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Farmland Biodiversity, Soil Health & Sustainability Assessment

A handbook of simple, on-farm indicators

Cathy Hawes, 2011
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The aim of this handbook is to describe indicators used to monitor the impact of agroecological management practices. These indicators are applied at the Hutton's Centre for Sustainable Cropping (CSC): a long-term research platform established in 2009 to design and implement an integrated, regenerative, agroecological cropping system and monitor the impact on biodiversity, soil quality, environmental footprint and financial returns.

For further information, visit https:\\csc.hutton.ac.uk

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Goals: delivering multiple benefits from farming

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Challenges

Agriculture is facing major challenges and multiple conflicting goals: meeting demands for food security, mitigating and adapting to climate change, reversing the loss of biodiversity caused by past agricultural intensification.

Farmers need new ways to manage land that will maximise productivity and efficiency while at the same time achieving sustainability and healthy environments in the long-term.

Opportunities

Concerns over agricultural intensification resulted in a trend towards organic production. However, efforts to balance biodiversity and production in these extensive systems exacerbates food security issues, especially where available land is limited.

Biodiversity-based farming presents a potential solution, where ecosystem services compensate for reductions in agrochemical inputs to maintain high yields. This increases efficiency and builds resilience into agroecological systems.

Transition towards agroecological production is a process of evidence-based learning and iterative improvement:

1. Define your end goals and select management options to meet them (see companion handbook "Agronomic practices for sustainable crop production").

2. Take baseline measures of indicators as described here.

3. Implement new management options and monitor the effect on indicators to identify whether targets are met.

4. Revise your management accordingly.

On-farm data can be entered directly into the Hutton CSC Toolkit via phone app or web site. This will provide feedback on your farm indicators relative to national averages and an assessment of overall sustainability.

Goals for management

- to produce high quality food and maintain yields, with
- less reliance on agrochemical inputs, using
- ecosystem services provided by arable biodiversity, creating
- optimised systems with minimal losses and therefore
- low environmental impact.

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Balancing biodiversity and crop production at the Hutton's Centre for Sustainable Cropping platform, Balruddery Farm, Scotland

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Baselining, indicators and why we need them

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Baselining

There is no absolute, quantifiable target for the many facets of agricultural sustainability. Rather, the goal is to improve on current states by enhancing biodiversity, soil health and system resilience.

To achieve this, we need to be able to define what the current state of a system is so that improvements can be quantified. This requires reliable baseline data to be gathered against which to benchmark future trends and assess progress towards the end goals.

Indicators for monitoring change

Our main objective is to achieve multiple environmental and ecological benefits from farming while maintaining productivity. To achieve this, we adopt a holistic approach, where trade-offs between different system components can be optimised.

Since it is impossible to measure the whole ecosystem, we need to identify a suite of easy to monitor, reliable and well understood indicators for each component: biodiversity, soil quality, management intensity and economics (financial margins)

This handbook describes the indicators of biodiversity and soil quality applied at the Hutton's Centre for Sustainable Cropping. A brief rationale is given for each, along with generic methods for sampling that can be applied in any arable field environment. Some methods require specialist techniques and equipment, but where possible, protocols have been adapted to provide reliable data without the need for extensive investment other than time.

Trends are outlined for each indicator, based on review and original research carried out at the Hutton (1999 - 2022). Finally, a framework is presented that combines these indicator data with farm records on agronomic inputs, crop yields and sale prices to give an overall assessment of sustainability at the farm scale.

The CSC Toolkit

- Select appropriate and realistic goals for management
- Identify measurable indicators for each component
- Measure the baseline state for each indicator
- Implement management practices to meet goals
- Monitor progress against your baseline
- Review results and adjust management accordingly

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Developing systems indicators for monitoring: from high tech drone imaging for crop health, to phone apps for measuring soil structure, and simple quadrat counts of weed biodiversity in an arable field.

Farmland habitats and crop diversification

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Background

Semi-natural habitats in agricultural land provide essential ecosystem services at the farm and landscape scales, as well as having high conservation value in their own right.

Loss of habitat is a major factor driving declines in farmland biodiversity and can be rectified by habitat creation, including new field margins, beetle banks, uncropped or conservation headlands, hedges and tree lines and riparian buffers.

Spatial heterogeneity in the distribution of habitats over the landscape is key to success and can be achieved through smaller field sizes, greater crop diversity and maximising the connectivity between habitat patches.

Together this contributes to system resilience and helps mitigate climate change by sequestration of carbon in areas of undisturbed soil.

Indicators

Semi-natural habitats

IACS data, farm maps and apps can be used as a source of data for a range of farm-wide diversity indicators:

Habitat richness - is the number of different semi-natural habitats (woodland, wetland, unimproved grassland etc.)

Habitat diversity - the proportion of the total farm covered by each habitat, used to generate diversity indices based on habitat richness and area.

Habitat connectivity - the inverse of the number of isolated patches of semi-natural habitat (those not connected to others, e.g., by hedgerow or tree line).

Cropping diversity

Habitat heterogeneity includes the fields themselves as the most dominate land use type. At the whole farm-scale, indicators include:

- average field size; number of different crops in rotation; unharvested crops (winter cover crops, bird cover)

and within field-scale measures:

- number of fields with companion crops, intercrops and cultivar mixes grown together within a cropping season.

Results

- Habitat heterogeneity provides the potential for coexistence between many different farmland species.
- Spatial scale/extent of habitat distribution needs to match species dispersal ability (e.g., bats vs. mice vs. microbes).
- Landscape connectivity across habitats beyond the farm boundary must be considered for on-farm management strategies to be most effective.

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Tree lines, field margins and diversified cropping at the Centre for Sustainable Cropping

CSC at Hutton's Balruddery Farm, near Dundee, Scotland

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Monitoring: field margin vegetation

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Background

Much of the arable plant diversity on a farm is found in the first metre of the crop edge. These field margins provide resource for beneficial insects (bees, butterflies and predators), small mammals and birds.

Habitat loss, fragmentation and intensive agriculture has had a major impact on the beneficial insects that rely on these resources, with consequences not only for the productivity and quality of agricultural crops but also for population sizes of wild plant species.

Increased availability of plant resources (shelter, flowers and seeds) within field margins helps support functionally diverse arable food webs.

Measurements

The set of measurements taken in field margins is designed to quantify both the diversity of field margin vegetation and its resource value for invertebrates and higher trophic groups.

Field margins are strips of perennial vegetation between the field boundary (wall, hedge, fence) and the cropped area of the field. These may be sown wildflower mixes, naturally regenerated plant communities or grass.

Margin area – width and length relative to field size (from maps, also part of the Habitat and Diversification section). Important for connectivity between semi-natural habitats, to reduce losses of pollutants and soil from fields and to enable movement of beneficial organisms between the margin and cropped area.

Structure – heterogeneity within a margin in terms of vegetation height, patches of different vegetation types and areas of bare ground. This structure is important for niche differentiation between organisms allowing similar species to coexist in the same location.

Composition – proportion of grasses to broadleaved species; functional types of plants (flower shapes for a diversity of pollinators); sequence of flowering through the year for extended seasonal resource availability.

Findings

- Margins reduce losses of soil and nutrients from cropped fields.
- Diverse field margins support insect food webs and have lower greenhouse gas emissions relative to standard grass margins.
- Impact of margins on the cropped field depends on dispersal ability: pollinators/predators > earthworms > mites/microbes.

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Engineered riparian buffers for multiple benefits: integrating Nature-Based Solutions at Balruddery Farm for flood risk management, soil quality and biodiversity.

CSC at Hutton's Balruddery Farm, near Dundee, Scotland

Field Margin Vegetation Survey

Version 1.0, 17 March 2022, developed from Farm-Scale Evaluations Roy et al. (2003)

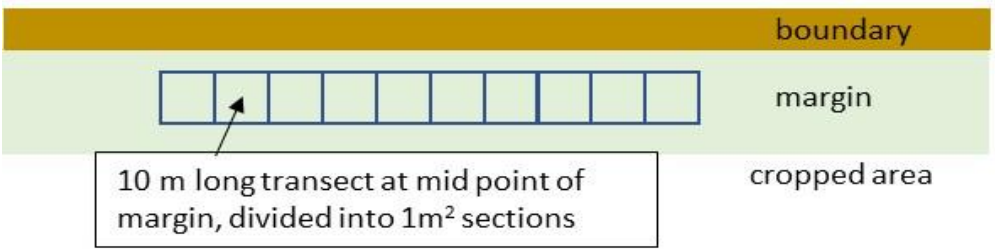
Equipment: 10 m tape marked at 1 m intervals, metre rule, record sheet, plastic sample bags and marker pen (for unknown plant IDs).

Timing: Survey 3 times a year to capture seasonal resource availability for beneficial insects and higher trophic groups. Timing to coincide with pollinator surveys where possible: May (flowering frequency only), June/July (flower, seed and cover assessments), Aug (flowering, seeding frequency only). NB cover assessments only need be done once in the season.

Location: Carry out surveys on two margins per field, selecting areas with a margin type (grass, wildflower etc.) that is representative of the field but with two contrasting aspects.

Procedure:

1. Record date and surveyor initials on the record sheet.
2. At each margin, identify a point at least 10 m from a field corner or gateway and lay the 10 m transect tape along the middle point of the margin:



3. Place the meter rule perpendicular to the transect tape to demarcate the first 1m section.
4. Within this section, estimate % cover of all plant species present (use morpho species or functional types if necessary). The total % can add up to more than 100.
5. Repeat for all 10 sections along the transect. Do steps 4 and 5 at the June/July survey only.
6. For flowering species, record how many of the 1m sections that each species in flower is found in. This gives flowering frequency per species out of a maximum of 10. Do this on all three survey visits.
7. Repeat for all species in seed to give seeding frequency per species out of a maximum of 10. Do this on the June/July and Aug survey visits.

Field Margin Plant Functional Types

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A. Compositae (daisies) 	B. Legume (vetch, clover)   
C. Umbellifer (carrots) 	D. Brassica (wild radish)  
E. Tiny or closed flrs 	F. Thistle-like  
G. Tubular/bell-shaped  	H. Open flowers  

Monitoring: arable weed flora

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Background

In-field arable weeds are an important resource for insects, supporting up to ten times more herbivores than the same mass of crop. Diversity in this weed layer is propagated through the food web to pollinators and natural enemies which then regulate crop pest numbers.

Weeds also deliver other ecosystem services: ground cover to reduce erosion losses and diverse carbon inputs to the soil, which improves organic matter quality for soil invertebrates and micro-organisms.

Tolerance of some weed cover within fields is therefore essential for the maintenance of within-field functions.

The challenge is to define the optimal biomass and composition of the weed flora that supports a healthy agroecosystem but without detrimental impact on crop yield or product quality.

Measurements

Weed distribution in fields is highly variable due to localised seed rain and limited dispersal away from the parent plant. The resulting heterogeneity requires systematic monitoring to ensure an accurate picture of overall diversity and abundance.

Quadrat counts of weed numbers for each species present are made over about 20 sample locations per field. Unless the whole field is to be mapped, sampling should follow a W-pattern to ensure a spread of points across headland and in-field areas.

Surveys are carried out in spring, summer and winter to capture species with different seasonal germination and flowering times.

These data are used to estimate species composition and can be converted into measures of functional diversity using classifications of species based on functional traits.

The target is to maintain ca.10% cover of beneficial broad-leaved species (e.g. *Viola, Veronica),* that have high wildlife value (insect pollinated, large seeds), but low competitive index (low growing, shade tolerant, limited seed dispersal).

Such precision management of weed species assemblages is a major challenge in agriculture today.

Findings

- Emerged weed species composition in arable fields depends on crop type (cereal or break) and season (autumn or winter).
- Build-up of competitive species can be managed through cover crops and rotation.
- Selection and maintenance of low levels of beneficial weeds to balance biodiversity and productivity will require novel precision technologies in the future.

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In-field Weed Biodiversity Survey

Version 1.0, 17 March 2022, developed from Farm-Scale Evaluations Heard et al. (2003)

Equipment: 50 x 50 cm quadrat, camera, record sheet, labels

Timing: Survey 2-3 times a year to capture seasonal resource availability for beneficial insects and higher trophic groups.

- spring (May, approx. 3 weeks after spring crops sown)
- summer (July, approx. 2 weeks before cereal harvests)
- autumn (Oct, at least 3 weeks after autumn sowing)

Location: Carry out assessments at a about 20 locations in each field/treatment ensuring the whole field is covered either in a W pattern or along 3 equidistant transects

Procedure:

1. Record date and surveyor initials on the record sheet.
2. At each field, note cultivation state (plough, stubble, sown, crop).
3. Place a 50 x 50 cm quadrat at each sample location.
4. Within the quadrat, estimate % cover of crop, broad leaved weeds and grass weeds. The total % can add up to more than 100.
5. Count the total number of broad leaved weeds present for all species.
6. Record whether a species is in flower by an * next to the number entered for that species on the record form.
7. Place a label with the GPS location and date clearly visible next to the quadrat and take a photo from directly above so that the quadrat (plus label) fills the frame.

Monitoring: arable weed seedbanks

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Background

Unlike the emerged weed flora in annually cropped fields, the soil seedbank provides a buffer against annual changes in weather or cropping practices. This buffering capacity is due to the persistence of seeds over long (decadal) periods in the soil and confers resilience and opportunity for arable systems to recover from the negative impacts of intensification.

Taxonomic and functional diversity of the seedbank can be used as an indicator of long-term transitions, both negative, as losses of biodiversity resulting from intensive herbicide use, and positive, where beneficial species are left to return seed to the soil. Increased dominance of beneficial species has an antagonistic effect on noxious weeds and can reduce dominance of competitive weeds in the long term.

Measurements

Aim – to determine the taxonomic and functional reserve of plant biodiversity held in the soil seedbank as an indicator of previous cropping and management history and the potential for future recovery.

Sampling – as with above-ground vegetation, the weed seedbank is spatially variable and soil samples must therefore be taken either across the whole field (e.g. in a grid pattern), or along a W transect covering headland and in-field areas, or stratified according to vegetation and soil properties where field maps are available.

Assessment – numbers of seeds for each species present are either counted directly following extraction of seeds from soil by sieving, or through counts of weed seedlings emerging from the soil following germination in a glasshouse environment. The latter method is used to give a more direct measure of number of viable seeds in the species assemblage.

Data – species composition can be used raw or converted into functional types based on species traits. Multivariate analysis at both levels of aggregation give good indicators of previous field histories, management intensity and potential future in-field biodiversity.

Findings

- Seedbank composition is resilient to change; fields have a characteristic ‘identity’ that persists over time.
- Seed abundance however responds rapidly, declining up to 50% a year if intensive management prevents re-seeding.
- Seedbanks provide the basis for above-ground biodiversity in annually disturbed systems.

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Soil sampling at the CSC for assessments of arable weed seedbank diversity by measurements of weed species emergence in the glasshouse.

Seedbank Emergence Assessment

Version 1.0, 17 March 2022, developed from the Farm-Scale Evaluations of GMHT crops

Equipment: 20 x 20 cm quadrat, trowel, labelled bags, 2 litre plant pot

Timing: Soil to be collected mid-March, before spring crops are sown

Location: Carry out assessments at a minimum of 20 locations in each half field, in a W shape to cover the whole field, or a grid (if large numbers of samples can be collected), or for example, 21 samples equidistantly spaced across 3 tramlines over the full field length.

Procedure:

1. At each sample location, remove any surface debris, dig a 20 x 20cm pit to 20 cm depth and mix the soil thoroughly
2. Fill up 2 litre pot to the top with loose, mixed soil (avoiding stones) and tip into a plastic bag labelled with the field code and GPS location
3. On return to the lab, sieve soil through a 10mm sieve
4. Fill one seed tray with sieved soil for seedbank assessments and add plant label with field code and sample location
5. Place all seed trays in a glasshouse for emergence assessments. A standard greenhouse will work fine, but at the Hutton the glasshouse conditions are set to:

   Light intensity: 300µmol.m^2.sec^{-1}
   Light duration: 12 hour day length
   Minimum Day temp: 18^{o}C
   Minimum Night temp: 15^{o}C
   Shade screens operative at 600µmol.m^2.sec^{-1} set and 22^{o}C

6. Water trays set out in glasshouse after the bulk soil sampling procedure. Ensure that soil in trays is kept constantly damp but not wet and not allowed to dry out.
7. Record the identity of each species on the record form attached as soon as it is big enough to correctly ID and handle.
8. Pot up any seedlings that cannot be identified and label with a unique ID number, the field code and GPS point. Record the ID number on emergence record sheet.
9. Continue assessments until no new seedlings emerge for at least 2 weeks.
10. Turn off watering and leave trays dry until August.
11. For a second flush, dampen trays, re-sieve through a 10 mm sieve and return soil to the original trays.
12. Repeat steps 1-4, recording seedling IDs as they emerge

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Monitoring: insect pollinators

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Background

Many pollinator species are in decline due to arable intensification, habitat loss and fragmentation.

Loss of pollinators from farmland affects crop productivity as well as the population sizes of native plant species. Rare plants share pollinators with more common plant species, the latter providing insects with a continuity of nectar and pollen resources over the season.

Monitoring the abundance of these common plant species within and around cultivated fields and assessing their quality as a resource for pollinators is described in the previous sections.

However, there is little quantitative evidence to show how effective these strategies are in improving the diversity, activity and abundance of pollinators in arable systems.

Measurements

Insect pollinators include a wide range of taxonomic and functional groups including Diptera (especially hoverflies), Hymenoptera (honeybees, bumblebees and solitary bees), Lepidoptera (butterflies and moths) and Coleoptera (pollen beetles). Due to their long co-evolutionary history, this broad diversity is reflected in a similar range of form and function in the plant floral resources that they feed on, but presents a challenge for accurate monitoring. All insect sampling techniques are biased towards certain groups over others, depending on the insect's life-history strategy, activity pattern and foraging technique.

There are two simple methods, that can provide reasonably cost-effective and representative data to show relative differences in pollinator diversity across habitats:

1. Transects walks: non-destructive surveys, giving opportunity for additional information on which plant species are foraged.

2. Pan traps: destructive sampling but providing a broader picture of flying insect communities active in a habitat.

Other more time-consuming methods or requiring more specialist taxonomic expertise include malaise trapping, light traps (nocturnal flying moths), and bait plants to measure pollination rates.

Findings

- Pollinator activity is greater where there is greater abundance and diversity of flowering plants.
- Spill-over effects can be detected in pollinator movement from field margins into cropped fields.
- Greater pollinator activity correlates with better seed set in test plants (e.g. field beans).

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Pan Traps for Monitoring Pollinators

Version 1.0, 17 March 2022, adapted from the UK Pollinator Monitoring Scheme (Defra)

Equipment for set-up:

36 sets of 3 pan-traps: 20 cm diam plastic bowls painted with UV reflective paint (white, UV blue, and UV yellow)

36 stakes/pan trap stands, with attachment for the 3 bowls, set at a height level with crop canopy flowering height

100ml per trap of dilute Teepol® detergent (1 cap to 5 L of water)

Equipment for collection:

72x 20cm squares of muslin, tea strainer/sieve, paint brush, plastic pot to drain water into, 72 ziplock plastic bags labelled with field and location.

Timing: Twice a year depending on weather conditions – spring (May) and summer (July).

Weather conditions for sampling:

- relatively sunny day (no more than 75% of cloud cover)
- a minimum temperature of 13°C if the sky is clear (less than 50% cloud)
- a minimum temperature of 15°C if the sky is cloudy (cloud cover more than 50%, but not more than 75%)
- wind speed under 5 on the Beaufort scale.

Location: Set traps at 6 locations in each field: 3 sets spaced equidistantly along 2 tramlines into the field

Sampling procedure:

1. At each field, record date, time, weather conditions and crop growth stage
2. At each of the 6 locations per half field, install the pan trap stand at crop canopy height, with one bowl of each colour, white, blue and yellow.
3. Fill each bowl with teepol solution (approx. 100 ml)
4. Set all traps by 10am and leave until 4 pm. If possible, use the intervening period to carry out weed assessments.
5. From 4pm, record the collection time for each field, and collect samples as follows:
6. Place muslin square into sieve and place sieve over collecting pot. Pour samples from all three bowls through sieve, emptying out excess water as necessary.
7. Ensure that all insects from each bowl are in the muslin and use the brush to pick out any remaining specimens.
8. Fold muslin gently in half and place in the labelled Ziploc bag, seal.
9. On return to the lab, place all samples in a -20°C freezer until processing

Sorting Pan Trap Samples

Version 1.0, 17 March 2022

Sorting

1. Take sample bags from the freezer and leave to defrost at room temperature for an hour
2. For each sample, remove the muslin square from the bag and open on a white tray.
3. With a pair of forceps, pick out all specimens and group by taxa in separate petri-dishes.
4. Count and record the number of individuals in each taxa.
5. Place specimens in a sample tube, labelled with the date, field name and sample ID.
6. Fill with ethanol diluted with water to 70%, keeping any insects that require further identification in separate labelled tubes.
7. Ensure lids are tight and store somewhere cool.

Identification

Level of taxonomic identification will depend on available expertise and specific purpose of the survey. Here's a useful guide for beginners: https://www.nhm.ac.uk/content/dam/nhmwww/take-part/identify-nature/beginners-uk-invertebrate-id-guide.pdf

At the most basic level, record the following –

- Parasitic wasps (Hymenoptera) either as one group or separate into
  - Ichneumonids
  - Others
- Diptera divided into
  - Hoverflies (Syrphids)
  - Other large (like houseflies or blue bottles)
  - Other medium (around 5 mm long)
  - Other small (like midges or fruit flies)
- Bees divided into
  - Bumblebees (see ID guide overleaf)
  - Solitary bees
  - Honey bees
- Coleoptera (beetles) divided into
  - Pollen beetles
  - Others

Pollinator transects for monitoring bees and butterflies

Version 1.0, 17 March 2022, developed from Farm-Scale Evaluations of GMHT crops, Defra 1999-2003

Equipment: Record sheet and ID guides. Recommended:
https://www.bumblebeeconservation.org/bumblebee-species-guide/
https://butterfly-conservation.org/butterflies/identify-a-butterfly

Timing: Monthly if possible between May and August, depending on weather conditions.

Location: Two field margins and two tramlines into the cropped area of each field to be surveyed.

Sampling procedure:

1. Carry out surveys between 10:00 and 16:00 when the temperature is above 13°C with at least 60% clear sky and above 17°C in any sky conditions, apart from heavy rain and strong wind.
2. At each field, record date, time, weather conditions and crop growth stage.
3. Start a minimum of 10m along the field margin from a corner or gateway.
4. Walk slowly along the field margin for 100m aiming to cover the transect distance in about 5 minutes.
5. As you walk the transect, record all bees (bumble bee colour types, honey bees, solitary bees) and butterflies (species) that are actively foraging or resting on plants within 2m of the crop edge into the field margin. Don't record individuals that are just flying past.
6. Repeat for the same distance and time, walking along a tramline perpendicular to the surveyed margin into the cropped area of the field.
7. Repeat steps 4-6 for the field margin/crop area on the opposite side of the field.

Bumblebees

Large, furry bees; mainly black with yellow banding; often with white, buff or red tail.

Honey bees

Slender with furry thorax but otherwise smooth; brownish with stripey abdomen.

Solitary bees

Small and highly variable in furriness, stripiness and colour; if it's not a bumble or honey bee, but is definitely a bee, it is probably one of these!

Monitoring: predators at the ground-surface

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Background

Arable fields and their immediate surroundings can support many invertebrates active in the soil, leaf litter and on the ground surface. Many of these species, (predatory ground beetles, rove beetles and spiders) have important ecological roles as food for mammals and birds, predators of pests (slugs and aphids), and consumers of arable weed seeds.

These invertebrate groups are sensitive to local habitat conditions, availability of their preferred food and the intensity of management, particularly crop protection inputs. Much is known about their ecology, and they are easy to sample across many different habitats.

The species composition of these invertebrate groups is therefore an ideal indicator of farmland habitat quality.

Measurements

No single sampling method can capture a complete and accurate picture of the invertebrate community in a particular habitat. However, selecting a suite of sampling strategies with a knowledge of their inherent bias in mind, can give sufficiently reliable measures for estimating biodiversity impact.

Pitfall trapping is a quick and easy method that samples ground-surface active predators including hunting spiders, ground beetles and rove beetles.

Sampling should be carried out in both spring and autumn to capture species with different breeding seasons and life history strategies.

This method does not measure abundance of the species present but is a combined measure of activity and population density: trapping is biased towards species that move rapidly with frequent changes in direction. Trap modifications can increase accuracy of population measures, but the basic trap gives a functional and ecologically relevant estimate of change in management or land use and a relative measure of diversity between sites.

Results

- Ground beetles, rove beetles and spiders are good indicators of broad habitat quality and farmland management, but
- Activity patterns and vegetation structure needs to be accounted for in assessing subtle differences across systems.
- Further work is required to quantify the extent to which different species disperse into fields and their functional role in pest and weed seed predation.

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Pitfall Traps for Monitoring Ground Predators

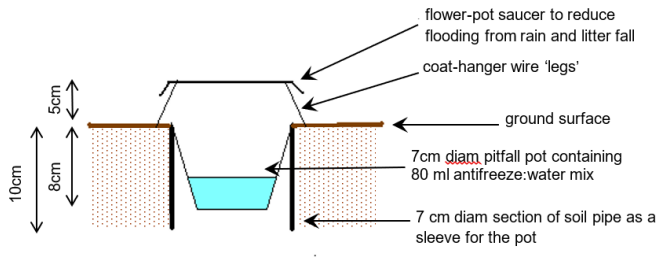
Version 1.0, 17 March 2022, developed from the Farm-scale Evaluations of GMHT Crops, Defra 1999-2003

Equipment for set-up:

Trowel, 20 pitfall traps and marker canes, tape measure.

Equipment for collection:

20 labelled pitfall pot lids.



Timing: Twice a year in April for spring breeding species and September for autumn breeding species. Traps should be set for exactly 2 weeks (14 days) during each collection period. If numbers of beetles trapped are low, the traps can be reset for a further 2 weeks.

Location: Set traps at 20 locations, spaced 10m apart in each field: 10 in a line along a field margin and 10 in a transect running perpendicular into the cropped area from the headland into the field centre.

Sampling procedure:

1. At each location, dig a 10 cm x 10 cm hole and insert the soil pipe sleeve, making sure that the rim is level with the ground surface and there are no gaps between the soil and the outer edge of the sleeve.
2. Place the pitfall cup containing about 80ml antifreeze (diluted to 50% with tap water) into the sleeve so the cup rim is flush with the ground surface and there are no gaps.
3. Cover with a flower pot, raised up above the ground by a couple of centimetres with coat hanger wire.
4. Mark location with a cane so that traps can be found easily on collection.
5. After two weeks, collect up the traps, placing a labelled lid on each pitfall cup to return to the lab for sorting
6. In the lab, pour the contents of each trap into a tray and pick through, placing all insect specimens in glass sample tubes filled with 70% ethanol and labelled with the date and sample location.
7. Samples can be stored in these tubes somewhere cool until identification.
8. Identify ground beetles (Carabidae) to species and rove beetles (Staphylinidae) and spiders to family. If time allows, number of mites and collembola can also be counted, unless tulgren funnels are being used as this is a more suitable method for estimating populations of these groups.

Monitoring: earthworms and other soil invertebrates

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Background

Soil biodiversity is essential for litter decomposition, pest suppression, nutrient cycling and uptake by plants, but in cultivated soils, these functions are reduced compared to natural ecosystems. Soil disturbance by cultivation particularly impacts larger taxa (earthworms) but all groups are affected by high rates of agrochemical use.

Earthworms are especially important in arable fields. They connect above and below ground processes and stimulate microbial activity in the soil. They maintain soil structure, cycle nutrients, increase organic matter and improve water infiltration.

Other arthropod groups, (springtails and mites), are also essential for soil food-web functioning, breaking down dead plant matter and providing resource for higher trophic groups.

Measurements

Traditional methods for sampling earthworms are based on in-field extractions using mustard solution (or other repellent chemicals), followed by time-consuming species identification in the lab. This is a major limitation in large-scale surveys needed to understand patterns in earthworm communities and how they are affected by agricultural management.

A simpler method is to hand sort a standardized volume of soil and ID adult worms just to functional group level. This provides less detailed information for a given field, but takes less time, allowing more sites to be surveyed. Data correlate well with standard extraction methods.

Smaller soil dwelling invertebrates such as springtails, mites, nematodes and the soil-dwelling larvae of surface-active beetles can be sampled using Berlese-Tulgren funnels to give a more complete representation of the soil and ground-surface food-web.

Trends in the data gathered will depend on the properties of local micro-habitat conditions, so measurements of the soil (moisture, texture, organic matter) and vegetation (ground cover, species composition) should be assessed alongside these indicator groups.

Findings

- More earthworms in conservation tillage systems and where organic matter amendments are high.
- Earthworm numbers are correlated with litter decomposition rates through interactions with soil micro-organisms.
- Smaller, less mobile soil invertebrates are sensitive indicators of local management intensity and soil conditions.

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Earthworm abundance and functional groups

Version 1.0, 17 March 2022, adapted from Jacqueline Stroud, SRUC, UkSoils.org

Equipment: 20 x 20 cm quadrat, trowel, white tray, pot for worms, record sheet, camera, ruler, labels

Timing: Sample 1. early/mid April (before sowing if possible)
Sample 2. October (after all crops harvested)

Field conditions: Sample when soil is damp (preferably after rain) but not frozen.

Location: Carry out assessments at 9 locations in each field arranged in a W pattern to achieve full coverage across the field.

Procedure:

1. Record soil conditions, date and surveyor initials on your record sheet.
2. At each field, the note cultivation state (plough, stubble, sown, crop).
3. At each sample point, count the number of middens and wormcasts in a 20 x 20 cm quadrat.
4. Dig out a 20 x 20 x 20 cm soil pit and place soil on a white tray.
5. Hand sort soil and place all worms into a pot.
6. Empty tray of soil into pit and brush clean.
7. Tip pot out onto the clean tray.
8. Record the number of juveniles and return them to the pit.
9. Place ruler and a label indicating sample point and date label next to adult worms and take photograph.
10. Record number of adults in each functional group (see over).

Earthworm ID guide

Version 1.0, 17 March 2022, adapted from Jacqueline Stroud, SRUC, UkSoils.org

Midden: pile of straw or stones over a permanent burrow

Wormcasts: digested soil produced by *Aporrectodea* and *Lumbricus* spp

Juveniles: no saddle

Adults: with saddle

Adult functional groups: 3 types

Surface worms

Size: small (matchstick, <8mm),

Colour: red

Habit: fast moving, found in leaf litter

Topsoil worms

Size: small-medium
Colour: pink, grey, green, mottled yellow

Habit: most common, found in topsoil

Deep burrowers

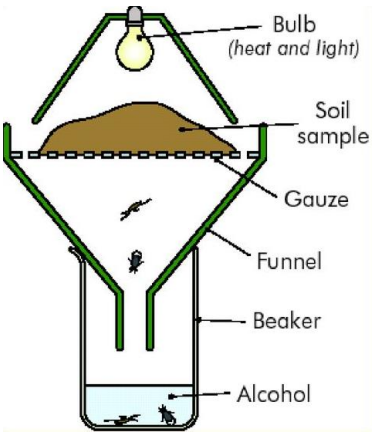
Size: large (>8mm)
Colour: red or black head

Habit: large, vertical burrows, middens

Tulgren funnel extraction of soil dwelling arthropods

Version 1.0, 17 March 2022

Equipment: trowel, labelled soil sample bags and a bank of 12 Berlese-tulgren funnels.



Timing: Sampling to be carried out in spring (April) and autumn (September) preferably at the same time as pitfall trapping and/or earthworm sampling if these are to be carried out as well. As for earthworms, sample when the soil is damp and warm enough for plant growth and soil invertebrate activity.

Location: Twelve sample points should be located across the field in a W pattern.

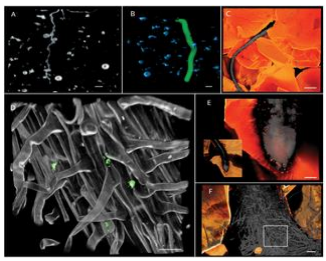
Sampling procedure:

- 24 hours prior to sampling, switch on the lights over the tulgren funnels and check they are working/warming up.
- Then, in the field, at each location, without disturbing or mixing the soil, take a 10 x 15 cm soil core from the top 15cm soil layer (i.e. to normal plough depth).
- Place entire core (including any surface litter) into a labelled, plastic sample bag
- Tie and return to the lab

Sample sorting:

- Sieve each core through a 10mm sieve and place the soil into one funnel sleeve and note which sample goes into which funnel.
- Repeat for each of the 12 samples.
- Place a labelled collecting tube filled with 70% ethanol under each funnel
- Leave for 48 hrs, checking level of ethanol and topping up if necessary
- Remove each sleeve, re-mix the soil and replace over the funnel for another 48h
- Store the tubes somewhere cool until sorting into groups.
- Count the number of springtails, mites and other insects extracted from each sample.

Monitoring: soil microbial function



https://doi.org/10.1371/journal.pone.0044276

The James Hutton Institute

Background

Earthworm activity, organic matter inputs and low input agriculture enhance the diversity of microbial soil-based networks, giving them greater resilience to disturbance and stresses.

Beneficial soil organisms include mycorrhizal fungi and rhizobia which form symbiotic associations with plants, improving availability and uptake of nutrients. Microbial activity is important for maintaining soil processes such as litter decomposition.

Microbial diversity also helps regulate pathogen populations by competition with neutral antagonists, which enhances the pest suppressive properties of the soil.

Microbial abundance and diversity is reduced by monoculture cropping, soil disturbance, agro-chemical inputs and fallow periods.

Methods

Assessment of the soil microbiome and quantification of specific microbial responses to field management requires specialist knowledge and laboratory techniques.

Simple indicators of the soil functions that are driven by the microbial community are therefore required.

Most indicators focus on litter decomposition processes, e.g. the community science projects:

"*Time for tea*" the Global Litter Decomposition Study, which uses the loss in weight of teabags buried for the duration of the growing season to calculate rates of litter decomposition. www.teacomposition.org)

"*Soil my Undies Challenge*" a fantastic KE exercise where farmers were encouraged to bury cotton underpants to test the level of biological activity in their soils. Breakdown of the cotton was greater in fields with 'healthy soil' management.

A more formalised protocol for measuring organic-matter decomposition represented by the breakdown of cellulose in standardised strips of woven cotton fabric can be used to give a quantifiable comparison across sites and fields.

Findings

- Faster rates of litter decomposition can be detected in fields with relatively high levels of organic matter (>3% carbon) and low disturbance (conservation tillage).
- Organic matter and less disturbance enhances the soil microbial community and therefore the rate of associated decomposition processes.

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Cotton strip assay for measuring litter decomposition

following https://conversations.echocommunity.org/t/simple-proxy-for-soil-life-cotton-strips/5480

Equipment: trowel, marker cane (so you can find your samples at the end of the season), 20 cotton strips per field, each sealed in a labelled mosquito net bag (so you know which strip is which when you dig them up again). The mosquito net is to stop larger insects shredding the cotton so that the decomposition rates estimated are just a result of microbial activity.

Timing: Bury cotton strips as soon after planting as possible in the spring (for spring sown crops) and around the same time in winter sown fields (April). Retrieve them after harvest when it's easy to access the field.

Location: 20 sample points should be located across the field in the usual W shape, but make sure they are away from tramlines so tractor movement and harvest operations don't get in the way.

Procedure:

1. Cut 20 strips of unbleached, 150 GSM cotton calico measuring 20x10 cm.
2. Dry at 70 ^{0}C for 24 hours to standardise the starting weights, then weigh and place each one inside a bag made out of mosquito netting (or similar fine mesh), labelled bag with a unique identifier, noted alongside the starting weight.
3. Bury each bag 4-5 cm below the ground surface and mark the location with a cane. Record the date of burial.
4. Leave for whole the growing season.
5. After harvest, locate and carefully dig up the samples, making sure not to damage the mesh bag. Record the date of retrieval.
6. Spread the bags out on a bench to dry, then gently roll something heavy over them to break up any clods of earth. Shake to remove as much soil as possible. Then rinse off any remaining debris in water.
7. Dry the bags again at 70 ^{0}C for 24 hours
8. Carefully tip out each cotton strip (or what's left of it) onto a dish and re-weigh (making a note of the weight of the empty dish).
9. Take the final weight away from the starting weight and the result is the amount of cotton that has decomposed. This gives a relative measure of overall microbial activity and can be used to detect change over time or differences between different fields.

Monitoring: soil carbon and structure

The James Hutton Institute

Background

Soil structure is determined largely by the level of disturbance and the organic matter content of the soil. Reduced disturbance by conservation tillage, plus organic matter inputs from dead plant matter, root exudates and external amendments (e.g. compost) provide better environment for both root growth and microbial activity.

Improved soil structure is critical in minimising losses through erosion, facilitating better drainage and water holding capacity, lessening extremes of waterlogging and drought.

Accumulating SOC not only benefits soil structure but contributes to enhanced productivity by microbial activity releasing plant available nitrogen. This helps reduce the GHG emissions resulting from mineral fertiliser production and application.

Methods

A simple Visual Evaluation of Soil Structure VESS was developed by SRUC and is widely used by farmers and agronomists to assess agricultural soil structure on a 5-point scale from friable to very compact.

The guide and colour chart can be downloaded from: https://www.sruc.ac.uk/media/xbrfn4x3/vess-colour-chart.pdf

Measuring soil organic matter is straightforward but requires access to a muffle furnace.

- Take 20 soil samples in a W-pattern across the field.
- Weigh each and dry in an oven at 70°C for 48 hrs or until there is no further loss in weight. This will give the soil moisture content at the time of sampling.
- Using a pestle and mortar, crush the soil from each sample into a fine powder.
- Sub-sample approximately 5g of ground soil and record the exact weight to 3 decimal places.
- Heat in a muffle furnace at 450°C for 4 hours and then re-weigh.
- The difference in weight before and after burning gives the organic matter content, which is roughly estimated as about 2x the carbon content.

Results

- Soil carbon content was increased from around 2% to 4% in 6 years at the Hutton's Centre for Sustainable Cropping platform by occasional tillage, compost amendments, cover cropping and crop residue incorporation.
- The increase in organic matter and improved soil structure correlates with more earthworms, faster rates of litter decomposition and an increase in microbial biomass.

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Measuring soil structure

For Visual Evaluation of Soil Structure, see:
https://www.sruc.ac.uk/media/xbrfn4x3/vess-colour-chart.pdf

Other methods include the Slake test and the Infiltration test
https://cropscience.bayer.co.uk/blog/articles/2020/02/test-soil-health/

Slake test measures the stability of soil aggregates and it can maintain its structure.

- Collect a handful of topsoil from the field to be tested
- Take two a glass jar and some wire mesh. Mould the mesh so it can hook over the top of each jar and is large enough to hold the chunk of soil inside the jar. Put the lump of soil into the mesh inside the jar and fill with water, submerging the soil in the process.
- Time how long it takes for the soil to disintegrate. Soil that disintegrates quickly has a poorer structure and lower organic matter content than one which remains intact
- Compare length of time to disintegrate for soil from different areas of the field and record the time taken at each location. Repeat over a number of years at the same locations to monitor the change in soil aggregate stability over time.

Infiltration test measures how fast water percolates through the soil, providing a good indicator of soil structure.

- Take a 150x150mm metal or plastic ring (marked at 85mm depth), a bottle of water, a beaker marked at 450ml and a stop watch.
- The testing area should not be saturated, so don't do this straight after heavy rain.
- Insert the tube into the ground to a depth of 85mm, using a mallet if necessary.
- Start the stop watch as you start pouring the 450ml water steadily into the cylinder and time how long it takes for the water to disappear
- Repeat in 5 – 10 locations across the field.
- Faster rates suggest good crumb structure and aggregation, such as a healthy soil with high organic matter. Slower rates suggest the presence of compaction, reduced porosity and lower organic matter.
- Repeat the test at the same time each year to monitor improvements.

Economics and whole-system sustainability

The James Hutton Institute

Background

Agricultural practices that benefit the environment in terms of soil quality and biodiversity, are often in conflict with management to maximise yield output. However, degradation of the farmland environment in which food is produced is unsustainable in the long-term.

To assess how well a cropping system meets these multiple goals, indicators of both economic and environmental sustainability need to be combined to generate a whole-system assessment of impact.

We have produced a qualitative model to do this which, alongside these handbooks for monitoring indicators and selecting best agronomic practices, forms the online CSC Toolkit for optimising sustainability in arable cropping.

(to be released 2023)

Methods

Environmental indicators include the biodiversity and soil health assessments outlined here which, together with data on agronomic practices and inputs (collated from farm records), impact overall system sustainability.

Economic indicators for the impact of a change in management at the field-scale include all input costs (agrochemicals, amendments, seed), plus fuel use, tractor time, yield, product quality and sale prices.

Using these data, a qualitative, multi-criteria sustainability assessment can then be carried out via the CSC Toolkit: all indicators are aggregated into a hierarchical framework where overall sustainability at the top is broken down into economic and environmental components, each of which are further subdivided into progressively smaller elements, down to individual measured indicators at the bottom.

This user-friendly tool highlights where the positive and negative impacts of different management choices through each branch of the model, allowing users to identify where improvements can be made. This provides the opportunity for iterative design, implementation, monitoring and assessment for gradual transition towards more sustainable cropping systems.

Results

- Initial losses in financial margins are expected in the transition to low input, regenerative agroecosystems.
- Ecological networks and soil biophysical quality stabilise over time, allowing less reliance on inputs with no yield penalty.
- Social and wider food system impacts need to be incorporated into the overall impact assessment.

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DEXi-CSC: A tool for whole-system sustainability assessment

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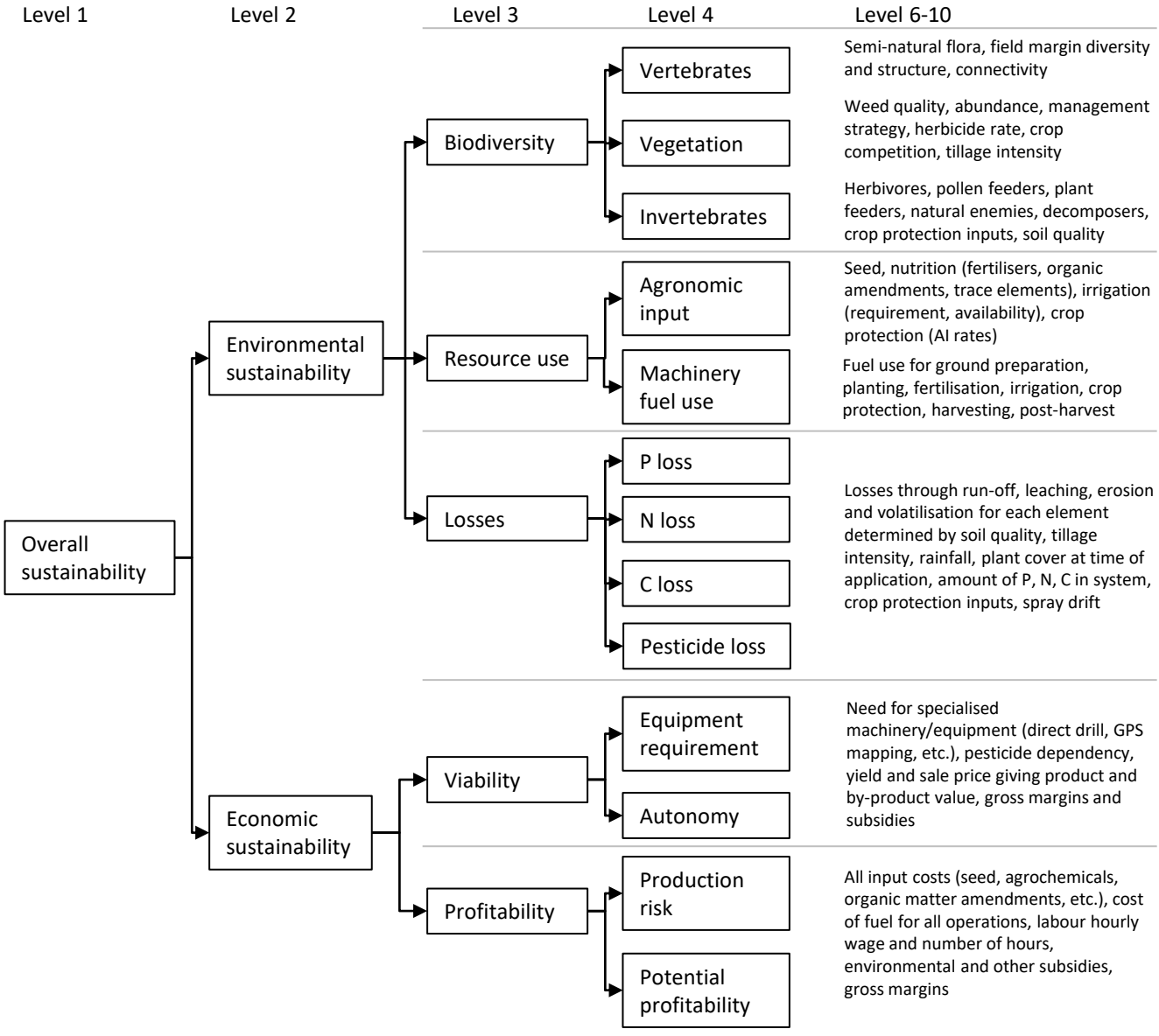

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