Florabank Guidelines

Module 13

Nursery Propagation of Tubestock and Restoration Planting

Shane Turner[1,2,3], Eric Bunn[2,3,4], Kerryn Chia[2], Wolfgang Lewandrowski[2], Amanda Shade[5], Mark Viler[6], Paul Gibson-Roy[7,8], Carole Elliott[2,3]

[1] School of Molecular & Life Sciences, Curtin University, WA
[2] Kings Park Science, Department of Biodiversity, Conservation and Attractions, WA
[3] School of Biological Sciences, The University of Western Australia, WA
[4] School of Pharmacy and Biomedical Sciences, Curtin University, WA
[5] Botanic Gardens and Parks Authority, Department of Biodiversity, Conservation and Attractions, WA
[6] Penrith City Council, Penrith NSW
[7] Kalbar Operations, Bairnsdale VIC
[8] Hawkesbury Institute for the Environment, Western Sydney University, NSW

How to cite these Guidelines

Commander LE (Ed.) (2021) 'Florabank Guidelines – best practice guidelines for native seed collection and use (2nd edn).' (Florabank Consortium: Australia)

How to cite this module

Turner S, Bunn E, Chia K, Lewandrowski W, Shade A, Viler M, Gibson-Roy P, Elliott C (2021) Florabank Guidelines Module 13 – Nursery Propagation of Tubestock and Restoration Planting. In 'Florabank Guidelines (2nd edn).' (Ed. LE Commander) (Florabank Consortium: Australia)

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The update of the Florabank Guidelines was funded by the New South Wales Government through its Environmental Trust, as part of the Healthy Seeds Project, and administered by the Australian Network for Plant Conservation (ANPC). It was overseen by the **Healthy Seeds Consortium** consisting of representatives from the ANPC, Australian Association of Bush Regenerators, Australian Seed Bank Partnership, Centre for Australian National Biodiversity Research, Greening Australia (GA), NSW Department of Planning Industry and Environment, Royal Botanic Gardens and Domain Trust, and the Society for Ecological Restoration Australasia. The **Florabank Consortium** which will oversee implementation of the Guidelines consists of the Australian National Botanic Gardens, ANPC, CSIRO and GA.

Key points

In some circumstances, producing plants under nursery conditions, then planting out, may be preferable to directly sowing seeds as part of a restoration program.

Species that produce relatively few seeds, poor quality seeds, seeds with complex dormancy or are critically endangered may be better propagated in a nursery to produce tubestock for field planting.

Relative to direct seeding, the production of tubestock is generally more expensive.

Good horticultural practice needs to be followed to produce high quality plants that are disease and pest free to improve survival and to prevent the spread of pests and weeds.

For field planting, tubestock needs to be in good health, not overgrown or root bound, and ideally hardened off to local conditions.

When field planting tubestock, good site preparation is essential to maximise survival and growth, with ongoing maintenance and monitoring strongly recommended.

Introduction

Using native seed and other plant **propagules** is a common and significant practice to produce nursery-grown **tubestock** for use in restoration (Corr 2003). This chapter focuses on the effective use of seed and other plant propagules and includes guidance on practical propagation approaches for the production of plants, the use of seeds in a horticultural setting, general seed sowing techniques and the horticultural maintenance of seedlings and young plants as they transition to restoration ready tubestock. For large-scale restoration, seed is by far the major raw material used in restoration due to the relatively low costs and quick results of **direct seeding** in comparison to other methods like the planting of tubestock (Ede et al. 2018). However, seeds can be expensive to purchase and may also be limited in availability for many species (Merritt & Dixon 2011; Palma & Laurance 2015) so in some circumstances, seeds may be better used for the production of nursery tubestock rather than in direct seeding. The following sections outline various approaches and requirements to support the efficient production of plants under horticultural conditions for use in restoration programs across Australia.

Why propagate tubestock for restoration?

Restoration is a growing challenge with landscape-scale targets for restoration or rehabilitation increasing annually in Australia. Consequently, hundreds of thousands of young plants (i.e. tubestock) from many different native species are required each year to support national restoration efforts (Stevens et al. 2016; Miller et al 2017). With such large-scale targets for restoration, the propagation and use of tubestock is a valuable approach for three key reasons:

- It is a more efficient use of propagules in situations of limited supply.
- It provides options for achieving restoration targets that include difficult to return species.
- It assists the transition of more individuals through vulnerable stages of early-development.

The consideration of tubestock in restoration planning and installation can improve the use of propagule resources, include more diverse species and provide more mature tubestock plants for achieving rapid restoration outcomes. While the production of tubestock is usually more expensive than direct seeding, higher survival rates in tubestock plantings can make it a more efficient use of propagule resources (Biodiversity Conservation Trust 2019).

When to direct seed and when to plant tubestock?

Direct seeding is a common approach used in restoration (Coore 2003, Gibson-Roy and Delpratt 2015). This is particularly the case in mining rehabilitation, where companies rehabilitate hundreds of hectares of land annually, mostly via direct seeding or respreading of topsoil (Miller et al. 2017) (see Module 14 – Direct Seeding). While under ideal conditions it can be convenient, quick and cheap (Palmerlee and Young 2010; Grossnickle & Ivetić 2017), direct seeding requires substantial volumes of seed. As well, where factors such as seed quality, site preparation, seeding approaches, and prevailing abiotic or biotic conditions are less than ideal, direct seeding can result in poor outcomes (Grossnickle & Ivetić 2017).

Direct seeding is commonly utilised to restore large, long cleared highly modified environments (i.e. abandoned agricultural land or mined environments) where little native vegetation remains and the soil seed bank is impoverished or non-existent (Biodiversity Conservation Trust 2019). However, when used to restore degraded bushland where significant stands of native vegetation are still present, direct seeding can be less successful due to increased competition pressure and higher levels of herbivory. Hence, for restoring degraded bushland, strategically planting tubestock into small clearings or disturbed areas may be more successful than direct seeding (Figure 1).

Figure 1. Left: Planting seedlings within existing vegetation on a coastal dune. (Photo: L. Commander)
Right: Planting within degraded bushland. (Photo: S. Turner)

When using direct seeding, the percentage of sown seeds that result in an established plant can be significantly less than 20% (Palma & Laurance 2015). This may not be such an issue for common species that are easy to collect and which germinate readily under a range of conditions (Koch 2007), but when seeds are expensive, relatively few in number, poor in quality (i.e. low viability) or from endangered/threatened species (Monks et al. 2019), seeds may be more strategically used by a nursery for the production of seedlings for field planting at a later date.

The main advantages and disadvantages of using tubestock in restoration are outlined below:

Advantages

- Precise numbers of well-developed plants are put in the field, reducing plant loss during transition between germination, emergence and young seedling stages.
- Plantings create an immediate visual impact.
- Plantings are well suited to volunteer and community building events.
- Desired numbers of plants for a restoration area can be accurately produced.
- Many species are easily produced in large numbers under nursery conditions with reduced levels of seed wastage.
- Other methods of propagating plants can be utilised for species which are difficult to propagate via seeds, using different types of vegetative material.
- Tubestock can be selectively planted in micro-habitats, or niche environments such as mounds, furrows, under canopy or in clearings.
- Planting can be timed to maximise plant growth according to monthly climate outlooks.

Disadvantages

- The cost of propagating tubestock can add considerably more expense to projects than that of only collecting and processing seed then using this for direct seeding.
- Planting tubestock can be labour intensive and significantly slower than direct seeding.
- The growth and survival of some plant types (e.g. trees) can be negatively impacted by poor nursery practice and root disturbance (if handled roughly) thus may be less resilient when planted in situ.
- Post-planting survival is directly affected by immediate environmental conditions that may be harsh/erratic or poorly managed.
- Nursery grown plants are often targets for increased herbivory due to highly palatable young foliage.
- Tubestock plants are susceptible to damage from vandalism or inadvertent events (e.g. road works, stock movements).
- Poor nursery hygiene can result in the transfer of foreign pests, weeds or diseases into a restoration site.

To assist with choosing which planting method best suits the circumstances, a decision tree capturing some of the key factors influencing planning, costing, implementation and project success is presented in Figure 2. On occasion (particularly in large cleared sites), a combination of direct seeding and tubestock planting is recommended - direct seeding for species for which large volumes of readily germinable seeds are available and tubestock planting for dominant/keystone trees, species for which seeds are limited or not available, or for taxa that are propagated via other means. Nonetheless, high quality tubestock can also fail under field conditions for many reasons such as drought, herbivory and poor planting approaches so the use of tubestock in restoration does not automatically guarantee success (Miller et al. 2017).

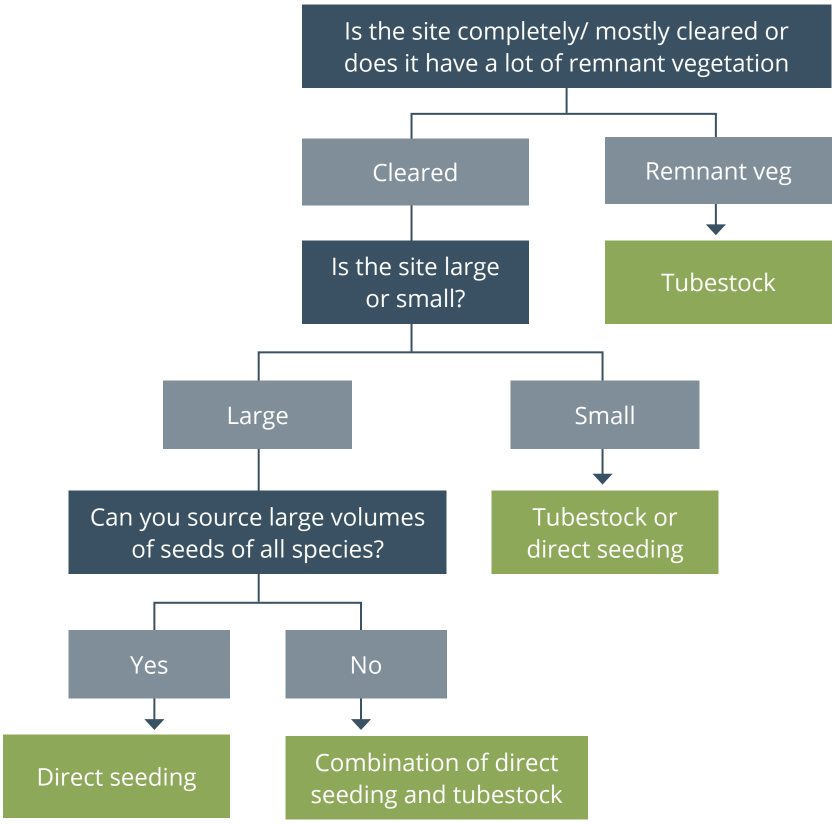


Figure 2. A decision tree that outlines situations when to undertake direct seeding and when to plant tubestock for restoration.

Nursery propagation of tubestock

Producing native tubestock for restoration purposes should be regarded as essentially the same as the production approaches used in the floriculture, horticulture or forestry industries (Davies et al. 2018). Native plants, like all plants, have basic cultural requirements in terms of water, light, temperature, nutrients and growing medium which must be met and optimised for healthy growth to be achieved (Beyl and Trigiano 2015). However, unlike most horticultural and forest crops that are generally comprised of a handful of relatively well-studied species, the requirements for cultivating native species are, in many cases, poorly known (Koch 2007). As such, nurseries that focus on native plant production need to follow good horticultural processes and adapt their approaches based on experience and observations. Related to this is good record management to keep track of which methods have worked and, just as importantly, which have been unsuccessful for particular species by using a nursery propagation schedule to record practices (see Module 4 – Record Keeping). Practitioners need to keep abreast of the relevant propagation literature and adopt the latest techniques as new information about horticultural developments or requirements comes to hand. Being part of local and national networks is a good way to share and keep up-to-date with the latest information and techniques.

The propagule most commonly used to produce tubestock is seed (see "Propagation of tubestock from seed" below), but there are alternative propagule resources for difficult to grow species that can be utilised for vegetative propagation (see "Other tubestock propagation approaches" below). These include aboveground material for cuttings or belowground material like bulbs or corms (see "Division and separation" below).

For the propagation of seed-derived tubestock, seed can be sourced either through commercial native seed companies, that typically collect from wild populations (or alternately from native plant crops see Module 7 – Seed Production), or from collection efforts undertaken by volunteers and community groups for use in localised community nurseries (see Module 6 – Seed Collection). Plant production through a nursery can assist species that are difficult to establish via practices of direct seeding or topsoil application, particularly for those with complex seed dormancy issues (i.e. *Persoonia* spp.) or those that do not form a soil seedbank because they are either recalcitrant (i.e. *Syzygium* spp.) or serotinous (*Eucalyptus* spp.). In addition, production in a nursery offers protection during vulnerable early-stage seedling development and growth. By contrast, directly sown seeds may sit in the soil for several months until conditions are suitable for in situ germination and therefore, may be at greater risk of dying (via predation or fungal attack) or movement to a less desirable location via wind or water displacement (James et al. 2013). Consequently, the use of tubestock can, under some circumstances, provide a more efficient use of propagules (i.e. more plants produced from fewer seeds) as well as improving establishment (i.e. higher survival) in situ (Greet et al. 2020).

Propagation of tubestock from seed and general growing conditions

Seed quality should first be assessed (i.e. to verify seed viability) before seed sowing is implemented (Hancock et al. 2020). This may save both time and money as many seeds have variable quality and thus may not be worth using (see Module 10 – Seed Quality Testing; Hancock et al. 2020). Species with recalcitrant (i.e. desiccation sensitive - Sommerville et al. 2017) seeds are ideally suited to immediate sowing in the nursery, as most cannot tolerate drying and storage for longer than several weeks. For example, rainforest seeds that cannot be dried may be germinated and maintained as a living collection for 1-3 years in a plant nursery (see Module 9 – Seed Drying and Storage & Box 2) (Dunphy et al. 2020). As well, up to 70% of native species will have some form of seed dormancy, which may cause significant problems if not addressed, as viable though dormant seeds will not germinate within a reasonable timeframe when sown (Sweedman & Merritt 2006; Stevens et al 2016; Erickson et al. 2016). Where identified or known, seed dormancy should be removed prior to sowing, using appropriate treatments (see Module 11 – Seed Germination and Dormancy).

Incubation temperatures for promoting germination

Promoting germination from non-dormant seeds requires trays to be incubated at temperatures that are similar to the time of year when germination is most likely to occur under natural conditions for that species. For example, for south-western WA most seeds germinate during the winter wet season so optimal temperatures for promoting germination are around 15 to 20°C – a range somewhat similar to much of southern Australia (Merritt et al. 2007). Depending on the species, temperatures warmer than 20°C can suppress and slow germination. For Mediterranean-climate and temperate species this may be a significant problem if most plant production is scheduled during the summer months so that tubestock is ready for planting in winter. In this case it is recommended to either use a cool room to replicate winter-like conditions or, if unavailable, restrict seed sowing to the cooler parts of the year when conditions are more conducive to germination. For other parts of Australia, warmer germination conditions may be needed. Species from northern Australia, subtropical and semi-arid regions usually require temperatures between 20 to 30°C to promote germination and in several cases even warmer conditions (>30°C) have been shown to be optimal for some species (Erickson et al. 2016). To achieve these types of conditions, seeds may need to be sown during summer and watered frequently (making sure that the seeds do not dry out) or sown as previously described and placed onto a heat bed set to 30°C or in a climate-controlled greenhouse or tunnelhouse.

Special note on implementing techniques to promote germination

Stratification

Where seeds require either cold or warm stratification to promote germination (see Module 11 – Seed Germination and Dormancy) a simple scalable approach to provide this treatment is to firstly sow seeds in appropriate growing media, then water. To apply a cold stratification treatment, place the seed trays into either a fridge or cool room set to ~5°C for approximately 4 to 6 weeks (make sure to water the seeds regularly during this time as they should not be allowed to dry out). For warm stratification, place seed trays onto a heat bed (see right) (Figure 3) set to 25 to 30°C for up to 6 weeks, again watering trays as needed so they do not dry out. After this time, the trays are removed and placed at a temperature matching the growing season of the species, often around 18°C (15 to 25°C range) either outside, in a greenhouse or in a temperature-controlled germination room. Germination and seedling emergence should follow shortly thereafter if the treatment has been effective (Davies et al. 2018).

Figure 3. A heat (~25°C) bed for raising seedlings. (Photo: L. Commander)

Smoke

A significant number of Australian species may require smoke treatment to promote germination (see Module 11 – Seed Germination and Dormancy; Merritt et al. 2007). For example, some species of *Stylidium* (trigger plants), *Grevillea* (spider flowers), Poaceae (grasses), Asteraceae (daisies) and *Solanum* (bush tomatoes) show improved germination when exposed to aerosol smoke or smoke water (Erickson et al. 2016; Stevens et al. 2016). There are several ways in which this can be done. A simple way to treat seeds with smoke is to soak them in a diluted (1 to 20%) smoke water solution for up to 24 hours. (smoke water can either be made in house (see http://anpsa.org.au/articles/smoke-germination.html for details) or purchased from a commercial source). Seeds are then sown while still wet or dried back until touch dry, then sown. Alternatively, seeds requiring smoke treatment can be sown into soil, then watered and left to settle for an hour or so then exposed to aerosol smoke for 1 hour using a smoke tent/ shed or food smoker (http://anpsa.org.au/articles/smoke-germination.html). Inside the sealed environment, smoke from a controlled fire source is pumped directly over the soil, which is similar to what happens during a bushfire. A simple variation of this technique is to use a bee hive smoker, which pushes small amounts of smoke directly over the soil surface for a similar length of time - an approach which is ideal for smaller nurseries, community groups or wildflower enthusiasts. After the smoke treatment, seeds are then lightly watered ensuring that the water does not wash too much of the smoke residue through the pots, as this may lessen the effectiveness of the treatment. Trays/pots are then treated like other pots with recently sown seeds (Davies et al. 2018).

Aging seed in soil

A relatively small but nevertheless interesting sub-group of native species produce seeds, indehiscent endocarps or nutlets that while viable are **deeply dormant** for poorly understood reasons. Consequently, these taxa have complex germination requirements making them very difficult to reliably germinate. Examples include *Hibbertia* spp. (guinea flowers), *Persoonia* spp. (snottygobbles), *Leucopogon* spp. (native heaths), *Eremophila* spp. (emu bushes) and *Leptocarpus* spp. and *Gahnia* spp. (rushes and sedges) (Merritt et al. 2007). Species of *Lepidosperma* are important in some ecosystems, especially for sand binding purposes, but have been extremely difficult to propagate and grow for restoration projects. Intensive research has revealed the secrets to success for several of these, though much more work is still needed (Kodym et al. 2010; Turner 2013).

Germination success can sometimes be achieved with many of these deeply dormant species through **ageing seed** to break dormancy, by burying them in soil in a nursery environment (Roche et al. 1997). While time consuming, ageing seed in soil can be surprisingly effective for producing seedlings if there is no other means to do so and it is recommended for some problematic species (Turner 2013; Chia et al. 2016). In such cases, cleaned seeds are sown into punnets or trays (nursery potting media or field-sourced soil) around the time that they are naturally shed and then placed outside to be exposed to local climatic conditions (e.g. extremes of temperature, and sporadic rainfall events). These climatic events appear to be critical to rendering seeds non-dormant, gradually over several months to years. It is recommended that trays be protected with mesh or wire that is placed over the top to reduce potential predation (e.g. rats, mice or birds), especially when seed trays are unattended for months at a time. As well, it is likely that they will also need to be weeded occasionally if left exposed for any length of time.

At the next phase, seeds may be exposed to aerosol smoke (as previously described) leading into the time of year when seeds are expected to germinate under natural conditions (i.e. autumn in southern Australia). Once the break of season commences (late March to May, in southern Australia), seed trays can be regularly watered (several times per week) to supplement natural rainfall, with trays monitored regularly for seedling emergence. Seedlings can be gently removed (**pricked out**) and potted on as described below, leaving behind the rest of the seeds, which may still be viable.

Some species show no signs of emergence until the second year of ageing in the soil, so require exposure to several different seasons to become non-dormant (Roche et al. 1997). This approach significantly enhanced seedling emergence for *Persoonia longifolia*, along with the addition of several short pulses of summer watering to simulate intermittent summer rainfall events, even though seed trays were kept largely dry during the summer months (Chia et al. 2016). The brief wetting and drying cycles enhance dormancy loss and promote germination once the winter wet season commences in April/May. It is reasonable to expect that other equally challenging native species may respond in a similar way.

Seed sowing approaches

Approaches to sowing seed requires considering seed size, understanding where in the soil profile species prefer to emerge from (surface or at depth) and their sensitivity to being disturbed when potting on to larger containers. In general, if seeds are relatively small (less than the size of a tomato seed), seeds can be surface sown then either covered with a thin layer of sieved (1-5 mm) aggregate (i.e. coarse river sand), sieved potting mix or vermiculite to a depth approximately several times the diameter of the seed or left on the soil surface if very small (< 1 mm) (Davies et al. 2018). For very small surface-sown seeds, there is some susceptibility to seeds floating or washing to the edges of containers which may result in clumps of germinated seedlings around the container edges and few seedlings in the centre (which may impact on their growth and ease of pricking out). For larger seeds such as *Acacia* (wattles; which can be pre-treated before sowing, see Module 11 – Seed Germination and Dormancy), these can either be pushed individually into the potting media or surface-sown and covered by a thin layer of media (0.5 to 1 time the diameter of the seed). Very large seeds, for example such as those found in Zamiaceae (cycads) (Figure 4), *Santalum* (sandalwood), and *Araucaria bidwillii* (bunya pine) should be half buried lying longitudinally, so seed is still partly visible on the potting media surface. In certain cases where species require light to germinate (i.e. some small seeded species like Asteraceae (daisy) or *Wahlenbergia* (native blue bells)), seeds are surface sown onto sufficiently moistened seed germination media (as described below), and left uncovered and exposed to light. Watering (with fine sprinkler heads) or misting is recommended for these and other surface-sown species and also careful monitoring so seeds do not dry out or become dislodged and lost from the media surface. These should also preferably be maintained under greenhouse conditions rather than outside (Davies et al. 2018).

Figure 4. Examples of the very large seeds of *Macrozamia fraseri* (left) and *Podocarpus drouynianus* (right) that have been sowed so they are half buried in the media for optimal seedling emergence. (Photo: C. Elliott)

On occasion, germinated seeds can be extracted from agar plates or filter paper in germination trays (Figure 5) and grown on in containers, however, these may be especially prone to failure if not acclimatised appropriately or are **etiolated**. Good hygiene practices are critical to the success of this process, as contaminants (e.g. fungi, bacteria) can easily kill these vulnerable seedlings in this environment (see Nursery hygiene practices below). Seeds germinated on agar (or germination papers) have come from a high humidity environment and maintained under constant conditions (i.e. temperature and reduced light), thus plant tissues may be quite soft and susceptible to collapse once moved. In such cases, the seedling may be planted into small pots, placed in hooded trays and slowly acclimatised within a controlled environment (i.e. fogging unit or climate-controlled propagation house). This process involves leaving the hoods on the trays with the vent closed for the first 2-3 days, after which the vents are partially opened decreasing the humidity within the tray. Gradually over time (i.e. every few days), the vents are opened fully with the hood eventually removed. This process can take several weeks depending on the external climatic conditions and the sensitivity of the seedlings.

Figure 5. Left: Seeds germinating on agar plates. (Photo: L. Commander)
Right: Close up of a germinating *Microseris lanceolata* seed on germination paper. (Photo: P. Gibson-Roy)

Choosing propagation containers

Depending on the circumstances and seed size, seeds can be sown into a range of different containers such as punnets, seedling trays or pots (Figure 6). Many large-seeded seedlings produce a rapidly developing radicle/root so require extra depth to minimise root malformation and defects ('J' rooting – Box 1). As well, sowing one seed per container for larger seeded species can help prevent seedling roots excessively intertwining making them difficult to separate without significant damage. Communal containers are generally better suited to small-seeded species which are easier to separate once they germinate (Beyl and Trigiano 2015). Alternatively, for species that do not like excessive root disturbance, small numbers of seeds can be sown into individual cell trays, which can be lifted out as a plug once the seedling has developed a sufficient root system (Figure 7). These methods are quick and space-efficient as only germinating seedlings are potted up, meaning less space and fewer resources are required (Davies et al. 2018).

Figure 6. Seeds can be sown into a range of different containers such as punnets, seedling trays or pots. (Photo: L. Commander)

Figure 7. Cuttings (*Grevillea preissii*) that have been struck into individual cell trays, which can be lifted out as a plug once the plant has developed a sufficient root system. (Photo: D. Blumer)

A significant disadvantage of using punnets, seed germination trays or individual cell trays is the requirement for increased handling, as young seedlings need to be removed from the initial pot (or pricked out) before growing too large (Figure 8). Seedlings that outgrow their initial containers can experience significant transplant shock when re-potted, root system damage when moved and 'J' rooting (Box 1) if seedlings are potted up carelessly (Beyl and Trigiano 2015). As an alternative, multiple seeds can be sown directly into larger pots (i.e. standard forestry pot) that will be used to support the ongoing growth of young plants without a disturbance event during early-development (Figure 9). Remember to always adequately label the containers to maintain the link between the detailed record database (see Module 4 – Record Keeping) and the live plants that are produced from propagule sources (i.e. parent plant or seed lot) to ensure accurate tracking of nursery stock through the plant production process (Figure 10).

Figure 8. Many seeds can be sown in punnets, which once germinated, are then gently separated and repotted. (Photo: L. Commander)

Figure 9. Seeds sown individually in forestry pots. (Photo: L. Commander)

Figure 10. Always label the containers to keep track of the species and seed lot. (Photo: L. Commander)

Box 1 – What is J-rooting and why must it be avoided?

Often seeds larger than a pea or from dominant tree species will produce a radicle (the newly developing root) which will grow rapidly into the seed sowing media before significant shoot development can be seen. Sowing these seeds in shallow trays may result in the roots reaching the base of a punnet or tray well before the leaves and shoot indicate it is ready for repotting. This may lead to root damage upon removal or a compromised root architecture that produces seedlings of poor health. In such cases, it is better to sow these seeds into deeper community pots or individually into larger forestry pots with sufficient room to facilitate normal early root growth and development (Thomas et al. 2008).

It is recommended to standardise pot size where possible. While cell trays (which come in various sizes) may be preferable for smaller, resilient and faster growing species such as *Atriplex* (salt bush) and Poaceae (grasses), forestry pots are suggested for larger sized species (Corr 2003). To produce more **advanced tubestock** such as trees, larger pots may prove to be advantageous, though plants may need to be older (>12 months) before planting out. Anti-spiral/root training/air pruning pots with porous/slotted sides designed to reduce root spiralling can enhance and encourage healthy root system development (Beyl and Trigiano, 2015) (Figure 11). The open slotted design “trains” roots to grow down and out rather than to simply spiral around themselves into a ball thus becoming root bound, which may cause significant health problems upon planting. Root spiralling is a common problem with nursery-grown plants and impacts plant performance in situ through lower survival, reduced plant growth and stunting or total plant loss (possibly some years later) as the root system fails to develop adequately to support above ground plant growth. The Australian Standard (AS 2303:2018) contains methods for ensuring the production of high-quality advanced tree stock.

Figure 11. A type of air-pruned pot that stimulates healthy root development. (Photo: L. Commander)

Biodegradable containers

At present, plant production mainly occurs in plastic pots that, in many cases, can be recycled or re-used, to prevent them ending up as waste in landfill. However, there are alternate container products now available that biodegrade (biopots) over several months to a year or two and support more sustainable plant production practices (Beyl and Trigiano, 2015). Besides increased sustainability, biodegradable pots can be advantageous for certain species that respond poorly to disturbances of the root zone when replanted. Biopots are generally made from pressed fibrous materials like peat, coconut fibre, wood pulp, rice or wheat husk (i.e. Fertil®, and Jiffy® pots) with some of the more recent types now available physically similar to plastic pots (Figure 12). These products are either used by removing the plants from pots at the time of planting (like plastic pots), and the remaining pot is then composted or the plant and the biodegradable pot are planted in situ together (as a unit) with the biopot degrading in the soil while the plant pushes out and grows into the surrounding substrate (Muriuki et al. 2014).

Implications for the use of biodegradable containers in nursery propagation processes involves understanding their performance dynamics, such as their higher porosity and water loss (when compared to standard plastic pots) in which case a more intensive irrigation regime may be required. Their rate of degradation (i.e. changes in structural integrity over time) under standard nursery conditions, may also cause problems when used long-term in a horticultural setting (e.g. >3 months). Some biodegradable pots may also be more sensitive to breakage (i.e. brittle) when used in conjunction with standard potting or planting machines (Beyl and Trigiano, 2015). Product testing that examines pot performance under nursery conditions and at the time of in situ planting, their impact on root development and long-term survival, as well as growth and development of biopot tubestock under field conditions should be conducted to determine their utility for use in restoration. Initial assessments of biopots have been encouraging with several potential benefits noted such as reduced transplant shock, increased survival and improved plant health (Figure 12) as well as significantly less reliance on plastic (Muriuki et al. 2014).

Figure 12. Left: *Conostylis aculeata* seedlings after several months growth in a standard plastic pot (left) compared to biodegradable coir pot (middle) and a 'plastic-like' rice husk pot (right). (Photo: S. Turner) Right: *Anigozanthos manglesii* seedlings grown in a standard plastic pot (left) or slotted biodegradable rice husk pot (right) after 6 months growth. (Photo: S. Turner)

Nursery propagation media

Seed germination media

Composite seed germination media usually consist of equal parts perlite, coarse sand and composted organic matter, so they exhibit desirable water retention and oxygen permeation qualities (Davies et al. 2018). This media may be a little different from standard potting media used for young plants and are typically composed of a finer particle size (i.e. sieved to remove coarser materials) and commonly devoid of fertilisers, as these basic substrates are simply used to support initial germination rather than plant growth (Davies et al. 2018). On occasion, it may be desirable to use a more complete potting media that supports both initial germination as well as seedling growth over the first few months, if seeds are sown directly into larger pots (i.e. forestry pots). In such cases, the choice of media type comes down to the species and the horticultural production approach that is planned for that particular species.

Potting media

Many native species naturally grow in well-draining soils, and so prefer a free-draining potting media that is not rich in organic matter or fertiliser (Handreck 2001) (Figure 13). Within each state there are a number of reputable suppliers of good quality native potting media (or mixes), which may be more expensive but are highly recommended. Sourcing potting media from reputable suppliers will ensure compliance with Australian standards for potting media (AS 4419:2018). A typical native potting media may be comprised of composted sawdust/pine bark/fines/coir and/or peat which is mixed with nursery sand and coarser washed river sand (Handreck 2001). Wetting agents can also be incorporated into the media to reduce hydrophobicity and abate rapid drying. Lime and dolomite can be added to adjust the acidity of the potting media which should be around pH 6. Slow release macro- and micro-nutrients are added to support and encourage healthy plant growth, which is particularly important if plants are likely to be in pots for a significant period prior to planting. There are various plant fertilisers available that have been specifically formulated for native species which may be unusually sensitive to nutrients such as phosphorus which can be toxic in high concentrations for Proteaceae species such as *Banksia*, *Grevillea* and *Telopea* (Waratah) (Handreck 2001). As well, perlite or vermiculite can be added to further improve drainage and porosity which can be important for species that are sensitive to having wet roots under nursery conditions (Davies et al. 2018).

Figure 13. Free-draining potting mix that is suitable for native plants. (Photo: L. Commander)

Potting media (and pots) may require pasteurisation or chemical treatment (sterilisation) prior to use to neutralise potential pathogenic organisms and weed seeds (Davies et al. 2018). This ensures healthy stock is produced and maintained, and meets most strict protocols that are required for planting (e.g. see section 6.4.3 in (Commander et al. 2018)). To minimise soil borne diseases, it is recommended to purchase potting media from reputable companies and to check prior to purchase how the supplier maintains an accredited level of growing media hygiene as recommended by the Nursery Industry Accreditation Scheme Australia (NIASA) Guidelines.

Many commercial soil suppliers can provide large volumes of potting media to suit most nursery production systems (e.g. 1m^3 bulka bags – large polypropylene bags or bulk soil deliveries in tipper trucks). Large volumes of potting media should be used in an appropriate time period (i.e. 1 to 2 months), to minimise the leaching or degradation of nutrients, or shifts in pH levels, which will compromise soil quality and increases the risk of deleterious impacts on plant growth and health (Handreck 2001). Options to manage potting media in the nursery include, adding slow release fertilisers to smaller batches of potting media at the time of use; and testing the pH levels of aging potting media (or even new supplies) to check that levels are suitable (i.e. pH 6).

When seeds are not enough – other tubestock propagation approaches

Vegetative propagation (**cuttings**, **division** and **separation**, **tissue culture** and **micropropagation**) can be a highly effective substitute for seed-based propagation when producing tubestock (Davies et al. 2018). As well, vegetative propagation can produce specific numbers of plants of specific genotypes to contribute to the genetic diversity at the site, which may be particularly important for critically endangered species where few individuals remain (Bunn et al. 2007; Commander et al. 2018). In such cases, the production of large numbers of plants can be achieved through other means such as cuttings, division, **layering** (particularly useful for prostrate and climbing species) and/or tissue culture. While generally more expensive, these approaches can be highly effective under some circumstances with each having inherent advantages and disadvantages which are broadly outlined below.

Cuttings

Material

Propagation via **cuttings**, which is the detached part of a stem from a parent plant, is a relatively common way to produce large numbers of plants for restoration projects and is particularly effective for species that are difficult to propagate from seeds, such as some species of *Baeckea*, *Boronia*, *Dampiera*, *Grevillea*, *Hibbertia*, *Myoporum*, and *Westringia* (Stewart 2012). One of the advantages of cutting derived tubestock is that many species reach reproductive maturity much more rapidly. Consequently, flowering and fruiting can occur within 12 months for cutting-derived tubestock rather than after several years as commonly observed for some seedling-derived tubestock (e.g. *Grevillea* and *Telopea* spp.) (Davies et al. 2018).

Cuttings should be taken from either actively growing shoots or from suitably hardened growth from the previous season. In some species this material may only be present at certain times of the year, such as spring or after significant rainfall events (Stewart 2012). When collecting cutting material from in situ populations, where possible, minimise the removal of flowering or fruiting material to ensure that pollination and seed development is not interrupted in the natural population. Consider collecting a variety of cutting material from all parts of the plant, especially if little is known about the species. Some species propagate more effectively from tip cuttings (softwood) while others do so from semi-hardwood, suckering growth or even hardwood. Material taken from lower branches, closer to the root zone and energy stores often strikes effectively. Coppice regrowth may be required if there is a need to source actively growing material (i.e. softwood or semi-hardwood) from older plants with limited (or non-existent) recent new growth. Coppicing (tree pollarding would also have a similar effect as well) involves pruning back the mature plant by removal of 1/3 to 2/3 of its above ground biomass to stimulate vigorous plant regrowth, which is then harvested several weeks to months later and used as the initiation material to produce softwood and semi-hardwood cuttings (Stewart 2012, Davies et al. 2018).

Collection

Cutting material is best collected in the morning as plants are more hydrated due to the cooler conditions. The use of ethylene-reducing 'vegetable bags' may be beneficial for temporary storage particularly if material has long transit time (>2 days). Plant material should be kept cool and moist in fridges or eskies, noting that material from some species may be damaged if kept too wet. Care should be taken not to accidentally crush material or freeze material if using old fridges/ car fridges and to avoid direct contact with freezer bricks/ice if using these to keep the material chilled during transportation. A simple but effective way to do this is to wrap the cutting material in several layers of moist (not dripping wet) newspaper for added insulation.

Propagation process

Tip cuttings are one of the more common ways to initiate cuttings which involves snipping the top 3 to 10 cm of the shoot and then removing the lower branches and leaves with only the top few remaining (Figure 14) (Stewart 2012). In comparison, some shrub species strike more effectively from semi-hardwood cuttings with the soft-tip removed. The bottom of the cutting can then be placed into a suitable rooting gel or rooting powder which contains an auxin (growth regulator – i.e. indole-3-butyric acid; IBA) to stimulate root development adventitiously (Figure 15) (Beyl and Trigiano, 2015). Commercially available root induction products include powders such as Yates® cutting powder and gels such as Rootex® or Clonex® which come specifically formulated for use on softwood, semi-hardwood and hardwood cuttings as needed. Good results may also be achieved from a liquid growth regulator solution called Esi-root® which contains equal proportions of IBA and NAA (naphthaleneacetic acid). Cuttings once processed, are soaked for 15 minutes in a diluted Esi-root® solution then placed into the propagation medium (for further information see Offord et al. (in press)).

Figure 14. Example of softwood tip cuttings (*Chamelacium* sp.) where the lower branches and leaves are removed with only the top few remaining on the cutting. (Photo: D. Blumer)

Figure 15. Propagation process of dipping the bottom of the cutting (*Chamelacium* sp.) into a suitable rooting gel before placing it into a hole, created by a dibbler (top left), in the propagation media and then backfilled to secure it in the media for striking. (Photo: D. Blumer)

To reduce double handling and transplant shock, cuttings from easy-to-strike species can be directly struck in larger pots (i.e. forestry pots) that are to be used for supporting later plant growth, using standard native potting media. To ensure at least one viable plant per pot, several cuttings can be implanted into the same container. Those that either do not strike, die, or remain weak are removed at a later stage simply by cutting off at the base. For more challenging species, 20 or so cuttings can be placed into a punnet containing a suitable propagation media using a dibbler to create a small hole (one to several cm deep) into which the cutting is placed. The mix is then backfilled and pushed firmly against the basal end of the cutting to remove any gaps and air pockets in the soil (Stewart 2012). These dibblers should be regularly decontaminated by soaking in a 0.5% bleach solution or simply replaced following their use. Besides the potting media described above (Figure 16), other substrate options for striking cuttings are pre-formed peat or rockwool plugs (Figure 17). This allows inspection of root formation with minimal disturbance and when potting-on struck cutting material root disturbance and transplant shock is greatly reduced.

To enhance root formation, the punnet/pots/plugs can be placed onto temperature-controlled bottom heating (i.e. heat mats or beds). Bottom heating is usually applied at between 20 and 25 °C but care needs to be taken to ensure the cutting material does not dry out (so regular inspection and watering is essential). For many species, good root development usually occurs within 4 to 6 weeks which should be visible as young actively growing roots emerging out of the bottom of the pot/punnet/plug (Figure 17) (Beyl and Trigiano, 2015). Alternatively, to check if roots are present, cuttings can be gently ‘tugged’ and if they remain firm it is likely that roots have formed.

Cuttings struck in a mist-house or other high humidity environments should be hardened off (i.e. placed in drier and cooler conditions) at least a few days before they are potted up. Once the cuttings have begun to be hardened off they can be carefully removed by tipping out the mix onto a clean work surface which is then gently broken up to separate the young plantlets (Figure 18). Damage to the developing root system should be minimised, with the cutting potted into a suitable pot using an appropriate potting media to support the growth of that species (as previously described) (Davies et al. 2018). Struck cuttings can then be placed into a sheltered environment such as a greenhouse or tunnelhouse to reduce transplant shock, and once clear signs of growth are evident, can be moved either outside or into a shadehouse (depending on the species, local environmental conditions and/or time of year). Once settled into their new pots, tubestock may benefit from regular application of a dilute liquid fertiliser or seaweed extract (Davies et al. 2018).

Figure 16. Left: Cuttings of a variety of species placed into punnets containing a mix of sand, peat and perlite. Punnets sit on a heat bed with misting nozzles to irrigate them. Right: Cuttings in rock wool plugs. (Photos: L. Commander)

Figure 17. Left: Individual cutting of *Rhodomyrtus psidioides* with developing root system in peat plug. (Photo: M. Viler)
Right: Roots extruding from the bottom of a rockwool plug after several weeks growth in a cutting of *Androcalva perlaria.* (Photo: S. Turner)

Figure 18. Cuttings grown in punnets are carefully removed by tipping out the mix onto a clean work surface, so the young plantlets can be gently separated and potted into larger containers. (Photo: D. Blumer)

Division and separation

Material

Division refers to the physical cutting of underground shoots (such as rhizomes) into smaller units with growing points, nodes and some roots on each unit (Stewart 2012). These underground shoots can be found in species such as *Dianella*, *Gahnia*, *Juncus* and *Schoenoplectus.* Separation is used to describe the removal and culture of discrete units such as bulbs (i.e. *Crinum* spp.) and corms (i.e. *Wurmbea* spp.), which are essentially miniature/immature versions of the parent plant (Davies et al. 2018). Plants that are suited to division and separation are commonly referred to as geophytes as most of their growing shoots (i.e. rhizomes, stolons, bulbs, corms) are either below or just above the soil surface. These growing shoots commonly develop their own root systems, sending up new shoots and leaves away from the mother plant. In *Triodia* spp. (hummock grasses), ramets (stolons) are produced which are relatively long aerial shoots that terminate in a new plantlet that develops roots when it touches the ground (Figure 19). The propagation of monocots via division can be very successful, particularly as many are difficult to propagate from seeds, such as some species of Cyperaceae, Restionaceae, Hemerocallidaceae and Iridaceae (Stewart 2012).

Collection and propagation

For division or separation, mature plants are firstly removed from either the pot (if maintained in a nursery environment as part of a container collection) or carefully dug up as a clump in situ. These clumps are then split or separated using either hands or a spade, or more precisely with scissors or secateurs, through identifying and separating the different growing points or bulbs. Strict hygiene practices (outlined below) need to be always followed when digging up plants in situ as there are risks of inadvertently transferring diseases to between sites or to the growing facility or nursery with potentially disastrous consequences (Commander et al. 2018). For example, the recent arrival and spread of the highly infectious pathogen Myrtle Rust (*Austropuccinia psidii*) across eastern Australia has been disastrous for nurseries, agroforestry and local environments with many species of Myrtaceae now seriously threatened with this new disease, so all due care and appropriate precautions should be taken at all times to reduce and eliminate the risks posed by destructive plant microorganisms (Makinson et al. 2020).

Figure 19. Examples of division. Left: *Triodia* ramets which have been cut from the parent plant and have developed roots. (Photo: L. Commander). Right: Developing root suckers of *Persoonia hindii* showing significant signs of growth and development. (Photo: M. Viler)

If harvesting/collecting material with roots, or other delicate organs, it is suggested to use plastic containers with sphagnum peat or a perlite/vermiculite mix to keep roots protected from bruising and drying out while processing. In *Triodia* spp., propagation is approached a little differently, with aerial shoots terminating in a ramet snipped off the mother plants, which are then placed in moist newspaper inside a plastic bag and kept in a warm (30°C) environment for 1 to 2 weeks, while they form roots (Figure 19). Likewise, for *Persoonia hindii* surface roots can be gently harvested and laid upon the propagation medium with shoot and root development occurring shortly thereafter (Figure 19). Once this occurs, the rooted plantlet can be potted directly.

Depending on the species, this form of propagation may be better undertaken while the donor plants are dormant or semi-dormant which can significantly improve survival as transplant shock is diminished (Stewart 2012). To further reduce transplant shock before planting or potting foliage should be trimmed or cut back by up to two-thirds (depending on the species). The new plantlets are usually potted into a standard native potting mix and then left to settle in the nursery for several months to a year to establish new growth. Once the plants are clearly growing and healthy, they can be used for in situ planting (Davies et al. 2018). Although this is usually quite a slow and laborious way to propagate large numbers of plants it can be surprisingly effective and may be the only realistic option for many species in some cases. As well, this method can be used for the establishment of mother stock plants to provide an ongoing source of propagation material to maintain rare species with broad genetic diversity under ex situ conditions when appropriately collected and managed.

Tissue culture and micropropagation

Tissue culture and micropropagation techniques use fragments of plant material, such as leaf, stem, root or embryo, to generate individual plants that are genetically identical to the parent plant (Figure 20). As a propagation technique, tissue culture is highly suited to the production of rare or threatened species in the following circumstances:

- Extreme rarity prohibits sufficient cutting material and/or seeds to be utilised.
- Prior knowledge with same or closely related species indicates cutting propagation unlikely to be successful.
- Life history/morphology of species makes enough suitable cutting material difficult or impossible to access without risking excessive disturbance or harm to the donor plant(s).

Tissue culture can also be useful for common though problematic species that exhibit poor seed set, low germination and/or complex germination and growth requirements such as some species of Cyperaceae, Restionaceae and terrestrial orchids (Willyams 2005; Bustam et al. 2014). Tissue culture may be useful in restoration in the following circumstances (Bunn et al. 2007; Beyl and Trigiano, 2015; Davies et al. 2018):

- Large-scale uniform production of specific desired genotypes (i.e. to enhance drought, disease or salinity tolerance).
- Production of large numbers of plants where genetic variability is not a major consideration/issue, such as highly clonal species that may seldom produce viable seeds (i.e. some native rushes).

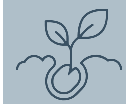
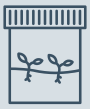
- Ability to utilise in vitro culture material for cryostorage and add multiple genotypes as opportunity arises for future restoration/translocation.
- Propagation and storage of species that cannot be kept under standard seed storage conditions (e.g. recalcitrant seeded species), or which have complex regeneration systems (e.g. ferns and terrestrial orchids).
- Culture of seeds with specific nutritional or other niche growth requirements (e.g. carnivorous plants, epiphytes).
- Co-culture of species requiring or preferring the presence of a symbiont (e.g. terrestrial orchids with mycorrhizal associations) (Figure 20).

A number of commercial tissue culture laboratories now exist across Australia. Rather than investing in the development and maintenance of a culturing facility (in addition to hiring and training specialist staff), it is likely to be more cost effective to engage with a reputable commercial laboratory, or form a partnership with a research organisation or botanic garden for the production of target species. Outsourcing propagation via tissue culture may be particularly appealing if some viable seeds have been sourced from a problematic species or some knowledge already exists as to the micropropagation requirements of the target taxa (i.e. native orchids).

Tissue culture can be viewed as complementary to other forms of plant propagation and only as a replacement under some circumstances (Kodym et al. 2014). While very effective in particular cases, the requirement for specialised consumables, equipment, staff and laboratory facilities can make tissue culture an expensive option (Offord et al. 2009) and thus it is often viewed as an option of last resort (Figure 21) (For further information see Sommerville et al. (2021)).

Figure 20. Left: Tissue culture can be used to produce individual plants by taking fragments of plant material and growing them in agar-based media under controlled conditions. Right: Growing mycorrhizal fungi on agar for future use in terrestrial orchid seed germination. (Photos: L. Commander)

Figure 21. A comparison of four different propagation methods showing their relative cost, time frames, infrastructure requirements, advantages and disadvantages, and an example.

Propagation method	Cost	Time frame for field ready plants	Equipment and facility support needed	Advantages	Disadvantages	Example
Seeds 	Low	Short (4-8 m)	Low	Tubestock with strong root systems	Only practical when seed is available and seed biology understood (i.e. seed quality, dormancy and germination requirements)	*Acacia* spp. *Eucalyptus* spp.
Cuttings 	Low-medium	Short (4-12 m)	Low to medium	Overcomes seed limitation bottlenecks Produces semi mature plants	Plants may not perform as well due to weaker root systems, not all plants strike from cuttings, slower than seeds	*Boronia* spp. *Eremophila* spp.
Division 	Medium	Short - medium (6-24 m)	Low to medium	Can work well with rhizomatous plants, overcomes seed limitation bottlenecks	Slow to establish, can take up a large amount of space, only applicable to a niche group of plants	*Dianella* spp. Rushes and sedges
Tissue culture 	High	Medium-long (>12 m)	High	Small amounts of material required, overcomes seed dormancy and other bottlenecks, very high multiplication rates	Some species difficult to initiate into culture, exhibit poor in vitro growth, low or no root induction and/or endure high death rates on deflasking	Threatened species, species with poor seed quality or complex dormancy

Nursery hygiene practices

A key consideration in creating effective propagation systems is the implementation of sound nursery hygiene practices which the broader horticultural industry defines as the nursery accreditation scheme (for more details on nursery accreditation please refer to the Nursery Industry Accreditation Scheme, Australia (NIASA) best management practice guidelines).
Good nursery hygiene ensures that the risks of disease, weeds and pests are minimised for plants growing within the nursery environment. In particular, there are several plant diseases that are highly virulent and can quickly cause significant stock damage if not effectively managed including Phytophthora dieback (*Phytophthora* spp.) and Myrtle Rust (*Austropuccinia psidii*).
Other plant diseases such as grey mould (*Botrytis* spp.), Pythium (*Pythium* spp.), Fusarium wilt (*Fusarium* spp.) and Rhizoctonia (*Rhizoctonia* spp.) are more of a problem for seedlings and younger plants growing in moist sheltered environments with higher humidity but can be found on occasion in other situations as well (Stewart 2012). Good nursery hygiene practices also protect against the unintended introduction of an exotic biological agent (such as Myrtle rust) into a restoration site which may have far reaching negative ecological consequences (Commander

et al. 2018). To implement good nursery hygiene, all pots and potting mixes should be sourced from either accredited suppliers or pasteurised prior to use. Plants where possible should be kept well above the ground on frames or benches (NIASA Guidelines 3rd edn. 2005) (Figure 22), and not left sitting on the ground unless on a suitable layer (10 to 20 mm) of non-organic substrate (e.g. sieved crushed rock to minimum 75 mm) or a well-draining hard surface (e.g. concrete). Only good quality water should be used for irrigation (i.e. free from water borne diseases, high salt concentrations or extreme pH levels). All pots and ground surfaces should be weeded regularly and pests suitably controlled, particularly leading up to field planting (Commander et al. 2018).

Figure 22. Plants are kept on benches above the ground. (Photo: L. Commander)

To further assist in controlling plant diseases, all equipment such as tweezers, secateurs, spades, benches and trays should be routinely sprayed with **sterilants** such as commercial biocides (e.g. Phytokleen®), 70% methylated spirits or concentrated bleach solutions (0.5 to 2% w/v). These are generally safe for use by people when used according to safety data sheet (SDS) guidelines, nevertheless due care should be taken when handling these products at all times (and instructions for their safe disposal should be strictly followed) (Davies et al. 2018).

Nursery infrastructure

At a most basic level, relatively modest nursery facilities are needed to produce small numbers of plants (approx. several hundred), meaning community groups with basic planning and resources can set up growing facilities and produce a broad range of easily propagated local species. The production of thousands (+) plants annually will require more advanced nursery facilities, which generally include dedicated propagation house, polyhouse, greenhouse, and/or shadehouse facilities along with suitable stand-out areas for hardening off plant stock (Figure 23). The use of this level of infrastructure improves plant quality and survival (Davies et al. 2018).

Figure 23. Hardening off sun loving species in preparation for in situ planting. (Photo: S. Turner)

Plant production requires growing containers of various formats and sizes (e.g. trays, punnets, forestry tubes) and potting media (often to specification and standards). These are readily available from wholesale and retail horticultural suppliers around the country. Work benches (often stainless steel) provide a suitable and ergonomic working surface for activities such as media mixing, potting, transplanting, seed sowing or preparing cuttings. Ideally these can be easily kept sterilised, clean and free from debris (Figure 13). Wire frame benches (often galvanised steel) are generally required to provide a stable and hygienic surface that plants (seedlings, young or mature) can be grown on. These allow free draining of water, easy cleaning and create a barrier between potential pathogens on the ground surface (Figure 22). Alternately, benches that maintain or deliver water directly to plant roots (i.e. wicking or bench surface flow) can be used for specialised species (e.g. wetland) or processes (e.g. striking cuttings) that require this type of access to water or high humidity.

If plants are not to be kept outside, a well-maintained propagation house must provide shelter from environmental extremes but also be well-lit and with some capacity for shading (either internal or external covers), particularly during summer periods (Beyl and Trigiano 2015). Propagation houses can be constructed from glass, polycarbonate or a polyethylene skin fitted over a rigid light weight frame which are relatively low cost to construct. It is recommended that some form of automatic cooling be used to moderate internal temperatures to keep them ideally below 30°C during the summer months (particularly for hotter parts of Australia). Heating mats usually set between 20 to 25°C can be used to enhance and maintain root growth of young plants, particularly during unwanted cooler periods (time of day, season or location). These heating mats supply bottom heat, which can stimulate heathy and rapid root growth enabling plants to have better field establishment once planted out (Beyl and Trigiano 2015).

In very simple nursery settings, plants can be hand-watered but this is time consuming and inefficient in terms of water use. Most nurseries have automated irrigation in their propagation and growing houses, and outside to water benches and outdoor growing areas. Simple home garden style irrigation systems can be relatively cheap to install, but are limited in their capacity and effectiveness, whereas complex computerised and automated systems are costly up front but deliver significant benefits in terms of plant maintenance, time and water efficiency. Irrigation water droplet size can be delivered through nozzles as coarse (e.g. used for outside), fine (e.g. used inside a propagation house) or mist (e.g. used to maintain high humidity or minimise seed movement or seedling damage for small-seeded species) depending on the situation (Beyl and Trigiano 2015). Plants should receive an appropriate amount of water to support healthy plant growth for that species. Too little water may result in sickly stunted plants and exacerbate soil hydrophobicity (as soils dry making them difficult to rehydrate). Conversely, excessive watering can also cause significant problems by leaching fertilisers and leading to anoxic soil conditions which may cause root problems and diseases. Soil hydration can be checked electronically via various commercially available soil sensors which can constantly monitor soil hydration status or simply through regular visual and physical (1 to 2 times per day) assessment of potting media and plant stock particularly during warm periods (Handreck 2001).

Timing propagation to match restoration scheduling

Propagation normally commences between 3-9 months before the planned in situ planting (depending on the species). This period gives tubestock adequate time to germinate or strike and grow to a size that is suitable for field planting. If seeds are sown too late or cuttings struck too late, their root system may still be underdeveloped and collapse when the plant is removed from the pot. It should be noted that for quick growing species or taxa with large root systems, plant roots can quickly become overgrown and root-bound if propagated too early (a common problem for *Acacia* and *Eucalyptus* spp.), so appropriate timing, good record keeping and stock management (i.e. visual checks of root growth) are important considerations (Corr 2003).

Generally speaking, it is important to ascertain the ideal planting time for the target installation location based on seasonal triggers for plant growth; and determine the time taken for each species or each propagation method to produce tubestock of suitable size for planting, and then generate a nursery schedule to meet these requirements. For example, most field plantings in south west WA occur during the winter wet period (May to August) after the arrival of the first winter rains (Stevens et al. 2016). Consequently, propagation of tubestock from seed usually begins in November and can continue into February for more rapidly growing species such as some grasses, legumes and *Eucalyptus,* so for most species the time from seed sowing to field planting is usually between 4 to 6 months. Field plantings in the Sydney basin and Southern Victoria are mostly conducted in early-mid autumn (April) to take advantage of warm soil temperatures and good rainfall, before the onset of cooler and drier winter conditions. Propagation of tubestock from seed usually begins a little earlier in September-January to allow sufficient time for growth.

In some cases, older/larger plants may be utilised in field plantings. The reasons for using what is often termed advanced stock include, the species may be slower growing (e.g. Cycads), show higher survival when planted as advanced stock (e.g. some rainforest species) or which are known to be logistically unfeasible to plant as smaller tubestock (i.e. within the desired time frames) due to insufficient site preparation, drought, or lack of planting approvals. In such cases, plants should be grown in appropriately sized containers and this will require potting-on into larger pots. This is done to support ongoing root system development so they are capable of supporting above ground biomass (i.e. leaves, shoots and stems) which ensure stock is ready for in situ planting without undue stress (Davies et al. 2018). Advance stock therefore needs to be appropriately maintained over longer periods under nursery conditions and this must continue to keep plants in optimal health (i.e. free from pests, weeds and diseases, not experiencing nutrient deficiencies) through the use of good horticultural practices.

Hardening off and acclimation of seedlings

Tubestock maintained under 'ideal growing conditions' might appear to be healthy, but their tissues are often 'soft' and very sensitive making them susceptible to extreme conditions and potentially more prone to pests and diseases. This is especially so if they are transferred quickly from nurseries and planted in situ (Corr 2003). To address this issue, tubestock are 'hardened off' before they are taken from nursery conditions and planted in the field. This process entails moving stock from a highly sheltered environment (e.g. growing houses) to less sheltered conditions (e.g. shade house), then finally to an exposed setting (e.g. stand out areas; Figure 23) over a period of several weeks. This process gradually prepares the plants for outside conditions which are more similar to what they will experience when planted in situ (in terms of light intensity, temperatures, humidity and evaporation). However, some species may not require this slow transition from sheltered to exposed conditions (such as some *Acacia* and *Eucalyptus* spp.) and can transition to outside growing conditions shortly after they have developed their first true leaves.

Delivering plants to site

It is essential to properly plan the logistics of transporting nursery stock to a planting site. This should consider the number and size range of the planting stock. This will ensure that suitable modes of transport are used to avoid damage and stress to the plants. For small numbers of plants utilities and/or box trailers are often sufficient, but for large stock numbers specialised trailers with built-in shelving may be required (Figure 24). Where possible, it may also be advantageous to transport young plants to a holding site close to their final destination several weeks prior to planting. This will aid transition to local conditions (particularly remote areas) which may be quite different from where they were grown, and so may make a significant difference to long-term plant survival and growth once planted out. Ideally such a temporary holding site is a makeshift plant nursery with basic irrigation and protection (e.g. fences) from herbivores such as rabbits, goats, and kangaroos.

Figure 24. Plan how the plants will be transported to the project site. (Photo. L. Commander)

Site preparation, field planting and follow up care and maintenance

While the production of healthy and actively growing tubestock is the end goal for any plant nursery – from a restoration perspective, it is just the beginning of the in situ restoration process. It is not uncommon for many plants that have arrived at a restoration site in optimal condition to die within a relatively short timeframe once planted in situ. Plants can die for a host of reasons (e.g. extreme drought and temperature, frost stress, herbivory, vandalism) some of which cannot be controlled, steps can nevertheless be taken to minimise initial planting losses, particularly during the first 12 months (Close and Davidson 2003). These steps may include supplementary watering, the use of tree guards, fencing, wind breaks, shade and the targeted use of herbicides, pesticides and fungicides (Corr 2003; Biodiversity Conservation Trust 2019). An example of this complete process is outlined in Commander et al. (2017), and demonstrates the site preparation, installation, experimentation, and monitoring that are involved in restoring a threatened species community.

Quality

A key factor in reducing tubestock loss is to ensure that the plants are in good health (e.g. showing no signs of disease, pests, or nutrient deficiencies) and properly hardened off (as previously described) at the time of planting. Plants should also be actively growing and neither too small nor too large for their pots. As well, excessive fertiliser should not be applied to the tubestock prior to in situ planting as this may result in rapid growth of soft green leaf material which may make them especially prone and vulnerable to herbivores and diseases.

Site preparation

Proper site preparation is paramount to improving planting success. Weeds should always be controlled prior to planting. Standing weed biomass can be reduced using chemical herbicides (selective, pre-emergent), slashing or burns prior to planting. Post-planting weed control is also critical and can be addressed through the use of herbicides (selective, and post-emergent) or by mechanical means (slashing and mowing) (England et al. 2013). Where possible, ripping the soil (using a tractor with tines working down to 50 cm soil depth) may also improve survival and growth (Figure 25) (Commander et al. 2013).

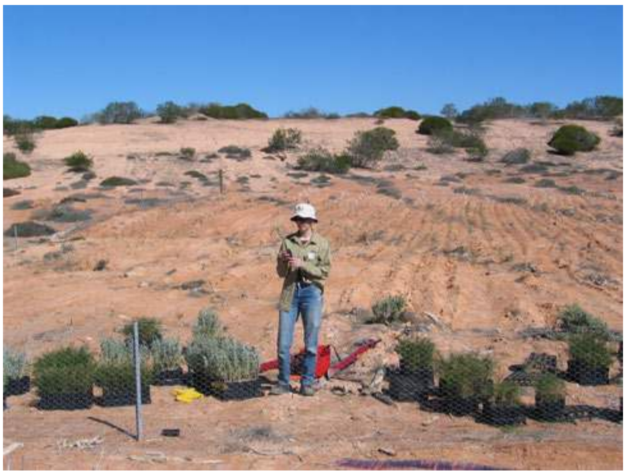


Figure 25. Planting seedlings into rip lines in heavily compacted soil. (Photo: L. Commander)

Ripping reduces soil compaction, thus improving water infiltration and enhancing root system development, which is critical for recently planted tubestock. In some circumstances, cross-ripping may be required in severely compacted soils and in heavier soils.

Planting

Figure 26. Potti pukis can be used for planting seedlings. (Photo: L. Commander)

Plants can be placed into the ground in several ways. For large-scale plantings such as in agricultural lands, automated planters using a tractor that rips the soil then places the seedlings into the resulting furrows is highly effective (England et al. 2013). For smaller scale plantings, potti pukis (steel planting device used for precision planting of tubestock) (Figure 26) with kidney trays (hold ~40 easily accessible seedlings while strapped to your upper body) or Hamilton Treeplanters, used for planting forest tubes, can be utilised by a small planting team to place hundreds of plants into the ground in only a few hours. In harsher environments such as mine sites, rocky landscapes or compacted soils, petrol driven augers can be highly effective in rapidly digging holes for planting into hard ground. For volunteer and community groups undertaking smaller scale restoration plantings, hand planting using trowels is often the preferred method. Some practitioners apply planting amendment products such as slow release fertilisers or water retaining granules in the planting hole to assist root development and plant establishment. As well, spraying with salicylic acid (SA) or other plant stress signalling compounds or an anti-transpirant such as Yates WiltNot® has also been shown to improve drought and heat tolerance in a number of species so may be worth considering (Pandey et al. 2017).

Plant protection

Once in the ground, individual tree guards (either plastic, mesh or shade cloth) or plant mats can be placed around each plant (Figure 27). These may confer several benefits to the young plant, for instance, they can protect them from predators and to some extent weeds while the plant becomes established. In hot conditions, shade cloth guards (50 to 70%) are preferable to plastic guards as they effectively moderate the environment around the plant reducing temperatures, soil evaporation, wind burn, and plant transpiration (Figure 27) (Close et al. 2009). Depending on the species tree guards should be removed after 3 to 12 months usually once the plant is well established and beginning to grow above the guard. It should be noted that if left for too long, guards may eventually compromise plant growth, as well as add to the accumulation of local environmental plastic pollution. However, biodegradable tree guards are now becoming widely available which facilitate both good air movement and sun protection though may cost a little more than standard plastic guards. In some circumstances, it may be possible to fence the entire site to exclude herbivores, until plants are well established rather than individually placing tree guards around each plant (Figure 28).

Figure 27. Left and middle: Shade cloth tree guards can be particularly effective in supporting plant establishment in harsh and extreme environment such as waste rock dumps in semi-arid mining environments. (Photos: K. Chia and S. Turner). Right: Open weave onion bags are also useful for full sun requiring species to deter local herbivores. (Photo: K. Chia)

Timing

The timing of planting is important. Planting is preferably aligned with the time of year that seedling recruitment usually occurs for those environments which have marked wet and dry seasons. For other environments where water availability is not such a limiting factor timing is more centred around avoiding climatic extremes such as frost, snow or severe temperatures (see Timing propagation to fit in with restoration schedule section above). In such situations planting is normally undertaken after the soil has received the first breaking rains, though for plantings that incorporate some form of irrigation this may be relatively less important (Figure 29).

Figure 28. *Symonanthus bancroftii* planting site with fencing, irrigation and wire mesh tree guards. (Photo: B. Dixon)

Irrigation

To reduce planting losses due to highly variable rainfall, where feasible, supplementary watering via an automated trickle system is recommended for restoration programs involving threatened species (Commander et al. 2018). Gravity fed trickle systems linked to 1,000 L tanks have been shown to be effective and tanks may only need to be refilled every week or two. To help plants adjust to the local environment, watering might only be programmed to occur 1-3 times per week (depending on environment), delivering a few hundred millilitres of water each time. Watering should only occur for the first 12 months or less (Close and Davidson 2003). Ideally, irrigation should be scaled back as soon as possible, so that plants can adjust to the local environmental conditions. This scaling back may roughly align with the end of the local wet season or if required, before the start of the next wet season.

To assist with the maintenance and monitoring of remote sites soil moisture/temperature/visual logging technology are now available that can issue alerts if equipment fails (i.e. automated irrigation systems) or if conditions become too harsh and remedial treatment is required (e.g. extra watering, pest management). While up-front cost may be high, the cost should be weighed against the cost of restoration investment and risk of failure which may be particularly important when managing threatened species translocations (Commander et al. 2018).

Figure 29. *Eremophila resinosa* translocation site showing irrigation pipes. (Photo: B. Dixon)

Monitoring

Once the plants have been installed in situ, they should be regularly monitored. The first monitoring should be completed within four weeks to gauge how plants have initially responded to the local environment (i.e. either survived or early decline) and coped with in situ planting. For the 12 months thereafter, monitoring to some degree should be undertaken every three months, then once per year around the same date (e.g. end of summer). At each monitoring event, the height, health, and reproductive stage (i.e. flowering or fruiting) of plants can be measured. Either all plants can be monitored (recommended when working with a small number of plants, or a threatened species) or a subsample of plants from the larger planting. Subsampling can be done through either selecting a random sample of plants at each restoration site that are used each time or through the use of permanently marked monitoring quadrats (e.g. 20 m x 20 m) that can be regularly revisited and assessed over many years. This provides valuable information that can be used to update and refine future restoration approaches that ultimately should improve restoration success in the years to come (Commander et al. 2018). The results of the monitoring can be compared with the targets, goals and objectives of the restoration project to determine whether or not they have been achieved, and if further action is required such as additional plantings (see Module 1 – Introduction).

Acknowledgements

Thanks to Julie Percival, ANBG and Lorraine Perrins, Royal Tasmanian Botanical Gardens. Thanks also to Amelia Martyn Yenson, Ross Rapmund, Warren Worboys, Meredith Cosgrove, Shannon Murphy and others for reviewing this module.

Glossary

Advanced tubestock: tubestock that is older (>12 months) than standard tubestock (3-9 months) at planting, and used for species that are slow growing or have better survival post-planting than younger tubestock.

Aging seed: keeping seeds in soil over a prolonged period of time to alleviate dormancy.

Auxin: a plant growth regulator which stimulates root development.

Coppicing: the pruning back to near ground level of a tree or shrub to stimulate new growth.

Cutting: the part of a stem from a parent plant used to propagate tubestock.

Deeply dormant: The state in which healthy seeds are exceptionally difficult to germinate, and which require an extended period of time within the soil seed bank to become non-dormant and germinate.

Dibbler: dedicated probe such as a wooden skewer which makes a hole not much larger than the stem of the cutting in propagation media for planting into.

Direct seeding: seeds are directly placed into or onto the soil in situ.

Division: a propagation technique whereby plants with rhizomes or stolons are divided up into many individual plants each with a separate root system and growing point.

Etiolated: plants that have elongated inter-nodes and are weak, commonly stimulated by a lack of light.

'J' rooting: Common root development problem that occurs when the young tap root is damaged, distorted or bent during transplantation which may compromise medium to long-term plant growth and development.

Hardwood cutting: a cutting taken from an older part of the plant, not new growth. The stem may be hard or inflexible.

In situ: in the natural, original or appropriate location (i.e. the field or natural population).

Layering: a propagation technique where stems are pinned to the ground, or wrapped in a substrate such as moss, to stimulate root formation. Stems are then cut from the plant once roots are formed.

Micropropagation: the process of producing plants vegetatively through tissue culture using small amounts of plant material cultured under aseptic conditions.

Plant growth regulator: a natural or synthetic chemical used to control and modify plant growth, such as the production of adventitious roots or multiple shoots.

Pollarding: removal of the top branches of a tree to encourage new growth.

Pricked out: process of gently separating and removing seedlings from where they emerged together in the same container to individual containers.

Propagule: Any part of a plant capable of growing into a new organism (e.g. spore, seed, cutting, rhizome, stolon, bulb, corm).

Recalcitrant: desiccation intolerant seeds, i.e. seeds that do not survive drying (c.f. orthodox).

Seed dormancy: A dormant seed (or other germination unit) is one that does not have the capacity to germinate in a specified period of time under any combination of normal physical environmental factors (temperature, light/dark, etc.) that otherwise would be favourable to support germination, i.e. after the seed becomes non-dormant.

Semi-hardwood: a cutting taken from an older part of the plant or new growth. The stem may be flexible and somewhat hard.

Separation: a propagation technique in which discrete units such as bulbs and corms are removed and cultured as separate plants.

Serotinous: species which store their seed within fruits (i.e. *Banksia* cones) in their canopy for an extended period of time after seed maturity.

Softwood cutting: a cutting taken from new growth on the plant. The stem may be soft, flexible and green.

Sterilants: chemical agents such as sodium hypochlorite, hydrogen peroxide and ethanol used to eliminate problematic microorganisms.

Tissue culture: a propagation technique in which fragments of plant material (e.g. leaf, stem, root or embryo) are grown in a sterile media containing nutrients to produce individual plants.

Tubestock: plant grown from propagules (see above) in containers, pots or tubes for in situ planting. Commonly used term for plants grown in forestry tubes or small containers. Similar terms include greenstock, plant stock or planting unit.

Online resources

Australian Native Plants Society (Australia) – Smoke Germination of Australian Plants
http://anpsa.org.au/articles/smoke-germination.html

Nursery Industry Accreditation Scheme Australia NIASA Guidelines
http://nurseryproductionfms.com.au/niasa-accreditation/

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