Edition 1.0
June 2026

g.co/hardware/biodiversity-monitors

A Manual for Biodiversity Monitoring at Supply Chain Sites

Google

Welcome!

We are happy you are here to learn more about what you can do to monitor biodiversity at supply chain sites.

Getting started is easy!

You don't need to be an environmental expert to follow the methods in this manual. Everything inside is tailored to be simple to set up, highly automated, and deeply rewarding.

Thank you for your dedication to your site, your community, and our shared planet. Let's explore what nature is waiting to return to our workplace!

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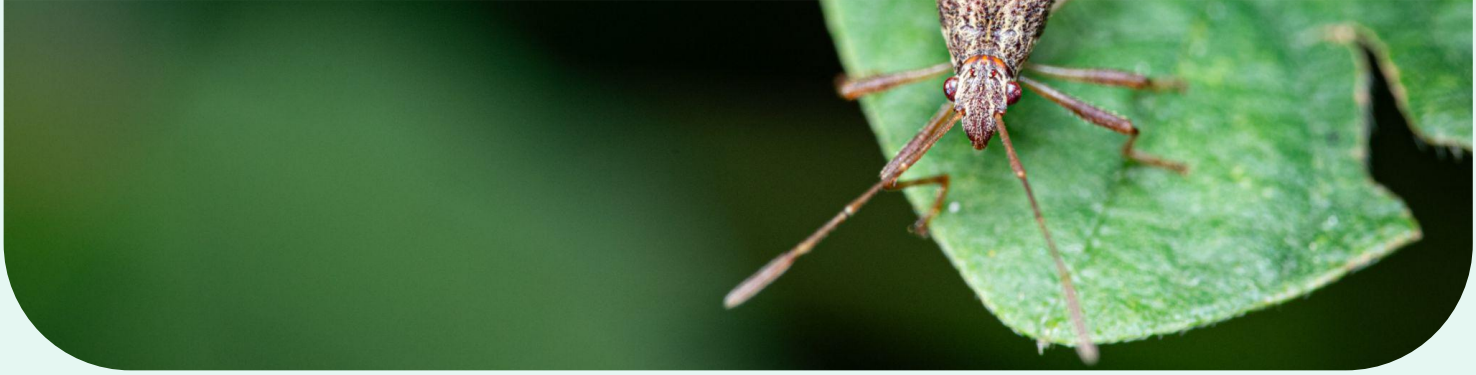

01 Introduction: Measuring biodiversity at supply chain sites

Taking action to improve habitat at supply chain sites is likely to lead to increases in the amount of biodiversity at a site.

Biodiversity monitoring, or tracking the arrival and presence of animals at a site, provides a way to understand the outcomes of projects. If projects are successful at creating supportive habitats, animals and plants may respond quickly, arriving at the site within months of new habitat creation. Data from biodiversity monitoring can offer powerful evidence of the return on investment that can be achieved from improving onsite conditions.

Here, we lay out a practical manual for monitoring that any site manager can deploy to demonstrate the return on investment associated with biodiversity projects. Recent advances in technology are creating new tools to passively monitor animals in the field at a fraction of the cost typically associated with biodiversity monitoring relying on human observers.

The methods in this manual rely primarily on low-cost, low-effort approaches that can be deployed by non-experts to efficiently measure the outcomes of projects. These approaches can be easily scaled across sites, and can be combined in various ways depending on the project goals or location.

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02 Designs for measuring biodiversity outcomes

The most powerful way to measure improvements in biodiversity is through baseline comparisons, in which monitoring is conducted both before and after projects take place.

This method allows for direct comparison of improved conditions to baseline conditions, enabling the tracking and identification of species that most likely responded to habitat improvement. While baseline comparisons are the gold standard for understanding how species respond to habitat improvement, they are not the only available method. If measuring biodiversity after projects have taken place is the only option available, this method can also provide insight into which species are being supported by the habitat at your site. If feasible, a final design can compare your site to other sites that are similar to your site before it was improved.

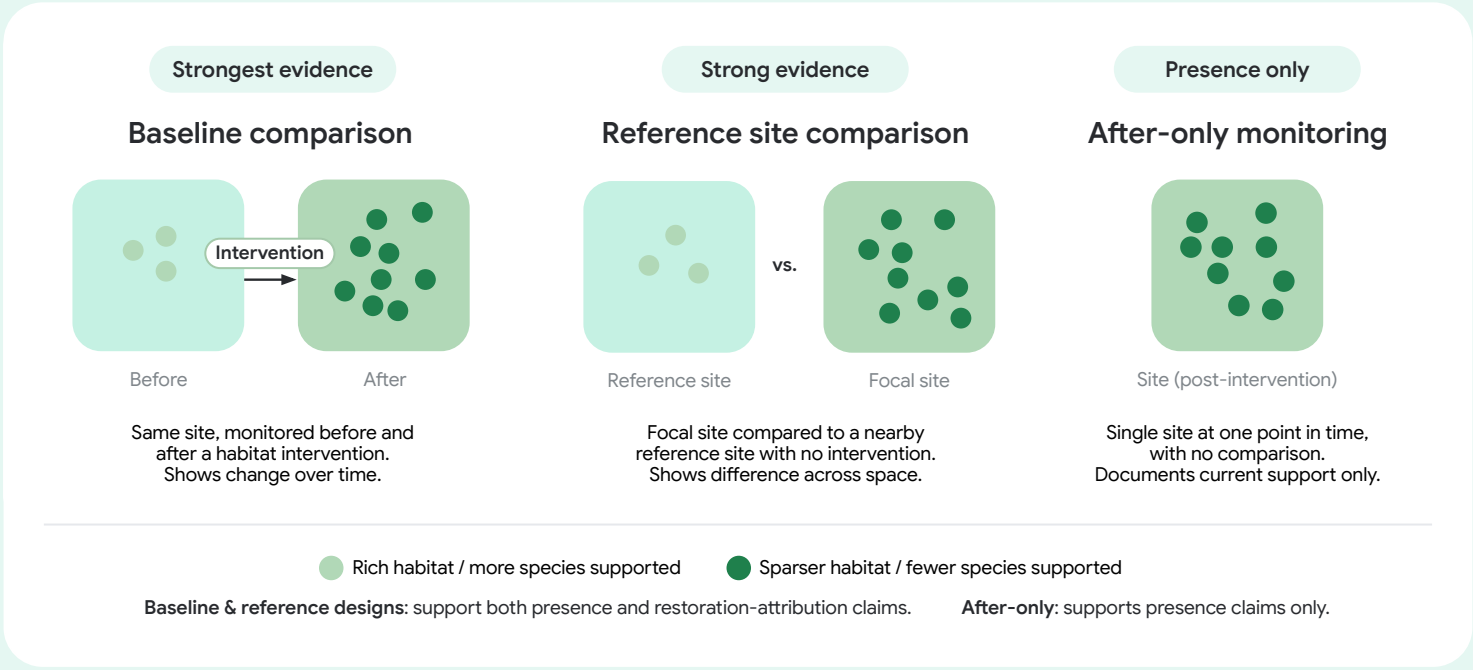


Figure 1. Three types of study designs provide options for biodiversity monitoring at supply chain sites after project interventions. While baseline comparison and reference site comparisons offer the strongest evidence, if after-only monitoring is the only feasible design, this option can still provide insight into what species are supported by habitat creation at the site.

03 Building your own monitoring stack

Recent advances in low-cost sensor hardware, open-source software, and DNA-based identification have made it feasible to conduct species surveys with little expert oversight in data collection.

For example, site-managed passive acoustic recorders, environmental DNA (eDNA) sampling, insect trapping, and motion-activated camera traps can be deployed and serviced by trained site staff rather than visiting specialists. The emergence of these tools is bringing biodiversity monitoring within reach for facilities managers at supply chain sites. It is important to note that these methods do not completely exclude expertise, as reference databases and final species validation may be required.

Beyond monitoring wildlife, measuring baseline weather conditions can also provide insight into how a supply chain site is changing as biodiversity projects are implemented. For example, in industrial zones, urban heat islands, or the elevation in temperature above ambient conditions in the surrounding landscape can be significant, and temperatures may be elevated relative to areas outside cities and industrial areas. Changes in temperature and humidity may occur rapidly after project installation. Simple weather monitoring devices can track these changes and show additional return on investment associated with projects that can also translate into benefits to employees at the project site.

3. Building your own monitoring stack

This manual recommends 4 possible options for monitoring animals at a site and local weather conditions.

Collectively, these options work together to comprehensively monitor many of the main wildlife groups that are most likely to occupy a site after biodiversity improvements, including mammals, birds, amphibians, and insects.

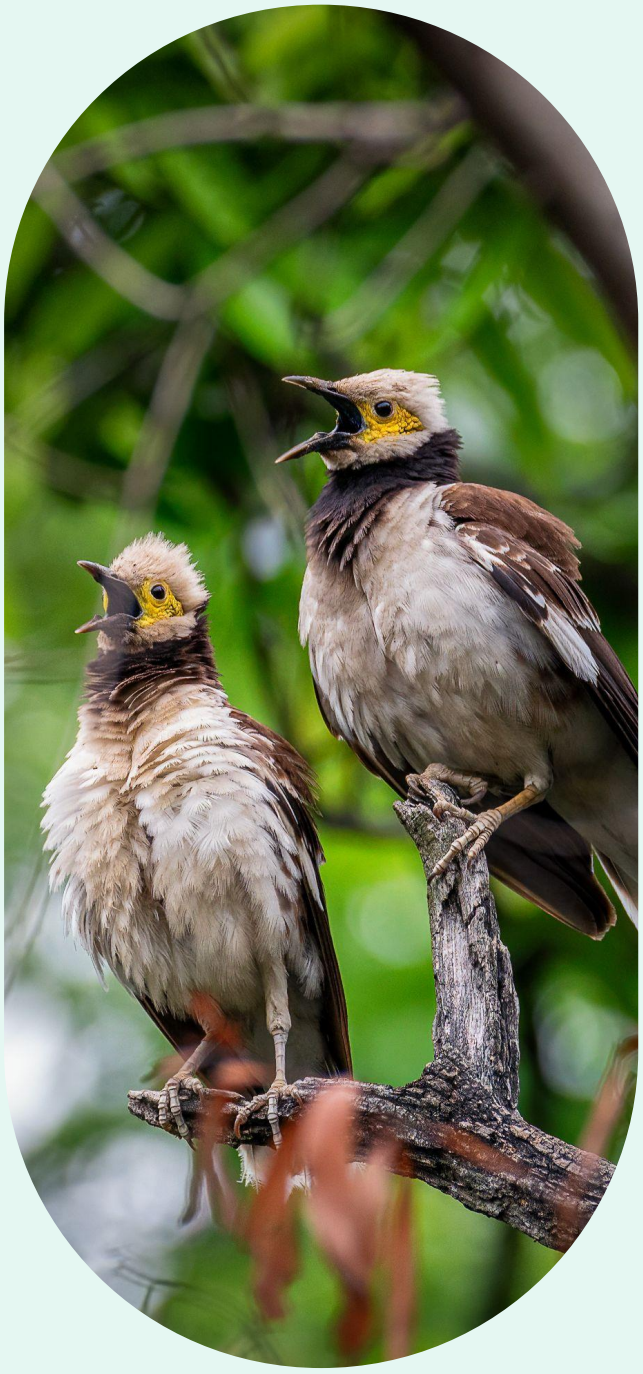
These methods will not necessarily monitor all wildlife, and each method has both bias and certain organisms it cannot efficiently track. For example, Passive Acoustic Monitoring methods are ineffective for birds that very infrequently make noise, such as vultures. Despite these drawbacks, these methods offer a low-cost solution that will, if deployed together, efficiently and effectively capture a wide range of species that are using your site.

Monitoring method	Description	Animal groups reached
3.a **Passive acoustic monitoring (PAM)**	Audio recorders deployed at fixed stations capture continuous soundscape data over weeks to months. Vocalizations are identified to species using AI tools (e.g., Perch, BirdNET).	Birds, frogs, bats
3.b & c **eDNA from plants/water**	DNA shed by insects onto plant surfaces is collected via leaf or flower swabs and sequenced to identify species present, without needing to capture or observe the insects directly.	Insects (Plants) Aquatic species (Water)
3.d **Camera traps**	Motion-triggered cameras at fixed stations photograph wildlife as they pass. Images are classified to species using ML tools (e.g., SpeciesNet).	Mammals
3.e **Temperature**	Environmental sensors collect site-level temperature and microclimate data over time, providing baseline environmental context but not species-level detection.	None — baseline weather data

3.a Privacy-enabled passive acoustic monitoring (PAM)

How it works

Passive acoustic monitoring (PAM) is a technique for monitoring birds using AI-assisted models to identify species from audio data. PAM provides a low-cost way to monitor the changes associated with biodiversity improvements at consumer electronics manufacturing sites. As native landscaping is integrated and biodiversity-conscious management increases, birds that were not formerly present may be expected to show up at the site. Monitoring the site both before and after biodiversity improvements allows projects to demonstrate how animals respond to landscape changes.



While several standard PAM devices exist for purchase, a major concern many potential users have is the privacy and corporate security issues related to the recording of human speech. Privacy-enabled PAM devices solve this issue by processing all audio recordings directly on the device, removing the need for storage of audio recordings. Key features of this approach include **no cloud storage of audio recordings and no recording of human speech - the only data stored is a species detection list.**

What it measures

Birds are the most commonly monitored group with PAM because nearly all birds have songs that are unique to their species, and these calls can be identified from audio recordings. Other types of animals that make sounds, such as amphibians and mammals, can also be identified using PAM devices, though data collected from animals beyond birds may be less reliably detected compared to birds.

3.a Privacy-enabled passive acoustic monitoring (PAM)

Components

The privacy-enabled PAM device runs on a Raspberry-Pi device (a small single-board computer) that requires external power. Core components of a PAM system include the following:

- Raspberry Pi 5 with a case and active cooling system. Note earlier iterations of the Raspberry Pi do not have enough RAM to run the Perch model on the device.
- Simple lapel microphone (like this or similar)
- Waterproof housing to protect the device from the elements (like this or similar)

Electrical Box
Houses Raspberry Pi and protects from weather

Raspberry Pi
Small computer. Runs AI model locally to record all birds heard within 100 feet of device.

Microphone
Connects to Raspberry Pi. Listens to bird song.

Plug
Power plug for Raspberry Pi.

Figure 2. Basic components of a privacy-enabled PAM setup, including a Raspberry pi, microphone and electrical box for protection from the elements.

3.a Privacy-enabled passive acoustic monitoring (PAM)

Installation guide

01. Preparing a Raspberry Pi for installation

To prepare for installation, an operating system must first be installed on the Raspberry Pi. Once installed, an AI detection model needs to be installed. There are at least two AI-models for detecting birds that can run on a Raspberry Pi: Google's Perch model, and the Cornell Lab of Ornithology's BirdNET model. Instructions for installing operating systems and AI Models on Raspberry Pi devices can be found here (GitHub repository for installing Google's Perch model on a Raspberry Pi is here). Note that the BirdNET model can only identify birds, whereas Google's Perch model identifies birds, amphibians, and mammals. For more information on installation, maintenance, and data management, contact Second Nature Ecology and Design directly for support.

3.a Privacy-enabled passive acoustic monitoring (PAM)

Installation guide

02. Finding a location and installing the device

The core considerations for placing the PAM devices are:

Power supply	The privacy-enabled PAM device requires external power. Therefore, the device needs to be either located near a building within reach of an outdoor outlet, or run using a solar panel/battery system.
Proximity to the greatest diversity of birds	The device should be placed in the areas where birds are most likely to sit and call/sing such as clumps of trees. If the site is large, multiple devices can be installed which will enable more complete detection of the composition of the bird community at your site.
Distance from human traffic	Select a location with a minimum of human traffic where birds are most often found. We highly recommend placing a sign near the device to alert people walking by that a listening device is present and reviewing local laws and regulations to determine notice requirements.

If using a solar panel, it is crucial to select a location that receives minimal shade throughout the day to ensure continuous operation. However, Raspberry Pi devices are vulnerable to overheating. To minimize this risk, place the solar panel in the sun and the device in an adjacent shady spot.

Figure 4. The PAM device is housed in an electrical box, mounted on a garden stake (left), and connected either to an outdoor outlet or to a solar panel system (middle). We highly recommend including a sign (right) to alert passersby that a listening device is present.

3.a Privacy-enabled passive acoustic monitoring (PAM)

Installation guide *(continued)*

03. Downloading and managing data

The privacy-enabled PAM device does not upload audio files or detections of birds to the internet. Therefore, the data must be retrieved manually. To download data, you can either bring the device into an office or you can download the data directly in the field. If you bring the device indoors, you can connect it directly to a computer monitor, mouse and keyboard, where you can download the data to a standard USB drive. To download the data in the field, you will need a tablet (like this or similar). An external mouse and keyboard can also be connected to the Raspberry Pi device using standard USB ports. The PAM device should be checked once a month to ensure that the device storage has not been reached and that it is still receiving power properly.

The PAM device listens for birds continuously, and will rapidly accumulate large volumes of data. We therefore recommend planning accordingly, identifying a home spreadsheet in Google Sheets or MS Excel where data will be stored, and establishing a data summary and analysis pipeline. Because no audio is stored, data outputs are stored as text files that are relatively compact, including the species identified, the time of the detection, and the confidence of the detection. For more information on confidence scores, see Wood & Kahl 2024.

PAM data can be summarized using standard pivot tables, a feature that groups and counts data into a summary, to create lists of species and numbers of times each species has been detected over time. This information can be compared before and after project installation to identify how bird communities have responded to improvements at the site. The data can also be used to track changes in bird communities over time, providing useful information to guide ongoing biodiversity-friendly management at the site.

Keep in mind that PAM AI models can be wrong and thus, it is important to periodically interpret your results with an ecologist to ensure the accuracy of your findings. Sometimes similar-sounding birds can be confused with one another, and machine, industrial and construction noise can sometimes be misidentified as bird song. False detections of birds that are not really present can be identified and isolated using a combination of confidence scores and ecological information (including the likelihood of presence of a particular species at your site). For more information, contact Second Nature Ecology and Design directly for support.

3.b eDNA monitoring

When animals move around a site, they leave traces of their DNA in the environment, including in water, and on vegetation.

Environmental DNA (eDNA)-based monitoring is a non-destructive (no animals need to be killed) sampling methodology which searches for these traces of wildlife DNA from the environment (soil, water, air, or leaves) and can provide evidence of species presence. This method is especially useful for taxa (a scientifically classified group or entity, such as species or genus) that do not vocalize, are nocturnal and/or cryptic, or that occur in habitats (e.g., water) that are difficult to survey. While eDNA monitoring can be established to cover many different taxa and environmental sources, this manual will focus on two approaches for monitoring species diversity: **1) leaf eDNA to monitor insect biodiversity** and **2) water eDNA to survey general biodiversity.**

It should be noted that any method involving eDNA sampling will require partnership with an (ideally local) DNA laboratory partner whose business model is DNA biodiversity monitoring for clients. One can find a local partner in major cities to avoid challenges with shipping genetic material across borders. To find companies, we recommend beginning with larger global companies using search terms like: commercial DNA monitoring for biodiversity, eDNA biodiversity monitoring, eDNA testing, eDNA sampling, etc.

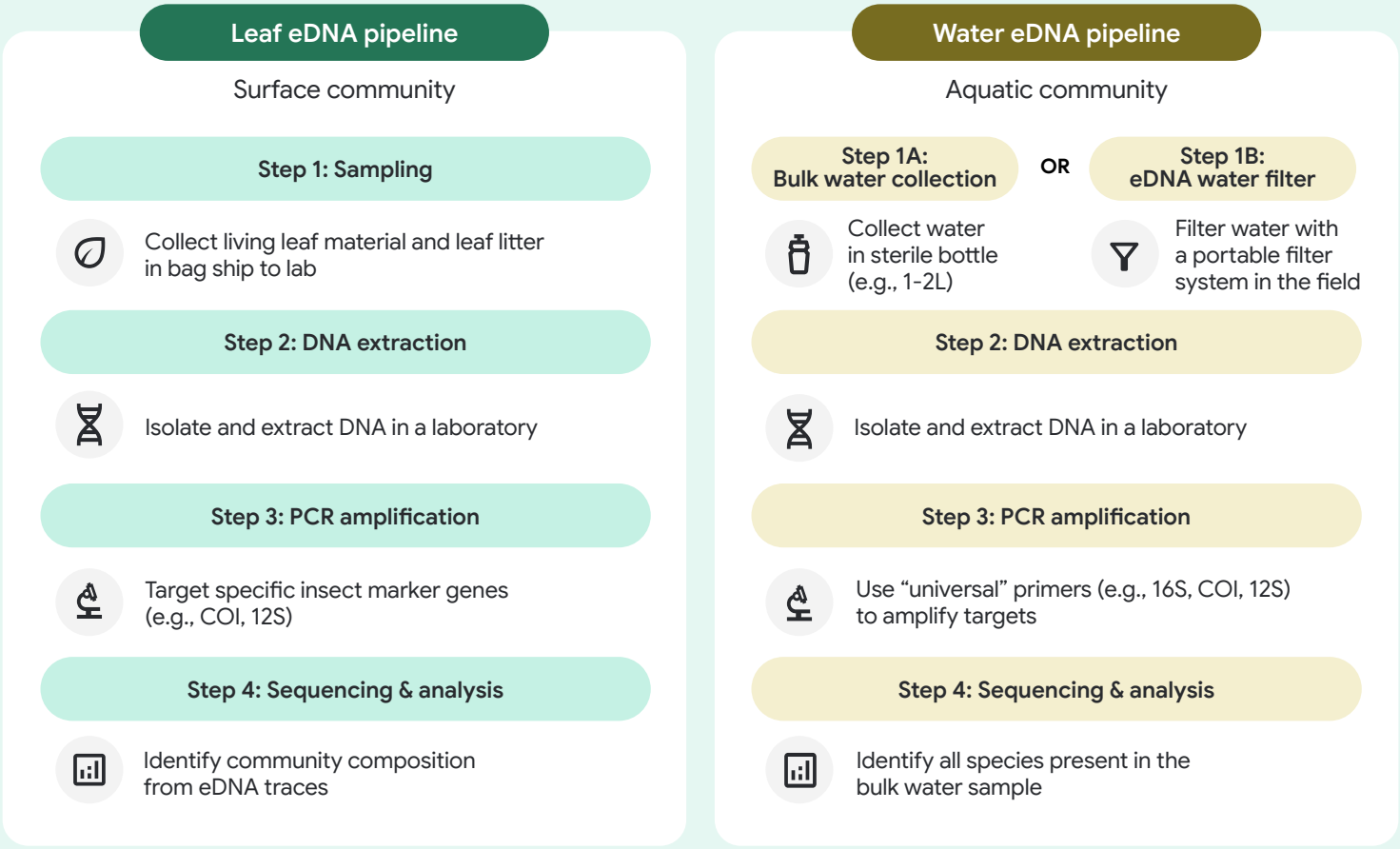


Figure 5. To estimate the biodiversity of insects and animals that live in water, eDNA monitoring is the best option. Leaf insect eDNA pipeline (left) and water eDNA pipeline (right) represent two complementary methods to measure on-site biodiversity.

3.b eDNA monitoring

Insect eDNA from plant surfaces

How it works

Environmental DNA approaches can be used to monitor insects through collection of plant samples in the environment. Insects are valuable additions to any biodiversity monitoring protocol because they: 1) make up a large fraction of the diversity of animals in most locations, 2) are very difficult to visually identify without expertise, 3) provide valuable local ecosystem services such as pollination, 4) respond more rapidly than any other taxon to habitat creation. Insects make up the majority of terrestrial animal biodiversity — globally, an estimated 5.5 million insect species exist, of which roughly 80% remain undescribed, and insects account for over half of all described animal species. Without insect sampling, any biodiversity assessment misses the majority of the on-site animal community.

Insects interact constantly with plants and leave behind trace DNA on leaves, stems, and flowers. By either collecting plant material or swabbing plant surfaces and sending the resulting material to a laboratory for DNA extraction, amplification, sequencing, and bioinformatic matching against reference databases, the arthropod community that has recently visited the vegetation can be reconstructed. The approach is non-destructive — no insects are killed — and a single sample can yield detections across many species and groups of insects simultaneously.

What it measures

The output is a species list of insects that have recently interacted with the sampled plant. For mitigation sites, this is particularly useful for tracking pollinator response to new native plantings, detecting the predatory and parasitoid arthropods that signal a maturing food web, and documenting shifts in the herbivore community between the baseline landscaping and the post-mitigation native planting.

3.b eDNA monitoring

Components

- Sterile sampling materials: nitrile gloves, sterile containers or zip-lock bags, distilled water (for surface washes) or sterile cotton swabs (for surface swabbing)
- Cooler with ice packs for transport to the laboratory or fridge for temporary storage
- A partnership with a local DNA biodiversity monitoring laboratory in the same country where supply chain sites are located that specializes in arthropod metabarcoding from plant material. The laboratory should advise on the preferred sampling protocol (washes vs. swabs vs whole leaves), preservation method, and shipping logistics before any field sampling begins, and will handle DNA extraction, sequencing, and bioinformatic species assignment.

Monitoring Technique

Sampling design and timing

Time sampling to peak insect activity, which usually occurs when the weather is warmest and most humid: spring & summer for temperate areas, the wet season for tropical areas, summer for high latitude regions. Sample foliage in the baseline plantings as well as you can given the landscaping. Collection can include leaves, flowers, leaf litter, and grasses to ensure the highest variety of vegetation and associated insects is sampled. Ideally leaf litter and material on the ground are included in all sampling to ensure ground-based insects are sampled. When possible, try to sample at least some of the same plant species in both baseline and post-mitigation collections, since plant identity strongly influences which arthropods visit. Do not collect samples for at least 2-3 days following a rain storm due to the washing away of DNA traces.

Collection technique

Common collection techniques include surface washes of leaves, surface swabs, and whole leaf collection. We recommend choosing between the main approaches in consultation with the local DNA company. Surface washes involve spraying distilled water across the whole plant body and collecting the runoff in a sterile container placed at the base of the plant — fully non-destructive and well suited to shrubs and herbaceous plants. Surface swabs involve rubbing a sterile cotton swab across leaves, stems, and flowers and then placing the swab into preservative as directed by the lab. Whole leaf collection is the simplest but requires a specific lab protocol allowing for entire pieces of vegetation to just be placed into sterile bags for processing in the lab. In all cases, change gloves and use fresh materials between sites to avoid cross-contamination, and record sampling location, plant species, date, and time.

Data analysis

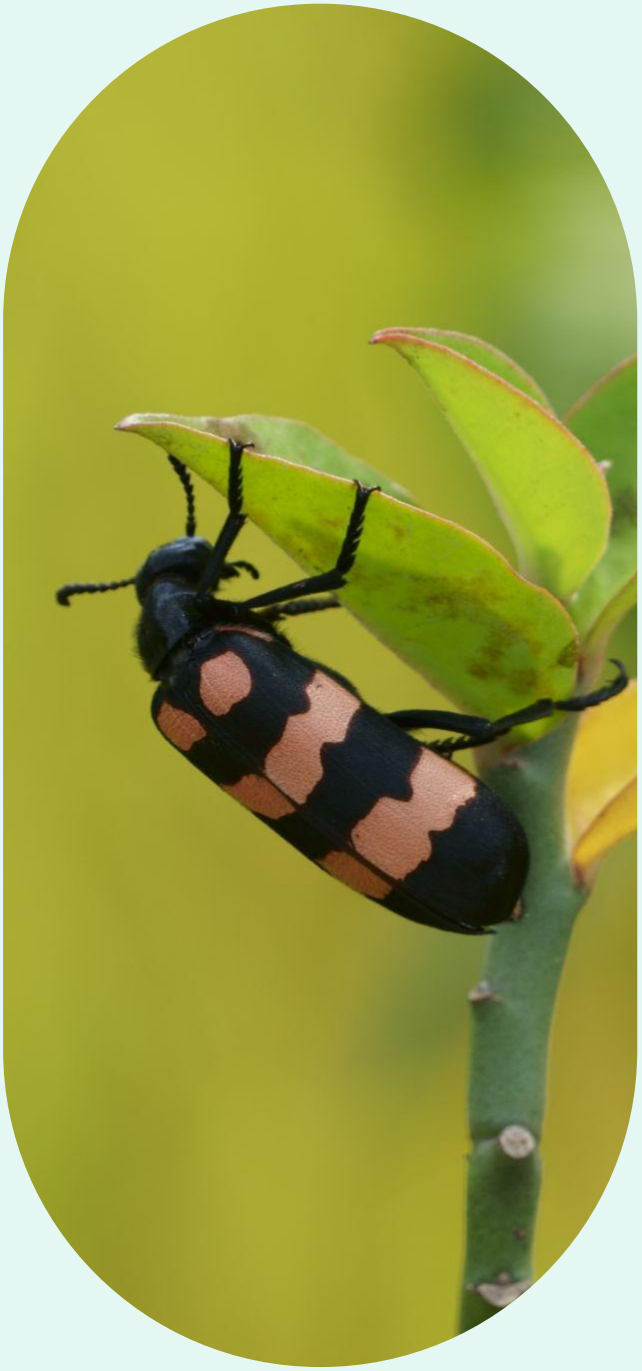
Ship preserved samples to the partner laboratory following their cold shipping and storage requirements. The lab returns a species list with read counts per taxon. Work with an ecologist to interpret the results — flagging likely false detections, separating species responding to interventions detections from common-species background, and comparing community composition between baseline and post-mitigation samples and across years as habitat matures.

3.b eDNA monitoring

Insect trap + DNA monitoring as an alternative

An alternative to using eDNA to monitor for insects is insect trapping DNA monitoring. Insects can be lethally collected in passive traps such as malaise or pitfall traps. Collections from these traps are pooled together into a single collection that can be shipped to a DNA laboratory where extraction and DNA sequencing are conducted. A single trap can capture hundreds to thousands of individuals at once, and a single sequencing run can identify many of them given sufficient reference library coverage. Many traps can be deployed at once, and this method may therefore capture a more representative sample of the insect community at your site than eDNA sampling using leaf collection methods.

While pitfall and malaise traps are standard protocol for insect sampling in research contexts, these methods are both more expensive and more labor intensive. While they are not recommended as part of the core, low-effort stack for beginners, a step-by-step implementation guide is provided in Section 4 (Appendix) for advanced monitoring sites. For more information, contact Second Nature Ecology and Design directly for support.



3.c Water eDNA from aquatic habitats

How it works

Animals that live in water are constantly shedding DNA, and collecting water samples is a simple and efficient way to gather information about aquatic biodiversity using eDNA approaches. Water-based eDNA collection is: 1) highly cost-effective per species detected, 2) captures cryptic and elusive species missed by visual surveys, 3) opens a single sampling channel to multiple wildlife groups (fish, amphibians, mammals, reptiles, birds, and aquatic invertebrates species).

Any animal that drinks at, bathes in, or otherwise interacts with a waterbody continuously sheds DNA into the water. By collecting water samples and either bulk-shipping to a laboratory or filtering on-site through a fine-pore membrane, then extracting, amplifying with broad-range primers, and sequencing the captured DNA against reference databases — the wildlife community that has recently used the water can be reconstructed.

3.c Water eDNA from aquatic habitats

What it measures

Water eDNA signal is strongest for taxa that spend extended time in the water (fish, amphibians, aquatic invertebrates), but reliably extends to terrestrial mammals and birds that visit the water source. This makes water eDNA particularly valuable at sites where a constructed pond, wetland, or retention feature acts as a wildlife magnet.

The output is a species list of taxa that have recently used the sampled water. For mitigation sites with constructed or restored aquatic habitat, this is particularly useful for:

- **Amphibians,** which are cryptic, often nocturnal, and visually active only briefly during breeding — but whose DNA persists in water at detectable levels even when no individuals are observed.
- **Fish,** when there is the belief the pond may be naturally colonized (fish can be ignored if the water is stocked).
- **Mammals** that visit the water — often elusive but reliably DNA-positive after drinking, bathing, or crossing the feature.
- **Birds,** particularly waterfowl and species that drink at the water's edge. Water eDNA for birds complements PAM by detecting species that visit the site without vocalizing.
- **Tracking community change** between baseline and post-mitigation sampling, and across years as the aquatic habitat matures and the species pool develops.

A single water sample, with the right primer set, can produce species lists across multiple taxonomic groups simultaneously — making this method one of the highest information-per-dollar monitoring channels available where suitable aquatic habitat exists on site.

3.c Water eDNA from aquatic habitats

Components

- Sterile sampling materials: nitrile gloves, sterile 1–2 L bottles for bulk water collection, or a portable filtration kit (sterile filter cartridges, syringe or peristaltic pump, sterile tubing) for on-site filtering.
- Cooler with ice packs for transport to the laboratory or fridge for temporary storage. Bulk water samples must be kept cold and processed quickly to limit DNA degradation; preserved filters tolerate transport better.
- A partnership with a local DNA biodiversity monitoring laboratory that specializes in aquatic eDNA metabarcoding. The laboratory should advise on the preferred sampling protocol (bulk water versus on-site filtration), filter type and pore size, preservation method, primer choice, number of replicates, and shipping logistics before any field sampling begins. The laboratory handles filter kit procurement, DNA extraction, sequencing, and bioinformatic species assignment.

Monitoring Technique

Sampling design and timing

Time sampling to peak wildlife activity at the water source — such as the breeding season for amphibians and the spring & summer months in general. Sample the same water body in both baseline (if a pre-construction water feature exists) and post-mitigation monitoring rounds. Collect multiple replicate samples across the waterbody rather than a single grab sample — DNA is not evenly distributed in water, and detection probability rises substantially with the number of replicates and the total volume sampled.

For terrestrial vertebrates that visit briefly (birds, mammals), preferentially sample near the water's edge, where drinking, bathing, and shoreline activity concentrate the DNA signal.

Collection technique

In consultation with the partner laboratory, choose between the main approaches:

- **Bulk water collection** involves filling sterile 1–2 L bottles with water from the target site, transporting cold to the laboratory, and filtering there. This is the simplest approach in the field but heavier to ship and carries higher risk of DNA degradation in transit, particularly in warm climates or where lab turnaround is slow.
- **On-site filtration** involves pumping water through a portable sterile filter cartridge in the field — using a syringe, peristaltic pump, or gravity-fed setup — then transporting only the filter (preserved in ethanol, buffer, or kept cold per the lab's specification) to the laboratory. The field equipment requirement is higher and must be supplied by the laboratory, but DNA is preserved at the moment of capture and shipping is far lighter. This is the preferred approach for remote sites or where shipping logistics are difficult.

In all cases, change gloves and use fresh sampling materials between sample locations to avoid cross-contamination, and record sampling location, water source characteristics, weather, time, water depth and temperature if measurable, and any wildlife observed at the time of sampling.

3.c Water eDNA from aquatic habitats

Data analysis

Ship preserved water samples or filters to the partner laboratory following their cold-shipping and storage requirements. The lab returns a species list with read counts per taxon. Work with an ecologist to interpret the results — flagging likely false positives (especially from contamination, domestic-animal DNA in agricultural settings, or upstream sources in flowing water), separating focal-species detections from common-species background, and comparing community composition between baseline and post-mitigation samples (where appropriate) and across years as the aquatic habitat matures. As with all eDNA methods, detection of a species confirms presence but absence of detection does not confirm absence — multiple replicates and repeated sampling over time strengthen confidence in community-level inferences.

3.d Camera traps

How it works

Camera traps are weatherproof, battery-powered cameras triggered by a passive infrared (PIR) sensor that detects differences in heat and motion. Animals walking in front of the camera will trigger the motion sensor, and the resulting images can be processed to provide information on species presence, including time and date of detection, and to build an overall understanding of the wildlife community.

Once triggered, the camera captures a still image, a burst of stills, or a short video clip, each stamped with date and time. Because cameras operate continuously (day and night) and require only periodic servicing for fresh batteries or SD cards, they can generate large volumes of data much more easily and cheaply than other methods such as live-trapping. Additionally, camera traps offer a non-invasive way to capture information related to biodiversity with essentially no disturbance to wildlife. Depending on the battery and capacity of the SD card, camera traps can be left to run for months before servicing is required.

What it measures

Camera traps are most reliable for detecting mammals, which travel along the ground and tend to follow game trails that put them directly in the camera's path. While camera traps can capture other wildlife such as birds, this data isn't reliable for comparisons, since non-mammalian species move in ways that cameras positioned along trails or fence lines won't consistently detect.

Camera placement

The data produced by camera traps is highly dependent on camera placement. Ideal placement is along movement corridors for mammals such as game trails, fence lines, gaps or holes in fencing, and dried riverbeds. Poor placement, for example, in a grassy field or facing branches that sway in the wind, will produce large volumes of empty imagery and use up SD storage that could otherwise capture species passing in front of the camera. Camera traps are also less likely to detect species that aren't warm-blooded, such as reptiles and amphibians, or that are too small to trigger the sensor, such as insects; though rodents can be captured depending on placement. As a result, the species list is generally biased toward mammals.

Components

A weatherproof PIR-triggered camera trap with infrared night illumination (commonly used models include the Bushnell, Reconyx, or Browning), a high-capacity SD card (32–128 GB is typical), and a fresh set of batteries (rechargeable lithium or alkaline AA, depending on the model). For publicly accessible sites, a lock box that protects the camera along with a python lock or steel security cable is recommended to deter theft.

3.d Camera traps

Installation Guide

01. Setup and selecting locations

Mount each camera between ~0.6-1.3 m high for small-to-medium-sized mammals, with the camera pointed level rather than angled down. Angled down may be appropriate if the camera must be placed higher to ensure it captures trail activity. Avoid facing the camera toward direct sun (east/west) or toward branches and grasses that move in the wind, as both will produce false triggers. If vegetation crowds the line of sight, clear low vegetation in front of the camera if appropriate; the goal is to reduce false triggers. When choosing a spot within the site, select features animals actually use, such as game trails, water sources, and fence lines and gaps, rather than open mid-field placements. Do not select a site frequented by workers (e.g., walkways, break areas, entry points, etc.).

3.d Camera traps

Installation Guide

Configure each camera before deployment, and use the same settings across all cameras at the site so detections are comparable. Set the camera's internal clock to the correct date and time so every image is properly stamped. Decide between single photos and burst mode: burst is useful for fast-moving species but fills the SD card faster, increasing service frequency. Set the cool-down period between triggers (e.g., 1–10 seconds) and trigger sensitivity (low, medium, or high). To maximize detections, use a short cool-down and high sensitivity, accepting the trade-off of higher storage use and battery drain. After mounting, crouch down and walk past the camera a few times, then pull the SD card and review the test images. Ideally, your entire body should be in frame if crouched. Use these images to calibrate the camera's placement and angle so that mammals at your site (e.g., foxes) will be captured as they move through. If the camera fails to trigger, raise the sensitivity and re-test.

Record each camera's ID, GPS coordinates, mount height, orientation, surrounding habitat, and deployment date/time. At each service visit, log the date, who serviced the camera, battery and SD card replacements, and any issues observed. Bring spare batteries and a fresh SD card to every visit. Generally, cameras should be left out for 30-45 days per season to capture a full picture of the wildlife, namely mammals, using the site.

Figure 6. Example deployment of a camera trap mounted on a tree facing a game trail (photo credit: Cesar Estien).

3.d Camera traps

Installation Guide

02. **Data retrieval**

Retrieve SD cards and replace batteries on a schedule matched to site activity. High-activity sites may need servicing every 4 weeks to keep cards from filling and batteries from draining, while low-activity sites can often go 8 weeks or longer between visits. Bring an empty SD card and a full set of batteries to every visit so the camera is never offline; the SD card pulled from the camera trap should be backed up to a hard drive before processing.

AI workflow for processing camera trap imagery

One challenge with camera trap methods for monitoring wildlife data is the volume of images produced. In addition to wildlife of interest, camera traps can also capture images of plants moving in the background, people walking past the camera, and common animals such as dogs and cats that are not the main focus of study. In addition, animals captured in camera traps must be identified as species, which can be challenging for non-experts in biodiverse environments. To overcome this challenge, a variety of AI tools are available that can separate non-animal images from animals, and can reliably identify animals in images as species. Below is a brief list of some of the available tools, as well as a suggested workflow for image processing.

The tools and what they do

- MegaDetector: This tool looks at a camera trap image and finds where the animals (or people/vehicles) are in the frame, but doesn't identify species. It's trusted for filtering out empty images that can make up the majority of camera trap data.
- SpeciesNet: This is an open-source classifier created by Google that identifies what species an animal is. It depends on MegaDetector to first locate the animal pixels, then classifies just that crop.
- AddaxAI: A free, open-source desktop GUI that runs MegaDetector and SpeciesNet locally on a batch of images (no cloud upload required). It can also blur people in images.
- Timelapse: A free Windows tool for reviewing camera trap images that enables a user to quickly sort and filter a large batch of images already processed by AddaxAI.

3.d Camera traps

How tools knit together in a typical workflow:

Once images are downloaded from an SD card on a camera trap, they can be run through the following workflow to separate non-animals from animals and to identify animals to species:

1. Run the folder through **AddaxAI**, which runs **MegaDetector** to find animals/people/vehicles and then **SpeciesNet** to classify the animals, producing a JSON results file.
2. Load images and the JSON file from step 1 into **Timelapse** for human review, where you can bulk-clear blanks and bulk-remove people. Note that while SpeciesNet has high accuracy for detecting and identifying common species, a small portion of images may still require expert verification to confirm the identification. These images are cases of species that may be confused with one another.
3. Export a CSV from Timelapse with your data, including the time, date, and species for every animal that walked past your camera trap.

3.e Measuring temperature ROI

While biodiversity monitoring captures how species respond to habitat improvements, abiotic monitoring (measurements of the physical environment, such as temperature and moisture) captures the underlying conditions that make those biodiversity gains possible.

For supply chain sites implementing mitigation actions such as opening sealed surfaces, installing native vegetation, or adding water features, temperature measurements often respond faster than biodiversity does. A newly planted native bed may not attract its full bird community for one or two growing seasons, but the cooling and humidifying effect from its canopy and shade can be measured within weeks. Temperature data therefore provides early evidence that an intervention is working, and gives site managers a quantifiable, repeatable way to communicate impact to internal and external stakeholders even before biodiversity responses are fully expressed.

How it works

Temperature and humidity loggers are small, battery-powered, weatherproof devices that record ambient air temperature at fixed intervals. Once deployed and configured, they run unattended for months, storing thousands of readings in onboard memory. Data is retrieved wirelessly via Bluetooth to a smartphone or tablet, where it can be exported as CSV files for analysis. By comparing readings from loggers placed in mitigation areas (e.g., over native plantings, near depaved surfaces, or beneath new tree canopy) against control loggers placed in untreated areas (e.g., open parking lots, mown lawn), site managers can quantify the cooling effect of their interventions.

3.e Measuring temperature ROI

What it measures

The primary measurements are air temperature and relative humidity. From these two values, one can also calculate a further variable of ecological and human-comfort relevance: **heat index** (a combined metric of how hot conditions actually feel to organisms). Together, these capture the full microclimate signal of a biodiversity intervention more completely than temperature alone.

The most useful metrics for evaluating mitigation ROI are:

- **Daytime peak temperature:** the strongest signal of cooling from shade, vegetation, and surface depaving.
- **Daytime mean relative humidity:** vegetated areas, particularly those with established native plantings and trees, raise humidity through evapotranspiration. This effect is often as strong or stronger than the temperature signal on hot, dry days.
- **Heat-index hours:** number of hours above a chosen threshold (e.g., 30 °C apparent temperature), which integrates both measurements and tracks worker comfort and ecological stress in a single number.
- **Diurnal range:** vegetated and infiltrating surfaces tend to dampen the swing between day and night temperatures, an effect often clearer than mean temperature alone.

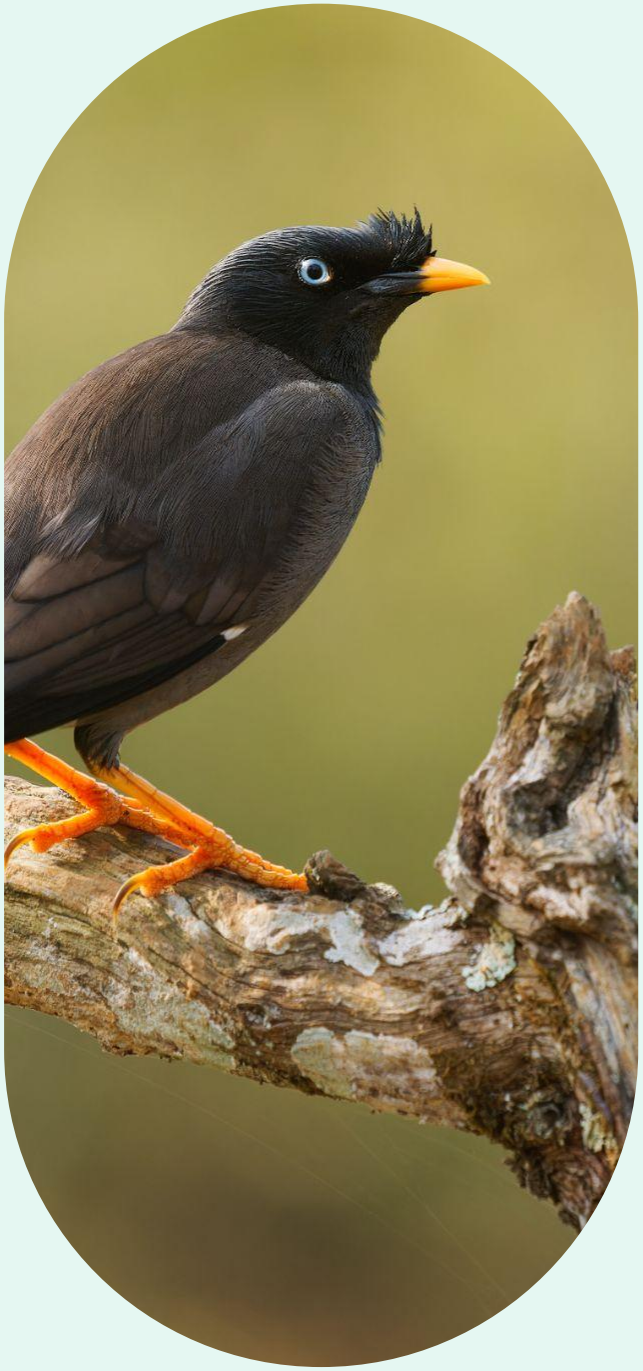
Temperature differentials of 2–10 °C and humidity differentials of 5–20% RH between paved and vegetated surfaces are commonly reported in the urban heat island literature.

3.e Measuring temperature ROI

Components

While many such systems exist, we have used the Kestrel DROP Wireless Temperature Monitor and Data Logger. At a minimum, a site should use a system that measures temperature such as the DROP D1. These devices are compact and weatherproof running on batteries and can be mounted to trees, posts, fences, or structures. They use a precision thermistor with ±0.5 °C accuracy. Using their app, which connects via Bluetooth within a ~30 m (100 ft) line-of-sight range and can manage up to 8 loggers simultaneously and download data.

For sites that also want to track humidity, you can use the DROP D2 (temperature + humidity) with no change to deployment workflow.



3.e Measuring temperature ROI

Installation Guide

01. Setup and selecting locations

Set the logging interval to be frequent enough for daytime peaks and nighttime lows (the periods most diagnostic of cooling effects) while conserving battery life. A robust monitoring design requires at least one logger in each treated area (e.g., a depaved patch, a new native planting, a tree canopy zone) and at least one matched control logger in a comparable but untreated area (e.g., adjacent parking lot, mown turf). To isolate the effect of the mitigation itself, all loggers should be mounted at the same height (typically 1.5–2 m), exposed to the same general sky conditions, and placed away from confounders such as building exhausts, AC condensers, or paved walls that radiate heat. Critically, the control logger placed in a paved area such as a parking lot must not be located beneath any artificial shade structure, as this would mute the very contrast the study is designed to detect.

02. Data analysis

Download the data via your logger's defined methodology (Kestrel users will use the Kestrel LINK app). Logger outputs are generally easily compared in spreadsheets or statistical software. Work with an ecologist or statistician to create accurate comparisons of treatment and control loggers, looking at measures such as mean daytime temperature, mean nighttime temperature, daily maximum, and number of hours above defined heat thresholds. For multi-year monitoring, the same metrics can be tracked over time to show that the effect strengthens as vegetation matures.

Figure 7. Example deployment of a DROP D1 mounted on a stake in a native planting (left) and on a parking-lot fence post as a control (right), at matched height.

04 Appendix: additional monitoring options

2

3

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4. Appendix: Insect trapping with DNA metabarcoding

Why use it

The two approaches to insect DNA monitoring answer slightly different questions and complement each other well. **Leaf-surface eDNA** is non-destructive (no insects are killed), low-effort in the field, lower effort for the staff conducting the sampling. However, there is a higher risk of false positives, and it is less applicable for sites with no baseline vegetation, and the detection window is short — DNA on leaves degrades within days under sun and rain.

Trapping with metabarcoding, by contrast, is destructive but captures a much broader and more quantitative slice of the community (more likely to capture ground insects). However, this comes at a higher cost in terms of labor and significant effort. Furthermore, while shipping plant materials internationally is difficult, shipping insects is almost impossible, thus mandating initial lab work be done by a local lab partner. For most mitigation sites, the strongest design is to use **both**. Sites with tighter budgets or non-lethal-sampling requirements may wish to start with leaf-surface eDNA alone; sites prioritizing the most complete possible inventory should start with trapping.

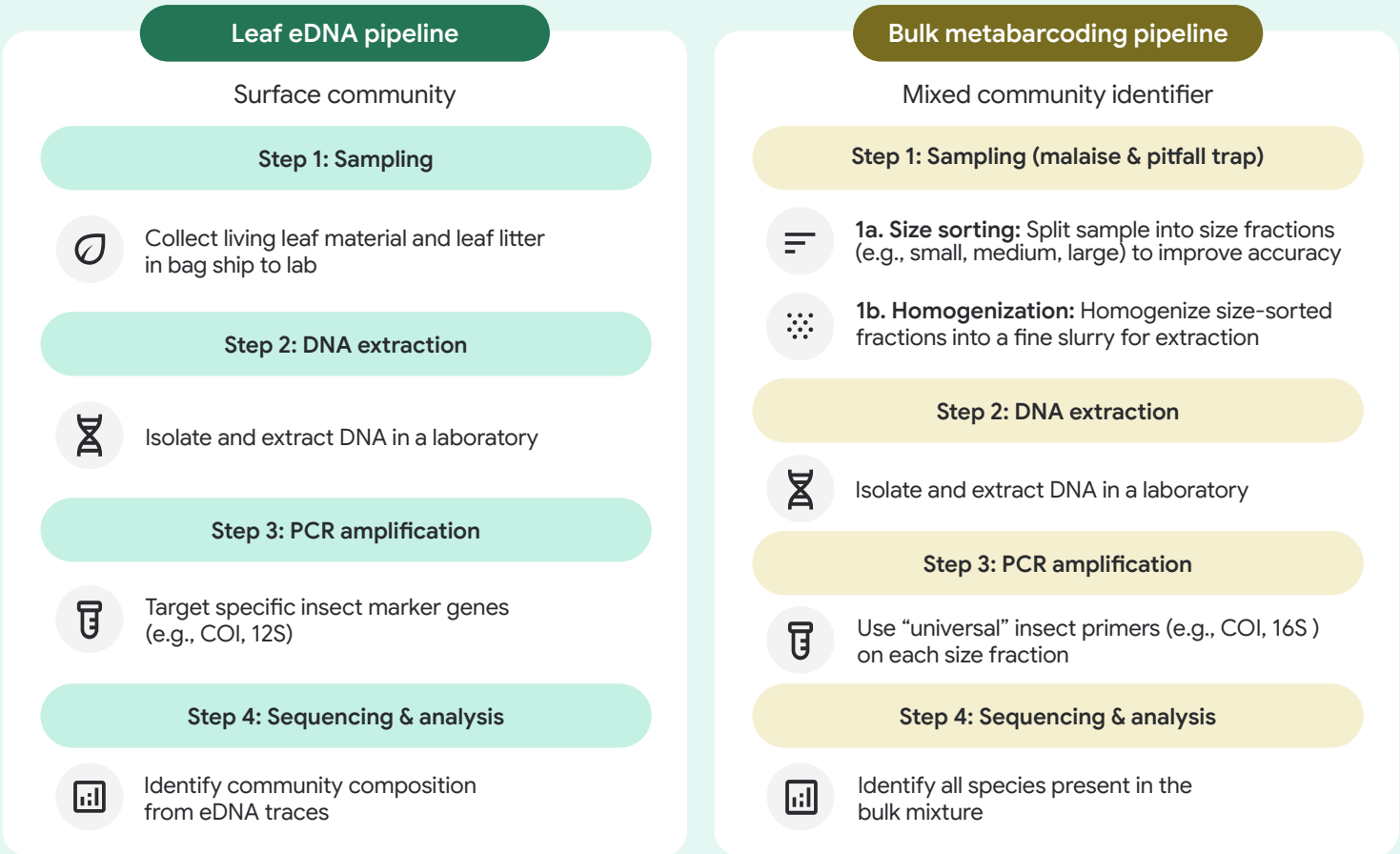

Figure 8. To estimate insect biodiversity ROI between baseline and post-mitigation DNA monitoring is the best bet. Leaf eDNA pipeline (left) and trap collection metabarcoding (right) represent two alternative methods to measure insect biodiversity.

4. Appendix: Insect trapping with DNA metabarcoding

How it works

Bulk insect samples are collected using standardized traps and shipped/picked up as a whole sample to a DNA laboratory. There, the entire sample is homogenized before following the same extraction and amplification protocol as with eDNA. A single trap can capture hundreds to thousands of individuals at once, and a single sequencing run can identify many of them given sufficient reference library coverage.

What it measures

Trapping with metabarcoding is currently the most comprehensive way to characterize the insect community at a site. Output is a species (or genus) list with read counts per taxon, from which species richness, presence of focal species, and changes in community composition over time can be derived.

4. Appendix: Insect trapping with DNA metabarcoding

Components

Two trap types are considered the standard:

1. Pitfall traps
Small containers (typically plastic cups 8–10 cm in diameter) buried flush with the ground and filled approximately one-third with preservative. Best for ground-active arthropods, including beetles and spiders.

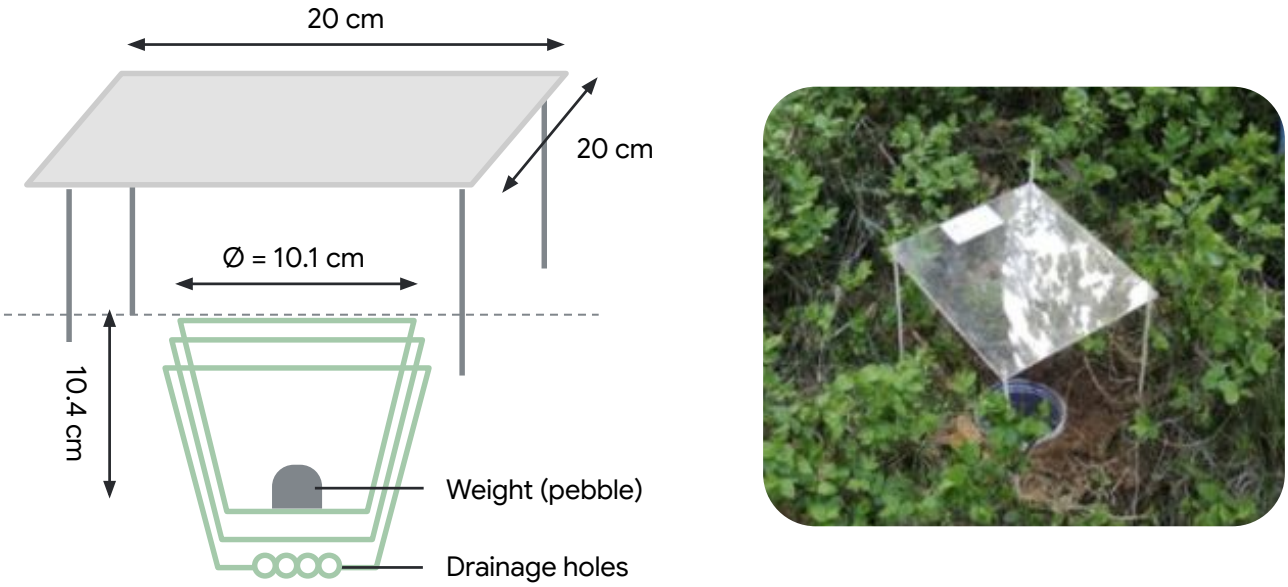

Figure 9a. Pitfall trap (credit Pereira et al., 2024) out in the field.

4. Appendix: Insect trapping with DNA metabarcoding

Components

2. Malaise traps

Tent-like structures of fine mesh designed to intercept flying insects. Insects strike the mesh, fly or climb upward, and are funneled into a collecting head containing preservative ethanol. Best for flying insects including flies, butterflies, and bees.

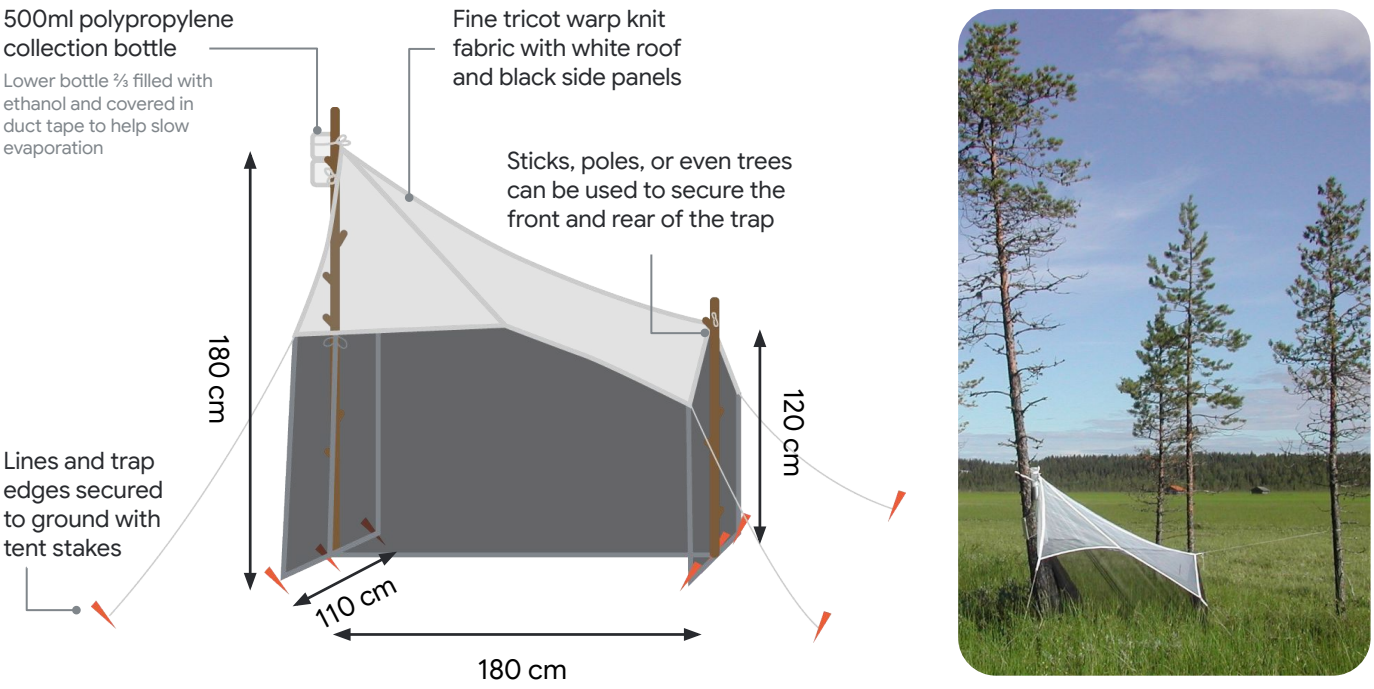

Figure 9b. Malaise trap (credit Karlsson et al. 2020) out in the field.

A partnership with a local DNA biodiversity monitoring laboratory is essential. The laboratory should advise on the preferred preservative, trap type, sampling duration, and shipping protocol before deployment, and will handle extraction, sequencing, and bioinformatic assignment.

4. Appendix: Insect trapping with DNA metabarcoding

Installation Guide

Trap general locations	During baseline sampling, traps should be set up to collect insects in the landscaped areas where mitigation will take place, or in the landscaped areas which are as close as possible. Do not set up traps on sealed surfaces.
Pitfall trap deployment	Traps should be placed throughout the habitat, beneath bushes/trees, along vegetation edges, and in more open bare ground to capture the widest variety of insects. Bury cups so the rim sits flush with the soil surface — any protruding lip will deter ground-active arthropods. Fill each cup to approximately one-third depth with the preservative specified by the laboratory (ethanol or, where vertebrate bycatch is a concern, a propylene-glycol/water mix, which is less attractive to small mammals and amphibians). Place a small raised rain cover roughly 5 cm above each trap on short stakes or wire to exclude rainfall and falling leaves. Leave deployed for 1–2 weeks, checking regularly to ensure cups do not overfill, then retrieve the contents into a labeled vessel for shipment.
Malaise trap deployment	Orient the trap perpendicular to expected insect flight paths — typically along habitat edges, between two contrasting vegetation types, or across a clearing within taller vegetation. Insects flying horizontally strike the mesh wall and fly upward toward the lit collecting head. The collecting bottle should contain high-percentage (≥95%) ethanol, or whatever the partner laboratory specifies. Deploy each trap for a standardized interval (typically 1–2 weeks), then decant the ethanol with the captured insects into a labelled vessel for shipment and replace with fresh ethanol if continuing to sample.
Data analysis	Ship preserved bulk samples to the local partner laboratory or have them pick them up following their requirements. The lab will return a species list with read counts per taxon. Work with an ecologist to compare species richness, focal-species presence, and community composition between baseline and post-mitigation samples and across years as habitat matures.

References
Wood, C.M., Kahl, S. 2024. Guidelines for appropriate use of BirdNET scores and other detector outputs. Journal of Ornithology. 165:777-782.

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A Manual for Biodiversity Monitoring

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