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A quantitative tool kit for assessing soil health indicators for resource limited laboratories

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ABSTRACT

Soil health assessment is crucial for sustainable agriculture; however, farmers and their advisors can face barriers accessing traditional laboratory-based testing. To address this, we developed a low-cost soil health kit using readily available materials to provide quantitative results for selected soil physical, chemical and biological properties. The kit includes ten indicators adapted from established laboratory methods. We first evaluated the kit's ability to detect differences in soil properties under different land uses, including rainforest, pasture, sugarcane, and banana farms. We then validated the kit's sensitivity to farm management impacts on soil health, using soils from commercial banana farms with varying levels of vegetated ground cover. Differences were detected in nine of the ten soil properties included in the kit (e.g., P, pH, bulk density) across land uses and significant differences were detected in seven of the ten soil properties (e.g., labile carbon, ACE protein, moisture, β-glucosidase) among banana production soils with varying levels of ground cover. Correlation analyses revealed that the kit's measurements of P, pH, and labile carbon significantly aligned with traditional laboratory methods ($R = 0.81$, $R = 0.83$, and $R = 0.86$, respectively), while there was a weaker correlation for nitrate nitrogen ($R = 0.62$). These findings highlight the kit's potential as a proxy to supplement standard laboratory measurements and its utility in assessing management-induced changes in soil health. The quantitative monitoring outcomes can support advisors, extension staff, and growers to make informed soil management decisions, particularly where access to laboratory facilities is limited or cost prohibitive.

1. Introduction

Soil degradation is a major global challenge, with approximately one-third of the world's soils affected by erosion, nutrient depletion, salinisation, and compaction due to intensive land use and inappropriate management practices (FAO, 2020). Preventing further degradation and restoring soil function are essential for sustaining agricultural productivity and ecosystem services, particularly as global food demand continues to increase. Achieving this requires reliable methods to monitor soil health and to evaluate the impacts of land management practices over time.

Soil health refers to the soil's ability to maintain productivity that sustains plants, animals, and humans while maintaining environmental quality (Doran and Parkin, 1994; Doran and Zeiss, 2000). Healthy soils support key ecosystem functions including nutrient cycling, water regulation, carbon sequestration, and resistance to erosion and disease (Fausak et al., 2024). Assessing soil health therefore requires indicators that are sensitive to management, linked to soil functions, and practical to measure. Doran and Zeiss (2000) proposed that soil health indicators should be responsive to management change, biologically meaningful, interpretable, and feasible for land managers to implement.

While new agricultural practices, such as regenerative and conservation agriculture, are often designed to enhance soil health, assessing their impact on soil conditions is typically not conducted by farmers, advisors and local agricultural researchers (Thomsen et al., 2019). This is mainly because standard soil health tests can be inconvenient, expensive, unreliable, or impractical (Friedman et al., 2001). Soil analysis in some regions comes with high service and shipping costs, particularly for remote areas, and results are often delayed, diminishing the value of the soil data. This highlights the need for affordable and accessible convenient local soil testing options to supplement traditional laboratory tests. Many of the currently available soil health testing methods worldwide only provide qualitative data (FAO, 2020), which when solely used can be subjective and incomplete. Submitting soil samples to conventional analytical laboratories can be the most practical option for many users; however, many laboratories do not offer

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biological and physical tests (e.g., Autoclave Citrate Extractable (ACE) protein, β-glucosidase activity, aggregate stability). An exception would be the Cornell University soil health testing services, which provides a comprehensive soil testing service for the USA (Moebius-Clune et al., 2016). However, smallholder growers who collectively make up much of the food supply chain in developing countries do not always have access to soil testing services.

To address the lack of accessible, holistic soil testing, we present the development and evaluation of a quantitative, low-cost soil health kit that adapts established laboratory methods into simplified protocols using readily available materials and minimal instrumentation. Here, the soil health kit refers to a coordinated set of compatible physical, chemical, and biological indicator measurements selected using predefined criteria and implemented with low-cost equipment suitable for basic laboratory or near-field analytical settings. In contrast to many existing field-based approaches that rely on qualitative or semi-quantitative interpretation, the soil health kit generates quantitative outputs for a targeted set of indicators that are directly linked to key soil functions. The indicators included bulk density, soil stability, moisture content, pH, nitrogen, phosphorus, labile carbon, ACE protein, β-glucosidase activity, and basal respiration (SituResp®). The indicator set was designed to capture multiple dimensions of soil function rather than rely on a single proxy, because different land uses and management changes may affect physical, chemical, and biological properties differently. These were selected based on their sensitivity to management, relevance to ecosystem processes, and suitability for use in basic or near-field laboratory settings. The soil health kit is intended to complement, rather than replace, standard laboratory analyses by providing convenient, interpretable measurements that support comparative assessment and identify trends warranting further investigation.

Accordingly, the objectives of this study were to: (i) evaluate the capacity of a low-cost, quantitative soil health kit to detect differences in soil physical, chemical, and biological properties across contrasting land uses, and (ii) assess its sensitivity to management-induced changes in soil health within commercial banana farming systems with differing levels of vegetated ground cover. In addition, selected kit-derived measurements were compared with analyses conducted by an accredited commercial laboratory to evaluate how closely simplified kit outputs aligned with standard laboratory procedures. We hypothesised that a carefully selected suite of quantitative indicators adapted from established laboratory techniques would be sufficiently sensitive to detect land use and management effects on soil health, and that these measurements could serve as a practical proxy to supplement conventional laboratory analyses, particularly where access to laboratory facilities is limited or cost prohibitive.

2. Methods

2.1. Selection of soil health indicators

The soil health kit was developed using a small number of reagents and inexpensive, off-the-shelf materials to deliver quantitative measurements of key soil physical, chemical, and biological indicators. Soil test indicators were selected based on their relevance to core soil functions, including structure maintenance, carbon transformation, nutrient cycling and water regulation. A set of ten criteria guided the selection process: namely portability, cost-effectiveness, quantitative output, sensitivity to management, minimal equipment requirements, ease of interpretation, safety, reproducibility, adaptability from established laboratory methods, and reliance on readily available materials. Based on these criteria, ten soil health indicators were adapted from validated laboratory methods, including bulk density, soil stability, soil moisture, pH, labile carbon, nitrogen (N), phosphorus (P), β-glucosidase activity, ACE protein, and basal microbial respiration measured using SituResp® (Table S1). Cost estimates associated with the kit were based on approximate consumable and equipment costs only and did not include labour, which may vary substantially among institutional and national contexts.

Bulk density and soil stability were included as physical soil health indicators representing different but related aspects of soil condition, including compaction, porosity, aggregation and resistance to erosion, with soil stability measured using a modified wet-sieving approach that allowed the same soil core to be used for both analyses (Kemper and Rosenau, 1986; Brown and Wherrett, 2020).

Biological activity was assessed through basal microbial respiration using SituResp®, a sensitive indicator of microbial metabolism responsive to management and environmental conditions (Thoumazeau et al., 2017). The β-glucosidase activity, an enzyme involved in cellulose degradation and organic matter turnover, was also measured using a modified colourimetric assay that substitutes toluene with Tween to reduce chemical hazards (Tabatabai, 1994; Pattison et al., 2018; Bonanomi et al., 2010). Measurement of ACE protein was also incorporated to provide an estimate of bioavailable organic nitrogen and enhance the biological relevance of nutrient assessments, consistent with its inclusion in comprehensive soil health testing frameworks (Hurisso et al., 2018; Moebius-Clune et al., 2016).

Chemical measures for N, P, pH and labile carbon were also included as indicators of nutrient availability and the soil's capacity to supply nutrients to crops, all of which are measurable with test kits. N and P indicate nutrient availability and fertility status of the soil (Dimkpa et al., 2017); the soil pH influences nutrient solubility and microbial function (Smith and Doran, 1996), while labile carbon represents the readily available energy source for soil microorganisms and is a sensitive indicator of management-induced changes in soil organic matter dynamics (Weil et al., 2003). Inclusion of these indicators also provides measurements that are widely interpretable and comparable with standard laboratory analyses.

All colourimetric assays were quantified using a single portable spectrophotometer (Go Direct SpectroVis Plus, Vernier), enabling consistent measurement across wavelengths from 380 to 950 nm and supporting standardised, reproducible data collection across field settings (https://www.vernier.com/product/go-direct-spectrovis-plus-spectrophotometer/).

2.2. Soil physical indicators

The soil bulk density was determined following the methodology outlined by McKenzie et al. (2002). The soil core was collected in the field using a metal ring of known volume, which was pressed into the soil. In the soil health kit (Figure S1A), 75 mm lengths of aluminium irrigation pipe were reused as sampling rings. The metal core ring was carefully positioned on the soil, and a piece of timber was placed on top of it. The timber was gently tapped with a hammer to ensure the metal ring was fully embedded in the soil. Afterward, the metal ring was removed, and the ends of the core were trimmed using a knife.

The weight of the fresh soil core, along with the pre-weighed metal core, was determined. The ring and soil core were placed in common kitchen toaster oven for drying at 105°C for 24 h. Once dried, the combined weight of the soil core and metal ring was again determined. To measure the moisture content, the same soil core used for bulk density determination was utilised. The moisture content was calculated by determining the fresh and dry weights of the soil (after subtracting the weight of the metal core from both). The soil moisture content was converted to a percentage using the following formula:

$$\textbf{Soil Moisture}(\%) = \frac{(\textit{fresh weight} - \textit{dry weight})}{\textit{dry weight}} x \quad 100$$

.

The bulk density was calculated using the dry weight of the soil core:

$$\textbf{Bulk density}(\mathbf{g.cm^3}) = \frac{dry\ weight}{volume \text{of metal ring}}$$

.

Soil stability was determined using a modified wet sieving method, as described by Almajmaie et al. (2017). For this measurement, the same dried soil core previously used for bulk density determination was placed on a 5 mm screen mesh basket and submerged in water for 10 min, to allow the soil to re-wet and for the aggregates to slake naturally. After 10 min, the screen mesh with the soil core were removed from the container of water. The slaked soil was collected by sieving the water using a funnel placed over a container, and the water passed through a pre-weighed filter paper. The filter paper was then oven-dried at 105°C for 30 min, weighed, and the weight recorded. The percentage of dry unstable soil and the stable soil were determined using the following formula:

$$\textbf{Unstable Soil}(\%) = \frac{[(\text{weight of dry soil on filter}) - (\text{weight of filter paper})]}{\text{weight of the air} - \text{dried soil core}} \times 100$$

Soil Stability (%) = 100 - Unstable Soil (%)

.

2.3. Soil health kit chemical indicators

To measure nitrogen (N) and phosphorus (P) concentrations, the HI3896Hanna Kit (https://hannainst.com.au/product/npk-soil-chemical-test-kit−10-tests-each-hi3896/) was used (Figure S1B). Chemical reagents were added to react with N and P to produce a coloured product following the manufacturer's instructions. The Hanna HI3896 kit provides an estimate of available mineral N and P in soil extracts, which in this study was compared with laboratory nitrate-N and Colwell P as the closest conventional laboratory measure. Soil samples (10 g each) were collected, sieved through a 5 mm mesh, and air-dried. A 1-gram subsample of soil was transferred into a 15 mL tube, followed by the addition of 7.5 mL of the Hanna Kit extraction solution as instructed by the manufacturer. The tube was gently shaken for one minute and allowed to stand for 5 min to facilitate the extraction process. Subsequently, 2.5 mL of the extract was pipetted into a 3.5 mL Vernier colourimeter cuvette (Scientrific Pty Ltd). One sachet of N reagent was added to the cuvettes containing N samples, while the P reagent was added to those with P samples as specified by the manufacturer. Each cuvette was sealed with parafilm, shaken for 30 s, and left to stand for 5 min. Absorbance readings were then taken using the portable spectrophotometer, Go Direct SpectroVis Plus Spectrophotometer (Vernier). The same procedure was repeated for cuvettes containing N and P standards, respectively. Standard curves were constructed for each element based on the peak absorbance values, 900 nm for P and 520 nm for N. The concentration of N and P in the soil samples was determined by applying the linear equation derived from the standard curve to the absorbance data.

To measure the labile carbon in the soil the potassium permanganate ($KMnO_4$) method described by Moody et al. (1997) was used (Figure S1D). For analysis, 1 mL of 0.1 M $CaCl_2$ was added to 5 g of sieved air-dried soil in a 50 mL centrifuge tube, followed by 25 mL of 33 mM $KMnO_4$. The mixture was vortexed briefly and mixed on a roller for 2 min, then centrifuged at 4000 rpm for 5 min. The supernatant (1 mL) was mixed with 49 mL deionised water, and 2.5 mL was pipetted into a cuvette for absorbance measurement at 550 nm using the portable, Go Direct SpectroVis Plus Spectrophotometer (Vernier). For labile carbon standards, 1 mL of each standard was added to 49 mL deionised water, and absorbance was measured at 550 nm.

To determine ACE protein as indicator of organic nitrogen, the method of Hurisso et al. (2018) was performed (Figure S1E). One gram of sieved soil was added to a 50 mL autoclavable tube, followed by 8 mL of 0.2 M sodium citrate buffer (pH 7.0). The tube was autoclaved at 121°C for 30 min, cooled, then centrifuged at 3100x*g* for 15 min. The clear supernatant was decanted into a new tube, and 66.6 μL of the supernatant was mixed with 2 mL of Bradford reagent in a cuvette. After a 30-minute incubation at room temperature, absorbance was measured at 595 nm using the Go Direct SpectroVis Plus Spectrophotometer (Vernier).

The soil pH was measured using a 1:5 ratio of soil and 0.01 M of $CaCl_2$ (Rayment and Higginson, 1992) (Figure S1D). The mixture was shaken for 1 h and the pH measured using a pH meter (Aqua-pH, TPS Australia).

2.4. Additional laboratory chemical indicators

Selected soil chemical properties were analysed by Nutrient Advantage Laboratory, Werribee, Victoria (https://www.nutrientadvantage.com.au/services/testing-services/soil-testing/) to provide standard laboratory measurements for comparison with the corresponding soil health kit outputs. Analysis used in this study included (with method codes), nitrate nitrogen (7C2b), organic carbon (Walkley and Black 6A1), pH (1:5 $CaCl_2$ 4A1) and phosphorus (Colwell 9B2).

2.5. Soil health kit biological indicators

To measure the basal respiration of soil microbes, a modified SituResp® (Thoumazeau et al., 2017) was performed (Figure S1G). The modification involved adding 5 mL of water into the soil to induce microbial respiration. A 100 g sub-sample of fresh soil was placed in a 250 mL jar, and 5 mL of distilled water was added to moisten the soil. The absorbance of the indicator gel in a cuvette was measured at 570 nm using the Go Direct SpectroVis Plus Spectrophotometer (Vernier) and recorded as absorbance at 'time 0'. The cuvette was placed in the jar of fresh soil, which was then sealed and left at ambient temperature (27°C) for 24 h. After 24 h, the absorbance of the indicator gel was measured again and recorded as absorbance 'time 24'. The amount of basal respiration was calculated using the equation in Thoumazeau et al., 2017):

$$\%CO2 = \alpha.\exp(\beta * \Delta abs)$$

where: Δabs = (Absorbance "time 24" – Absorbance "time 0")

$\alpha = 0.444$

$\beta = 3.3988$

The β-glucosidase activity in the soil was assessed using the colourimetric assay described by Lapis-Gaza and Pattison (2021) (Figure S1F). One gram of 2 mm sieved soil was added to a 50 mL centrifuge tube with 4.0 mL McIlvaine buffer, 0.25 mL Tween solution, and 1.0 mL ρ-Nitrophenyl ß-D-glucopyranoside (PNG) solution. The tube was sealed, mixed, and incubated at 30°C for 1 h. A second tube was prepared with distilled water instead of PNG solution. After 1 h, 10 mL of tris-calcium chloride stopping buffer (50 mL of 0.5 M $CaCl_2$

added to 200 mL of 0.5 M THAM (Tris (hydromethyl) aminomethane) and 250 mL of deionised water) was added to both tubes, which were centrifuged at 4000 rpm for 10 min. Two millilitres of supernatant were pipetted into a cuvette and absorbance measured at 410 nm using the Go Direct SpectroVis Plus Spectrophotometer (Vernier).

2.6. Experimental field sites

To evaluate whether the soil health indicators could distinguish among different land uses, soil samples were collected from four contrasting land use types following a methodology similar to that described by Birt et al. (2024). That is, at each site nine points were marked in a 3 m × 3 m grid covering 9 m^2. A 10 cm deep core was taken from each subsite, pooled and homogenised into a composite sample for the different land use, specifically from sites under rainforest, pasture, sugarcane and banana land uses along a linear transect near South Johnstone, Queensland Australia. All sites were within 500 m of one another and four sites from each land use were collected. Roots or other living material were removed. Samples were then transported to the laboratory in a cool box with ice until further processing on the same day.

To determine the sensitivity of the tests to farm management practices, soil samples were also collected from nine commercial banana farms, in the Cassowary coast region, Queensland Australia. The major soil types used for banana production in the Innisfail sub-region, which included brown dermosols and red ferrosols as described by Orr et al. (2023) were included in this study. Banana farms in this region require moderate inputs of nitrogen (N) and low inputs of phosphorus (P), which are regulated to 400 kg $N.ha^{-1}$ $.year^{-1}$ and 60 kg $P.ha^{-1}$. $year^{-1}$, respectively (Orr et al., 2023). The farms were selected on the basis of differences in vegetated ground cover within their banana fields as a proxy for overall crop management intensity. Three separate farms with low (<33%), medium (33–66%) or high (>66%) ground cover formed replicates. At each farm, soil was collected from three sites within the paddock as previously described. Each site was represented by a composite sample formed from three subsampling areas. The soil samples collected were used for analysis using the quantitative soil health kit and for standard laboratory analysis by Nutrient Advantage Laboratory, Werribee, Victoria as described previously.

2.7. Statistical analysis

Soil indicators from the various land uses were analysed to assess their sensitivity to changes in land management practices. A one-way analysis of variance (ANOVA) and a Fisher's protected least significant difference post-hoc test were performed using Genstat 24th edition (VSN International, 2024) to determine whether variations in land use resulted in significant differences in soil test results. To minimise collinearity among predictor variables, principal components analysis (PCA) was conducted on soil characteristics using the R platform (R Core Team, 2020).

Assessment of soil indicators from soil under banana production with varying ground cover density was performed to determine the soil health kit's sensitivity to changes in farm practices. A one-way analysis of variance (ANOVA) and a Fisher's protected least significant difference post-hoc test were performed using Genstat 24th edition (VSN International, 2024) to determine whether variations in ground cover density resulted in significant differences in soil test results. To reduce the number of predictor variables, principal components analysis (PCA) was conducted on soil characteristics using the R platform (R Core Team, 2020). To examine the relationships between the different soil heath indicators, a Pearson correlation analysis was conducted using the R platform (R Core Team, 2020).

To assess the consistency and reliability of the soil health kit measurements, Pearson's correlation analysis and a paired *t*-test were conducted using R platform (R Core Team, 2020) was conducted to examine the relationship between soil metrics obtained with the soil health kit and those derived from standard laboratory tests. The correlation analysis was performed on banana production soils with varying ground cover density.

Additionally, data obtained from the various soil health indicators were standardised using z-score normalisation (standardisation) using the R platform (R Core Team, 2020). Each variable was transformed so that it has a mean of 0 and a standard deviation of 1, to allow a direct comparison among indicators. For each soil health parameter, the z-score was calculated as:

$$Z = \frac{X - \mu}{\sigma}$$

where:

X= observed value for a given sample
μ= mean of that parameter across all samples
σ= standard deviation of that parameter across all samples
Z= standardised value (z-score)

.

Standardisation was performed separately for each indicator across all samples to preserve relative differences among sites. The resulting z-scores represented how far each observation deviated from the overall mean in units of standard deviation. Higher positive z-scores indicate comparatively greater values of a given soil health indicator, whereas lower scores indicate comparatively lower values. The z-score normalised data were scaled to a 0–1 range and visualised using a radar chart created with the "fmsb" package in the R platform (R Core Team, 2020), enabling relative differences among categories to be compared.

3. Results

3.1. Soil health kit measures differentiated land use impacts

The indicators included in the soil health kit clearly distinguished soils from less intensively managed systems (rainforest and pasture) from those under more intensive agricultural use (banana and sugarcane). Significant differences ($P < 0.05$) were detected in nine of the ten indicators measured across the different land uses (Table 1). Rainforest and pasture land uses were characterised by stronger biological and

Table 1
Mean values for soil indicators and *P* value among the four different land uses.

Soil Tool Kit Analyses		Land Use			*P*
Soil indicator	**Rainforest**	**Pasture**	**Sugarcane**	**Banana**	**value**
Physical					
Soil Stability (%)	98.1 a	99.1 a	78.3 b	94.7 a	**< .001**
Bulk Density (g. cm^{-3})	0.65c	0.87 b	0.95 b	1.17 a	**< .001**
Moisture (%)	32.1 a	32.1 a	21.4 b	27.8 b	**0.041**
Chemical					
P ($\mu g.g^{-1}$)	0.19 b	0.13 b	0.31 a	0.38 a	**< .001**
N ($\mu g.g^{-1}$)	1.23 b	1.72 a	1.66 a	1.81 a	**0.002**
pH $_{(1:5\ soil:0.01\ M\ CaCl2)}$	4.36 d	4.82c	5.68 a	5.14 b	**< .001**
Labile carbon (mg. g $soil^{-1}$)	1.09 a	0.83 b	0.47c	0.36c	**< .001**
ACE protein (mg.g $soil^{-1}$)	0.10 a	0.08 b	0.06c	0.06c	**< .001**
Biological					
β-glucosidase ($\mu g.g\ soil^{-1}hr^{-1}$)	44.2 a	25.2 b	11.2 d	13.9c	**0.005**
Basal respiration (% CO_2)	0.68.	0.93.	0.51.	0.83.	0.321

Note: Means within the row followed by the same letter are not different at $P < 0.05$.

physical attributes. For example, in soil under rainforest the primary distinguishing factors were greater labile carbon (1.09 mg.g soil^{-1}), ACE protein (0.10 mg.g soil^{-1}), and β-glucosidase activity (44.2 μg.g soil^{-1}. hr^{-1}), compared to other land uses (Table 1**)**. For pasture soils, greater soil stability (99.1%) and moisture content (32.1%) were the key differentiators.

Conversely, soils under banana and sugarcane land uses were more strongly associated with elevated nutrient status and reduced soil structural or biological functions **(**Table 1**)**. Banana production soils were distinguished primarily by greater nitrogen and phosphorus content (1.81 and 0.38 μg.g^{-1}, respectively), while sugarcane producing soils were characterised by higher pH (5.68), lower soil moisture (21.4%) and soil stability (78.3%) **(**Table 1**)**.

The PCA and radar plot together provided a clearer summary of these land-use differences (Fig. 1). The principal component analysis explained 77.2% of the total variation in land use, from the soil health kit indicators (Fig. 1A). The first principal component (PC1) along the x axis could explain 57.7% of the variation and was made up of biological soil properties but also included lower soil bulk density (Fig. 1**A**). The first principal component largely represented the variation of the rainforest from the banana land uses. Conversely, PC2 explained 19.5% of the variation in land use using soil health kit measures and was composed of soil physical properties as well as respiration (Fig. 1**A**). The second component largely corresponded with the discrimination between pasture and sugarcane soils.

The radar plot, based on the normalised differences in soil indicators, complemented this analysis by showing the overall soil-function profiles of each land use (Fig. 1B). Rainforest and pasture land uses had broader profiles associated with organic matter cycling, moisture, and stability, whereas banana and sugarcane soils showed profiles more strongly associated with nutrient enrichment and greater bulk density (Fig. 1B). The radar plot highlighted the distinct soil property profiles associated with each land use type and underscored the impacts of land use on the different soil indicators. For example, the soil under rainforest had high moisture content and higher organic matter recycling potential based on the high amounts of ACE protein, labile C and β-glucosidase activity while banana production soils have higher N and P content and bulk density.

3.2. Soil health kit measures differentiated ground cover management among banana farms

To further evaluate the sensitivity of soil measures in detecting farm practice-induced differences in soil health, we examined banana production soils with varying amounts of ground cover. The mean values of various soil properties (Table 2) indicated significant differences

Table 2
Mean values for soil properties under different ground management and the *P* value among the three management practices.

Soil Tool Kit Analyses	Ground cover management			
Soil Indicator	**Low (<33%)**	**Medium (33–66%)**	**High (>66%)**	***P* value**
Physical				
Aggregate stability (%)	86.9 b	98.9 a	98.3 a	**0.016**
Bulk Density (g.cm^{-3})	1.18.	1.10.	1.14.	0.415
Moisture (%)	21.4 b	26.9 ab	25.2 a	**0.049**
Chemical				
P (μg.g^{-1})	0.92.	0.80.	0.97.	0.299
N (μg.g^{-1})	2.33 a	2.23 a	2.06 b	**0.018**
pH (1:5 soil:0.01 M CaCl2)	6.02 c	6.29 a	6.11 b	**< .001**
ACE protein (mg.g soil^{-1})	0.03 b	0.04 b	0.06 a	**< .001**
Labile carbon (mg.g soil^{-1})	0.53 b	0.81 a	0.82 a	**0.039**
Biological				
Basal respiration (% CO_2)	0.61.	0.63.	0.65.	0.425
β-glucosidase (μg.g soil^{-1}.hr^{-1})	29.8 b	17.2 b	46.3 a	**0.021**
Laboratory Analyses				
Colwell P (mg.kg^{-1})	228.0 a	191.4 a	78.8 b	**0.013**
Nitrate-N (mg.kg^{-1})	32.0.	27.7.	7.6.	0.358
pH ($CaCl_2$) (0−14)	5.79.	5.67.	5.61.	0.607
Organic Carbon (%)	1.79 b	2.28 b	2.92 a	**0.017**

Note: Means within the row followed by the same letter are not different at $p < 0.05$.

($P < 0.05$) among soils from banana farms with low, medium, and high ground cover. Soils with high and medium ground cover exhibited higher soil stability (98.3% and 98.9%, respectively), moisture content (25.2% and 26.9%, respectively), and labile carbon content (0.82 mg.g soil^{-1}and 0.81 mg.g soil^{-1}, respectively) relative to soils with low ground cover. Additionally, soils with high ground cover demonstrated elevated ACE protein levels (0.06 mg.g^{-1} soil), higher β-glucosidase activity (46.3 μg.g soil^{-1}.hr^{-1}), and increased organic carbon content (2.92%). In contrast, soils with medium ground cover had a less acidic pH (6.29). Soils with low ground cover, on the other hand, showed higher nitrogen content (2.33 μg.g^{-1}), Colwell P (228 mg.kg^{-1}), and had a more acidic pH (6.02) than those with greater ground cover. The soils with low amounts of vegetated ground cover also exhibited the lowest values for soil stability (86.9%), moisture content (21.4%), ACE protein (0.03 mg.g soil^{-1}), labile carbon (0.53 mg.g soil^{-1}), and organic carbon (1.79%) (Table 2). No significant differences were observed in bulk density, phosphorus content, basal respiration, nitrate, or laboratory-measured pH (Table 2).

The PCA and radar plot again highlighted a clearer summary of differences in soil indicators associated with different ground cover in banana fields (Fig. 2). The PCA revealed that 58.3% of the total variation

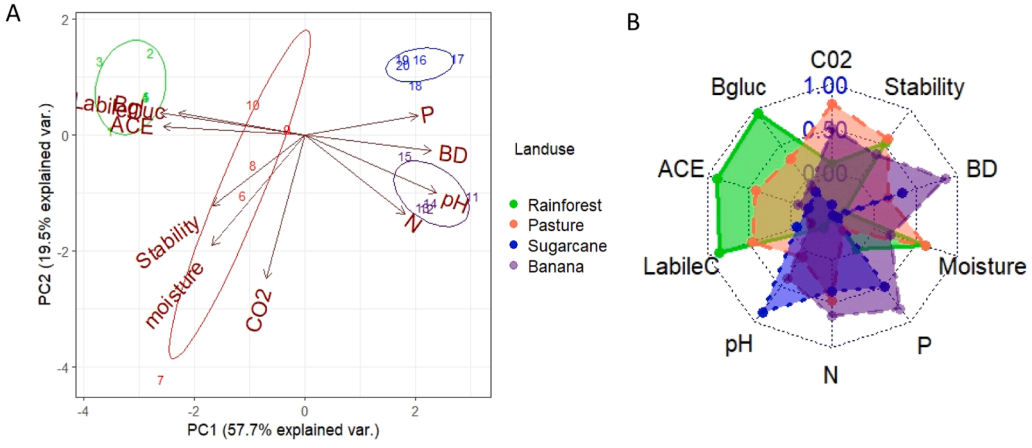

Fig. 1. Variations in soil properties across different land uses detected using the soil tool kit. (A) Principal component analysis (PCA) biplot and (B) normalised radar plot showing the differences in soil health of soils from rainforest, pasture, sugarcane and banana farm.

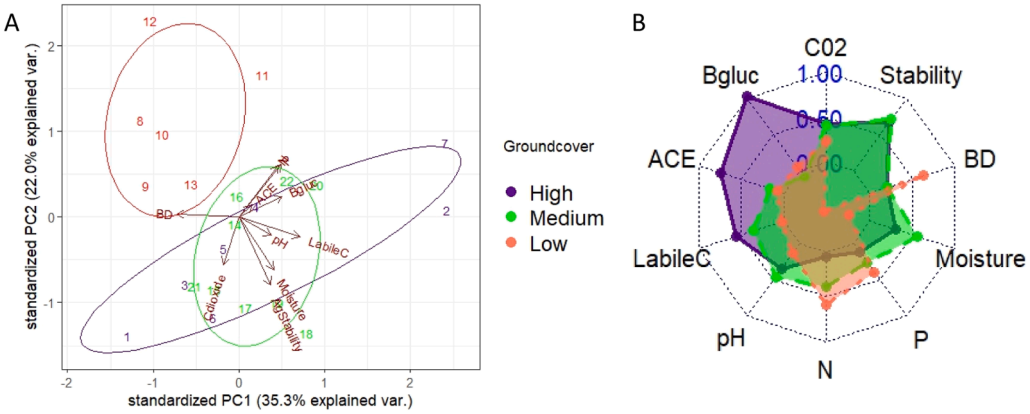

Fig. 2. Management-induced changes detected via the soil properties measured in the soil tool kit. (A) Principal Component Analysis (PCA) biplot and (B) normalised radar plot showing the differences in soil health of banana farm soils with low, medium and high ground covers.

in ground cover density could be predicted from soil health kit indicators. PC1 explained 35.3% of the variation and was primarily influenced by soil properties such as bulk density, labile carbon, pH, ACE protein, and β-glucosidase activity (Fig. 2**A**). In contrast, the second principal component (PC2) accounted for 22% of the variation in ground cover density based on soil health kit measures. PC2 incorporated a combination of soil physical and chemical properties, but also included respiration (Fig. 2**A**). It was particularly effective in distinguishing soils with low ground cover from those with high and medium ground cover.

To gain additional insight into the differences in soil properties related to ground cover density, a radar plot derived from normalised Z-scores was used (Fig. 2**B**). The radar plot demonstrated that physical properties along with the nitrogen and phosphorus content were the key factors that differentiated soils with low ground cover from the soils with high and medium ground cover. On the other hand, biochemical indicators such as β-glucosidase activity, ACE protein and labile carbon, were the key parameters that distinguished soils with high ground cover from those with medium cover (Fig. 2**B**).

To explore associations among the soil properties measured in the soil health kit, Pearson correlation analysis was performed and is presented in Fig. 3. The bulk density was negatively correlated with labile carbon. In contrast, soil stability was positively correlated with moisture content. Similarly, nitrogen content was positively correlated to phosphorus content (Fig. 3).

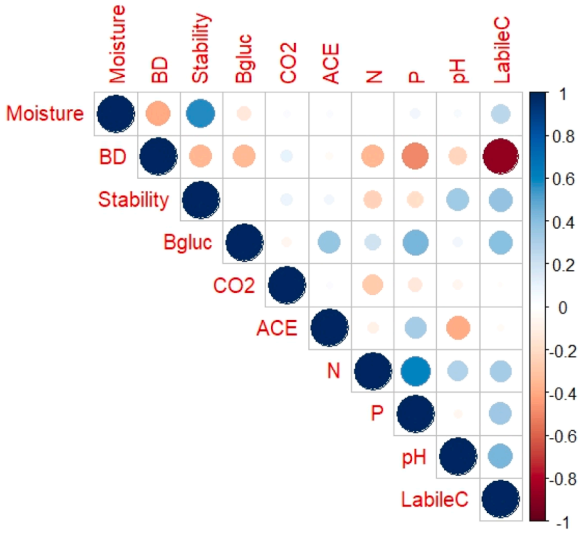

Fig. 3. Correlation matrix among soil health indicators measured from the banana soils with varying ground cover density. The areas of circles show the value of corresponding Pearson correlation coefficients with significance at the 0.05 probability level. Correlation values are indicated by circle; positive correlations are displayed in blue and negative correlations in red. Colour intensity (light to dark) and the size of the circle (small to big) are proportional to the correlation coefficients (0–1 for the positive coefficient and 0 to −1 for negative coefficient) where, for example, the correlation coefficients on the principal diagonal are equal to 1 (it was represented in dark blue and biggest size of circle). The legend on the right side of the correlogram shows Pearson's correlation coefficients with their corresponding colours.

3.3. *Comparison of soil health kit versus standard laboratory measured soil properties*

Across the soils under banana production with varying amounts of ground cover, positive relationships were observed between selected soil properties measured using the soil health kit and the corresponding standard analyses conducted by an accredited commercial laboratory. Notably, the soil health kit P and laboratory measured Colwell P were highly correlated ($R = 0.810$ and $P < 0.001$) (Fig. 4**A** and Table 3). Similarly, a significant correlation was observed between the pH values measured by the soil health kit and those obtained in the laboratory ($R = 0.835$ and $P < 0.001$) (Fig. 4**B** and Table 3). Additionally, the labile carbon determined by the soil health kit was correlated ($R = 0.865$ and $P < 0.001$) (Table 3) with the soil organic carbon determined using the laboratory procedure (Fig. 4**C**). However, although the nitrogen (N) determined from the soil health kit and laboratory measured nitrate nitrogen were still correlated, the Pearson correlation coefficient was relatively lower, at 0.62 (Fig. 4**D** and Table 3).

In addition to analytical comparison with selected laboratory measures, the soil health kit showed cost advantages for use by resource limited laboratories. These estimates were based on consumables only and excluded labour and initial equipment purchase costs, which are reported separately in the supplementary material (Table 4). The estimated up-front equipment cost for the full kit was approximately USD 2753.50, while reagent costs totalled approximately USD 560 for the consumables listed, with many reagents sufficient for more than 100 tests (Tables S2 and S3). On a per-sample basis, the combined consumable cost of the ten-indicator kit was estimated at USD 15.80, compared with approximately USD 336.00 for equivalent conventional laboratory analyses where available (Table 4). Individual kit tests were also low cost, ranging from USD 0.05 for bulk density and moisture to USD 5.00 for N and P. Test times ranged from 5 min for N and P to longer batch-based procedures for moisture, bulk density, ACE protein, and basal respiration, with several assays able to be processed in batches of 6–10 samples. These data indicate that, although some kit components still require bench-based steps and are not field tests, the soil health kit substantially reduces per-sample analytical cost while enabling a broader range of physical, chemical, and biological indicators to be

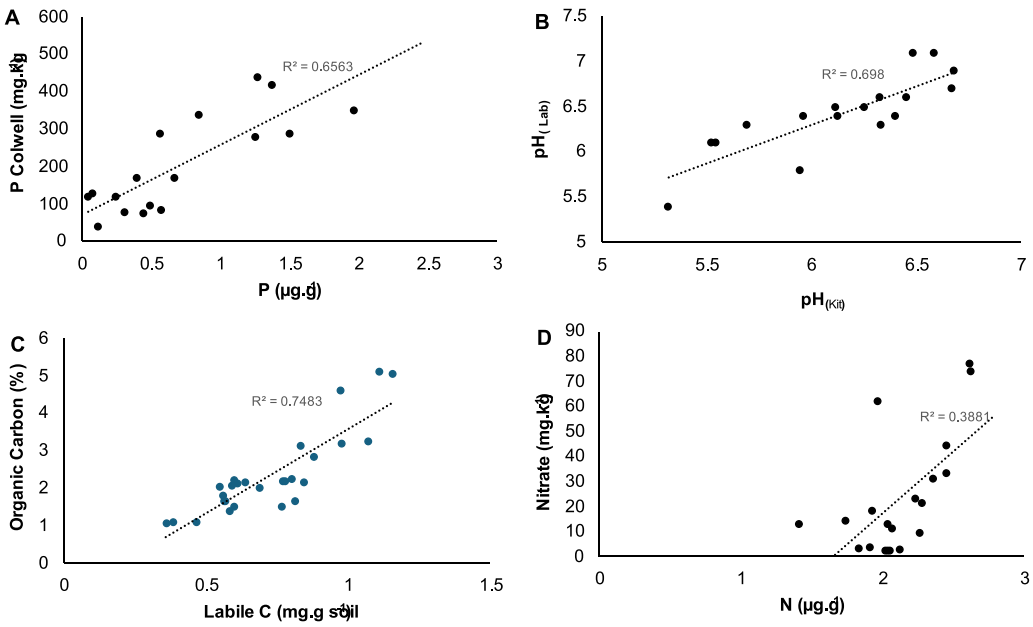

Fig. 4. Regression relationships between selected soil health kit measurements and standard laboratory analyses: (A) soil kit P (µg.g^{-1}) versus laboratory Colwell P (mg.kg^{-1}); (B) soil kit $pH_{(Kit)}$ (1:5 0.01 M $CaCl_2$) versus laboratory $pH_{(Lab)}$ (1:5 soil:0.01 M $CaCl_2$); (C) soil kit labile carbon (mg.g soil^{-1}) versus laboratory organic carbon (%); and (D) soil kit available mineral N (µg.g soil^{-1}) versus laboratory nitrate-N (mg.kg^{-1}). Standard laboratory analyses were conducted by Nutrient Advantage Laboratory, Werribee, Victoria.

Table 3
Pearson's correlation and the *P* value between soil kit test and laboratory test results for selected chemical soil properties.

Soil Kit	Laboratory Analyses	Pearson R Coefficient	*P* value
P (µg.g^{-1})	Colwell P (mg.kg^{-1})	0.810	< 0.001
pH (Kit)	pH (Lab)	0.835	< 0.001
Labile C (mg.g soil^{-1})	Organic Carbon (%)	0.865	< 0.001
N (µg.g^{-1})	Nitrate (mg.kg^{-1})	0.623	< 0.001

measured in laboratories with limited resources.

4. Discussion

This study addressed three questions: whether a low-cost quantitative soil health kit could distinguish soils under contrasting land uses, whether it could detect management-related differences within commercial banana systems, and whether selected kit measurements aligned with standard laboratory analyses. Across these three objectives, the results showed that the kit detected clear land use effects, respond to variation in vegetated ground cover within banana farms, and generate several measurements that were strongly correlated with accredited laboratory results. Together, these findings support the value of the kit as a practical quantitative tool for soil health assessment where access to laboratory infrastructure is limited or where comparative assessment is needed.

By establishing a set of pre-determined criteria, the targeted set of quantitative physical, chemical, and biological soil measurements effectively detected management induced changes in soil health across contrasting land uses and within a single cropping system. By integrating indicators linked to soil structure, nutrient availability, and organic matter cycling, the soil health kit captured functional differences in soil properties that are directly relevant to land management decisions. Importantly, several kit derived measures aligned closely with standard laboratory analyses, supporting the scientific validity of the kit to provide estimates as an accessible soil health tool.

Table 4
Approximate cost and time taken per soil test using the soil kit compared with the estimated costs of equivalent soil tests from a commercial laboratory provider.

Soil Tool Kit Analyses	Approximate cost per test (USD)		
Soil indicator	Soil kit		Conventional laboratory
	Cost*	Time (hh:mm)‡	
Physical			
Soil Stability (%)	$0.20	1:30	$85.00
Bulk Density (g.cm^{-3})	$0.05	24:30‡	$50.00
Moisture (%)	$0.05	24:30‡	$21.00
Chemical			
P (µg.g^{-1})	$5.00	0:05	$21.00
N (µg.g^{-1})	$5.00	0:05	$25.00
pH (1:5 soil:0.01 M CaCl2)	$0.20	1:30	$11.00
Labile carbon (mg.g soil^{-1})	$0.50	0:30	$43.00
ACE protein (mg.g soil^{-1})	$2.00	2:00‡	$30.00⌘
Biological			
β-glucosidase (µg.g soil^{-1}.hr^{-1})	$2.00	1:30‡	Not offered
Basal respiration (% CO_2)	$0.80	24:30‡	$50.00§
Total	**$15.80**		**$336.00**

* These estimates include consumable costs only and exclude labour and initial equipment purchase costs, which are reported separately in the supplementary material. (Table S1) ⁑ Does not include reagent preparation time ‡ Multiple samples can be processed in batches of 6–10 ⌘ Offered by Cornell Service Lab in the USA § Different methodologies available from laboratories for different prices.

4.1. Detecting land use–driven changes in soil properties

A finding from this study was the contrast between rainforest and pasture soils and the more intensively managed land use systems, banana and sugarcane soils. The soil health kit showed that rainforest and pasture soils were associated with stronger biological activity, greater moisture retention and soil structural condition (Table 1; Fig. 1). It was not surprising that rainforest soils had greater biochemical properties such as labile C, β-glucosidase and ACE protein. These properties are

central to organic matter degradation and nutrient recycling which underpin ecosystem resilience and contribute to water retention and soil aggregation (Nunes et al., 2021).

Conversely, soils under banana and sugarcane land uses were more closely associated with higher nutrient concentrations and generally weaker biological indicators (Table 1; Fig. 1). Soils under banana land use had the greatest nitrogen and phosphorus concentrations, and sugarcane soils were associated with higher pH but lower soil moisture and lower stability. The radar plots (Fig. 1B) support this pattern by showing that land uses separated clearly according to combined biological and physical gradients, confirming that the kit captured differences in soil conditions across contrasting land use systems. The radar pattern indicates a chemically enriched yet biologically constrained soils, likely resulting from frequent agrochemical inputs, with a strong dependence on mineral fertiliser (Adingo et al., 2021) and physical disturbance associated with tillage (Sokolowski et al., 2020). While such management practices can enhance short-term nutrient availability for crop production (Zhang et al., 2021), they often diminish organic matter inputs and impair microbially mediated nutrient cycling over longer timescales (Burton et al., 2022).

The ability of the soil health kit to simultaneously capture these contrasting chemical and biological changes highlights its strength as an integrative tool. Rather than focusing on nutrient status alone, the kit reveals how land use alters holistic soil functions, providing additional insights that are often overlooked by conventional soil nutrient testing.

4.2. Sensitivity to management intensity within banana systems

Within banana production systems, vegetated ground cover served as a useful proxy for management intensity, influencing soil biological, chemical, and physical properties. Soils with higher ground cover showed enhanced labile carbon, ACE protein, and β-glucosidase activity, indicating greater organic matter inputs and more active microbial communities. These results align with findings by Adetunji et al. (2020), who reported that cover crops improve soil health by increasing organic matter, and Xu et al. (2024), who observed greater enzyme activity in banana production soils with ground cover relative to bare soils. The heightened biological activity associated with high ground cover suggests a more dynamic and resilient soil microbial community that facilitates nutrient cycling.

Vegetated ground cover also influenced soil physical properties, with increased moisture retention and soil stability observed under medium and high cover. These effects likely result from reduced evaporation (Mohammed, 2013), physical protection of the soil surface, and root-driven aggregation (Blanco-Canqui and Ruis, 2020), which collectively improved soil structure and resistance to erosion. Such improvements have direct implications for water-use efficiency and resilience to extreme weather events, particularly in crops like bananas.

Different ground cover density was also associated with differences in soil chemical properties, reflecting farm management intensity gradients. Soils with low ground cover contained higher nitrogen and phosphorus concentrations and tended to be more acidic. According to Zhang et al. (2022), increased nitrogen availability can lead to soil acidification through nitrification of ammonium fertiliser, a process that accumulates hydrogen ions when nitrate is lost from the soil profile. These chemical differences were consistent with differences in management intensity, for which ground cover was used as a proxy, but farm-specific fertiliser inputs were not quantified in this study.

Overall, the greater biological and biochemical properties found associated with greater vegetated ground cover in banana production soils highlight the importance of retaining vegetation as an important soil health practice in banana. Moreover, the soil health kit used in this research was effective particularly in separating soils with high and low ground cover. Although it was less sensitive in differentiating between high and medium ground cover soils, the soil health kit's ability to detect significant variations supports its value as a practical tool for monitoring soil health in agricultural systems.

4.3. Soil health kit results correlate with standard laboratory measures

Strong correlations between soil health kit measurements and accredited laboratory analyses for pH, phosphorus, and organic carbon confirm that the kit provides reliable quantitative estimates of key soil properties. These relationships indicate that, although simplified, the adapted methods preserve the underlying signal captured by more complex laboratory procedures. The weaker correlation observed for nitrogen reflects known challenges in estimating dynamic nitrogen pools using rapid kit-based approaches and highlights the need to interpret the soil health kit results within their methodological limits. Similarly, the comparison between kit-measured labile carbon and laboratory-measured organic carbon should be interpreted as a relationship between a sensitive active carbon fraction and total organic carbon, rather than as equivalent measures. The soil health kit is therefore best suited as a diagnostic tool for identifying nutrient limitations or potential excesses rather than for developing precise crop nutrient recommendations.

From a practical perspective, the soil health kit offers significant advantages for advisors, extension staff, researchers, and resource-limited laboratories that require convenient, repeatable, and low-cost soil assessment. For applied researchers, the soil kit indicators add scientific validity to the outcomes, ensuring that field-detected trends are not artefacts, but are aligned with established analytical benchmarks, supporting the soil health kit's use as a complementary tool alongside laboratory analyses. For growers, advisors, and applied researchers, these indicators may support comparative on-farm assessment and management discussion. Additionally, the soil health kit is well suited to comparative use, such as tracking management trends over time, supporting demonstration activities, and identifying soils or sites that warrant more detailed laboratory testing. The kit's affordability and use in basic laboratory settings support repeated measurements, enabling users to track trends in soil condition rather than solely relying on infrequent laboratory tests. This is particularly relevant where growers, advisors, or industry programs require quantifiable evidence to support environmental certification claims about how their management decisions impact soil functions and in regions where access to laboratory infrastructure is limited or costly.

A practical strength of the soil health kit was its cost-efficiency relative to conventional laboratory testing. Although the kit requires an initial equipment investment, the estimated consumable cost for the full ten-indicator assessment was only USD 15.80 per sample, compared with approximately USD 336.00 for equivalent laboratory analyses where these services were available. Importantly, several biological indicators included in the kit, such as β-glucosidase, are not routinely offered by standard commercial laboratories, increasing the comparative value of the kit for more holistic soil health assessment. However, the efficiency gains of the kit varied among indicators. Some measures, such as N and P, could be completed rapidly, whereas others still required labour and basic laboratory infrastructure for drying, incubation, centrifugation, or batch processing. The kit should therefore not be considered a fully in-field, rapid test for all parameters, but rather a low-cost quantitative platform that makes broader soil health assessment feasible outside fully equipped analytical laboratories.

While this approach offers clear advantages in accessibility and practicality, it also has limitations that warrant consideration. The soil health kit provides estimates rather than high-precision measurements, and its indicators represent snapshots in time rather than continuous monitoring of soil dynamics. Some measurements, particularly those related to soil moisture and nitrogen availability, may be influenced by short-term environmental variability and may not fully capture complex soil water dynamics and nutrient pools. Additionally, the soil health kit was evaluated within a specific climatic and soil context, and its broader applicability across soil types, crops and regions requires further testing.

Where precise nutrient recommendations, regulatory compliance, or high-precision diagnostics are required, the soil health kit should be used alongside, rather than instead of, accredited laboratory analyses. Recognising these limitations is essential to ensuring that the soil health kit outputs are interpreted appropriately and used to complement, rather than substitute, conventional soil analyses, particularly for nutrients.

Beyond their analytical performance, the results demonstrate that the soil health kit produces readily interpretable outputs that are well suited to demonstration, extension, and near-field assessment settings. The consistent patterns observed across land uses and ground cover treatments indicate that the indicators respond in predictable and functionally meaningful ways to management. For example, indicators of organic matter cycling such as labile carbon, ACE protein and β-glucosidase activity were greater in systems with higher organic inputs, while soil physical indicators such as moisture content and stability reflected differences in soil cover and disturbance, allowing results to be interpreted in terms of underlying soil processes. Because several quantitative measurements can be obtained using relatively accessible procedures and within operationally useful timeframes, the soil health kit is well suited to near-field and basic-laboratory use, enabling soil health concepts to be demonstrated to growers, advisors and land managers, and linked to farm management practices and field observations.

A key strength of the present kit is that it combines physical, chemical, and biological indicators into one low-cost quantitative framework, whereas many soil health assessment approaches remain qualitative or focus on only one aspect of soil condition. Its main limitations are reduced analytical precision compared with accredited laboratories, lower reliability for some dynamic nutrients such as nitrogen, and the need for further validation across a wider range of soils, climates, and production systems. Taken together, these findings demonstrate that the soil health kit effectively captures meaningful variation in soil carbon dynamics, nutrient availability, and general soil condition. The strong agreement with laboratory benchmarks indicates that the kit is both scientifically robust and practically reliable. Although the soil health kit does not replace accredited laboratory analyses, it provides a practical and affordable way to generate quantitative soil health information for screening, demonstration, and routine monitoring, particularly where laboratory access is limited, costly, or slow.

5. Conclusion

The soil health kit presented in this study offers a practical, cost-effective method for monitoring soil health across various land management scenarios. It effectively detected management induced changes, demonstrating differences in soil physical, chemical and biological properties. The soil health kit also detected variability in farm management practices, such as in vegetated ground cover levels in banana farm soils and could provide quantitative data to emphasise the role of ground covers in improving soil health. In particular, soil functions such as organic matter cycling, nutrient availability and soil structural stability could be quantified and monitored, through changes in labile carbon, microbial activity, moisture retention, and soil stability. When compared to traditional laboratory methods, the soil health kit demonstrated its potential as an affordable alternative for soil health monitoring, particularly for assessing soil functions related to organic matter degradation, soil structural stability and nutrient availability and recycling. While useful for monitoring management differences, caution is advised in substituting the soil health kit estimates for established laboratory measurements particularly for developing farm nutrition plans. Overall, we demonstrated that the soil health kit is a valuable diagnostic tool for monitoring and managing soil health, especially in settings where traditional, more expensive soil testing may be less accessible. Its application may support more informed soil management and sustainable agricultural practices in settings where conventional soil testing is less accessible, benefiting advisors, researchers, growers, and the farming environment.

CRediT authorship contribution statement

Hazel L. Gaza: Writing – original draft, Methodology, Investigation, Formal analysis. **Anthony B Pattison:** Writing – review & editing, Project administration, Funding acquisition, Conceptualization.

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Competing interests

The authors declare no competing interests.

Additional information

None.

Declaration of Competing Interest

The authors declare that they have no known competing financial interests or personal relationships that could have appeared to influence the work reported in this paper.

Appendix A. Supporting information

Supplementary data associated with this article can be found in the online version at doi:10.1016/j.soilad.2026.100129.

Data Availability

Data will be made available on request.

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