to cool and put into plastic planting bags of about 9 x 4 x 5 cm.

2. Soak the peat pellets in a bucket of water for an hour before use.
3. Remove the swollen peat pellets from water when they attain their final volume.
4. Pierce each pellet with a stick or top of a marker.
5. Write a label for each plant indicating genotype, number and date of transplanting.
6. Remove cover from the tissue culture tube.
7. Use a small, flat wooden stick and very gently and carefully loosen the edge of the culture medium from the culture container taking care not to break the shoot or the roots.
8. Hold the culture tube with the right hand (the tube opening facing downwards) and gently tap it against the left hand until the plantlet is half way out of the tube.
9. When the plantlet is out of the tuber, do not hold it by the stem because this will increase the chances of breaking the whole root system from the stem. Allow the plantlet to rest on your palm. To remove the culture medium attached to the root system, place your palm with the plantlet in the water of the second bucket and shake gently.
10. Place the plantlet in the hole of the jiffy pellet and press gently on the topside of the pellet to close the hole. Insert the label and place the planted jiffy in a humidity chamber.
11. Spray the humidity chamber generously with water and then close the top by tying the plastic sheet with rope and securing it with the masking tape.
12. From 10–14 days after transplanting, puncture three holes, 1 cm wide, at each side of the humidity chamber with the tip of a pen.
13. Two to three days later, reduce the humidity in the chamber by cutting an opening (a half circle window of 14 cm diameter) at the lower side of the chamber. Fill the wash bottle with clean water and drop about 6–8 grains of NPK fertilizer in so that it dissolves. Water the plants with the solution. An adequate amount of water must be given, but not in excess. Spray the chamber to maintain humidity. Check the plants daily and water when necessary.
14. Two days after cutting the first window, cut a second window on the opposite side of the first window. By now the humidity of the chamber is near ambient conditions. Check and water the plants daily.
15. At 21–24 days after transplanting, the plants will have formed new leaves and roots. The plants can remain in the chamber for another 2–3 weeks and then be directly transplanted in a seedbed at 25 x 25 cm apart, or to polythene bags filled with soil. If the exterior net of the pellet is not biodegradable, it should be removed immediately before planting.

16. After transplanting in seedbeds, pots or bags, all agronomic practices such as weeding and staking should be done. At 6–8 months after transplanting in the seedbed, the plants will senesce, and the tubers can be harvested. Depending on the establishment of the plants, seed tubers of 5 to 250 g will be obtained (Ng, 2002).

AGRONOMIC PRACTICES

Fertile field plots should be used that have a satisfactory rotation record. A legume following a root crop should help to achieve this, such as *Mucuna* sp., which is commonly used in West Africa.

MOTHER TUBERS

Mother tubers are the tubers from which the minisetts are cut. The selection of the best mother tubers is the most critical step in minisett technology. The mother tubers **should**:

- weigh about 500–1 000 g – larger mother tubers are less able to produce sprouts, thus minisetts from larger tubers do not sprout early due to their lower meristem activity;
- be of a diameter that allows for little cortex or flesh to accompany the cut minisett – mother tubers of 8–12 cm diameter are preferable.

On the other hand, mother tubers **should not**:

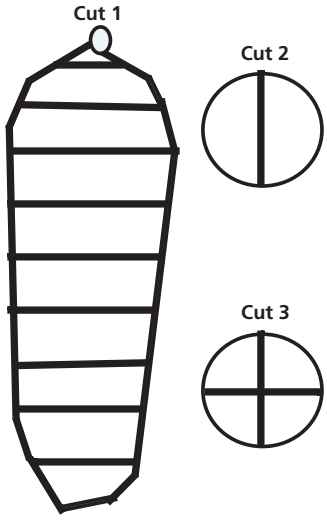
- come from "milked" or "double harvest" tubers – such tubers are not physiologically mature and give poor minisett performance as regards sprouting and the percentage of minisetts that fail to sprout and rot;
- be dormant – active buds or sprouts should be observed on parts of the mother tuber, and should occur 2–4 months after harvest, depending on the variety and storage conditions;
- be shrivelled or shrunken in the skin or peel,
- show symptoms soft or dry rots;
- have symptoms of storage pests, such as white cottony spots produced by sap-sucking bugs.

Cutting the mother tuber into minisetts

The mother tuber is cut into transverse cylindrical slices using a sharp kitchen knife (Figure 6). Each slice is laid flat and cut in 2, 3 or 4 sections depending on the diameter of the slice so as to obtain the desired weight or size.

Minisetts usually weigh 25–50 g but higher mortality has influenced some workers to prefer larger sizes. Setts of 100–150 g have been tried for different varieties so as to achieve the desired seed yam weight.

FIGURE 6
Transverse cuts followed by cutting into sectors to achieve the desired sett size



Each species reacts differently to the minisett technique. *Dioscorea alata* does well with smaller setts of less than 50 g but *D. rotundata* does better with setts of 40–100 g. However, varietal differences also exist. For example, 15 clones of *D. rotundata* gave an average multiplication of 1:5.5 compared with 1:12.9 for 15 clones of *D. alata* (Okonkwo *et al*, 2004).

Overall, trials for any specific variety should involve a range of setts weighing from 25 to 100 g to provide an understanding of the interaction of the variety and the environmental conditions under which the crop will grow. The most important determinant of the size of the minisett is the distribution of the resulting seed yams. For example, if it is determined that 200 g is the maximum weight for a good seed yam to producing ware yams, it is necessary to assess the number of seed yams of that weight that result from the seed yam production effort. In other words, it is not a matter of just the total number of seed yams or the total weight of seed yams *per se*. Thus, managing the seed yam production system to yield the greatest number of seed yams of 200 g should be the target of all seed yam production enterprises.

FACILITIES, EQUIPMENT AND APPROPRIATE STEPS FOR TREATMENT OF MINISETTS

Treating minisetts requires specific facilities and equipment:

- hand gloves, plastic baskets, plastic net bags or sacks, a container for the solution, plastic sheets on which the treated minisetts will be spread to air-dry;
- approved chemicals, up-to-date fungicides, insecticides, up-to-date nematicides or a mix of these in recommended ratios for treating yam minisetts, according to approved protocols;
- wood ash, to be used alone or in combination with a chemical by some farmers to achieve early drying of the cut surfaces.

Steps for treatment of minisetts include

- preparing a solution or suspension of chemical(s) into which the minisetts will be dipped;

- putting a batch of minisetts in the basket and dipping them into the solution or suspension in the basin for one to three minutes;
- spreading treated minisetts on a plastic sheet to allow the cut surfaces to air dry for at least two hours without exposing them to direct sunshine or, as an option, leave them to dry overnight (Plate 28 and 29).

AKORODA, 2007.

Plate 28
Freshly cut tuber pieces.

AKORODA, 2007.

Plate 29
Cut surfaces are dusted, or dipped into a slurry of chemicals to prevent rot.
Note that each sett has an outer skin area that will produce a sprout.

The efficacy of single or mixed usage of wood ash, neem leaf powder or other up-to-date fungicide with insecticides is yet to be determined as to residue in the soil, in the plant or the resulting ware tuber. Their persistence would need examination under the production environments so as to guarantee their safe full-scale utilization.

Pre-sprouting

Pre-sprouting of minisetts allows the best to be taken to the field and gives a better field coverage than direct planting if the pre-spouting takes place in nursery soil, or in cured sawdust in either baskets or perforated transparent polythene bags of appropriate thickness.

The key point in pre-sprouting is to set up favourable conditions that encourage sprouting. This means obtaining a slightly higher temperature with a high relative humidity.

When polythene bags are used, sprouts appear in two to four weeks depending on the variety, size of minisett, and the water and temperature regime maintained in the shade or store where the bagged minisetts are kept for pre-sprouting. Advantages of polythene pre-sprouting include easy vision of the development of sprouts, and cheap and portable material that occupy less space and can be stacked beside the earlier sprouting material. Additionally, the sawdust does not dry out within the polythene bag thus eliminating the need for frequent watering.

Transplanting

Field costs are reduced if all plant stands survive until harvest. Therefore, it is first necessary to select the well-sprouted minisetts from the nursery, basket or polybag and to leave the rest so that they will have more time to develop.

Direct field planting of cut setts that have been treated adequately with chemicals to control rots and pests is ideal. Experience shows that the survival of planted minisetts is increased by good handling of the best minisetts and if they are transplanted in the field within 90 days.

Good agronomic field management practices should be adopted to obtain high yields of seed yams. These should include:

- scheduling dates for timely field actions;
- using fertilizer and organic materials for nutrients, and also monitoring market prices for fertilizers and seed yam;
- selecting the seed mother tuber for health and variety;
- staking the plant early – once the plant growth has reached 1 m;
- harvesting only after shoots have completely senesced;
- protecting seed tubers from rodents, intruders and theft;
- making regular and frequent visits to check and verify any changes and dangers posed by pests or by environmental hazards such as rain or sun.

Good agronomic practices also include rotations and the use of organic materials and fertilizers to enrich the soil from season to season. Field plots to be used for seed yam production should be fertile. A suitable rotation of crops to avoid monocropping is advised. A legume following a root crop would help achieve this. *Mucuna* sp. is the common choice in parts of Benin, Nigeria and Togo in West Africa.

STAKING AND TRAINING THE VINES (FIGURE 7)

Both staking and training may be necessary in the savanna zones where bamboo branches usually are used with strings to lead the vines to the stake. Up to ten vines may be linked to a central stake depending on its size and height. Stakes are 1–1.5 m tall depending on the ecology and the availability of staking materials. In the forest zones, bamboo branches are usually regarded as sufficient.

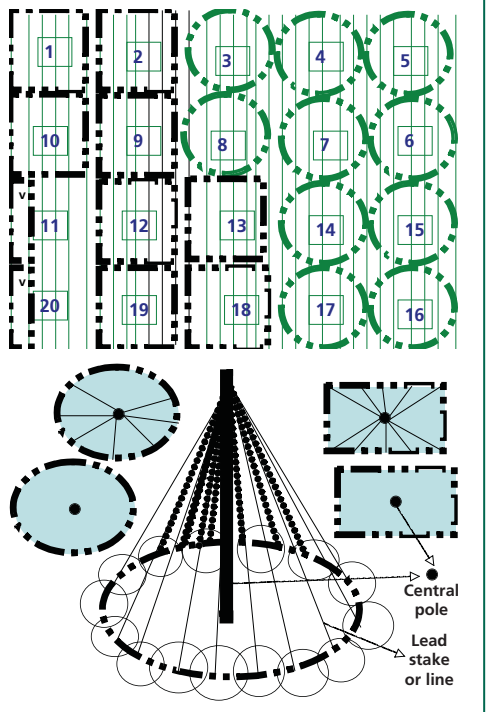
The staking in the field is designed to minimize the number of stakes, which thereby reduces costs of labour and materials. Seed yams mature in 5–8 months, depending on variety, soil, staking, the amount of rainfall and its distribution, and the time of transplanting after the start of the season.

SEED CROP MONITORING

At field production stage, the plants are inspected for leaf-borne diseases including viruses, anthracnose and blight. The five- to eight-month growth period

require threes inspections – at one and five months after transplanting (MAT) and at harvest. Harvested tubers are checked for nematode symptoms. No infested tubers should be stored because they deteriorate in storage and infest other healthy tubers. Any infected tuber identified during the storage period must be removed.

FIGURE 7
Plants along and between the rows are centred on a central strong stake with small stakes or ropes leading from each of the plants. In this way fewer stakes are needed per unit area



INSPECTION METHODS

The presence of government or of a local seed grower agency/association, authorized by national law to inspect and check seed is assumed. Otherwise public or private agencies can be formed in local government areas to help seed yam producers attain higher standards.

1 MAT

After planting, some of the seed setts do not germinate and do not produce young shoots. The degree of missing plant stands should be verified after 1 MAT in order to estimate the maximum number of plant stands that will mature at the expected harvest date. Any further supply of the missing plants will introduce plants that would normally mature at a later date. At this point 1 percent of plants should be inspected for the level of virus symptoms and for the percentage of other varieties in the plot. Any diseased plants should be uprooted, removed and replaced with a well-developed plant from the nursery. After a month of planting, the plants in the nursery need not be retained.

3 MAT

Once tubers begin to develop, rodents start to attack. It is necessary to seek methods to reduce and prevent their access such as use of wire netting fences, traps or chemical baits. Methods that allow reuse of the materials from season to season are the most efficient and economical.

5–6 MAT/harvest

Lay the tubers to be stored on a tarpaulin spread out on the ground and examine them for signs of peel defects, scratches, surface cuts, rots, bulges of the skin or

deep cuts with soil coating. This inspection of seed tubers is seeking evidence of bacterial or fungal rots, insect bore holes and sprout incidence.

HARVESTING

Soil conditions influence the ease of harvesting. Wood or metal poles are used together with hoes to retrieve seed yams from the ridges or mounds on which the minisetts are planted.

Estimated multiplication rate

The single sett multiplies by a rate of 2–12. However, the rate is commonly about 5, but this depends on the handling of the operation, season, variety and the fertility of the soil as well as when in the season the minisetts were established after planting.

Post-harvest treatment

It is important to present the seed yams in the best state for storage, packaging or sale:

- remove all soil from seed yams so they can be easily assessed for size and skin feature;
- dip seed yams in a solution of insecticide before storing, but not before sale;
- put seed yams in a basket or net bag and tie them to the barn using a rope;
- make periodic inspections in the barn, and remove tuber sprouts;
- for transport, put all seed yams in baskets, sacks or plastic packages and under cover to minimize bruising of skins or damage to sprouts, and to protect from heat, moisture and rodents.

STORAGE

Seed yam tubers are stored similarly to ware tubers. Barns or trees in rows are used, tied with bamboo cross bars. Net bags containing seed yams are hung on each bamboo cross bar. The aeration under the tree shade helps conserve high relative humidity. Shady conditions also develop low evaporative gradients. Barns are of varied sizes, but the basic unit is 10 m x 10 m, and multiples can be designed according to the quantity of seed tubers to be stored and the spacing of planted trees.

MULTIPLICATION PROGRAMME PROTOCOL

Mother tubers are selected

These are cut into small pieces, each with a portion of brown skin or peel. The optimum weight depends on the target weight of the resulting seed yam tuber at harvest.

Multiplication rates

Multiplication rates vary but, under good agronomic practices, they will be around 1:10. The smaller the sett weight, the higher the multiplication rate. Usually, microsetts or minisetts are small (5–50 g). Heavier minisetts (50–150 g or more) are also used, as they will have less damage and loss during sprouting in the nursery, basket or polythene bag. Bigger minisetts also achieve larger seed yam tubers in the expected weight ranges of 150–250 g. Consequently, as each minisett planted increases in size and weight, the rate of multiplication is reduced.

TABLE 26
Summary table of standards

Weight of yam set (seed tuber)	200 g
Varietal purity	98 %
Viability	100 %
Tolerances for all pests and diseases	10 %

There is enormous variability in the size of seed yam tubers. From tissue culture to the final target of planting materials is given as 5 to 250 g from plantlets (Ng, 2002).

QUALITY DECLARED STANDARDS

Local authorities can establish minimum standards by choosing among those listed below. These standards would serve as a basis to enhance seed quality from local producers but they would also be realistic and achievable (Table 26).

- Set weight of seed tubers: 200 g but not more than 250 g.
- Package tubers in batches of 25 kg each.
- Produce seed yams from the same variety and use very uniform setts of 25–50 g.
- Store only healthy seed yams tubers.
- Inspect field for disease and variety purity.
- Inspect harvested tubers for health and seed yam size.
- Set 98 percent purity of the specified variety on the label as the standard.
- Set physiological and physical status at 100 percent viability at date of label.
- Give statement of the results of field inspections as percentage observed among plants of the sample counted and assessed.
- Add other criteria for standards as deemed fit by the local authority, based on the prevalent pests and diseases.
- Allow tolerances of up to 10 percent for most variables. But less is preferable.
- Ensure tolerances are less than those that can be observed in "common seeds", i.e. regular commercial planting material not derived from a monitored, controlled QDS programme.

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Annex 1

List of main contributors

Crop	Main contributors	Organization
Andean tubers: Oca *(Oxalis tuberosa)* Ulluco *(Ullucus tuberosus)* Mashua *(Tropaeolum tuberosum)*	Carlos Arbizu	CIP Lima, Peru
Bananas, plantains and other Musaceae *(Musa* spp.)	Thierry Lescot	CIRAD Montpellier, France
	Charles Staver	Bioversity International Montpellier, France
Cassava *(Manihot esculenta)*	Hernan Ceballos Fernando Calle	CIAT Cali, Colombia
Cocoyam (*Xanthosoma sagittifolium*)	Juan Perez Ponce	Vitrobio Valencia S.L. Valencia, Spain
Garlic (*Allium sativum*)	Alejandrina Robledo Victor M. Villalobos	SAGARPA Mexico
Hausa potato (*Solenostemon rotundifolius*)	Elizabeth Acheampong	University of Ghana Accra, Ghana
Konjac (*Amorphophallus konjac*)	Sivasubramanian Edison	Central Tuber Crops Research Institute Trivandrum, India
Potato *(Solanum tuberosum)*	Ian Barker Enrique Chujoy	CIP Lima, Peru
Sweetpotato (*Ipomoea batatas)*	Robert Mwanga	National Agricultural Research Organization Namulonge, Uganda
	Segundo Fuentes	CIP Lima, Peru
Taro (*Colocasia esculenta*)	Mary Taylor	Secretariat of the Pacific Community Suva, Fiji
Yams *(Dioscorea* sp.*)*	Malachy Akoroda	University of Ibadan Ibadan, Nigeria

Annex 2
Glossary

Apical meristem
A region of the tip of each shoot and root of a plant in which cell division continually occurs to produce new stem and root tissue, respectively.

Asexual propagation
Vegetative reproduction (e.g. bulbs, tubers, corms) that does not involve the formation or union of gametes of different sexes.

Axillary bud (= lateral bud)
A bud found at the axil of a leaf.

Bract
A modified leaf that subtends flowers or inflorescences and appears to be a petal.

Bruise
Damage underlying plant or fruit tissue, visible as a soft discoloured area on the unbroken surface and caused by pressure or impact.

Bud
A region of meristematic tissue with the potential for developing into leaves, shoots, flowers or combinations of these, generally protected by modified scale leaves.

Bulb
A short modified underground stem which has one or more buds enclosed in fleshy modified leaves or scales which supply energy to the bud(s) when they start to grow.

Bulbil (= bulblet)
A small, deciduous bulb (or tuber) formed in the axil of a leaf and functioning to propagate the plant vegetatively.

Callus
Actively dividing non-organized masses of undifferentiated or differentiated cells often developing from injury (wounding) or, in tissue culture, from the presence of growth regulators.

Callus culture
A technique of plant tissue culture, usually using solidified medium and initiated by inoculation of small explants.

Centre of origin
The geographic locations where particular domesticated plant species originated.

Clonal propagation
Asexual propagation of many new plants (ramets) from an individual (mother plant); all have the same genotype.

Clone
Group of genetically identical cells or individuals which are all derived from one selected individual by asexual propagation.

Clove
One of the small bulbs (as in garlic) developed in the axils of the scales of a large bulb.

Corm
A bulblike stem which, unlike a true bulb, is solid and sends down a root when the new growing season begins.

Cormel
A new, small corm arising from a mature corm.

Cultivar
An assemblage of plants that has been selected for a particular attribute or combination of attributes. It is clearly distinct, uniform and stable in its characteristics and, when propagated by appropriate means, retains those characteristics (International Code of Nomenclature of Cultivated Plants, 2004, Art. 2.2).

Cutting
A detached plant part, such as a portion of leaf, stem, root or bud which, when removed from a plant and with appropriate treatment, can regenerate into a complete plant.

Disease free
A plant or animal certified through specific tests as being free of specified pathogens. Should be interpreted to mean "free of any *known* disease" as "new" diseases may yet be discovered to be present.

Disease indexing
Assay of organisms for the presence of known pathogens according to standard testing procedures.

Disease resistance
The genetically determined ability to prevent the reproduction of a pathogen, thereby remaining healthy.

Disinfection
Attempted elimination by chemical means of internal micro-organisms (particularly pathogens) from a culture or sample.

Disinfestation
The elimination or inhibition of the activity of surface adhering micro-organisms and removal of insects.

Dormancy
A period in life of an animal or plant during which growth and/or development slow or completely cease.

ELISA
Acronym of **E**nzyme **L**inked **I**mmuno **S**orbent **A**ssay, an assay that relies on an enzymatic conversion reaction and is used to detect the presence of specific substances (such as enzymes or viruses).

Explant
A portion of a plant aseptically excised and prepared for culture in a nutrient medium.

FOC
Fusarium oxysporum f.sp. *cubense* (with reference to banana).

Hardening off
Process that conditions plants for survival when transplanted outdoors.

Haulm
Stems or stalks of grain, beans, peas, potatoes or grasses, especially after harvesting.

In vitro
Outside the organism or in an artificial environment. Applied for example to cell tissues or organs cultured in glass or plastic containers.

Inoculum
In tissue culture, a small piece of tissue cut from callus, an explant from a tissue or organ, or a small amount of cell material from a suspension culture is transferred into a fresh medium for continued growth of the culture.

Meristem
Undifferentiated but determined plant tissue in which the cells are capable of active division and differentiation into specialized tissues such as shoots and roots.

Meristem culture
A tissue culture containing meristematic dome tissue without adjacent leaf primordia or stem tissue. The term may also imply the culture of meristemoidal regions of plants, or meristematic growth in culture.

Micropropagation
Miniaturized *in vitro* multiplication and/or regeneration of plant material under aseptic and controlled environmental conditions.

Microtuber
Miniature tuber, produced in tissue culture, which can readily be regenerated into a normal tuberous plant.

Minituber
Small tubers (5–15 mm diameter) formed on shoot cultures or cuttings of tuber-forming crops such as potato.

Mother plant (= donor plant = ortet)
The plant from which a clone or propagule is obtained, also a mother tuber in a yam.

Negative selection
Selection against individuals possessing a certain character. *Opposite:* positive selection.

Node
Slightly swollen structure on the stem, where leaves and buds arise and where branches originate. Stems have nodes but roots do not.

Off-type
Not conforming to the phenotype of the breed/variety.

Ortet
(see: mother plant).

Pathogen free
Uncontaminated with pathogen.

Peepers
In potatoes, the "eyes".

PIBS
Acronym for *Plants issues de bourgeons secondaires* (shoots emerging from secondary buds).

Pit
A hollow or indentation especially in the surface of an organism.

Pith
The soft, central portion of a plant stem inside the vascular cylinder.

Plantlet
A small, rooted shoot regenerated from cell culture following embryogenesis or organogenesis. Plantlets can normally develop into normal plants when transplanted to soil or other substrates.

Positive selection
(See: *Negative selection*)

Propagule
Any part or piece of a plant or plant organ used for propagation.

Rejuvenation
Reversion from adult to juvenile stage.

Roguing
To identify and remove inferior, diseased or non-typical individual plants from a crop.

Runner (= stolon)
A lateral stem that grows horizontally along the ground surface and gives rise to new plants either from axillary or terminal buds.

Scape
The stem-like, flowering stalk of a plant with leaves clustered around the base of its stem.

Seed-borne disease
A pathogen transmitted by seeds.

Seedling
A young plant, especially one grown in a nursery for transplanting or from a seed.

Sett
A small bulb, corm, tuber or a piece of tuber used for plant propagation.

Sexual reproduction
The process whereby two gametes fuse to form one fertilized egg (zygote).

Slip
A part (sometimes a root, leaf or bud) removed from a plant to propagate a new plant through rooting or grafting.

Sprout
The new growth of a plant.

Stake
A stem cutting of cassava.

Sucker
A shoot that arises from an underground root or stem.

Up-to-date nematicide
An agricultural up-to-date pesticide for the control of nematodes (eelworms).

Vector
An organism, usually an insect, that carries and transmits pathogens.

Vine
Climbing plant: a plant that supports itself by climbing, twining or creeping along a surface.

Virus indexing
A procedure to determine the presence or absence of virus in plant material.

Volunteer
A plant arising in a crop that originates from material (e.g. a tuber or seed) remaining in the soil from a previous crop.

Wilt
A disorder of plants marked by loss of turgidity in soft tissues with subsequent drooping and often shrivelling. May be caused by water stress and/or disease.

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Vegetatively propagated crop species contribute significantly to the agriculture and food production sectors of many developing countries and regions and to their food security. The current availability and dissemination of advanced reproduction technologies, in particular for micropropagation, have increased the scope for improvement and development of disease-free planting materials for these crops. However, in spite of their potential, they have received little attention in formal seed quality regulatory systems. Thus, these protocols and standards were developed to serve as practical and useful tools for seed producers and technicians at the community level as well as for national seed services and the agricultural research community in developing countries. Improved quality of planting materials will contribute significantly to improved agricultural production and productivity and to global food security.

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