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Department of Planning, Industry and Environment

Monitoring Manual for Invasive and Native Flora

Guidance for field monitoring and reporting

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Cover photo: Ox-eye daisy (*Leucanthemum vulgare*), Kosciuszko National Park. Pete Turner/DPIE

Authorship and citation

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This document should be cited as: Watson GM, French KO, Burley AL, Brading MB and Hamilton MA 2021, *Monitoring Manual for Invasive and Native Flora: Guidance for field monitoring and reporting*, NSW Department of Planning, Industry and Environment, Sydney.

Published by:

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ISBN 978-1-922767-05-9
EES 2021/0368
January 2022

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Introduction

Purpose of this manual

Monitoring is essential for conservation management and assessment of impacts to biodiversity, threatened species and ecological communities, and the effectiveness of management interventions to protect them. It is critical to understanding drivers of population change, testing appropriate management techniques and effectiveness, and for disseminating robust and appropriate information about species and communities. Monitoring allows us to examine if the management we are doing is achieving the desired outcome or if we need to make changes to see that happen. It provides an opportunity to adaptively manage to improve biodiversity outcomes.

Given there is no concise monitoring guide available to measure the response of biodiversity and weeds to weed management, this manual is based on the previous *Monitoring manual for bitou bush control and native plant recovery* (Hughes et al. 2009), other texts, previous monitoring programs, and the authors' knowledge of ecological monitoring techniques. This manual should be used to monitor the response of invasive weeds, native plants and ecological communities before, during and after weed control programs.

Legislative context

There are several pieces of legislation that necessitate the effectiveness of weed control to be monitored and reported, in order to protect biodiversity, including the NSW *Biodiversity Conservation Act 2016*, NSW *Biosecurity Act 2015* and the NSW *Local Land Services Act 2013*, and beyond. This manual has been revised to address this need, to provide a resource for stakeholders to monitor the response of native biodiversity to the control of invasive weeds.

Why do we need this manual?

Weeds in New South Wales

Invasive species are amongst the greatest threat to biodiversity in Australia. There are over 1650 introduced weed species in New South Wales, with seven weeds or weed groups identified under the Biodiversity Conservation Act as key threatening processes to biodiversity, and at least 300 species identified as having significant environmental impacts (NSW Invasive Species Plan 2018–2021, DPI 2018a). Without control efforts, many weed species can form monocultures, displacing and out-competing native species (DPI 2018a).

What is monitoring?

Monitoring is the repeated collection and analysis of observations (e.g. density or cover), that can provide information on changes in order to answer questions about management. The important part of this definition is that monitoring data is collected for an explicit reason; for example, to assess if your control program is meeting a management objective. Monitoring and subsequent analysis is what you do to determine if your weed control actions have been effective. Effective monitoring must consider what to measure, how best and how often to measure, how to record those observations, and how long to continue monitoring.

Why is monitoring important?

Monitoring is often perceived as resource intensive and an expensive (or unnecessary) component of land management work. Particularly within the weed management field it can often be seen as diverting funds and resources away from the control effort. But if the responses of weeds, native plants and communities to management actions are not monitored nothing can be learnt about the effectiveness of the actions nor whether other management actions are needed. **This is the underlying reason for all monitoring: to ensure what you are doing is achieving the outcome you intended.**

It is recognised that some forms of monitoring can be costly due to the high expense of labour and the time involved. However, not all monitoring is time consuming or costly, especially if a simple metric can be identified that is quick to assess and only the required data is collected. Once a program is established, ongoing monitoring costs are much lower. It is also acknowledged that resources are simply not available to monitor all weed control programs, especially those where the response of weeds to management is well known; where impacts to biota are not immediate; where the effectiveness of management can be evaluated visually during a site visit; and/or for lower priority programs. Monitoring, as per management, should be prioritised to areas of greatest need.

Monitoring provides you with the tools to assess whether your management actions are being effective and, if not, provides information to help you determine what changes you should make. Often, management regimes are implemented, and it is assumed the intended outcome will eventuate; monitoring removes the guesswork. From a land manager's perspective, the challenge is how to evaluate whether management objectives are being met with limited resources. Monitoring is therefore an essential part of adaptive land management that, when done properly, allows a transparent assessment of program outcomes, and saves time and money in the future.

How will this manual help you?

This guidance should be used to design your own monitoring plan, relative to your expertise, resources and a timeframe over which you'll be able to see the results of your control program. With a specific monitoring program, you will be able to adapt your control program to be most effective for your desired outcomes.

Structure of this manual

The techniques presented within this manual range from simple qualitative assessments for preliminary observations to more in-depth, robust monitoring methods to demonstrate outcomes. This allows land managers to adopt the techniques most suitable to their objectives, outcomes, skills and resources.

We have developed a two-tiered approach to monitoring, reflected in this manual as chapter one, **preliminary observations** and chapter two, **advanced outcomes**. These chapters build upon one another in the level of skills and resources required, and in the complexity of the monitoring question they are designed to address.

The chapters and the sections within them are written for use as independent units and you need **only use the section(s) relevant to your needs**. Table 1 can help you choose which sections you need.

Table 1 Essential (✓) and optional (+) monitoring techniques presented in this manual
In the advanced outcomes chapter at least one of the sampling techniques should be used, others are optional.

Section in manual		Chapter the technique relates to	
Technique	**Pages**	**1. Preliminary observations**	**2. Advanced outcomes**
Mapping	7 – 10	✓	✓
Observational data	10 – 16	✓	✓
Photopoint data	16 – 18	✓	✓
Control and monitoring activities	19 – 20	✓	✓
Line-intercept sampling	48 – 42		✓/+
Quadrat sampling	42 – 45		✓/+
T-square sampling	46 – 48		✓/+

Weed control: A staged approach

At many sites the abundance and area infested by a target weed species (or sometimes multiple weed species) is such that it cannot be controlled in a single control event/action. Thus, control at these sites needs to occur in stages.

The **first stage** is the removal of the target weed species and other weed species from the immediate vicinity of the native species, population or ecological community at risk. This will reduce the impact and/or direct threat in the short term.

The **second stage** is the expansion of stage one to cover a larger area of the target weed infestation at the site. In this stage, the removal of the target weed species should be prioritised to areas containing suitable habitat for the priority native species, populations and ecological communities to expand into in the future. Priority species, populations and ecological communities are those directly impacted by the weed infestation at the site. If such suitable habitat areas do not exist, concentrate on the highest risk dispersal pathways. This will decrease the threat by providing a larger buffer zone between the weeds and the threatened entity. Stage two also involves the follow-up control of seedlings that germinate within all previously controlled areas (including stage one areas).

The **third and subsequent stages** involve the further expansion of earlier stages with the aim of removing all of the target weed species from the site and surrounding areas, to prevent re-invasion. This stage also includes the **continual follow-up control** of the target weed species seedlings in all previously controlled stages/areas of the site (areas where control stages one and two occurred).

This staged approach is beneficial for a number of reasons:

- Control can be focused initially on areas where impact to native species, populations or ecological communities is greatest.
- Control is focused on an area for which there are sufficient resources available.
- Follow-up control constraints are considered.
- A plan can be drawn up to manage large infestations over time.

The staged approach should:

- be planned before control is undertaken, with all stages clearly marked and the timing of each stage determined, and preferably incorporated into a site management plan, and
- only control areas for which there are resources available to undertake the subsequent stages, including **most importantly the follow-up treatment** of seedlings; irrespective of the initial control measure implemented, follow-up treatments are required to control recruitment (as described by Winkler et al. 2008).

1. Preliminary observations

The techniques and methods described in this chapter are central to the design of your monitoring program, allowing comparisons across sites (Table 2). These techniques are largely qualitative and may not detect changes in the cover or density of the target weed species or the priority native species, so make sure your particular management or monitoring question will be answered by these techniques.

Table 2 Advantages and disadvantages of the monitoring techniques that provide preliminary observations

Technique	Uses	Advantages	Disadvantages
1. Mapping	Recording broad changes in the distribution of native and invasive flora and ecological communities	Shows the spatial relationship between key species and communities. Displays changes in affected areas and guides future control programs	Cannot be used to quantitatively demonstrate recovery of key native species or communities after weed control. Precision will vary with map scale and methods used to create boundaries. Difficult to do so accurately
2. Observational data a. Estimates of cover and density	Estimating canopy cover and density of the key native and invasive flora	Fast, simple. More informative than presence/absence observations	Potentially imprecise and subjective, with unknown bias between different observers. Cannot detect small changes in cover/density or species richness. Only a limited number of species can be monitored
2. Observational data b. Ecological observations	Recording basic ecological information on the species found at a site	Provides qualitative information on the state of target species and response to the target weed species and its control	May be subjective, given differences between observers. Information gathered will vary with experience of observer
3. Photopoints	Recording changes in native and invasive species abundance and changes to ecological communities with images	Fast. Can show visual changes that would be difficult to record numerically. Useful to combine with observational data sites and for plot relocation	Low importance compared to other monitoring techniques. Only provides a qualitative, visual demonstration of change. Limited area monitored with no data. May not accurately reflect changes. Very limited value for broader reporting
4. Assessment of control and monitoring techniques	Documenting which control activities were used and the costs of implementing controls	Provides a detailed record of activities and associated costs. These costs are often undocumented (but useful for planning future programs)	Cannot be used to demonstrate recovery of priority species or communities after invasive species control

1.1 Define the objective of your observational monitoring program

Defining your objective is the key to successful monitoring using observational data, as different objectives may require different locations or orientations. Therefore, define your objective carefully **prior** to establishing locations in the field.

Observational monitoring can allow you to:

- provide an assessment of the abundance (using cover and/or density indices) of the target weed species, before and after control
- establish the presence or abundance (using cover and/or density indices) of native or invasive species (those under threat from the target weed species or of interest to you for another reason) over time
- monitor the growth and health of individual native plants or species
- identify broad or simple changes in ecological communities, and/or
- observe the increase or decrease in abundance (using cover and/or density indices) of other weeds that have the potential to invade after control of the target weed.

1.2 Which species should you monitor?

After deciding on the objectives, the species (target weed, priority native species, and other weeds) to be monitored are then identified. For example, if the aim of your control and monitoring program is controlling lantana and monitoring the recovery of native vegetation after control, your target weed species is lantana. Your priority native species are those threatened by lantana at your site (or a selection of them if there are many, which can be used as a proxy to indicate recovery). Other weed species are those identified as likely to reinvade the control site and compete with the priority native species following control of the target weed species. If your priority native species are rare or threatened, you should also monitor surrounding common and fast-growing native species as these are most likely to respond to weed control in the first instance.

1.3 How often should you monitor?

Sites should be monitored just before any control is undertaken so you have an accurate picture of what the site was like before undertaking management. The lowest frequency of repeat monitoring would be to monitor again just before the next control event occurs, if occurring annually. You can also monitor soon after each control event to assess the effectiveness of the control of the weeds; the response of native plants to control will take longer. Generally, the frequency of monitoring is influenced by target weed and priority species growth rates, the expected length of the control program and time to achieve outcomes, and available resources.

If you have already started control efforts, we recommend you record the history (to the best of your knowledge) of those efforts and begin monitoring as soon as possible. The history of the site and previous control efforts can then assist you in the interpretation of your monitoring results.

1.4 How long should you monitor?

If your aim is to monitor the restoration of an ecological community back to a pre-invaded state, monitoring should continue until that state is reached, and then for some time afterwards to ensure the vegetation complex is stable. If you are monitoring the health and growth of a particular native species, monitoring could continue until that plant reaches a certain size (e.g. above being affected by the target weed species), until the plant dies (because of weed exclusion, fire, erosion, etc.), or until the threat of weeds has been removed from the vicinity. If a particular weed infestation is the focus of your monitoring, monitoring should continue until the target weed, and other secondary weeds, have been removed and the native vegetation has recolonised the site. Broadly speaking, most of these objectives require medium to long-term monitoring, with a recommended three-year minimum, continuing until your objectives are met and as long as your resources allow.

The decision to discontinue monitoring should be made as part of an adaptive management and monitoring program. Strategic endpoints for monitoring should be defined before monitoring begins; for example, the removal of the target weed species and other weeds from a site or reaching a low target weed abundance. You may find it beneficial to keep photo monitoring markers in place even after monitoring has finished. This will ensure sites can be revisited in the future to check if and how the vegetation has changed.

> While re-randomised plots can change each visit and provide broader conclusions, once a fixed plot has been established it cannot be changed. Therefore, care must be taken in choosing the location and subjects for monitoring. For further information on re-randomised and fixed plots refer to Chapter 2 – Advanced outcomes.

1.5 Mapping

A standardised map forms the basis for all the monitoring techniques outlined in this manual. A map is needed to define the area where you plan to control the target weed species and to record the details of your monitoring program. Monitoring the biodiversity you are aiming to conserve is an essential component of any management project and knowing the location of these species is therefore critical. If updated regularly, a map can also be used to visually assess your progress over time.

The following are mapping programs and platforms that can be used to produce digital or hard-copy maps (see More information for links):

- ArcMap (one of the most commonly used, and most powerful geographic information system (GIS) applications)
- QGIS (free and open source software)
- Google Earth
- US Geological Survey
- NSW Spatial Services
- Atlas of Living Australia BioCollect.

If you are new to digital mapping, there are a wealth of online tutorials available and community support for most mapping platforms, which you may find beneficial. These platforms also provide users with a lot of options in terms of mapping and analysis tools, if you are a more advanced user.

There are a number of mobile phone applications that can be used to digitise data in the field (e.g. Avenza, Collector for ArcGIS, Survey123 for ArcGIS, Google Maps or a range of free apps). The GPS within most smartphones is quite accurate, and almost certainly an improvement on a physical map.

If you don’t know the extent of your native or invasive flora distributions, you must carry out surveys to delimit the population boundaries and densities.

1.5.1 What to include on your map

The final map you produce should contain the following components as a series of layers:

- Layer 1 – base map (aerial photo or topographic map) showing orientation (i.e. north), a scale, legend and title (additional contextual information such as roads and rivers may also be included)
- Layer 2 – a site boundary that identifies and defines the bounds of your site
- Layer 3 – the location of any priority native species or threatened plant populations at risk from the target weed species. Note, many plant community types are now mapped and this data can be sourced from the SEED portal
- Layer 4 – the location of all weed infestations you are interested in managing
- Layer 5 – the area/s where you plan to undertake control, considering a staged approach to weed control and the year/s you plan to implement each stage
- Layer 6 – the location of any monitoring programs being implemented (e.g. photopoints, transects and/or quadrats).

Your map needs to be updated whenever control or monitoring is done at your site. The old versions could be kept to show the effect of control over time. Two example maps are provided below.

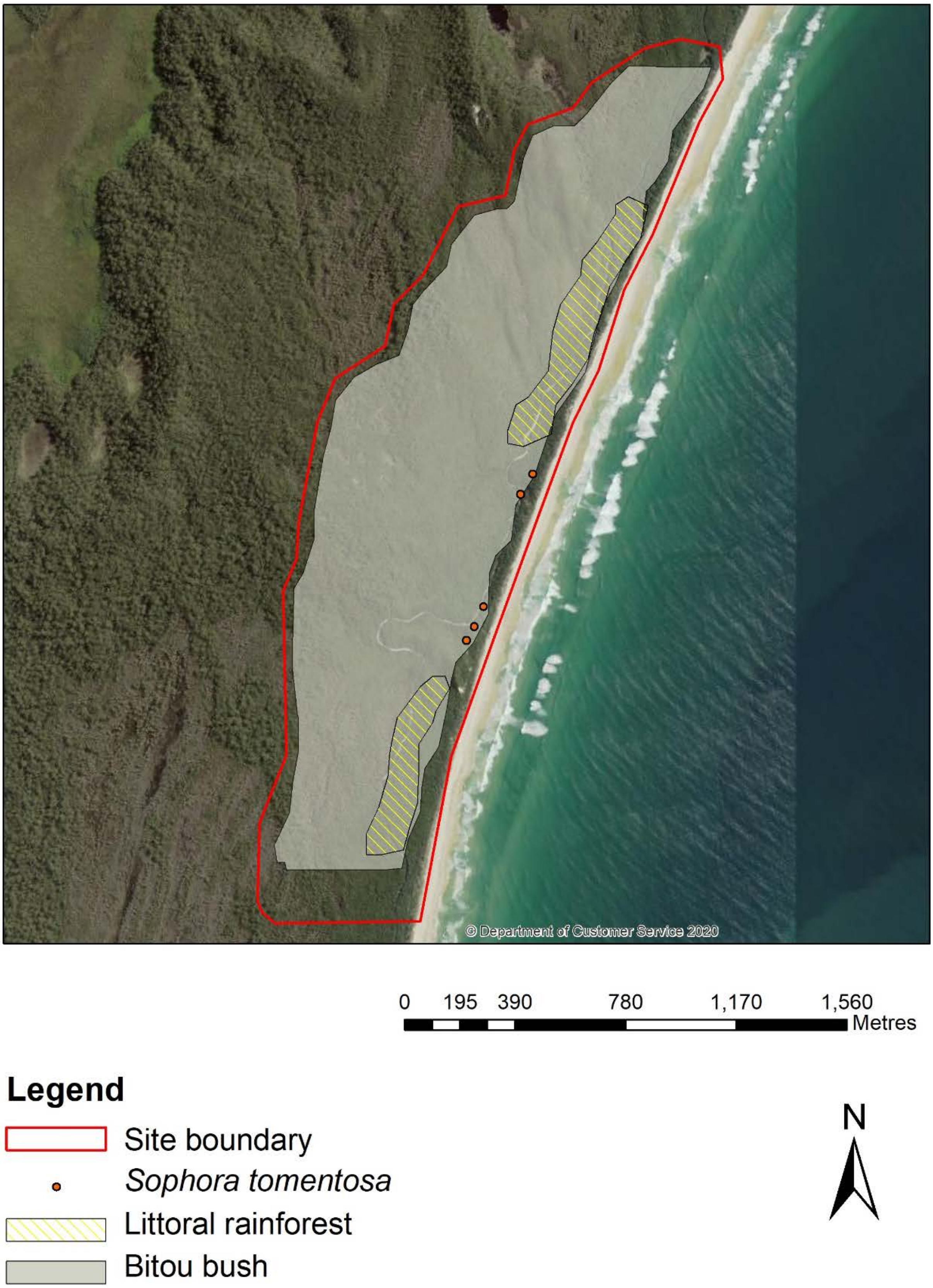

Figure 1 **An example of mapping layers demonstrating a bitou bush infestation (Layers 1–4)**

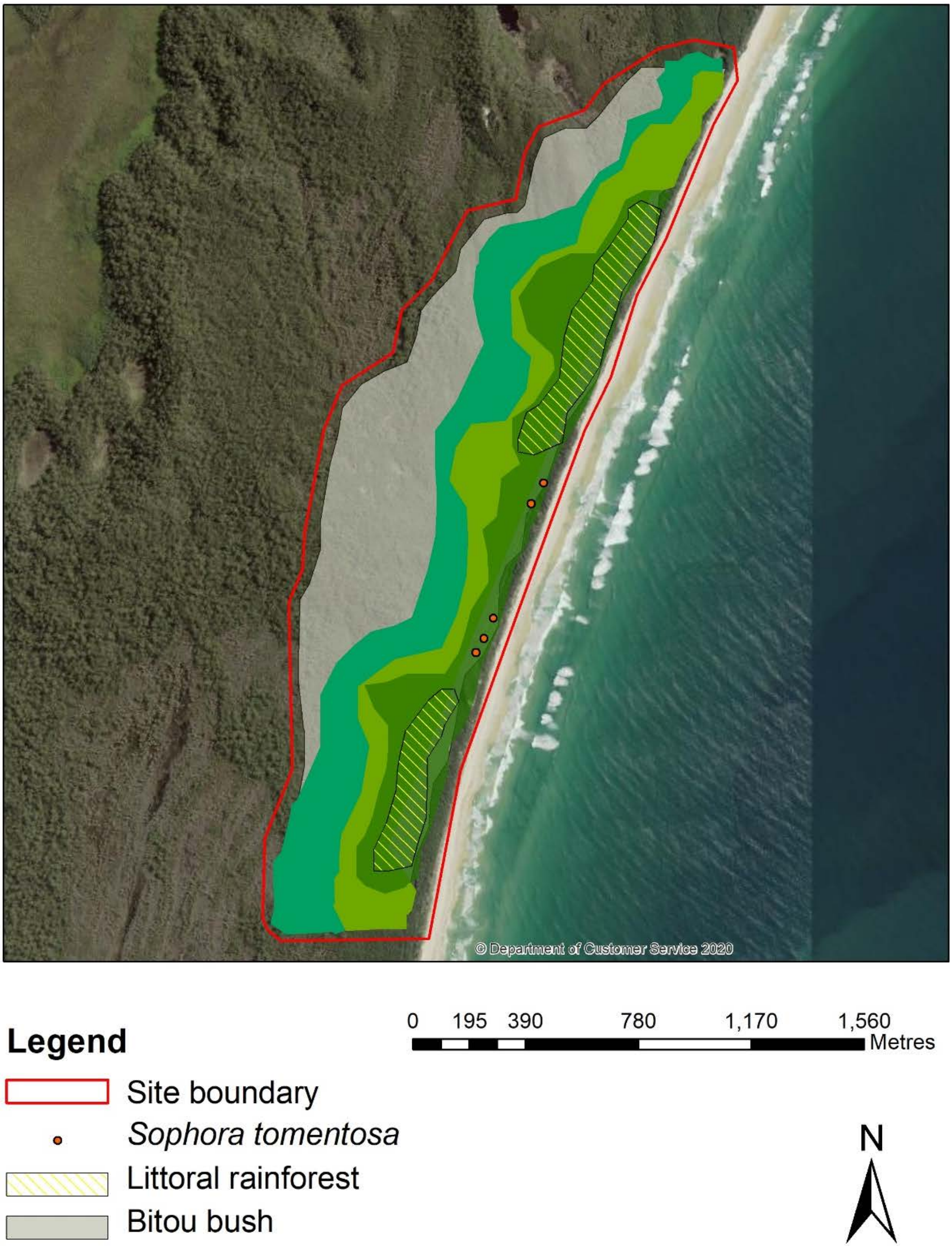

Figure 2 **The control areas and stages for planned control program (Layers 1–5)**

1.5.2 Density classes for mapping

Standard protocols for mapping the density of Weeds of National Significance (WoNS) in Australia have been developed and agreed to by all governments involved (Table 3, McNaught et al. 2008). Where possible you should use these so your distribution map is consistent with and can feed into other broader maps.

Weed cover should be assessed depending on the species habit (McNaught et al. 2008):

- actual surface area covered – useful for grasses and herbs
- projected canopy cover – useful for vines, tall shrubs and trees
- number of stems per hectare – useful for trees.

Table 3 Adapted density classes for weed mapping using standard Weeds of National Significance protocols (PDF 1.2MB) (McNaught et al. 2008)

Class number	Class description
1.	Absent
2.	Less than 1%
3.	1% to 10%
4.	11% to 50%
5.	51% to 70%
6.	Greater than 70%
7.	Present (density unknown)
8.	Not known (or uncertain)
9.	Not assessed
Additional optional classes for aquatic weeds	
10.	Scattered
11.	100% covered

The ‘not known’ class should be used if you think the weed is present but it was not observed (e.g. in a plot), as an incorrect recording of ‘absent’ would be undesirable. The ‘not assessed’ class simple distinguishes absence of data from absence of weeds (McNaught et al. 2008).

A problem may arise when trying to classify recently controlled infestations. For example, a heavy infestation (80% cover) may reduce to less than 10% cover immediately after control and may be 40% by the time of the next control. For consistency and accuracy, always record the density pre-treatment.

Don’t underestimate the value of ‘absence’ data, which provides information on where the target weed species is not present. This is valuable information and should be recorded with as much rigour as all other density classes.

1.6 Observational data

Observational monitoring is achieved through the collation of a standard set of information or data, which includes:

- estimates of vegetation cover and density at different life stages (of both key native and weed species), and
- additional ecological information.

Furthermore, a range of basic ecological data can be collected; for example, reproductive status, response to control, health, and whether the number of individuals is increasing or decreasing over time. The importance of collecting this type of information is evident in that very limited knowledge is generally available about some of the native species at risk from weeds.

The key advantage of this type of data collection is that virtually no special skills are required and it takes very little time to collect. For a more comprehensive assessment it would be helpful to have some botanical knowledge, but if you are only concerned with a subset of priority native and weed species it may only be necessary to be able to recognise these species in the field.

In this section, data collection is limited to a subset of the flora; that is, target weeds, other weeds and priority native species. As full floristic surveys are not part of this section, data collection is more rapid but is limiting as changes in species richness (the number of species) cannot be determined.

Monitoring datasheets have been designed to standardise and facilitate field data collection, and to minimise the variability between observers that typically occurs when collecting qualitative data. There are separate, standard proformas to collect observational data, details of photopoint plots, and control and monitoring details. We have provided blank datasheets in Appendix A for you to use.

Each datasheet should have, as a minimum, the following information recorded:

- site location (a unique identifier)
- date
- observers
- weed control history (current control stage and time since last control event)
- plot size and name (unique)
- coordinates
- a site description, and
- a disturbance history of the site.

It is also important to record the monitoring method used and the area sampled at the site.

Other useful information to include is the elevation above sea level, the inclination of the land and the aspect (measured in the direction of the downward slope). If a photopoint is taken at the site, record the photopoint ID.

> No matter how you monitor, you must be careful not to crush small plants and seedlings in the area where you intend to record information, e.g. the monitoring plot.

1.6.1 Establishing monitoring plots

Plots can be established as fixed, where the same area is sampled each time, or re-randomised, where plots are randomly located each time you sample. For preliminary observations where the amount of time, money and resources available are low, establishing permanent plots may help reduce the amount of time spent in the field. You may also need to use a permanent plot if your target species only occurs in one place.

Relocating plots randomly each time you visit is more time consuming per visit but allows you to sample a larger area and increases the utility of statistical analysis. The method you choose for establishing plots is dependent on the time and resources available and how to best meet your monitoring objective.

Once you have chosen the location for your observational plot you should give it a unique number/name and complete the remaining information in the appropriate datasheet (see Appendix A for datasheets).

The ideal **size, shape** and **number** of sampling units to use will depend on the size and density of individuals within the community being sampled and your purpose in monitoring. Sampling units must be large enough to contain a number of individuals, but small enough that the individuals present can be counted and measured. The ideal dimensions and number of sampling units will therefore vary between vegetation growth forms. If you are just monitoring a small population of an endangered plant, perhaps there is only room for one plot, but if you are looking for broader improvements across a number of species or over a larger area, three or more plots are more appropriate. See the **advanced outcomes** chapter for more information on sampling methods (including plot sizes for different scenarios); in particular, Section 2.5.3 about re-randomised and fixed sampling.

1.6.2 Site description

What constitutes a site is the area you are proposing to manage, however it needs to be a manageable size. Consider access, the extent and distribution of weed species, and the native species you're protecting. This is the management unit you are working to restore. It could be the area behind a beach, a small reserve or a vegetation type within a national park or catchment.

By including a detailed description of the structural vegetation and prominent landscape features of each plot you are monitoring, you will be able to capture subjective information about your site.

Vegetation can generally be classified into three stratums or layers within the area being surveyed. The tallest stratum refers to the tallest significant growth form present, usually the canopy trees. The mid-stratum contains all layers between the tallest stratum and 1 metre in height. The lower stratum (or ground layer) includes all grasses and vegetation up to 1 metre tall. As a very simple site description you should record the **dominant plant species** within each stratum and their **growth form**; for example, tree, shrub, grass.

Prominent landscape features might include any distinguishing features of the vegetation; for example, mallee formation of trees or swampland, or landscape features such as rock outcrops or mountain slopes. This section can also be used to make additional notes about the site or survey plot.

If there is evidence of previous disturbance, record the disturbance type (see Table 4), the time since disturbance (if known), the accuracy of this time since disturbance (e.g. exact date known, ±5 years, etc.), the severity of this disturbance (how much damage was caused by the disturbance), and how much of the site was affected.

Table 4 Examples of types of disturbance you might record as part of your site description and ways to describe their severity

Disturbance	Severity
Fire	Low, medium, high
Flood	Slight, moderate, high
Grazing	Slight, moderate, high
Clearing	Slightly modified, moderately modified, highly modified
Logging	Selective, integrated, clearfell
Drought	Slight, moderate, high
Soil erosion	Slight, moderate, high
Slope instability	Slight, moderate, high
Trampling (people)	Slight, moderate, high
Trampling (vehicular)	Slight, moderate, high
Other, e.g. wind, storm	Slight, moderate, high

1.6.3 Estimates of cover and density (indices of abundance)

Vegetation cover is a visual estimate of the proportion of the plot that is occupied by each species, while **vegetation density** is the actual or estimated number of individuals within the plot per square metre. Both measures are informative; however, to better assess the response to your control program you also need to consider the age dynamics of the target species over time (e.g. the number of seedlings, juvenile, adult and dead plants). For example, if the density of the target weed species (i.e. total number of individuals) increases after control, you might conclude your control has not been effective. This single measure is misleading, however, knowing that the increase is due to new seedlings provides a reflection of your control measures, whereas an increase in numbers does not.

For plots, a tape measure can easily be rolled out along your site, using a compass to ensure your corners are at 90° (as shapes other than squares and rectangles will have variable areas, making sites of different sizes difficult to compare). For fixed quadrats, measurements can be taken from a permanently located marker such as a sighter post for photopoints. If using photopoints as well, orientate the plot away from the camera post and in the direction of the photo.

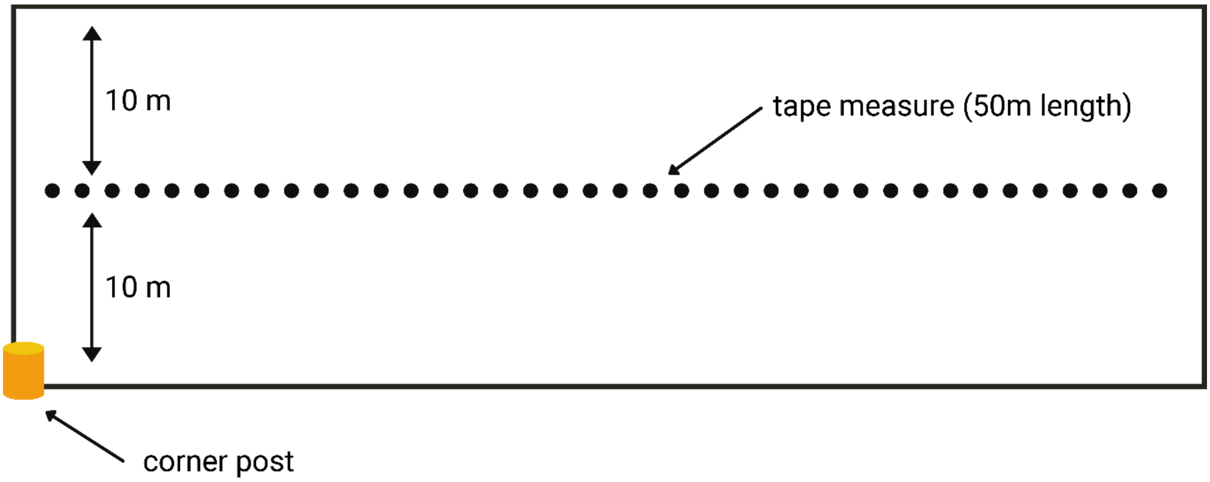

Figure 3 **An example of a rectangle plot that may be used for estimates of cover and density**

For a 50 metre x 20 metre plot, the tape measure can be extended along the middle of the plot and observations recorded within 4 metres either side of the tape (Figure 3). This will give you a plot covering a total area of 1000 square metres. The area you survey will depend on the growth form of your target species. For trees, you will need to cover a much larger area than for herbs.

There are seven cover classes used in this manual for estimating overall cover, which are aligned with the classes used in the Weeds of National Significance (WoNS) protocols (McNaught et al. 2008) but with slightly increased detail at high levels of cover (Table 5). These cover classes should be used as guidelines when it comes to determining the percentage cover of vegetation you see in the field.

Table 5 Vegetation cover classes, based on the Braun–Blanquet cover–abundance scale

Refer to Mueller-Dombois and Ellenburg (1974) for more information.

WoNS density class	Cover class	% cover range	Grasses, forbs, etc.	Shrubs	Trees
5	**7 – Very dense**	>75%	Individual plants are touching or overlapping – forming a monoculture with few other species present. Individuals may be very densely clumped.	The crowns or edges of individual plants of the target species are either touching or overlapping one another – forming a monoculture with few other species present.	As for shrubs.
5	**6 – Dense**	51–75%	Individual plants are slightly separated or touching – not forming a monoculture, with other plants present or bare ground. Individuals may be densely clumped.	The crowns or edges of individual plants of the target species are either slightly separated or touching – not forming a monoculture, with other plants present or bare ground.	As for shrubs.
4	**5 – Sparse**	26–50%	Individual plants are separated or rarely touching. Individuals may be moderately clumped. Other plants present or bare ground.	The crowns or edges of individual plants of the target species are clearly separated. Other plants present or bare ground.	As for shrubs.
3	**3 – Very sparse**	6–25%	Individual plants are well separated. Other plant species dominate and typically occur between the target species. Small clumps may occur.	The crowns or edges of individual plants of the target species are well-separated. Other plant species dominate and typically occur between the target species.	As for shrubs except the degree of crown separation is higher.
2	**2 – Isolated plants**	1–5%	Individuals are scarce or scattered. Small clumps may occur.	Individual plants of the target species are scarce or scattered.	As for shrubs except the individual plants of the target species are further apart.
1	**1 – Absent**	0%	The target species is not present.	As for grasses, forbs, etc.	As for grasses, forbs, etc.

1.6.4 Age dynamics of the target species

You can choose to record the age dynamics of your target species using cover or density estimates; however, these two measures give quite different results. If using cover, you record the proportion of your plot that is covered by each life stage (Table 6). If using density, you record the number of individuals of each life stage within your plot.

Table 6 Record life stages of shrubs and trees according to the following categories

Category	Description
Seedling	Newly germinated/emerged seed which still has cotyledons and is yet to establish
Juvenile	An established plant that is not yet reproductive
Adult	An established and reproductive plant
Dead plants	A dead plant that has not yet decayed

If you have overlapping layers, the sum of cover values for all four categories combined may be more than the overall percentage cover. This will occur, for example, if you have seedlings growing underneath an adult plant. In contrast, the density estimates are the estimated number of individuals in your plot in each of the four life stage categories. Plants that are re-sprouting after fire or some other damage should be included in the adult life stage. If you would prefer a more accurate measure of cover or density, you will need to use the techniques in the chapter on **advanced outcomes**.

1.6.5 Additional ecological information

To further describe the condition of your plot, you can note the reproductive status, describe the dominant ecological community present, and any other weed species present that may invade your site post-control. If you have recently conducted control works on your site, you should also note the response to control of the target weed species.

Ecological communities

Record structural features of the ecological communities present, including dominant species and growth form (refer to Keith 2004 for descriptions of native communities in New South Wales and the ACT or the National Vegetation Information System for Major Vegetation Group mapping layers).

An example for a Themeda grassland community would be ‘Closed tussock grassland dominated by *Themeda triandra*, co-occurring species include *Hibbertia scandens* and *Viola banksii*. *Banksia integrifolia* and *Monotoca elliptica* occur as occasional emergents in the community.’

Response to control

For each weed species targeted for control, record the technique used for control and the response of each species to the control (Table 7). This will allow you to determine the effectiveness of your control methods and adapt your management technique if required.

Similarly, information on the response of native species to accidental (off-target) herbicide application is limited, and the method of removal of invasive species may influence native community response (Flory & Clay 2009). Recording the status of native species after control will supplement what is already known, which will be useful for the wider community, as well as for interpreting your own results.

There are seven categories established to record the response of plant species (native or exotic) to control (Table 7; following Broese van Groenou & Downey 2006, and John Toth unpublished data).

Table 7 Categories to measure the response of plant species (native or exotic) to herbicide spray as an effect of weed control

Damage	Definition
ND (no damage)	There has been no damage to the target plant population as a consequence of control
L (low damage)	≤25% of the target plant population has been damaged, with no dead plants
M (moderate damage)	>25% of the target plant population has been damaged, with no dead plants
LD (low dead)	≤25% of the target plant population is dead
MD (moderate dead)	25% of the target plant population is dead
AD (all dead)	All individuals of the target plant population are dead
U (unsure)	You are unsure of the damage to your target plant population; detail the reasons why you are unsure

If you suspect there has been off-target damage or there has been a report of off-target damage, you should measure plant responses at the time intervals outlined in the bitou bush aerial spraying guidelines (Broese van Groenou & Downey 2006), being eight weeks and six months after herbicide application. In addition to these responses, information must also be collated on the herbicide application, specifically:

- the herbicide and application rate
- the interval between application and the observation (e.g. two months)
- a follow-up sample to determine long-term response.

The NSW Department of Primary Industries and in particular the NSW Weed Control Handbook may be a useful source for this information. If you are unsure of what type of herbicide or control method to use and how it should be applied, check with the Department of Primary Industries and/or use the WeedWise app.

Unmanaged weed species

There may be weed species at your site that are a low priority or are not currently being managed. These species may invade post-control, so it is useful to monitor their abundance. For consistency, use the same cover classes as your target weed species (Table 6) and note if this abundance is the same, an increase, or a decrease since the last monitoring event.

1.7 Photopoints

Photopoints are an effective visual aid for communicating changes at your site. Photos can also be useful to relocate observational plots if plot markers go missing. Observational data collected at each photopoint will further support changes observed in the photos and your monitoring and reporting.

Note: Some organisations require photo monitoring for funding purposes, so ensure you know whether this form of monitoring is essential or optional for your requirements.

1.7.1 Where to put photopoints

Comparison of photopoints over time can be used as a simple form of monitoring but is very limited in its information. The success of photopoints for monitoring is based on the establishment of permanent photo locations, which can be revisited at regular intervals. Photopoints should be positioned in areas that will provide information to determine changes in weed abundance and community composition; for example, locating photopoints in areas where primary weed control is to occur. Planning and thought are therefore needed prior to the establishment of a photopoint to determine the exact nature of the photos and what they will reflect over time. For example, a picture of a weed infestation (being green vegetation) at the beginning of the program and one of (also green) native vegetation at the end may not adequately reflect the changes that occurred. In addition, planning is needed to track photos and match them over time. A descriptive set of notes for each photo as well as location labels are needed to achieve this.

Photopoint plots should try to utilise permanent fixtures that are already within the landscape, such as a sign, fence post, large rocks, easily distinguishable tree, or feature on the horizon. You should also record the compass bearing to your plot from prominent landmarks. Keep permanent fixtures such as star pickets or timber stakes out of view if vandalism is likely to be a problem.

1.7.2 What information to record

Record the exact location with GPS coordinates, compass bearings and post markers (so the camera is at the same height). Record the time, date, management area, site number, photopoint ID and brief description of what is seen in the photograph such as whether the photo is pre-control, or after primary control.

Take photographs at a similar time of day (i.e. either morning or afternoon, light cloudy days are ideal when sunlight is weaker and shadows from vegetation are less pronounced). Use the same camera and lens with the same zoom settings each time and ensure the aspect is consistent throughout time. Landscape orientation is preferable to portrait in most situations unless you’re demonstrating changes in either mid, upper or emergent stratums.

Other tips:

- take along copies of the original photos for reference when returning to the photopoints, to help match the field of view contents
- consider proximity to tracks or roads for future accessibility
- consider potential vegetation regrowth that may obscure the clarity of future photos taken from the same location
- consider integrating photopoints with your other monitoring methods
- if taking multiple images as part of a panorama, record bearings from north for each image and take sequential photographs (from either left to right or right to left) allowing for each image to overlap so that no gaps exist in the panorama (OEH 2018).

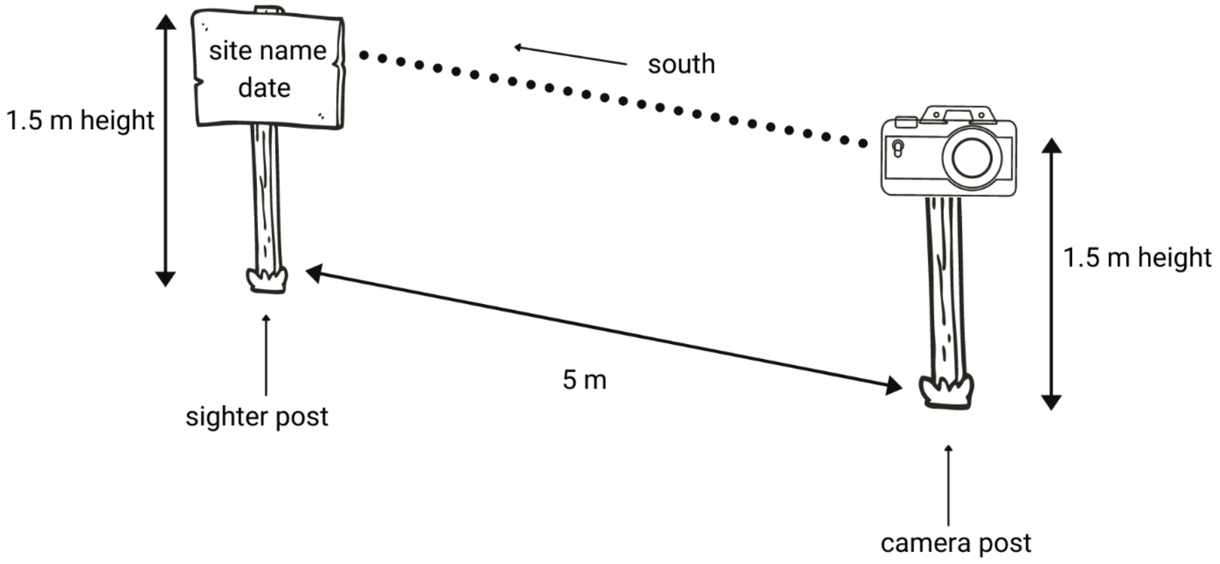

Figure 4 **An example of how to set up a permanent photopoint marker, including a camera post to place the camera at the same height each visit and a sighter post in the central focal point of the camera's field of view**

The sighter post is used to display the site name and date so this information is captured within the photo. A distance of at least five metres and choosing a cardinal direction is recommended, although this isn't necessary so long as the distance and direction you chose are noted.

1.7.3 How often to take photos

Photos should be taken at predetermined intervals (e.g. six-monthly) rather than indiscriminately over the course of your program, so that you have a documented timeframe in which the changes occurred. Also, you need to consider the period of time needed to show the response of the subject of the photo or your objective. For example, to show the kill-rate of initial weed control you may only need a before and after photo taken six months apart. However, if you want to show the response of a long-lived native species to control you may need photos spanning five years or longer. Further, annual species' responses will need to be photographed at an appropriate time to measure occurrence.

We recommend photos be taken prior to the initial target weed species control at the photopoint location, and at six-monthly intervals over the course of the monitoring regime at your site. The six-monthly intervals will give you two samples between your annual control programs and give a much greater comparison between pre- and post-control over time, and the response of native species and target weed species.

1.7.4 Filing

Photopoints are most useful when comparing the change in a site over a long period of time, therefore it is essential you file and store your photos in a way that will make sense years after they are taken.

Make sure you file your digital photographs in the same way as you record their ID in the field so they can be easily cross-referenced. We recommend labelling photos with management area, site number, date, time, photopoint ID and the subject of the photo. The subject of the photo should include the species being targeted or monitored and time since control (or pre-control if it is the first set of photos). Back up photographs to at least one other location and, if stored on an external hard drive, record the details of the contact person. A photopoint datasheet has also been supplied in Appendix A.

1.8 Keeping records of your control and monitoring activities

It is important to document what you do during your control program, along with the associated costs of implementing control and monitoring activities. Too often, such information is not documented; however, this is just as important as any other data as it indicates the effort required to get the response. If requesting or applying for funding, having this information prepared can assist your project and demonstrate the efficiency and effectiveness of your program as well as the outcomes. Information on control and monitoring activities needs to be collated on an annual basis, therefore time and dollars spent should be expressed as values per annum.

At the minimum, you must record the persons undertaking control activities, control methods, the cost of your control activities, information on your monitoring program (site, technique and methodology) and the cost of your monitoring activities over the course of a year.

1.8.1 Control activities

When applying herbicides to control target weeds, you should be aware of and adhere to the requirements of the NSW Pesticides Regulation 2017, including mandatory training, contractor licensing and record keeping requirements. Record keeping requirements are detailed in clause 36 of the Pesticides Regulation and include details of the species controlled, the control technique (e.g. foliar spray, cut and paint, hand removal, etc.), the specifications of any herbicides used (e.g. Roundup, Glyphosate, 2L/ha) and details of the person/s who undertook the control activities (i.e. the contractor or staff member names, organisation and contact information).

There is a regulatory exemption to record keeping, where small amounts of pesticides (normally used in the home and garden) are applied (see clause 4 of the Pesticides Regulation). For National Parks and Wildlife Service (NPWS) staff, records must be kept for every pesticide application on park estate regardless of exemptions. The records can be made electronically or in hard copy as soon as possible, but within 48 hours, and must be kept for three years. Records must be available to provide to the NSW Environment Protection Authority on request. NPWS staff should familiarise themselves with the latest *NPWS Pesticides Standard Operation Procedures* and make sure their pesticide training is current before undertaking herbicide application.

A scientific licence is also required under Part 2 of the Biodiversity Conservation Act for any activity which may harm a threatened species, an endangered population or a threatened ecological community, or damage their habitat. For further information about licencing requirements and the application process, head to the Scientific licences page on the NPWS website.

1.8.2 Documenting the cost of control and monitoring

Documenting the monetary and in-kind costs incurred in the control of weed species and the monitoring of weed and native species is useful to relate this information to your weed and biodiversity outcomes. For example, your biological monitoring may reveal a positive weed and biodiversity outcome, but it is informative to know what *effort* was required to achieve this outcome. A lack of long-term, specific expenditure data from individual weed control programs reduces the ability to deliver adequate policy initiatives, to accurately cost and prepare budgets for subsequent control/monitoring programs, and influence management. The specifics include the costs of materials, contractors and in-kind contributions as well as administrative costs. This information is useful to collect each year, including the costs of follow-up control.

The number of hours and hourly rate of labour costs for control work can be recorded, as well as the costs of any materials or activities performed by contractors (e.g. herbicides used, contractors to perform aerial spraying). For example, if three people worked 20 hours each, this totals 60 hours. Work performed both onsite and off-site can be recorded. This value can include the number of hours spent planning your control activities. The ‘cost’ of in-kind hours contributed to a control program is a valuable source of information on the effort required to control weeds at your site. For example, if a community group with eight members met once a month for four hours, the annual number of in-kind hours accrued would be 384.

You may also document the costs incurred in monitoring the response to control works of the target weed species and native vegetation. This includes the costs of paid labour, materials (e.g. tape measures, posts, etc.), contractors and in-kind contributions (e.g. from a volunteer group), as well as administrative costs (e.g. time spent planning the monitoring program and data entry). Monitoring costs can be significant, especially if more advanced methods are employed, due to the high cost of labour and time involved. Knowing how much time is required to effectively monitor a site is a useful measure for planning and budgeting for the effort required for future programs.

2. Advanced outcomes

This chapter uses a number of quantitative sampling techniques and concepts to collect data that are more objective and accurate than the data collection methods outlined in the preliminary observations chapter. These additional techniques and concepts increase the rigour and repeatability of data collection and reduce the likelihood of any variation in the data being due to different observers collecting data. However, these techniques require more resources, botanical knowledge and time.

This chapter provides detailed descriptions on how to monitor the response of (i) the target weed species, (ii) native species and ecological communities, and (iii) other weeds that are not part of the control program. There are three steps to designing a monitoring program with demonstrable outcomes:

1. ***Select a sampling method*** based on the category of vegetation you wish to monitor and its growth form.
2. ***Establish unbiased sampling units*** (e.g. plots) based on how you want your sampling units to be arranged, with starting points to be fully random or pseudo-random.
3. ***Sample your vegetation using***
    a. line-intercept transects
    b. quadrats
    c. T-square estimates.

These steps and techniques do not require extensive resources or further reading (background reading is provided for those who wish to know more); instead, you only need to follow the instructions appropriate to the vegetative growth life form you are monitoring. There is scope, however, for you to select between several different methods, and you will need to think carefully about the objectives of the monitoring program, what you want to monitor and the monitoring metric (e.g. cover, number of plants, etc.), and how rigorous your results need to be before you start, as well as any constraints such as time, funding or resources.

2.1 Requisite methods

To undertake these monitoring techniques, you first need to complete the **mapping** section (1.5) of the **preliminary observations** monitoring techniques. The maps and GIS project you create will include the location of your target weed species and the priority native species and ecological communities within your site, your proposed weed control program, and the location(s) of the monitoring program you develop using the techniques outlined in this chapter.

There is no need for you to collect the observational data as recommended in the first chapter, as the advanced methods in this second chapter essentially incorporate observational methods in a more rigorous manner. Photopoints could still have value as a visual record of the changes in your site, which can be a useful quick measure to show funding bodies the broad results of your actions, but these can't be used to quantify your success (refer to Section 1.7 for further details on photopoint monitoring).

For any control and monitoring activity you should complete the **control and monitoring costing** section in the **preliminary observations** chapter. This collates information on the time and funds spent controlling weeds and monitoring vegetation responses. Records of control and monitoring activities are important to evaluate their costs.

2.2 Sampling methods

The sampling methods described in this chapter are based around three methods of data collection (hereafter referred to as sampling methods): line-intercept transects, quadrat sampling and T-square sampling (Table 8). These methods were deemed most suitable for the scope of this particular manual, however other methods are still valid and may also be used (for other examples, see Surveying threatened plants and their habitats (DPIE 2020b) or Monitoring threatened species and ecological communities (Legge et al. 2018)).

Table 8 A description of sampling methods

Technique	Uses	Advantages	Disadvantages
Line-intercept transects	One-dimensional plots (i.e. a single line, often a tape measure) that are used to measure canopy cover, density and presence/absence	Estimates of cover are generally less subjective as actual cover is measured. Best for measuring low vegetation such as shrubs and matted plants	Less effective for measuring cover of grasses and some forbs, as well as trees and taller shrubs. Measures of density using this method can be ambiguous
Quadrats	Two-dimensional plots that can be square, rectangular or round, that can be used to measure cover, density, species richness and presence/absence	This is an effective method of sampling most vegetation types, if an appropriate quadrat size and number of replicates are chosen	Sampling biases can occur dependent on the census technique used. This isn't an appropriate way to measure scattered trees
T-square sampling	This is a plotless form of sampling, with no boundaries on the sampling unit. It is a nearest-neighbour sampling method for estimating density	Useful for monitoring large or scattered trees when quadrat sampling is impractical	Density is the only index of abundance that can be measured with this method

2.3 Measuring vegetation

Plants vary greatly in size and vegetation communities differ greatly in their structure. This presents challenges when deciding at what scale to sample and what measures to use. Furthermore, a range of characteristics of these plants and ecological communities can be sampled within the one plot, each of which provides a slightly different measure of response to weed control. Vegetation measures in this manual are grouped into **indices of abundance**, **demographic measures**, and measures of the **response to control**.

2.3.1 Indices of abundance

The following are the primary techniques used to assess vegetation responses to weed control in this chapter. While it is possible to measure the actual abundance (number of individuals) of a particular species within a monitoring area, the size of a monitoring area or the number of individuals present often makes this impractical. Other measures that provide an index (i.e. an indication) of abundance are therefore commonly used. A wide range of sampling techniques are available to estimate an index of abundance, three of which are referred to here (Table 9). Of these, **cover** and **density** are the preferred indices of abundance as they provide more detailed data than **presence/absence**.

The **richness** and **diversity** of native species in relation to the target weed species within your management area can provide an indicator of community resilience, as low species richness and diversity can reduce the resilience of a community to invasion and is a poor biodiversity outcome. Species richness is one measure to use for monitoring the response of ecological communities.

Table 9 Ways to estimate an index of plant abundance and composition discussed in this chapter

Data type	Definition
Cover	the proportion of ground within a sampling unit (transect or quadrat) that is covered by the vertical projection of all individuals of a species
Density	the number of individuals of a species in a sampling unit (a transect, quadrat or the monitoring area for T-square sampling), quoted per area
Presence/absence	a record of whether a species is found in a sampling unit or not
Species richness	a count of the number of distinct species within a sampling unit
Species diversity	an index of the 'evenness' of species or their relative abundance

Cover

Cover (also called canopy cover) is the vertical projection of vegetation from the ground as viewed from above. Cover is a commonly used measure as it enables all species to be compared irrespective of their size or abundance (e.g. from small but abundant to large but rare). Unlike **density**, cover is closely aligned with biomass or annual production. Cover does not require the ability to distinguish between individuals of the same species. It can be, however, sensitive to changes over the growing season. Although the lack of native deciduous species makes this less of an issue in Australia, sampling should be repeated at the same stage of each growing season.

There are a number of ways to estimate cover. When using **quadrats**, an observer estimates the proportion occupied by a species. In addition to estimating the actual percentage of the quadrat covered by a species, the observer can categorise cover estimates, for example into cover categories. For invasive species, we recommend providing an actual estimate (to the precision you're comfortable estimating, e.g. to nearest 1%, 5%, etc.) rather than using the modified Braun-Blanquet method (adapted from Poore 1955) as actual values can be converted to these broader indices for mapping or can be analysed separately.

While estimating cover in quadrats can be faster in comparison to counting each individual of a species, the estimate is subjective and different observers or one observer at different times may make different estimates. This variability can be somewhat reduced with training, practice, working in pairs or more, and being aware of what area (in square metres) equates to a specific percentage cover, e.g. 10% equates to one square metre in a 2 x 5 metre plot. Having a defined set of values to use consistently for sampling (e.g. boundary rules, covered in the quadrats section of this chapter) may also help to reduce variation as observers become accustomed to estimating within the value boundaries.

Bias may also result from species conspicuousness, with more conspicuous species more likely to be observed than less conspicuous ones. Those forming clumps of individuals with broader leaves are also more likely to be given a higher estimate than those that are dispersed with fine leaves. Again, careful sampling and observation should overcome this.

For all methods it should also be noted that overlapping cover of individuals of the same species should not be counted twice but should be recorded as continuous cover. If however, two or more different plant species overlap then the cover of each species should be recorded separately; this may give more than 100% cover if there are many overlapping canopies.

In addition to measuring the cover of plant species, you can also measure the cover of bare ground or leaf litter, and that of different life history categories. These variables are important in some ecological communities.

Density

Density is the number of individuals per unit area. For plants, it is generally calculated as the number of individual plants per square metre or per hectare. Because it is a per-area measure, density estimates can be compared across sampling units, even if the size and shape of sampling units vary.

Density can only be calculated if individual plants are readily and consistently identifiable. Density measurements are not suitable if plants form dense mats or clumps due to the difficulty of distinguishing between plants. Density will also be impractical for very sparsely distributed or rare species. Conversely, counting many individuals for density can be very time consuming and difficult unless you use very small quadrats. Note also that, like cover estimates, density estimates can be biased towards more conspicuous species.

Density estimates will detect changes in abundance via recruitment or mortality. They are unable, however, to detect changes in condition; for example, new growth or a decline in foliage or health. In addition to estimating the overall density of plants within an area, the density of different life history categories (e.g. seedlings, juveniles, and adults; reproductive or non-reproductive) can also be calculated. These estimates provide more sensitivity to some changes in the population.

Presence/absence

Presence/absence is a simple record of whether a species is found within a sampling unit or not (this will also result in a floristic survey, except where you have a targeted list of species you wish to sample). The key advantage of this method is that no special skills are required, other than the ability to identify the species. This technique is also very fast, although care must be taken to search thoroughly for all species (or a select list of species you are interested in, e.g. the priority native species). Presence/absence is insensitive to changes other than the appearance or complete disappearance of a species from a sampling unit. Therefore, while presence/absence provides information on **species richness** within a sampling unit, it provides no further information on the **abundance**, **cover** or **density** of species.

Species richness

Species richness is an index of the number of species within a sampling unit and is the most basic and important measure. It provides a simple indication of how weed species are affecting native species richness, and whether there is an increase in overall native species richness following weed control. Usually, native and weed species richness increases following weed management as disturbance occurs and extra light encourages germination. Long-term increases in native species richness are however the aim. Although conceptually simple, species richness relies on a full floristic survey of the sampling unit, and care must be taken to correctly identify all the species within the sampling unit and to apply equal effort in each plot to do so.

It may be difficult to calculate species richness if: 1) there are a large number of species, 2) many species are rare, 3) species are cryptic or absent above ground at certain times of the year (e.g. orchids), or 4) the vegetation is dense, making it difficult to assess all areas of the sampling unit.

Species richness can mask important differences in the abundance of different species, however. For example, a quadrat containing one dominant species and 19 rare species is very different to a quadrat containing 20 species with similar abundance, yet it has the same species richness (20 species).

Species diversity

Species diversity is a measure of the 'evenness' of species. Species diversity incorporates a measure of **species richness**; however, it also provides additional information on community structure by providing an index of the abundance of the range of species present within a community. A diversity index will indicate, for example, whether a community is dominated by only one species with all other species being relatively rare, or whether the abundance of all species is relatively even (see the example below, Figure 5).

There are many different diversity indices, each with their proponents and detractors (see Ludwig & Reynolds 1988, Magurran 1988, Krebs 2014). A commonly used species diversity index is the Shannon index (*H′*):

$$H' = -\sum_{i=1}^{S} p_i \ln p_i$$

where S = the number of species (also called species richness)

p_i = the relative abundance of each species, calculated as the proportion of individuals of a given species to the total number of individuals (of all species) in the community **(n_i/N)**

n = the number of individuals of species i

N = the total number of all individuals

For example, Figure 5 contains a list of the species found at two sites and the abundance of each species. At Site 1, species 1 dominates with all other species being relatively rare, while at Site 2 there is a more even abundance of all species. There are an equal number of species (10 species) and overall abundance of plants (124 individuals) at both sites.

These calculations give an *H′* index of 0.82 for Site 1, and 2.29 for Site 2. Site 2 can therefore be said to have higher species diversity than Site 1, although both sites have an equal number of species (species richness, 10 species at each site) and an overall equal number of individuals (124 individuals; Figure 5).

	A	B	C	D	E	F	G	H	I
1	Site 1					Site 2			
2	**Species**	**Abundance**	**pi**	**pi x ln(pi)**		**Species**	**Abundance**	**pi**	**pi x ln(pi)**
3	Species 1	103	0.8306	-0.1541		Species 1	12	0.096774	-0.2260
4	Species 2	2	0.0161	-0.0666		Species 2	14	0.112903	-0.2463
5	Species 3	3	0.0242	-0.0900		Species 3	12	0.096774	-0.2260
6	Species 4	2	0.0161	-0.0666		Species 4	13	0.104839	-0.2364
7	Species 5	1	0.0081	-0.0389		Species 5	16	0.129032	-0.2642
8	Species 6	2	0.0161	-0.0666		Species 6	14	0.112903	-0.2463
9	Species 7	2	0.0161	-0.0666		Species 7	13	0.104839	-0.2364
10	Species 8	3	0.0242	-0.0900		Species 8	11	0.08871	-0.2149
11	Species 9	4	0.0323	-0.1108		Species 9	9	0.072581	-0.1904
12	Species 10	2	0.0161	-0.0666		Species 10	10	0.080645	-0.2030
13	**Sum**	124	**Diversity**	0.8167		**Sum**	124	**Diversity**	2.2900

Figure 5 **An example of how two sites with the same number of species (richness) and the same number of individuals (abundance) can have different species evenness**

2.3.2 Demographic measures

Demographic measures should be recorded if resources and time allow as they provide additional information on how the vegetation is responding to weed control. If you have time to survey only a limited number of demographic measures, we recommend you focus on measures of life history (Table 10).

Table 10 Demographic measures to monitor vegetation response to weed control, with priority measures noted with an asterisk (*)

Demographic measure	Definition
Life history*	Incorporates measures of seedlings, juveniles, adults and dead plants
Diameter at breast height (DBH)	The diameter of a tree trunk at breast height, approximately 1.3 metres above ground level. The diameter of the main stems on shrubs should be measured at the base of the shrub
Height	The maximum height (or length) of the plant

2.3.3 Life history

Measures of the life history of a species (e.g. the percentage cover and/or the number of seedlings, juveniles, adults and dead plants) should be used to supplement general measures of cover and density. Measuring the life history of a species is much more time consuming than conventional estimates of cover or density for the species as a whole. However, recording the life history of species provides detailed information on the effectiveness of your control methods on the target weed species (e.g. adult plants are killed but there is seedling recruitment) and the recovery of native species (e.g. if the increased cover is because adult plants are growing or if there is native seedling recruitment). At its simplest, you could separate a species into seedling and non-seedling classes.

You can choose to record the life history of your weed or specific target native species using **cover** or **density** estimates; however, these two measures give quite different results. If using cover, you will record the proportion of your plot that is covered by each life history category of each species.

Note: If you have overlapping layers the sum of cover values for all four life histories combined may be more than the overall percentage cover. This will occur, for example, if you have seedlings growing underneath an adult plant. In contrast, density estimates are the actual or estimated number of individuals in your plot in each of the four life history categories. Details for recording life history are provided in the relevant section for each of the methods.

2.3.4 Diameter at breast height

DBH is a standard method of measuring the diameter of trees or shrubs. It is measured at 1.3 metres above ground level, and on the uphill side if the plant is on a slope. The diameter of the main stems on shrubs should be measured at the base of the shrub. For multi-stemmed trees with a height greater than 1.5 metres, if the stem forks below 1.3 metres then measure each stem individually, but if the stem forks above 1.3 metres measure the stem below the fork and treat it as if it were single-stemmed.

DBH can be measured using callipers, or a diameter tape. A low-cost option is to use a regular tape measure to record the circumference of a stem, then calculate the diameter by dividing the circumference by pi (π: 3.1415…).

2.3.5 Height

Measuring the height of a plant gives you an additional measure of vigour. You can either measure the height of the tallest individual of a species in your plot, or you can calculate the average height of that species in your plot; if you are measuring the average height you should measure the height of at least five randomly chosen individuals. Whether you measure the maximum or average height will give you different information about a species. To decide which one to measure (although you can measure both), you should think about the likely changes a species will make after control, and how this measure fits with the aim of your monitoring program.

The heights of small shrubs and groundcover species (e.g. grasses) are easily measured or estimated, however trees are more difficult. Two methods to estimate tree height are outlined below.

Using a clinometer

It may be worthwhile to invest in a clinometer or laser rangefinder for your monitoring program. In this case, you should follow the product manual to accurately measure tree height. In general, the following methods should be sufficient for most clinometers.

Stand on the same contour as the bottom of the tree (while there is no predetermined distance you must stand away from the tree, you must be able to see both the top and bottom of the tree easily) and measure the angle from the horizontal (eye-height on the tree) to the top of the tree using a clinometer (e.g. θ = 38°; Figure 6). Measure the horizontal distance from you to the tree (in Figure 6, for example, this distance 'x' equals 20 metres).

Use trigonometry (on your calculator or a statistical program such as Excel) to calculate the height of the tree, minus your eye-height (distance 'y' in Figure 6). Add the height of your eyes above ground level to 'y' to calculate the total height of the tree. In Figure 6, this is 1.6 metres.

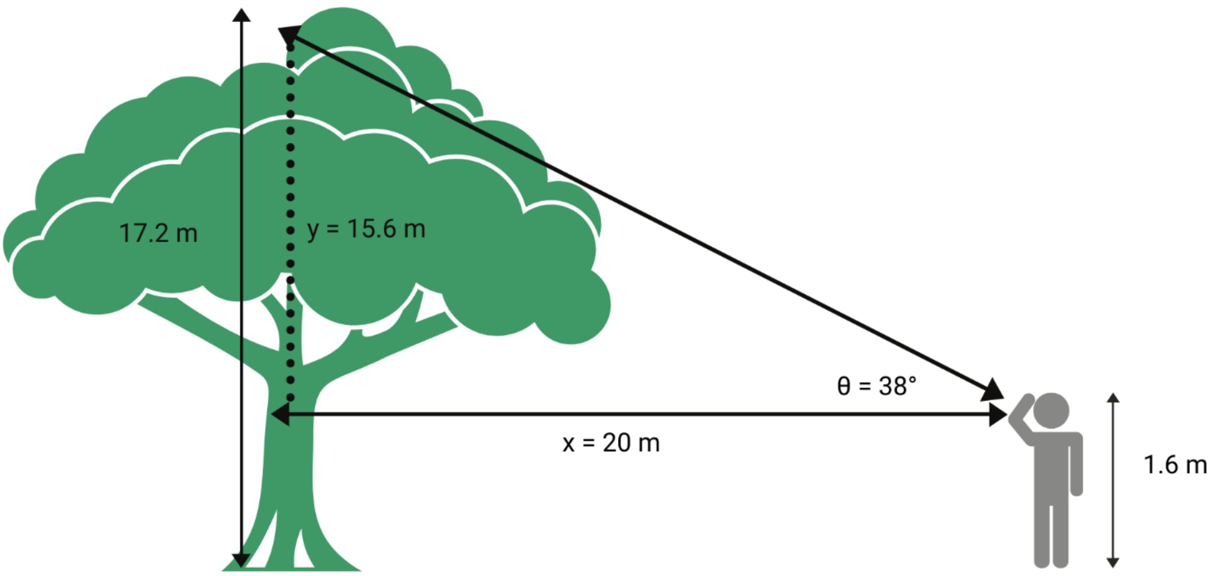

Figure 6 **Estimating the height of a tree using a clinometer and trigonometry**

Using the formula below, you know that x = 20 metres and θ = 38°. Solving this formula for 'y' gives you a value of 15.6 metres. Add the height of your eyes above ground level (1.6 metres) to the distance 'y' to calculate the total tree height: total tree height = 1.6 metres + 15.6 metres = 17.2 metres.

Therefore, the height of the tree in Figure 6 is approximately 17.2 metres.

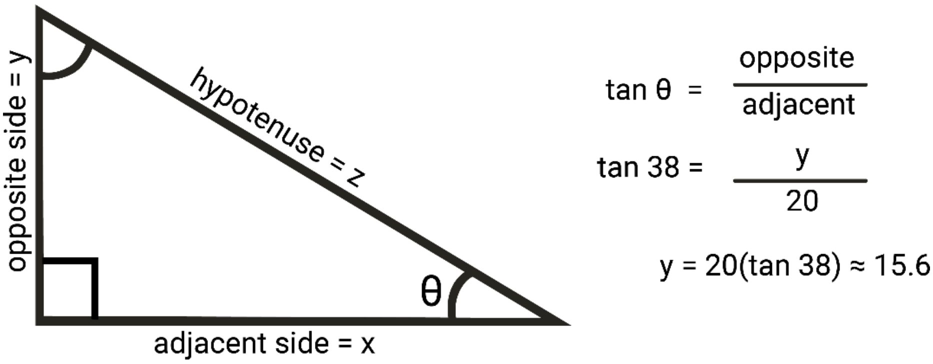

Figure 7 **Formula used to calculate tree height**

Using what you've got

If you don't have access to a clinometer, you could use a ruler. Make a mark at 10% along the length of your ruler (i.e. at the 3 centimetre mark for a 30 centimetre ruler; Figure 8). Walk away from the tree until the top and bottom of the ruler lines up with the top and bottom of the tree (Figure 8). There is no need to stay on the same contour as the tree.

Mark the point on the tree that corresponds to the 10% mark on the ruler; measure the height of this point above ground level (e.g. 1.7 metres in Figure 8). To calculate the total height of the tree, multiply this measurement by 10. For example, if the 10% mark aligns with a point 1.7 metres above ground level, the total tree height = 1.7 metres x 10 = 17 metres.

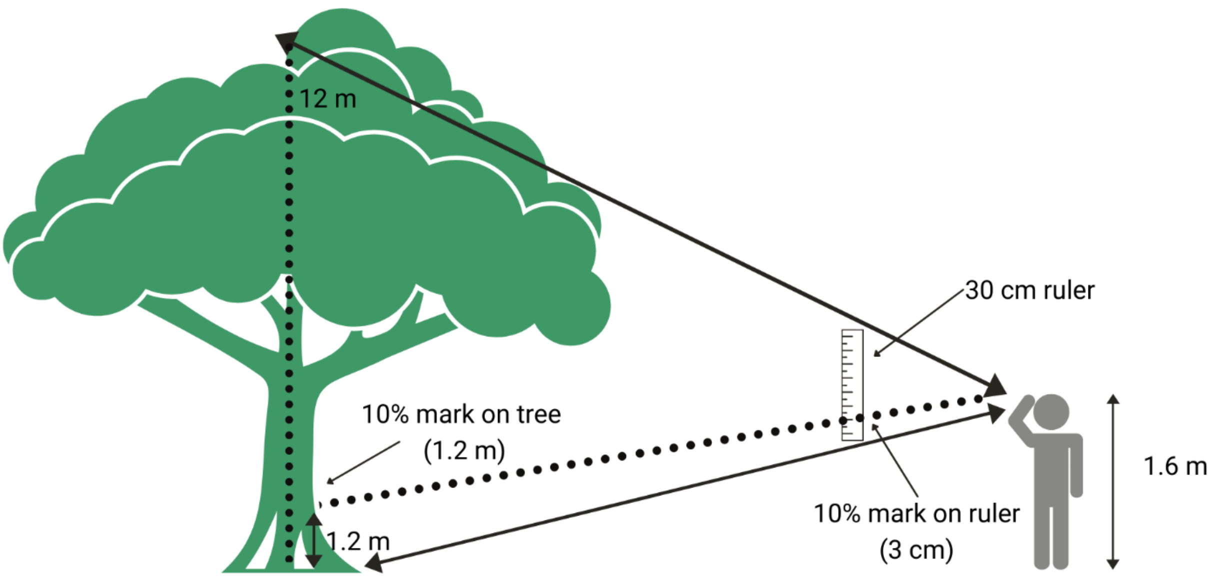

Figure 8 **Estimating the height of a tree using a 30 centimetre ruler**

2.4 Response to control

For each weed species targeted for control, record the technique used for control and the response of each species to the control (Table 11). This will allow you to determine the effectiveness of your control methods and adapt your management technique if required.

Similarly, information on the response of native species to accidental (off-target) herbicide application is limited, and the method of removal of invasive species may influence native community response (Flory & Clay 2009). Recording the status of native species after control will supplement what is already known, which will be useful for the wider community, as well as for interpreting your own results.

There are seven categories established to record the response of plant species (native or exotic) to control (Table 11; following Broese van Groenou & Downey 2006, and John Toth unpublished data).

Table 11 Categories to measure the response of plant species (native or exotic) to herbicide spray as an effect of weed control

Damage	Definition
No damage	There has been no damage to the target plant population as a consequence of control
L (low damage)	≤25% of the target plant population has been damaged, with no dead plants
M (moderate damage)	>25% of the target plant population has been damaged, with no dead plants
LD (low dead)	≤25% of the target plant population is dead
MD (moderate dead)	25% of the target plant population is dead
AD (all dead)	All individuals of the target plant population are dead
Unsure	You are unsure of the damage to your target plant population; detail the reasons why you are unsure

If you suspect there has been off-target damage or there has been a report of off-target damage, you should measure plant responses at the time intervals outlined in the bitou bush aerial spraying guidelines (Broese van Groenou & Downey 2006), being eight weeks and six months after herbicide application. In addition to these responses, information must also be collated on the herbicide application, specifically:

- the herbicide and application rate
- the interval between application and the observation (e.g. two months)
- a follow-up sample to determine long-term response.

The NSW Department of Primary Industries and in particular the NSW Weed Control Handbook (DPI 2018b) may be a useful source for this information. If you are unsure which herbicide or control method to use and how it should be applied, check with the Department of Primary Industries and/or use the WeedWise app.

2.5 Understanding experimental design for monitoring

There are some key concepts to consider before establishing sampling units at your site.

2.5.1 Replication

Replication refers to the number of sampling units chosen for your site. Lack of spatial (and temporal) replication in sampling is one of the most common mistakes when monitoring. Replication is essential to ensure what is measured and calculated across sampling units is *representative* of the site as a whole. Too few sampling units provide an imprecise or inaccurate estimate for the whole study population and, no matter how carefully or randomly chosen, may not be representative. Replication allows you to measure the precision of the generalisations you make in your study and also enables the use of standard errors or confidence limits. Table 12 provides some advice on the minimum number of samples required to provide adequate replication for each method.

Note that the level of replication referred to in this chapter is that within a site. Because sampling units are not replicated across sites, no generalisations can be made for areas outside of your monitored site.

The NSW Biodiversity Assessment Method (DPIE 2020a) requires that replication increase with the size and environmental variability of your sampling area to capture an accurate representation of your site, noting that more replicates will be needed if the vegetation is variable across the site. If you plan to survey multiple habitats within your site, you should undertake replicates within each habitat type, avoiding ecotones, vehicle tracks and other disturbed areas.

Table 12 The minimum number of replicates required by the NSW Biodiversity Assessment Method (DPIE 2020a), determined by the size of your site

Site size	Minimum number of replicates*
<5 – 20 hectares	3 quadrats/transects
20 – 50 hectares	4 quadrats/transects
50 – 100 hectares	5 quadrats/transects
100 – 250 hectares	6 quadrats/transects
<250 – 1000 hectares	7 quadrats/transects
>1000 hectares	8 quadrats/transects

*The recommended number of replicates assumes only one vegetation community with no variation in condition. If multiple communities of varying condition exist at your site, multiply accordingly.

2.5.2 Independence

Independence means that the sampling units are placed far enough apart so measurements are not spatially correlated, i.e. what happens in one quadrat will not affect the result in another simply because they are located near one another. The minimum distance needed to ensure independence of random units will depend on the type of vegetation you are monitoring; a smaller distance is needed for sampling small life forms such as herbs or grasses than for sampling shrubs, and an even larger distance is needed for sampling trees.

When determining the distance between transects, you should also consider the need for **interspersion** throughout your study area. That is, if you can establish only a few transects of sampling units, the distance between transects should be larger than if many transects were being established.

To ensure independence, we recommend you separate sampling units by at least the distance of their longest side. For example, sampling units of 2 x 10 metre quadrats would be separated by a minimum of 10 metres, while 2 x 25 metre quadrats would be separated by at least 25 metres. This same distance rule should be used to determine the minimum distance between transects of sampling units, e.g. line-intercept transects of 25 metres should also be separated by 25 metres.

2.5.3 Sampling design

Once a monitoring question is decided, the first steps to building a monitoring program are to select your sampling design and the appropriate statistical analyses to address your monitoring question/s. Much of this choice depends on the question/s you are asking, the size of the area you need to monitor and the resources available to support statistical analysis and on-ground monitoring.

Appropriate statistical methods should be decided prior to the start of on-ground monitoring, and in conjunction with developing your sampling design. It is important to understand the limitations of the statistical methods you choose, to support your sampling design. Consider contacting statistical experts when developing your program, as they can advise on the most robust methods and their limitations. Government agencies and research organisations have staff with high levels of statistical expertise. Community groups can seek advice from grant managers, who may be able to provide access to statistical expertise.

The ability to count or measure all individuals in a population of interest is rare in ecological systems. It often requires the species' distribution to be small and well-defined, habitat to be accessible and individuals to be conspicuous. Counting or measuring all individuals is called a *population census* (see Sections 2.6.1 – 2.6.4).

More often we will sample populations (or sites that they occupy) by measuring only a (usually small) subset of the population to estimate parameters such as population size, population abundance or site occupancy. Those estimates will likely differ from a census of the whole population because we are measuring only a subset. This difference is called *sampling error*. Moreover, estimates from different samples of the same population are likely to differ from each other. Sampling error is due to making assertions using only sample data, rather than values from the complete population. Many statistical texts address how to ensure that sampling of a population is both unbiased (so that statistical assumptions will hold) and of sufficient size (to limit sampling error).

Any trend (over time) in sites composing an unbiased sample will be representative of the trend (over time) in the population, provided the sample sites remain a valid, unbiased sample of the population.

The objective of monitoring is to determine how population or community parameters (size, abundance, occupancy) change over time; for example, in response to management. One question that often arises is: should a new representative sample of sites or individuals be measured each time (re-randomised sampling) or should the same sample of sites or individuals (fixed sampling) be measured repeatedly?

The decision of whether to re-randomise or use fixed sampling depends on several things, including: monitoring objectives; size of the population or community of interest; available resources; and access to statistical expertise. The statistical techniques and the types of inferences you can draw about the species or population of interest will differ for fixed and re-randomised designs. A thorough analysis of appropriate statistical techniques is beyond the scope of this monitoring manual. You should consult with statistical experts during the design planning stage to ensure the results of your monitoring program can be used to reliably inform future management action.

Re-randomised sampling (temporary plots) involves the placement of new, randomly selected plots each time you sample. As an example, if your objective is to detect a change in plant species abundance and diversity (e.g. of an ecological community over a large area) over time, then in the first year you may randomly select locations for 20 plots within your monitoring site and take measurements of the species in each plot. In the second year of sampling, you would randomly select another 20 locations for your plots and measure the number of species in each of these plots. You would continue to select 20 new randomly selected plots each time you sample. This provides you with a new and independent sample of the parameters of interest each time.

Fixed sampling (permanent plots) involves establishing plots at the start of a monitoring program and sampling these same plots throughout the program. As with temporary plots, fixed plots should be selected in an unbiased manner, e.g. randomly. For example, in the first year of sampling you would randomly select 20 locations and take measurements of the species in each plot. Plots are permanently marked, typically physically using star pickets and spatially with a handheld GPS unit. In subsequent sampling events, measurements are taken from the same established plots as the first sampling. In this example, the two samples are dependent in space, but are independent in time.

Fixed sampling can offer logistical advantages if sample sites are permanently marked and site access is well established; however, permanent markers can be difficult to relocate (even with a GPS), and searching for plots can be time consuming. Taking photopoints of plots and plot markers can reduce this problem. If a fixed plot is lost, a new one must be established, and this can impact statistical analyses and overall conclusions. While initial time resources are high to mark out fixed plots, less time is needed over the monitoring program because new plots do not need to be established each sampling period. Field sampling time is also reduced because species in the fixed plots have already been identified.

If the area of interest is difficult to access or if the vegetation type makes relocating fixed plots time consuming, re-randomised plots may be more appropriate. Re-randomised plots have minimal field setup time, as you do not need to permanently mark plots or find fixed plots each time you sample; however, sampling time can increase as you will need to identify new plots, and new species in those plots, each time you sample. Taking a baseline species list or using BioNet flora data can help reduce this issue. Additional effort and skills (e.g. with GIS) are also required to re-randomise plots in the office prior to each sampling event.

2.6 Selecting a sampling method

To simplify the process of developing a monitoring program, the techniques prescribed are based on the growth form of the species you wish to monitor. These categories have been created to assist you with choosing the correct sampling method:

- plant species (native or invasive)
  - non-rare
  - rare
  - cryptic
- ecological communities.

2.6.1 Monitoring a plant species

The sampling methods for monitoring plant species are described based on the growth form of each species (native or invasive). Species are grouped into three growth form sub-categories:

1. **Tree** – a woody plant usually greater than 5 metres high, which can be single or multi-stemmed dependent on the type of tree

2a. **Shrub** – a many-branched or single-stemmed woody plant, 2–8 metres high for tall shrubs, 1–2 metres high for small shrubs and less than one metre for low shrubs

2b. **Vine/scrambler** – a climbing or scrambling plant with long, typically thin stems that are not self-supportive. Those that climb rely on external support to obtain height, often through the use of hooks or tendrils. Maximum height is generally dependent on the height of the climbing support available. Also called climbers or twiners

2c. **Fern** – a non-woody plant with fronds, growing primarily in damp or moist habitats. Reproduction is typically by spores borne in sporangia on the underside of fronds or in nut-like structures

3a. **Herb** – a plant that does not produce a woody stem, although it may seem woody at the base. Examples include forbs, lilies and orchids

3b. **Grass/sedge** – a non-woody plant often forming tussocks or groups of closely aggregated stems, usually less than one metre high. These plants typically have small flowers enclosed by bracts that form spikelets. Examples include the Poaceae and Cyperaceae families.

2.6.2 Sampling methods for non-rare plant species

Use Table 13 to determine which method you will use to monitor species. Two options are provided within each of the survey types: the first is the preferred option and the second an alternative option (highlighted grey).

The aims of your monitoring program and the site characteristics such as access, topography, vegetation density and ease of setting up plots will determine which option you choose. This will require you, as the land manager, to determine what best suits your needs. The tree category is the exception, as only preferred methods are presented; select your method based on the density of tree species in your monitoring area.

Table 13 Example sampling methods for monitoring programs targeting individual plant species (native and invasive)

An example is a threatened herb within a grassland. For threatened ecological communities refer to Table 14.

Growth form	Sampling unit	Abundance index	Example sampling unit dimensions (metres)
1. Trees	Quadrats if common (>5 per ha^{-1})	Cover or density	20 x 50 (1000 m^2)
	T-square sampling if uncommon (<5 per ha^{-1})	Density	N/A
2. Shrubs, climbers/ scramblers and ferns	Line-intercept transects	Cover	25, 50*
	Quadrats	Cover or density	2 x 25 (50 m^2)
3. Herbs, grasses/ sedges	Quadrats	Cover or density	2 x 2, 5 x 5 , 2 x 10
	Line-intercept transects	Cover	10, 25*

Note: The presence/absence of species can also be measured using quadrats and line-intercept transects; however, as this measure provides only limited information it is not preferred.

* For dense vegetation, where long transects are difficult to establish in a straight line, shorter transects can be used but more should be established.

2.6.3 Sampling for rare species

For rare species sampling may not be required; if you have a good idea of the population extent it may be possible to conduct a population census, where all individuals are located and monitored. A whole population census is best practice and where possible, should be the aim. A rare species field datasheet has been provided in Appendix A. To standardise such censuses, it is important to apply equal effort across a site or have rules in place. For example, you may wish to scale the time spent at each location by the size of the area to be assessed, i.e. searching a larger site for more time than a smaller site, rather than applying the same timeframe to both sites. Where this is not possible, sampling the population is required. Rare species may exist only in one or a few restricted geographic areas or habitats or occur in low numbers over a relatively broad area.

Occasionally a particular site or location may be the stronghold for a particular rare species and the species may be **locally abundant**. If this is the case for the species you wish to monitor, it would be possible to sample the species according to the methods outlined above. Often, however, rare species exist in very low numbers and the methods for sampling non-rare species are not always suitable. For example, if there are only five individuals of a rare species within your site, sampling using re-randomised quadrats will most likely not detect them.

It is assumed that if you are doing weed control to protect a rare species, you already know the locations of these individuals; therefore, fixed monitoring plots should be utilised to monitor these individuals over time. Once individuals of the rare species to be monitored are detected, sample according to (Table 13). You can choose to monitor all the individuals located, or if there are too many to monitor practically, randomly select a subset of individuals to monitor. It may also be important to set up quadrats surrounding known populations to assess if new plants establish following your weed control.

For further information, refer to the Surveying threatened plants and their habitats (DPIE 2020b) or Monitoring threatened species and ecological communities (Legge et al. 2018).

2.6.4 Sampling for cryptic species

Particular care must be taken if the species you are monitoring is cryptic. Cryptic species are those that are difficult to find or detect in the landscape. Causes of crypsis can be divided into two categories, which are sampled in different ways:

- **Category A** – lifecycle, seasonality (e.g. many orchids are only visible above ground when flowering), and similarity between species (e.g. species are only easily distinguishable when flowering). Sample as for non-rare plant species, but based on the species' ecology or when the species is most easily sampled/detectable (e.g. seasonality)
- **Category B** – camouflage (e.g. growth form is hard to detect or distinguish) or size (e.g. very small plants). Conduct a pilot study by undertaking a systematic search of your monitoring area and identify patches or individual plant locations. Sample the located individuals as per non-rare plant species, if random or systematic samples can be placed within the area containing the cryptic species. Otherwise, sample according to rare species monitoring.

> Note: Samples targeted to species patches will not be random and cannot be analysed as such.

2.6.5 Monitoring an ecological community

Ecological communities are associations of particular plant species that exist at particular relative abundance. Monitoring ecological communities is hence inherently more complex than monitoring single or several target species of interest. Ecological communities are groups of species that occur together in a particular area, with the particular flora composition of a community determining its structure. The exact species composition of an ecological community can vary between sites due to both natural (e.g. limited ranges or adaptations to different climatic regimes) and anthropogenic (e.g. the effects of urbanisation or land clearing) factors.

Sampling methods for ecological communities

Two survey types are described here for sampling ecological communities (Table 14), a survey of (i) the whole community (**a floristic survey**), or (ii) a sub-set of the community (3–5 species within each life form category; **dominant survey**). Of course, if the whole community is large, a re-randomised sampling approach is appropriate using randomly placed quadrats or transects, rather than fixed quadrats.

While conducting a full floristic survey will provide the most detailed information on a community's recovery after weed control, the complexity of many ecological communities means this will be time consuming and require advanced botanical knowledge to correctly identify the species present. Surveying the dominant life forms (Table 14) within the ecological community you are monitoring will allow you to gauge ecosystem recovery after weed control with fewer resources required than a floristic survey, but will miss recovery of rarer species that might be really important. The specific aims of your monitoring program, available time or money, plant identification skills, and the characteristics of your site such as access, topography and ease of setting up plots will determine which option you choose. This will require you, as the land manager, to determine what best suits your needs.

Note: If structural stratification is an important feature of your community (e.g. littoral rainforest), you should sample within each strata.

Table 14 Sampling methods for monitoring programs targeting ecological communities, with the option to complete a full floristic survey or survey the dominant life forms within the community

Community structure	Sampling method	Sampling unit	Abundance index	Example sampling unit dimensions (metres)
1. Forest or woodland	Floristic survey	Quadrats	Cover or density	20 x 50 (1000 m²)
	Dominant survey	T-square sampling	Density	N/A
2. Shrubland, scrub, heath and vine thickets	Floristic survey	Quadrats	Cover or density	2 x 25 (50 m²)
		Line-intercept transects	Cover	25, 50*
	Dominant survey	Line-intercept transects	Cover	25, 50*
		Quadrats	Cover or density	2 x 25 (50 m²)
3. Herbfield, grasslands and other low height communities	Floristic survey	Quadrats	Cover or density	2 x 2, 5 x 5, 2 x 10
		Line-intercept transects	Cover	10, 25*
	Dominant survey	Line-intercept transects	Cover	10, 25*
		Quadrats	Cover or density	2 x 2, 5 x 5, 2 x 10

* For dense vegetation, where long transects are difficult to establish in a straight line, shorter transects can be used but more should be established.

2.7 Random sampling: the methodology

Random sampling forms the basis of most statistical techniques and is needed when you can only sample small parts of your whole site/population. Sampling units (plots) need to be placed without bias to account for the variation present across your site; if sampling units are biased your results may be misleading. Random sampling can be used for sampling units that are established using either re-randomised plots (randomly located each monitoring event) or fixed plots (randomly located for the first monitoring event and revisited thereafter in the same locations).

2.7.1 Random sampling

Fully-random sampling units are placed in the landscape using randomly generated coordinates, where the selection of coordinates is constrained only by the boundary of your sampling area. In some situations, however, vegetation density and/or terrain can make locating and re-visiting fully randomised sampling units difficult or overly time consuming. In these instances, it is possible to use a pseudo-random method to place sampling units.

A common method used to ensure unbiased placements or starting points for sampling is to overlay a grid on a map of your management site, clearly marking the monitoring area, then using a random number generator to determine the x and y coordinates of your sampling locations (Figure 9 and Figure 10). If you're proficient in GIS, there are also several ways to quickly generate random points that can then be converted to a file (e.g. gpx) that is read by and displayed on your GPS or smart phone.

2.7.2 Pseudo-random sampling

Pseudo-random sampling units use pre-existing trails or roads as a starting point, with sampling units placed at randomly selected intervals along this transect and potentially also at randomly selected distances from it.

As for random sampling above, you may wish to overlay a grid on the map of your site and use a random number generator to determine the placement of your sampling regime. In addition to randomly determining the interval between quadrats or intercepts, you may also choose to randomly determine the distance from the pathway or trail that you place your sampling units (Figure 11 and Figure 12).

Another way to pseudo-randomly locate plots is to generate more points than the required plots and then cull those points that are unsuitable, e.g. due to dense vegetation, dangerous to sample or too difficult to get to.

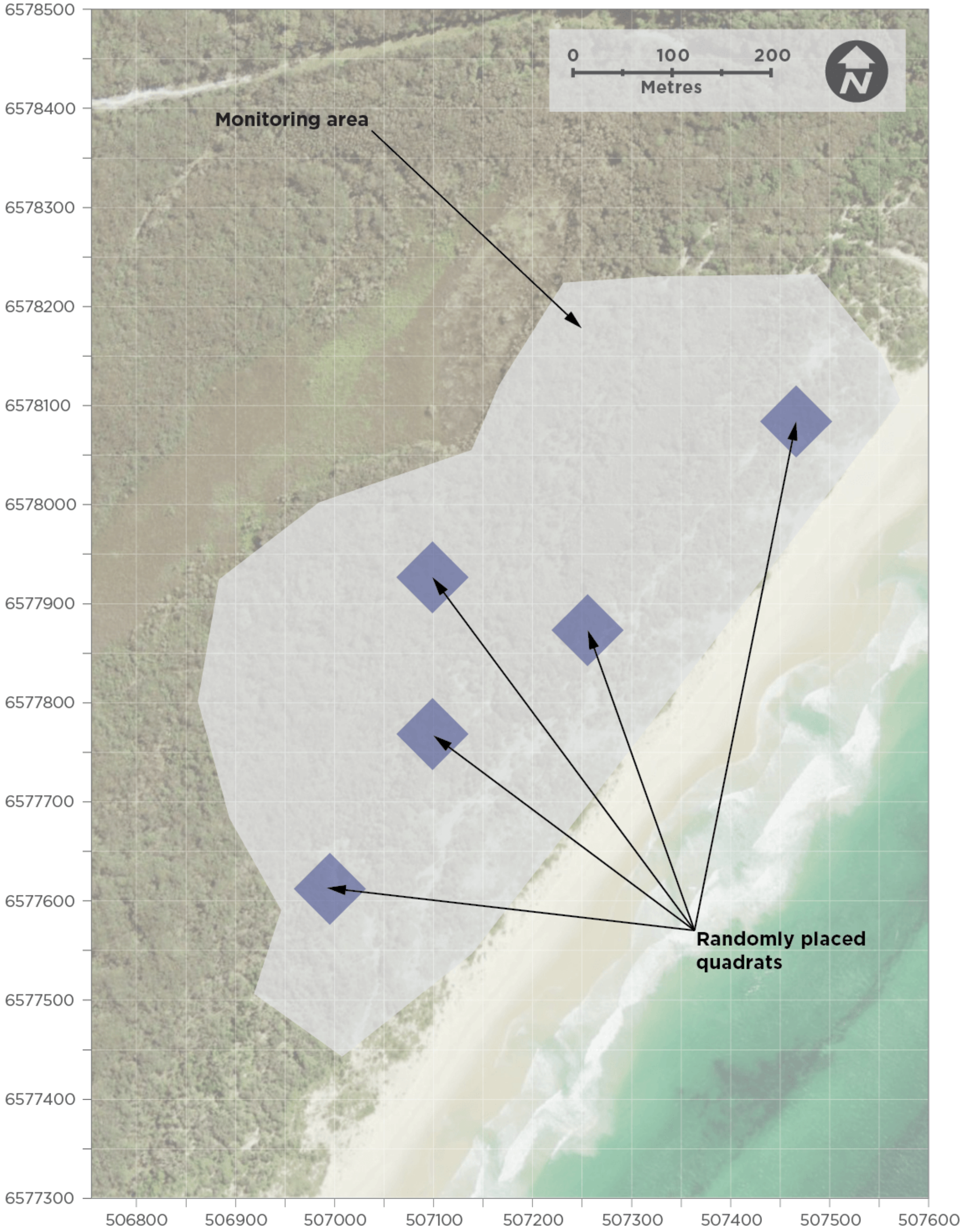

Figure 9 **Example of randomly placed quadrats, selected using a random number generator and a grid digitally placed over the mapped site**

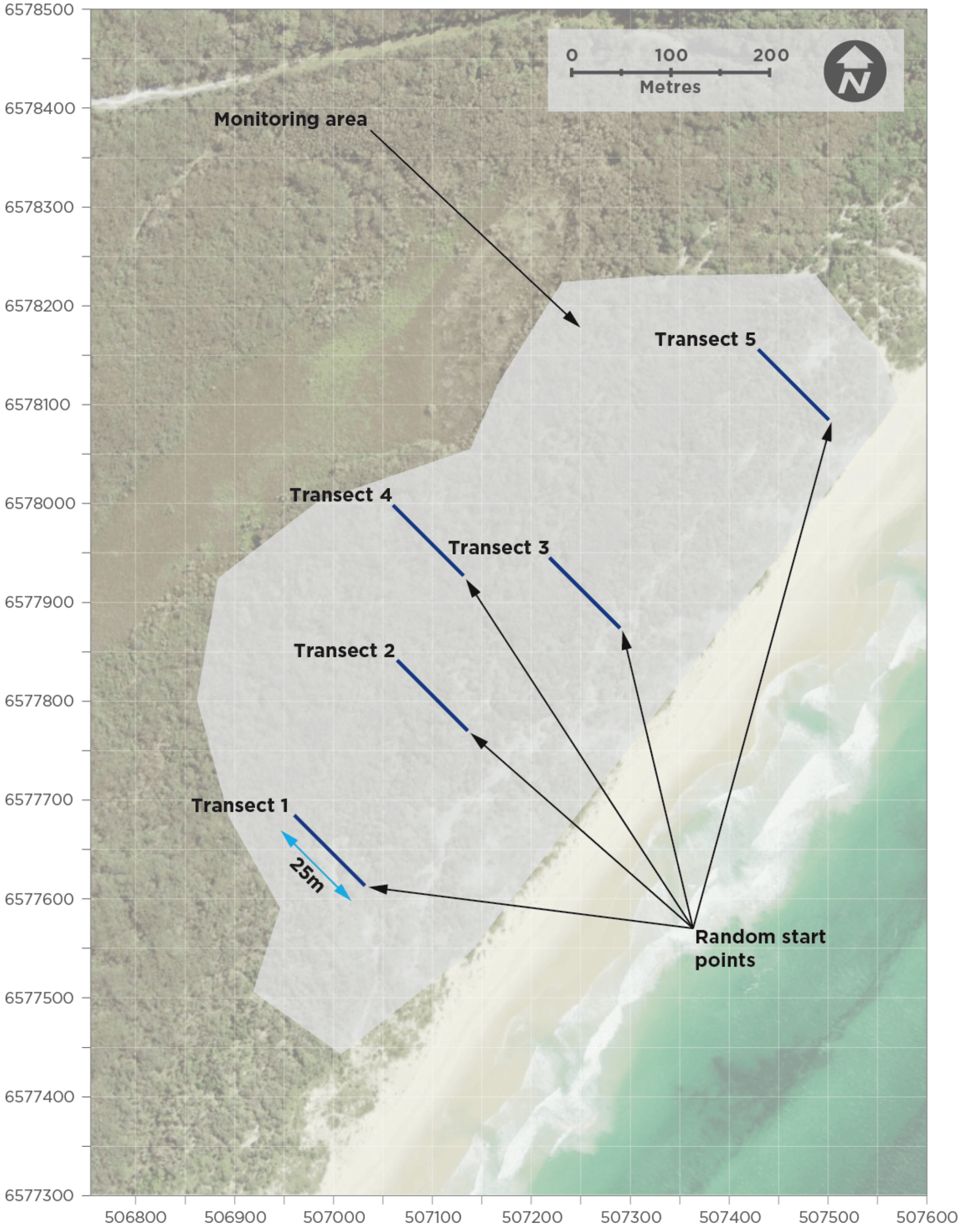

Figure 10 **Example of randomly placed line-intercept transects, selected using a random number generator and a grid digitally placed over the mapped site**

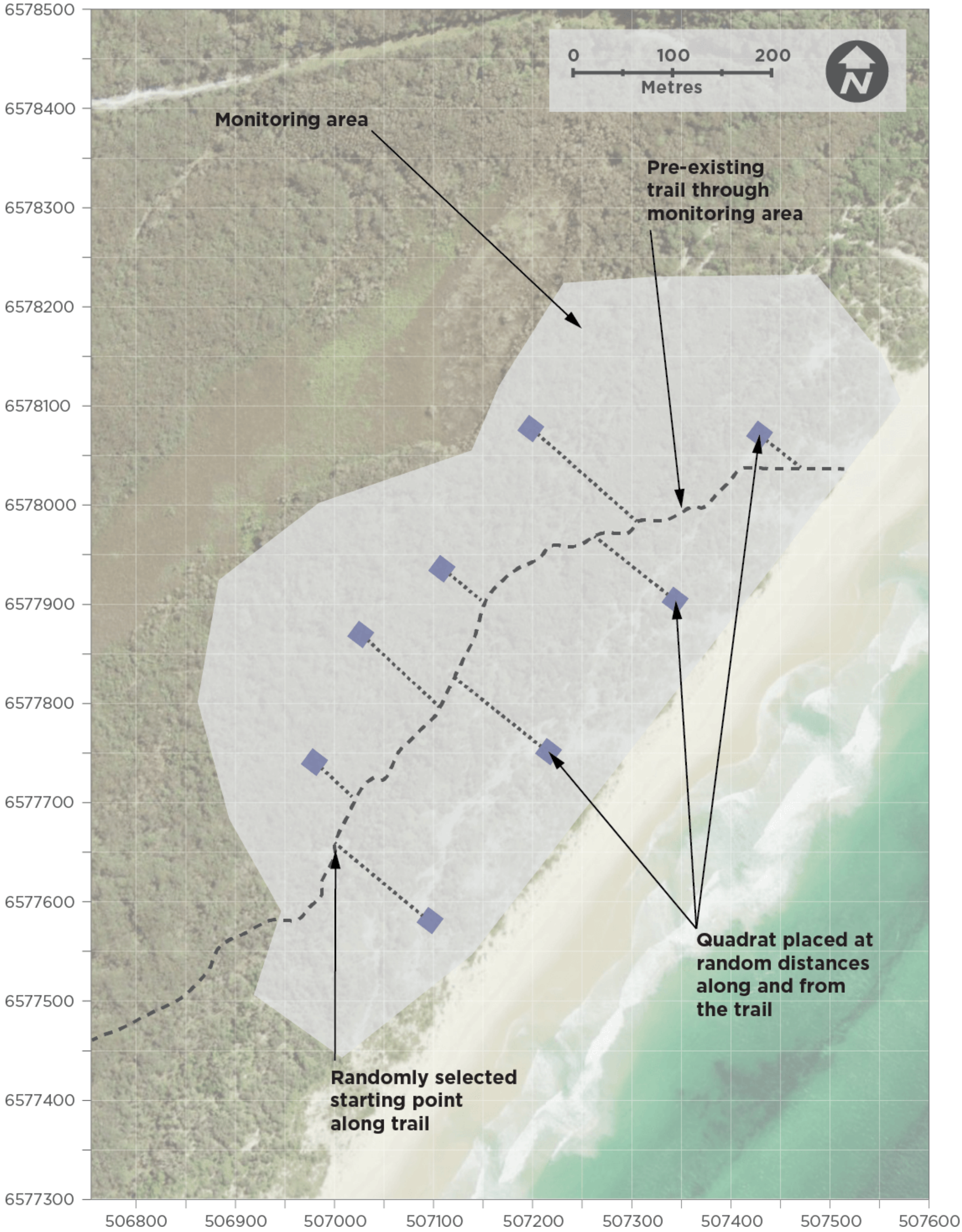

Figure 11 **Example of pseudo-randomly placed quadrats, selected using a random number generator and a grid digitally placed over the mapped site**

In this example, quadrats have been placed at random intervals along the trail and at random distances away from the trail.

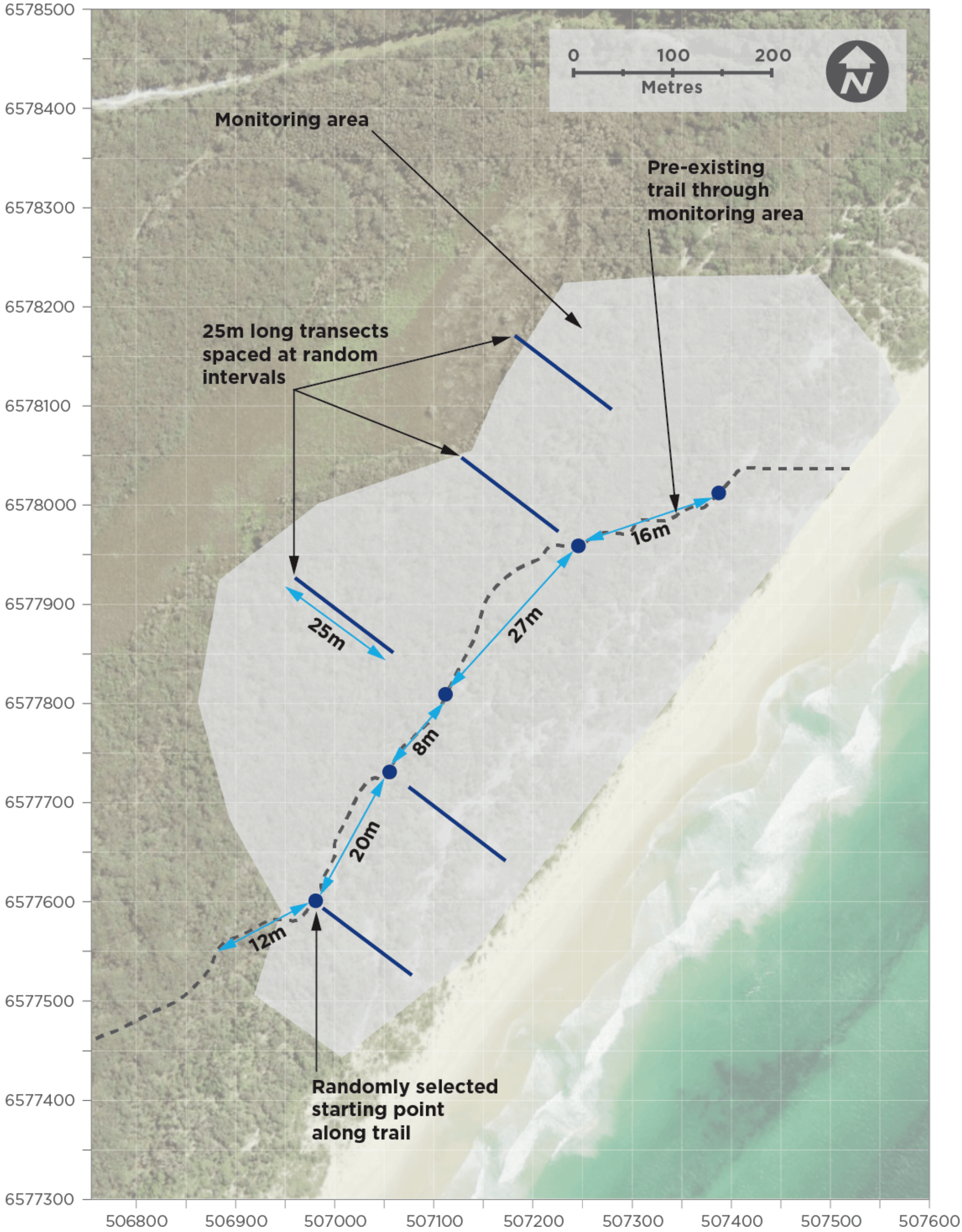

Figure 12 **Example of pseudo-randomly placed line-intercept transects, selected using a random number generator and a grid digitally placed over the mapped site**

In this example, transects have been placed at random intervals along the trail and at random distances away from the trail.

2.7.3 Stratification

Stratification involves dividing the monitoring area into non-overlapping areas (strata), and selecting a simple random sample (or samples) from each of the strata. Stratified sampling is appropriate if there is some underlying structure to the population, for example if different soil types result in different communities, if fires have burnt some patches of vegetation but not others, or if the units to be sampled are not distributed evenly throughout the study area. Variation at your monitoring site is common, from the weed species present, to the biota at risk and many other environmental variables that influence plant species abundance, composition and diversity. It is often useful to carry out a pre-sampling survey to locate populations and ecological communities and their extent so randomly located plots can adequately sample these entities and the natural variation within each of these entities.

Some common methods of stratification include spatial location, such as altitude, vegetation type, and the soil type. The population is expected to be relatively uniform within each stratum. There is a tendency to stratify sampling according to vegetation type; for example, few people would combine monitoring results of *Themeda* grasslands with littoral rainforest and expect them to be comparable. Similarly, you wouldn't compare the vegetation on a ridge with the vegetation in a gully. In some instances, however, care needs to be taken that the study area is suitably stratified (for example, into areas with different fire histories).

Stratified samples will have a smaller standard error than those obtained with a fully random sample if the individuals within strata are more similar than individuals in general.

2.8 How to collect data

In comparison to the observational data discussed in the **preliminary observations** chapter, data collected from monitoring methods presented in this chapter will allow you to **demonstrate outcomes** of your weed control program.

We have prepared datasheets for quadrat, line-intercept and T-square sampling methods, provided in Appendix A, however you may also choose to design your own. Each datasheet should have, as a minimum, the following information recorded:

- plot name or unique identifier
- plot location
- date
- observers
- weed control history (time since last control event)
- plot size
- plot location coordinates
- a site description and a disturbance history of the site.

It is also important to record details of the monitoring program, such as whether re-randomised sampling was used, which indices of abundance were used, and the area sampled at the site.

Before you begin monitoring, you must determine:

- what your monitoring question is, e.g. 'Is my weed management benefiting species x?'
- which species (native and invasive) you will focus on, or are you measuring overall species richness
- which sampling methodology is best suited to your project
- if you will be using fixed or re-randomised sampling units
- for random sampling units, if you will be using random, pseudo-random or stratified sampling:
    - how you will ensure independence of your sampling units, and
    - how you will ensure adequate replication of your sampling units.

You should also record the **cost of control and monitoring activities**, refer back to the **preliminary observations** chapter (Section 1.8) for more on how to collect this information.

2.8.1 Line-intercept transects

Line-intercept transects are one-dimensional plots (i.e. a single line, often a tape measure) that are primarily used to measure canopy cover, though presence/absence can also be measured. Cover measured with line-intercept transects is generally less subjective than those in quadrats because actual cover is measured, rather than estimated. Line-intercept transects are best used to measure low vegetation, being most accurate where the vegetation height is comparable to the height of the tape, and they are commonly used to survey changes in vegetation especially at sites that are linear.

Two measures of abundance can be made using the line-intercept method: cover and presence/absence. Density measures using this technique produce a measure that is ambiguous and dependent on the size of the species being measured, and so is not recommended.

From a species perspective, the line-intercept technique lends itself readily to measuring the cover of shrubs and matted plants; it is less effective for plants with lacy or narrow canopies such as grasses and some forbs, and for trees and taller shrubs that are high above the transect. Transects are also more difficult to establish and sample in dense vegetation as the densely clustered plants and foliage prevents access and obtaining a straight line for the transect. Line-intercept transects can be used quickly for plants with low but densely clustered cover, and they are more practical in sparse vegetation than quadrats. Plants that are sparse, small and well-distributed along a line will require more meticulous inspection of the transect.

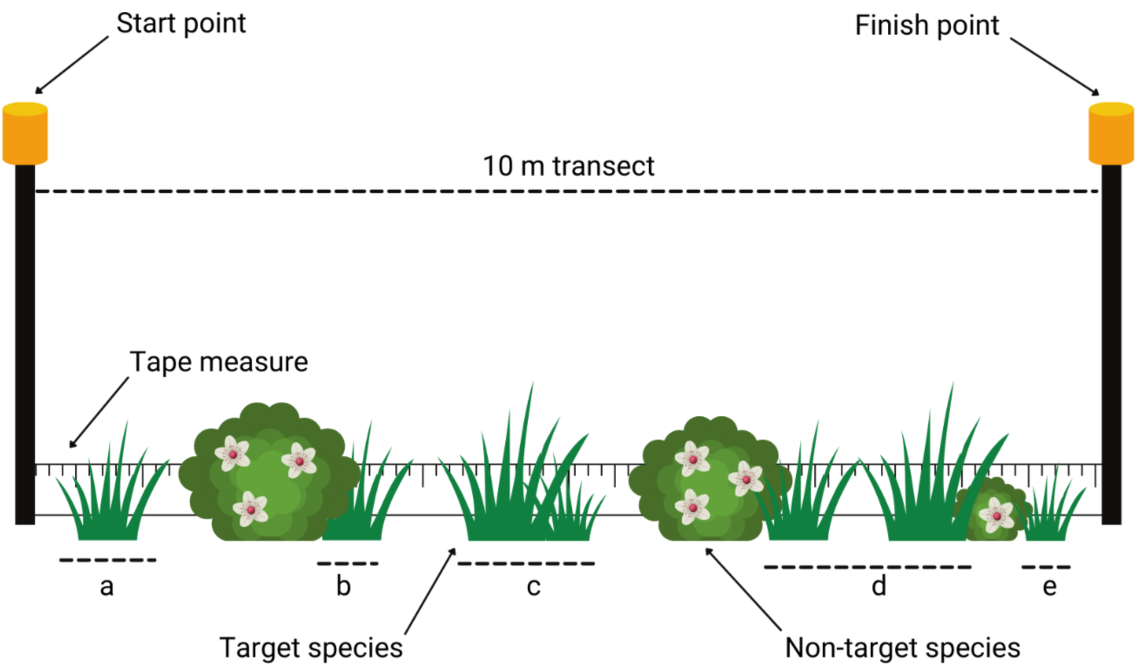

Figure 13 **An example of a 10 metre line-intercept transect in the field**

The grass is the target species, for which information will be collected in the field. The species with white flowers is a non-target species, therefore no information will be collected about this plant. Note: This diagram is not to scale.

Line-intercept transects are commonly used for shrubs that are less than 1.5 metres tall because a tape can be suspended above the shrub canopy and the interception easily measured. Irrespective of the height of the transect, however, you should abide by the definition of cover as the vertical projection on the ground.

In addition to the site information on your datasheet, you should include whether your sampling is random or pseudo-random, your transect orientation, a unique identifier for each transect, the transect length, the minimum distance between transects and the coordinates of each transect's starting point.

Note: The list of species selected must be the same for all samples (i.e. for each transect and year). You may wish to add species, but you can't remove any (i.e. stop sampling them).

Indices of abundance

Using line-intercept transects, two measures can be used to estimate an index of abundance for target species: percentage cover and presence/absence. Select the one most appropriate to your needs. Note: The presence/absence data will give you less robust data and limit you to calculating the proportion of plots a species is present at, for example.

Line-intercepts should not be used to sample/monitor tree species (see Table 13).

You must determine a maximum gap between canopies or foliage of individuals for each species (the gap width) before you collect any data. The maximum gap width is the maximum distance between the canopies of consecutive plants below which you will consider the canopy to be continuous (irrespective of the number of individuals). Thus, gaps in the canopy shorter than this distance are recorded as continuous cover and those greater are recorded as separate entries or cover intercepts. An appropriate maximum gap width for a shrub may be 10 centimetres (Figure 14). For some priority native species, a shorter distance might be more appropriate, but this needs to be specified and documented before you start sampling.

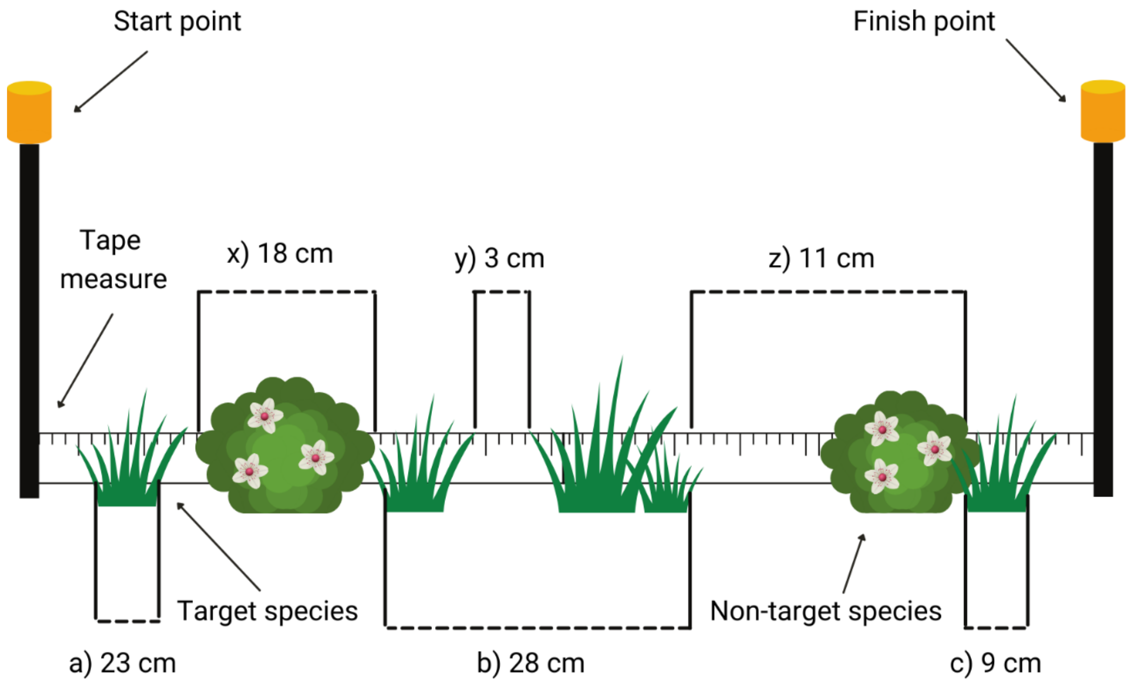

Figure 14 **In this example, the grass is the target species, for which a maximum gap width of 10 centimetres was determined**

The gap marked 'y' does not exceed the maximum gap width, so the target species is measured as having continuous cover across section 'b'. The sections 'x' and 'z' do exceed the maximum gap width and so sections 'a', 'b' and 'c' would each be recorded as a separate cover intercept. Note: This diagram is not to scale.

Percentage cover

Cover (also called canopy cover) is the vertical projection of vegetation from the ground as viewed from above. Cover is a commonly used measure as it enables all species to be compared irrespective of their size or abundance (e.g. from small but abundant to large but rare). Unlike **density**, cover is closely aligned with biomass or annual production. Cover does not need you to be able to distinguish between individuals of the same species. It can, however, be sensitive to changes over the growing season. Although the lack of native deciduous species makes this less of an issue in Australia, sampling should be repeated at the same stage of the growing season.

In the field, run the measuring tape from the start to the endpoint of the transect, then walk along the transect and record where on the tape your target species begins to intercept and where it ends. The total length of each intercept can be calculated, and you will also have the raw distances to ensure errors of calculation can be fixed. Intercepts at the beginning and end of each transect either start on 0 metres (zero) or end at the maximum transect length (e.g. 10 metres), irrespective of whether the vegetation cover continues beyond the transect. Overlapping cover of the same species should not be counted twice but should be recorded as continuous cover.

To save time, it is advised that you collect the data simultaneously for each target species rather than repeating the process (Figure 15). Ensure you record the entire intercept length of each target species irrespective of any canopy overlap by a different species.

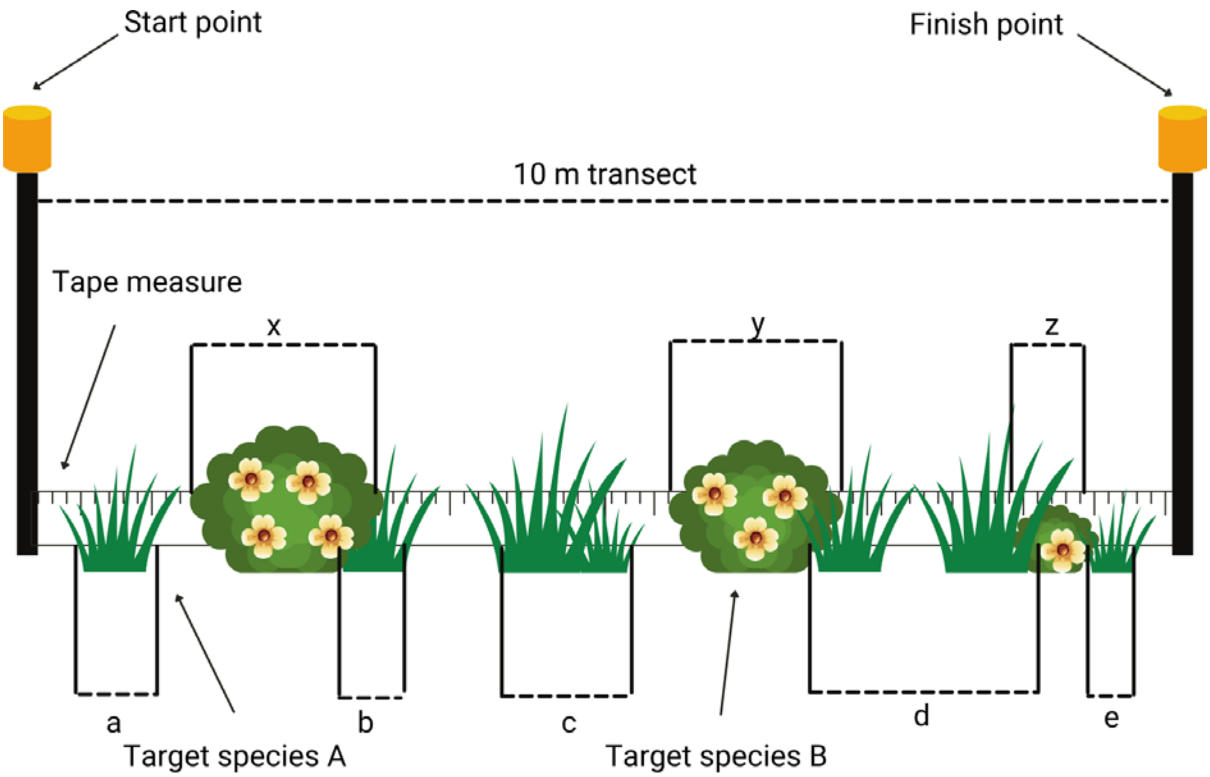

Figure 15 **Combined sampling of two target species using the line-intercept method**

The intercept distances for target species A (a, b, c, d and e) are measured independently of those for target species B (x, y and z), but are recorded in the one walk along the transect. Note: This diagram is not to scale.

Presence/absence. This method of data collection is very simple but doesn't yield very informative data unless performing a floristic survey. In the field, walk along the transect and record the presence of each target species that intercepts the tape measure. If it is present, no matter how abundant, record the species' name.

Demographic data

Life history. For a more detailed measure of how your target species is responding to weed control, recording how each life history category (i.e. seedlings, juveniles, adults or dead plants; Table 6) is intercepting with your transect will provide you with more in-depth information.

For a more rigorous measure of life history, you can also measure the line-intercept cover of each life history category separately for each target species. Note, however, that this might be very time consuming. You will need to consider each life history class separately when determining continuous cover (i.e. continuous cover only applies for the same life stage).

Height. If applicable for your target species, you can record the height of the tallest individual of each target species per intercept.

Response to control. Record the response to your control works of your target species as a whole across your transect, using the seven categories (Table 11).

Data evaluation

Data should be entered into a spreadsheet or database for preservation as soon as possible after collection. If using a spreadsheet program (e.g. Excel), record the file name and location on the top of the datasheet so you can easily find the files later.

Complete data analysis soon after data collection. This will enable problems to be identified early and ensures that questions requiring additional field visits can be identified. It is also more likely that questions that arise when entering field data can be answered because the field work is still fresh in your memory.

2.8.2 Quadrats

Quadrats are two-dimensional plots that are usually square or rectangular. Quadrats can be used to sample any type of vegetation. Cover, density, species richness and presence/absence can all be measured within quadrats. Long, thin quadrats are often referred to as transects or belt transects, though these differ from line-intercept transects.

Biases in quadrat sampling relate to the census technique used. For cover and density, more conspicuous species are likely to be observed and those forming clumps of individuals with broader leaves are likely to be given a higher estimate than those that are less conspicuous, dispersed or with fine leaves. Careful sampling should overcome this bias.

Very large quadrats can be difficult and time consuming to set up and measure. If species have a non-random distribution over the study area (a common occurrence for plants) then estimates of abundance from a single quadrat will be heavily influenced by the size of the quadrat. Yet, larger quadrats will capture more of the patchiness in the vegetation than smaller quadrats. For this reason, selecting an appropriate number of replicates with the resources available to you is important, as well as the appropriate quadrat size and spacing according to the growth form.

To establish your quadrats in the field, locate the starting points of your random quadrats or the pathway that will determine your pseudo-random quadrats using your site map and a GPS receiver/smart phone or tape measure. If you are establishing a fixed plot, flagging tape or fluorescent-coloured paint can be used to mark the top of either the star pickets or wooden stakes for easy recognition in the field. Brightly coloured string can be used to encircle the star pickets or wooden stakes to further delineate the quadrat boundaries when collecting data. This should be done just prior to recording data for each quadrat and the string should then be removed. If establishing re-randomised plots (not fixed), you can lay out a tape around the perimeter of your quadrat and begin recording your species responses. To ensure your quadrat corners are 90° use a compass to determine the direction/bearing for each side.

In addition to the standard information you should collect, you should also record information on your boundary rules (see below) of quadrats, what corner/s are marked with stakes (if not all four), whether you are using random or pseudo-random sampling methods, the dimensions of your quadrat and the coordinates of your pre-designated quadrat corner.

Boundary rules

Many plants will lie on the boundary lines of the quadrat and you will need to decide how to classify what is included in the quadrat and what is not (Figure 16). If estimating cover, generally all cover within the bounds of the plot is considered in, irrespective of where the individual is rooted. For measuring density, some commonly recommended boundary rules are:

- Plants are considered 'in' if any part of the plant is touching the quadrat boundary along two pre-selected and adjacent sides (of a square or rectangular quadrat); all other plants must lie completely within the quadrat boundaries to be included. You should predetermine which two adjacent sides will be interpreted in which way (i.e. 'in' or 'out'). In Figure 16, for example, the two bolded lines either side of the pre-selected corner (tagged) are the 'in' lines. This approach accurately estimates density and is the recommended method to reduce boundary bias (Elzinga et al. 2001).
- Plants are considered 'in' if more than 50% of the plant boundary (basal or canopy) is within the plot. This method accurately measures density, however subjective observer decisions are required, and it may be particularly difficult for irregularly shaped plants.

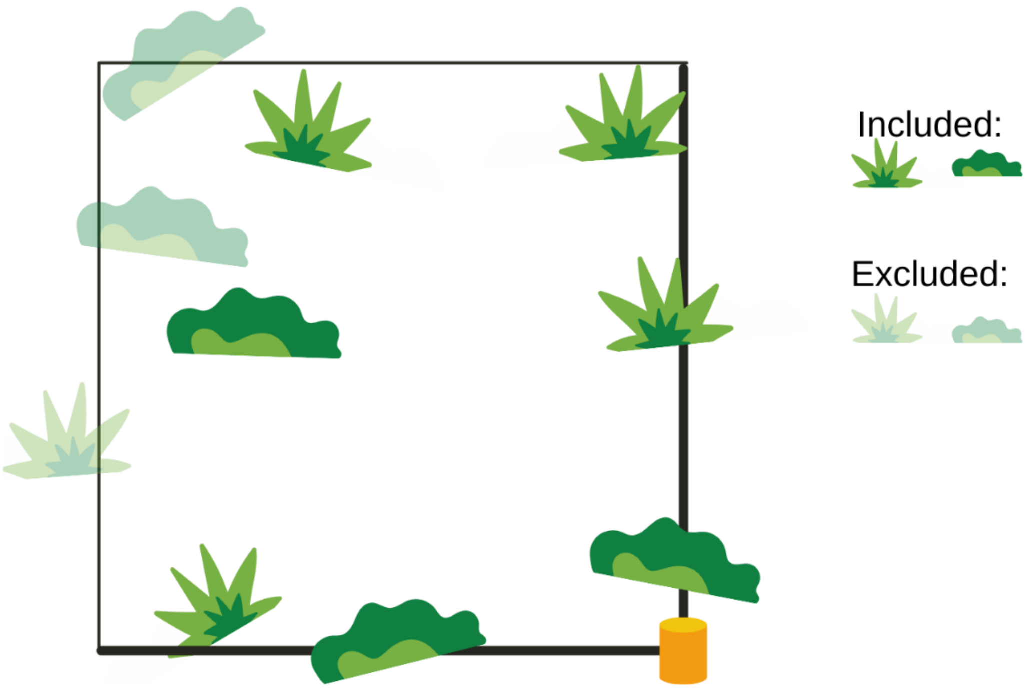

Figure 16 An illustration of a possible boundary rule for two target species sampled in a quadrat

Individuals included in the density estimate are completely within the quadrat or intersecting the two selected boundary lines (the bolded lines adjacent to the pre-selected corner picket in yellow). Transparent individuals are excluded.

Some other commonly used, but not recommended, boundary rules are:

- Only plants that are completely within the plot are included. This method underestimates true density.
- All plants that touch the boundary line are counted, even if most of the plant lies outside the quadrat. This method overestimates density.

Note that density estimates will vary slightly, depending on which boundary rule you apply. Therefore, the boundary rules used for the first monitoring event must be used for all subsequent monitoring events.

Indices of abundance

Using quadrats, three measures can be used to estimate an index of abundance for target species: percentage cover, density and presence/absence. Select the most appropriate for your needs and time/money.

Quadrats should only be used to monitor tree species if they are common (see Table 13) or as part of a floristic survey of an ecological community (see Table 14 and associated text).

Percentage cover. Visually estimate the proportion of the quadrat occupied by each of the target species. We suggest you measure actual cover for each target species, which can be converted to other indices. Which method you choose will affect how you analyse your data. Overlapping cover of the same species should only be recorded once (i.e. the maximum cover of a species in a plot would be 100%).

Repeat this process for all target species. If performing a full floristic survey, it is common to traverse your plot slowly and build your species list and get an idea of species' cover; once complete, the cover or cover class is assigned to each species. Ensure you record the entire cover of each species irrespective of any canopy overlap by a different species. Thus, the combined cover value of all target species may exceed 100%.

Density. Count the number of individuals of each target species that occur within the quadrat (adhering to your boundary rules) and record this on your datasheet. These counts will need to be converted to densities (individuals per square metre), which can easily be computed (e.g. using Excel).

Note: It may be difficult or too time consuming to measure the density of some species (e.g. clumping grasses or mat-like plants). For these species, you can estimate the number, or count patches or clumps (remember to record this if you do so).

Presence/absence. Search within the quadrat for a target species (adhering to your boundary rules). If it is present, no matter how abundant, record the species' name on your datasheet. All species not observed and recorded are assumed absent. You can use presence/absence sampling to complete a floristic survey.

Demographic data

Life history. If using **cover**, for each species estimate the percentage of your quadrat that each life history category (seedling, juvenile, adult or dead) covers. Note, the percentage cover across all life history categories can sum to more than 100% if you have overlapping layers of plants, e.g. seedlings growing under adults.

If using **counts** (abundance), count the number of individuals in each life history category. These counts will need to be converted to densities (individuals per square metre), which can easily be computed (e.g. using Excel).

If using **presence/absence** to record life history, simply record each life history category for each target species.

Response to control. Record the response to your control works of your target species as a whole across your quadrat using the seven categories (Table 11).

Diameter at breast height (DBH). As trees are slow growing, recording the diameter at breast height is optional as differences may not be detectable unless your monitoring program spans decades.

Record the diameter at breast height (metres) of at least five randomly selected individuals of your target tree species in each quadrat. For example, generate five sets of coordinates using the random number generator. Measure the DBH of the individual at these coordinates or, if there is no target individual at these coordinates, the height of the nearest individual. If an individual is selected more than once, generate additional random coordinates to select a new individual.

If there are fewer than five individuals of a target tree species present, measure the DBH of all individuals. Note, quadrats should only be used to sample tree species if they are common (see Table 13).

Height. As for DBH above, measuring the height of trees is optional. Record the height of the tallest individual of each target species in your quadrat. Alternatively, record the height of at least five randomly selected individuals of each target species. These could be the same trees as you measured for DBH.

Data evaluation

Data should be entered into a spreadsheet or database for preservation as soon as possible after collection. If using a spreadsheet program (e.g. Excel), record the file name and location on the top of the datasheet so you can easily find the files later.

Complete data analysis soon after data collection. This will enable problems to be identified early, and ensures that questions requiring additional field visits can be identified. It is also more likely that questions that arise when entering field data can be answered because the field work is still fresh in your memory.

2.8.3 T-square sampling

T-square sampling is a form of plotless sampling, i.e. there are no marked boundaries to the sampling unit, other than the boundaries of your monitoring area. T-square sampling is a nearest-neighbour sampling method for estimating density. It uses the distance between a randomly selected starting point and the nearest plant, together with the distance between that plant and its nearest neighbour, both measured along a straight line from the starting point, to estimate density. T-square sampling is used to monitor large or scattered trees when quadrat sampling is impractical, although it can also be used for other growth forms, especially if vegetation is at low densities. Density is the only index of abundance that can be measured with this method. Note that the density of trees is unlikely to change quickly, however, so this technique is more useful for comparing densities *between* areas than changes over time.

The core area in which T-square sampling occurs should be slightly smaller than the total monitoring area, to allow for the possibility that the nearest individuals to some of the random starting points might be outside the core area. Ideally, the number of random starting points should be greater than 10 (Sutherland 2006).

T-square sampling does not suffer the same degree of bias when individuals are distributed non-randomly as several other nearest-neighbour measures; however, it is still advisable to apply a test of randomness to the data. A simple test for randomness is:

$$t^i = \{\sum_i [x_i^2 / (x_i^2 + y_i^2 / 2)] - m/2\}\sqrt{(12/m)}$$

where: x_i = the distance from the random starting point to the nearest tree

y_i^2 = the distance from the nearest tree (x_i) to its nearest neighbour

m = the number of random starting points

If t^i is greater than +2 then the distribution is significantly more regular than a random distribution (i.e. dispersed); if it is less than –2 then it is significantly clumped; if it is between –2 and +2 it is considered random. This is an approximate test only; refer to Diggle (1983) for further details.

If individuals are at such low densities that they are not within sight, or the vegetation so dense that line of sight is affected, this method can be inaccurate as the nearest neighbour may not have been selected. This method is only useful for single species and not full floristic surveys.

Indices of abundance

Density is the only index of abundance that can be measured using T-square sampling. From each random point, measure the distance to the nearest individual (of any age) of a target species (labelled ‘x’ in Figure 17); record this first distance. The nearest individual could be in any direction and thus may require some searching. Distances may be measured in the field or via waypoints recorded on a GPS or smartphone and calculated on the phone or in GIS. Individual trees can be labelled in a way that ensures you can recognise which are ‘nearest neighbours’ (i.e. A1, A2, B1, B2, etc.).

Determine the direction of the nearest individual using a compass and record this bearing. Note: North is zero (0) degrees.

From the first individual, measure the distance (labelled ‘y’ in Figure 17) to its nearest neighbour of the same species (of any age) when moving in a forward direction (i.e. along the same or similar compass bearing; Figure 17); record this second distance. Only these two distances are measured for each starting point per target species. Repeat this for replicate estimates of that species.

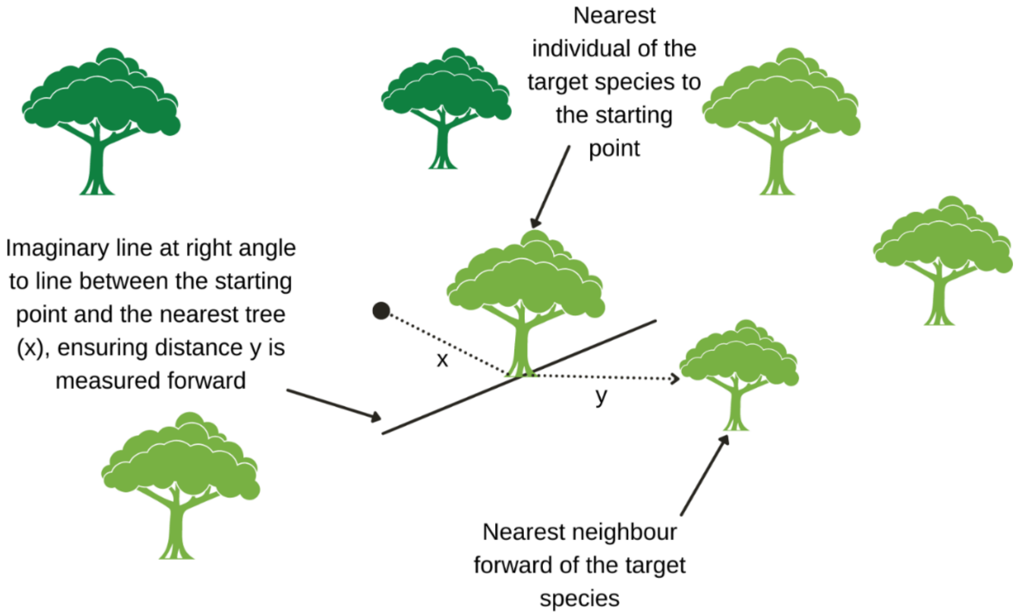

Figure 17 **Example of how to conduct T-square sampling to monitor a target tree species**

If you are monitoring more than one target species using this method, collect the data for each target species independently using the methods outlined above. Ensure the starting points for each target species (e.g. species B; Figure 18) are determined independently (i.e. using a completely new set of random numbers). Record data for each different target species sampled on the same datasheet, except where the target species occur in different monitoring/management areas or ecological communities.

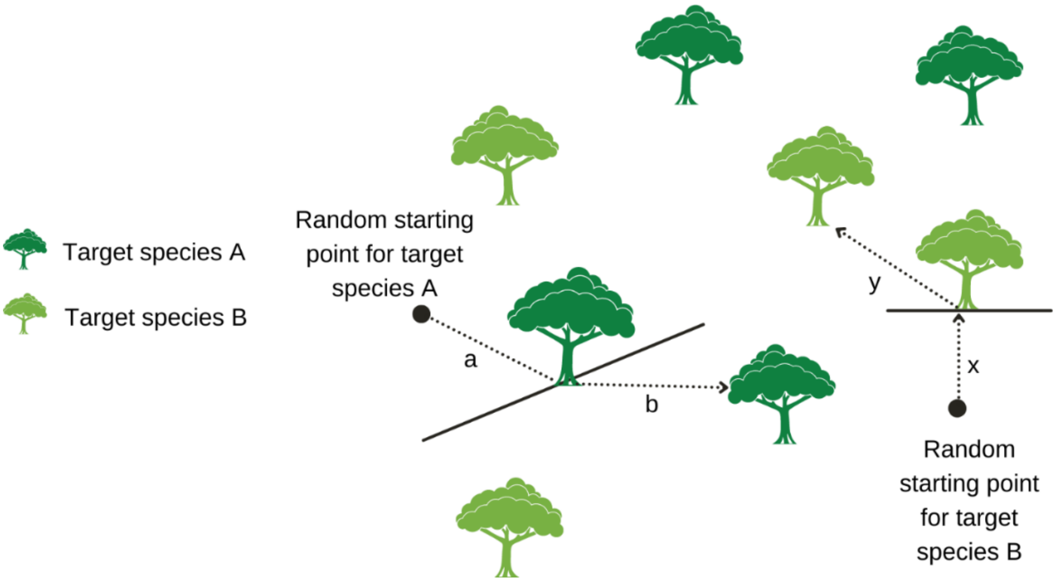

Figure 18 **Combined sampling of two target species using T-square sampling**
A different random starting point is selected for each species.

Demographic data

Life history. Record the life history category (juvenile, adult or dead) of each individual. This method may not be suitable for seedling life stages due to detection and identification difficulties.

Diameter at breast height (DBH). If DBH is a metric useful to answer your monitoring question, it can be recorded during sampling. Record the diameter at breast height of each individual. If any of the individuals is a seedling or juvenile (sapling) a DBH measure may not be appropriate, record n/a instead.

Height. If plant height is a metric useful to answer your monitoring question, it can be recorded during sampling. Record the height of each individual.

Response to control. It is not possible with T-square sampling to gain a good indication of responses of the population as only two live individuals are assessed per starting point. However, it is still possible to record information on these two individuals.

To estimate the response to control, use the categories outlined in Table 11.

Data evaluation

Using T-square sampling you will have collected data that can be used to obtain a density estimate of the species you have measured at your site. Data should be entered into a spreadsheet or database for preservation as soon as possible after collection. If using a spreadsheet program (e.g. Excel), record the file name and location on the top of the datasheet so you can easily find the files later.

Complete data analysis soon after data collection. This will enable problems to be identified early and ensures that questions requiring additional field visits can be identified. It is also more likely that questions that arise when entering field data can be answered because the field work is still fresh in your memory.

2.9 Contributing your data to state and national datasets

Data sharing can assist land managers to more effectively allocate resources, assess effectiveness of investments, contribute to research, and improvement management outcomes. In New South Wales, you can contribute your records to BioNet Atlas. While all records can be submitted as 'species sightings' data, if you have collected your records via a systematic survey, we encourage you to contribute your survey data via the systematic survey modules. Advice on gaining access to BioNet Atlas, an overview of the steps involved in contributing sightings or survey data, and related resources are available on the Contribute data to BioNet Atlas webpage, or contact the BioNet team.

At the national level, data can also be submitted to the Atlas of Living Australia (ALA). See the How to submit a data set webpage, or contact the ALA Data Management Team.

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Appendix A – Field datasheets

This appendix contains the following datasheets for you to print out and use in the field:

1. Observation Monitoring Field Datasheet
2. Photopoint Monitoring Field Datasheet
3. Rare Species Field Datasheet
4. Line-Intercept Transect Field Datasheet
5. Quadrat Sampling Field Datasheet
6. T-square Distance Measure Field Datasheet

Observation Monitoring Field Datasheet

Site description

Plot name		**Site name**	
Management area		**Control stage**	(e.g. stages 1-3 in your site plan)
Date		**Observers**	
Time since control		**Area sampled**	

Landform (one per site)

	Elevation (metres above sea level)	Slope (degrees)	Aspect (e.g. degrees or bearing)
Topography			

Description of site

Describe the structural features of the site, including vegetation characteristics, structure and any prominent landscape features. If known, the disturbance history of the site should also be listed here with time since disturbance, accuracy of this information, severity and area affected (in square metres or hectares).

Page 1 of 3

Observation Monitoring Field Datasheet, continued

Cover / density estimates

Fill out either cover or density, depending on what is appropriate for your monitoring program

	Target species	Cover class (tick cover category or estimate actual cover to nearest whole number) 0%	1–5%	6–25%	26–50%	51–75%	>75%	Density estimate Number
1								
2								
3								
4								
5								
6								
7								
8								
9								
10								

Age dynamics of the target species

Method used: Cover ☐ Density ☐

Optional: if measuring age dynamics, you can fill this section out instead of the above, to avoid overlap.

Estimate the percentage cover or number of individuals within each life stage category

	Target species	Seedlings		Juveniles		Adults		Dead plants		Total
1			+		+		+		=	
2			+		+		+		=	
3			+		+		+		=	
4			+		+		+		=	
5			+		+		+		=	
6			+		+		+		=	
7			+		+		+		=	
8			+		+		+		=	
9			+		+		+		=	
10			+		+		+		=	

Page 2 of 3

Observation Monitoring Field Datasheet, continued

Ecological communities - description

Describe the structural features of the ecological communities including dominant species

Response to control

Control technique used

Response categories:

	ND = no damage	
L = ≤25% damage, none dead	LD = ≤25% dead plants	AD = all dead
M = >25% damage, none dead	MD = >25% dead	U - unsure

	Target species	Response	Comments
1			
2			
3			
4			
5			
6			
7			
8			
9			
10			

Unmanaged weed species

List other weed species that are not currently being managed

	Target species	Cover class (tick box) 0%	1–5%	6–25%	26–50%	51–75%	>75%	Change since last monitoring increase	decrease	no change
1										
2										
3										
4										
5										

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Photopoint Monitoring Field Datasheet

Photopoint location — Photo ID | Easting | Northing | Datum and zone

Camera and lens model | Zoom/focal length details (if any)

Description of location & subject of photo

Photographer's name | Time of day | Date

Direction from the camera post (e.g. north) | Sighter post distance ____ metres

---

Photopoint location — Photo ID | Easting | Northing | Datum and zone

Camera and lens model | Zoom/focal length details (if any)

Description of location & subject of photo

Photographer's name | Time of day | Date

Direction from the camera post (e.g. north) | Sighter post distance ____ metres

---

Photopoint location — Photo ID | Easting | Northing | Datum and zone

Camera and lens model | Zoom/focal length details (if any)

Description of location & subject of photo

Photographer's name | Time of day | Date

Direction from the camera post (e.g. north) | Sighter post distance ____ metres

---

Photo storage details

	Filename	Location (e.g. network drive and folder name)
1		
2		
3		

Contact person — Name | Details

Page 1 of 1

Rare Species Field Datasheet

Site name		**Datum**		**Date**	
Management area		**File name**			
Observers		**Photopoint ID**			
Ecological community					
Rare species name					
Conservation status					

Notes

	Life history				Health status						
plant number	seedling	juvenile	adult	dead	sick	diseased	healthy	Height (m)	DBH (cm)	Waypoint no.	Notes

Page 1 of 2

Rare Species Field Datasheet, continued

	Life history				Health status						
plant number	seedling	juvenile	adult	dead	sick	diseased	healthy	Height (m)	DBH (cm)	Waypoint no.	Notes

Page 2 of 2

Line-intercept Transect Field Datasheet

Plot name ____ **Site name** ____ **Date** ____

Management area ____ **Ecological community** ____ **Easting** ____

Observers ____ **Northing** ____

Starting points: **Fully-random** ☐ **Pseudo-random** ☐ **Starting distance** ____ **(m)** **Time since control** ____ **Photopoint ID** ____

Systematic transects

Length of: Transects ____ (m) Sub-transects ____ (m)

Distance between: Transects ____ (m)

Orientation of: Transects ____ Sub-transects ____

No. transects ____ **No. sub-transects** ____

Sub-transect start pts

Trans no.	**1**	**2**	**3**	**4**	**5**	
A						(m)
B						(m)

Random transects

Transect length ____ **(m)**

Transect orientation ____

Notes

Floristic survey ☐ **Max gap width (cm)** ☐ **Intercept measurements (m)** **Life history dynamics** **Cover (%)** ☐ **Total length (m)** ☐

Transect (no.)	**Sub-transect (no.)**	**Target species name**	**Start**	**End**	**Seedling**	**Juvenile**	**Adult**	**Dead**	**Height (m)**	**Response to control**

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Line-intercept Transect Field Datasheet, continued

			Intercept measurements (m)		Life history dynamics					
					Cover (%)		Total length (m)			
Transect (no.)	Sub-transect (no.)	Target species name	Start	End	Seedling	Juvenile	Adult	Dead	Height (m)	Response to control

Page 2 of 2

Quadrat Sampling Field Datasheet

Plot name ______ Site name ______ Date ______

Management area ______

Ecological community ______

Observers ______

Boundary rule and notes:

Starting points: fully-random ☐ pseudo-random ☐ starting distance ______ (m)

Time since control ______ Photopoint no. ______

Random quadrats

Min. distance between quadrats: ______ (m)

Min. no. of quadrats: ______

Dimensions: ______ (m)

Eastings ______

Northings ______

Floristic survey ☐

Life history dynamics

Cover (%) ☐ Count ☐

Quadrat (no.)	Target species name	Cover	Count	Seedling	Juvenile	Adult	Dead	DBH (cm) 1	DBH (cm) 2	DBH (cm) 3	DBH (cm) 4	DBH (cm) 5	Height (m) (optional) 1	Height (m) (optional) 2	Height (m) (optional) 3	Height (m) (optional) 4	Height (m) (optional) 5	Response to control

Page 1 of 2

Quadrat Sampling Field Datasheet, continued

Quadrat (no.)	Target species name	Cover	Count	Life history dynamics Cover (%) ☐ Count ☐				DBH (cm)					Height (m) (optional)					Response to control
				Seedling	Juvenile	Adult	Dead	1	2	3	4	5	1	2	3	4	5	

Page 2 of 2

T-square Distance Measure Field Datasheet

Plot name

Site name

Date

Management area

Ecological community

Notes

Observers

Starting points:

Fully-random

Pseudo-random

Photopoint ID

Start point no.	Easting	Northing	Tree species	Direction to 1st tree (degrees from North)	Distances (m)		DBH (m)		Height (m)		Life history dynamics (tree numbers)				Response to control
					X	Y	1st tree	2nd tree	1st tree	2nd tree	Seedling	Juvenile	Adult	Dead	

Page 1 of 2

T-square Distance Measure Field Datasheet, continued

Start point no.	Easting	Northing	Tree species	Direction to 1st tree (degrees from North)	Distances (m)		DBH (m)		Height (m)		Life history dynamics (tree numbers)				Response to control
					X	Y	1st tree	2nd	1st tree	2nd	Seedling	Juvenile	Adult	Dead	

Page 2 of 2