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Assessment of soil properties and earthworms in organic and conventional farming systems after seven years of dairy farm conversions in New Zealand

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\section*{ABSTRACT}

Environmental degradation and consumer awareness are raising concerns about the sustainability of conventional farming while increasing interest in organic farming as an alternative food and fiber production. Well-replicated studies during the transition are necessary for testing the causes of observed changes. To test soil property changes following conversion, we collected data from 18 dairy farms (nine converting and nine that remained under conventional methods) in the Waikato, Taranaki, and Manawatu regions of New Zealand. Soil properties on the converting group were compared with matched farms that continued with conventional methods. Converting to organic did not result in increased total carbon or nitrogen, but phosphorus decreased by 42%. Bulk density decreased by 3.5\% in converted farms but increased by 9.8\% in conventional farms. Earthworm densities were higher in organic farms but there was no significant change in soil microbial parameters. Total nitrogen was lower where microbial respiration was higher but there was no evidence of a link between earthworms and soil nutrient levels. This observation challenges whether the observed changes in studies of farms that have already converted are indeed caused by organic farming methods themselves. Long-term studies are needed before the broader implications of conversion can be fully assessed.

\section*{Introduction}

The supply of agricultural products is essential to feed an increasing world population. While conventional farming practices have greatly increased global food supply, they undermine the ecosystem services on which agriculture depends (Arvanitoyannis and Giakoundis 2006; Moller et al. 2008; Tilman et al. 2002; Van Calker et al. 2005). Conventional agriculture depends
heavily on continued supply of external inputs (fertilizer, energy, and water inputs) and technology which may threaten biodiversity and ecosystem services (MacLeod and Moller 2006). Dairy farming is particularly intensive and has been blamed for the deteriorating health of agro-ecosystems (Hooda et al. 2000; Houlebrooke et al. 2004; Ribbe et al. 2008). Because of this unsustainability of intensive agriculture, organic farming is increasingly gaining worldwide acceptance (Willer and Kilcher 2009; Willer, Yussefi, and Sorensen 2010). In New Zealand, more dairy farms are converting to organic production (Reider 2007). Despite its increasing popularity, the sustainability and ecological claims of organic agriculture have been challenged (Condron et al. 2000; Leifeld 2012). Thus, there is need for rigorous research to support these assertions. Several studies have compared soils on already converted organic with conventional farms (Araújo et al. 2008; Crittenden et al. 2014; Hathaway-Jenkins et al. 2011; Hole et al. 2005).

To ensure long-term agricultural sustainability, it is critical to maintain and restore soil quality (Lal 2009) in ways that do not compromise environmental integrity (Vitousek et al. 1997). Organic agriculture is based on renewable resources and management of biological and ecological resources aimed at ensuring the long-term preservation of the environment (Reganold and Wachter 2016). It is therefore expected to be less detrimental to the natural resource base than conventional farming (Rigby and Cáceres 2001). This method can offer alternative approaches to improve soil quality (Stockdale et al. 2001) through their positive impacts on soil physical, chemical and biological quality (Allen and Zink 1998; Biederman and Whisenant 2009; Lal 2010). Organic farming methods promote soil biodiversity (Bengtsson, Ahnstrom, and Weibull 2005; Hole et al. 2005) that result in positive effects on soil processes that impact nutrient availability (Bradford et al. 2002; Partsch, Milcu, and Scheu 2006). Therefore, changes in the diversity, abundance, and activity of soil organisms (e.g. earthworms and soil microbes) are expected to be important drivers of change in the soil when farms convert to organic (Wardle et al. 2004).

Individual farms are the key site of action for sustainable farming. Therefore, when discussing and searching for sustainable agricultural practices, it should start from the farm and the farmer who is the manager and sole decision-maker. In 2004 and 2005, Fonterra (New Zealand’s main dairy cooperative) accelerated recruitment of new organic milk suppliers by offering a 7% premium to help through the three-year conversion period before they became fully certified. By using a Before-After-Control-Impact (BACI) study design, we followed soil property changes before and after this incentivized conversion to test whether the act of converting triggers increased soil quality and biota. We sampled 18 farms, nine converting farms matched with nine farms that remained under conventional methods.
Changing to organic farming methods through organic soil inputs may trigger several direct and indirect soil system effects (Allen and Zink 1998; Biederman and Whisenant 2009; Lal 2010). If so, soil nutrient levels, earthworm, and microbial measurements could change as farms shift from conventional to organic management.

- Therefore, we hypothesized that soil nutrient levels will increase in organic farms compared to their conventional counterparts (Hypothesis 1).

Organic farming in which insecticides, herbicides, and inorganic fertilizers are entirely avoided is promoted as a system with reduced environmental impacts (Butler, Vickery, and Norris 2007; Hansen, Alrøe, and Kristensen 2001) that benefits farmland soil biodiversity (Bengtsson, Ahnstrom, and Weibull 2005; Carey, Benge, and Haynes 2009; Gabriel et al. 2013).

- We, therefore, hypothesized that converting to organic will enhance earthworm and microbial density and biomass (Hypothesis 2).

As earthworms and soil microbes have a positive impact on soil quality (Edwards 2004; Perkins 2003; Syers and Springett 1984), we hypothesized that:

- Earthworm and soil microbe measurements will be positively associated with increased nutrient levels (Hypothesis 3).

All these predictions assumed that seven years of conversion are enough for soil nutrient levels and soil biota changes to emerge.

**Materials and methods**

**Study area and selection of study farms**

Study farms were selected from Fonterra’s assisted conversion scheme in the Waikato, Taranaki, and Manawatu regions of North Island, New Zealand (Figure 1). Experimental farms comprised of nine pairs (two nearby farms, of which one was converting to organic and the other a reference farm that continued farming in a conventional way throughout the study). Farms within each pair were between 2 and 10 km apart, had similar landforms, soil type, and climatic conditions. These weeded out disruptive effects of local ecology, and accounted for the high variability of individual farms. The organic farms had just indicated their willingness to convert and were in the process of seeking “organic” certification from either BioGro New Zealand or AsureQuality New Zealand.
Organic is a labeling term used on products produced in accordance with production standards based on minimizing the use of external inputs (synthetic fertilizers, pesticides, genetic modifications, etc.). Farmers voluntarily follow these standards to maintain the integrity of organically produced products. A key driver to these conversions was the initiative by Fonterra to increase their organic milk supply. The certification process starts with the farmer indicating an intention to convert by registering with a certification body. This is followed by a transition stage involving the collection of detailed information and auditing of the farmer’s operations including inputs and outputs. It is only after compliance to the quality standards is verified or when any non-conformities are closed out that the certification certificate is issued. In spite of these, organically produced products must meet the same food safety standards that apply to all other food products. Once certified, these farms neither use nitrogen or superphosphate fertilizers nor do they apply pesticides or medicate animals with antibiotics. All conventional farms used urea (nitrogen) and superphosphate fertilizers during the study period. They also applied antibiotics, synthetic insecticides, and herbicides.

Soils in the Waikato study farms are formed in layers of peat with minor additions of silty volcanic ash. Since they are poorly drained, they require...
maintenance of the drainage system to prevent flooding but care must be taken not to over drain them to preserve the peat resource as they have a low bearing strength. These soils are suited to pastoral farming. In the New Zealand Soil Classification (Hewitt 1998) the soils are classified as Acid Fibric Organic, while in the USDA Soil Taxonomy (Soil Survey Staff 1998), the soils are Hemic Medifibrist.

On the other hand, the soils in the Taraniki/Manawatu study farms are formed from moderately weathered quartzo-feldspathic loess and tephric loess blown from the aggrading beds of rivers. These soils are poorly drained with a moderate permeability in the topsoil and slow permeability in the subsoil. The soils have a high structural vulnerability that requires careful management to maintain soil quality and productive potential. The soils are suited for cropping and grassland farming. In the New Zealand Soil Classification (Hewitt 1998) the soils are classified as Argillic-fragic Perch-gley Pallic, while in the USDA Soil Taxonomy (Soil Survey Staff 1998), the soils are Aeric Kandiaqualf.

**Farm management**

Conversion information on management practices was collected from each farm.

**Fertilizer inputs**

The use of nitrogen and potassium were significantly different between organic and conventional farms. In both cases, the organic farm used less (Figure 2). There was no consistent difference in phosphorus application between the farming systems but it was strongly related to the farms geographical location.

**Stocking rate**

The difference in stocking rate was statistically significant with organic farms having 0.67 cows less per hectare (Figure 3).

**Labor**

Total hours worked to manage each property were averaged out over the 2006/2007 and 2007/2008 seasons. There were no significant differences between organic and conventional in hours worked per 100 ha and the number of hours worked per week per staff member (Figure 4). However, there was a significant difference in staff hours per cow per year ($p = 0.028$).
Before-after-control-impact study design

We designed this experiment based on the “Before-After-Control-Impact” (BACI) strategy (Conquest 2000) to test soil property changes resulting after conversion to organic farming methods. Several studies have compared the impact of organic conversion on neighboring farms (Kitchen et al. 2003; Nguyen, Haynes, and Goh 1995). An important difference in our study is the longitudinal comparison over the conversion period. The design provided an opportunity to test whether the experimental farms changed through time (‘before’ vs. ‘after’) relative to each other. This emphasis on the interaction effect rather than the main effects avoided the complication that each individual farm or group may already have had different soil qualities before conversion took place. This provided a key test of the overall impact of conversion.

Figure 2. The use of N and K were significantly different between organic and conventional farms, with less usage in organic farms. The application of P was strongly related to geographical location but there were no consistent differences between the organic and conventional farms.

Figure 3. There was a significant difference in stocking rate between organic and conventional farms with organic farms having 0.67 cows less per hectare (maximum cows milked per ha).
Selection of study paddocks and ‘soil sampling locations’ (SSLs)

Samples were collected from fixed Soil Sampling Locations (SSLs) in 2005 (the year that half the farms began organic production methods); 2007 and again in 2011. These samples are referred to as ‘Before’ and ‘After’, respectively. Sampling was carried out on the same day (for paired flat farms), or successive days (for paired hill country farms). Within each pair, sampling in the selected farms was stratified to dominant landforms (‘flat’, ‘hill crest’, and ‘slope’) to help control for landscape variation in soil parameters. We extended this theme of farming to the landform in order to achieve a better and more durable match between the environmental attributes of an area of land and its use. We differentiated our landforms on the basis of relief, topographic position and form. This strengthened our ability to compare across the farming systems and to detect trends in successive measurements.

Since farmers manage their farms in separate paddocks (individual farm fields), we selected three paddocks in flat farms and three paddocks within each of the two most extensive ‘landforms’ (flat, slope, or crest) occurring in each pair of hilly farms. Thus, we sampled three paddocks in each flat farm and six paddocks in each farm with hilly conditions in order to account for the high variability of individual farms. Paddocks that exhibited unusual land uses (e.g. airstrips and silage paddocks) were excluded. Three random ‘Soil Sampling Locations’ (SSLs) were positioned within each focal paddock using a table of random numbers to select grid coordinates, all of which met the following criteria: ≥30 m from nearest neighbor coordinates, trees, fences, gateways, and water troughs. These restrictions sought to minimize variance between samples and avoid areas where stock congregate. Within each selected paddock, three SSLs were randomly selected for actual soil sampling and their GPS locations recorded for subsequent sampling.
Soil and earthworm sampling

From each SSL, we collected three types of samples. First, two sets each consisting of 10 sample cores each of 7.5 cm deep and 2.5 cm wide were collected. One set was used for soil chemical analysis, and the other for microbial assays. Secondly, a soil sample of 15 cm deep was collected using a 7.5 cm diameter cylindrical corer for bulk density measurement. Finally, we extracted earthworms from a 20 cm × 20 cm × 20 cm soil layer cut using a spade; thus, results were expressed as earthworm density (individuals m$^{-2}$) and biomass (g m$^{-2}$). Earthworms were searched by sorting and crumbling the soil matrix by hand (Edwards and Lofty 1977), followed by separation of the collections and determination of species identities. Separate pooled samples per species were weighed using an electronic balance accurate to 0.1 g.

Laboratory analyses

In the laboratory, one set of the two samples was used to measure soil biological activity, thus microbial biomass carbon and nitrogen by the irradiation and incubation methods (Schinner et al. 1995). Microbial biomass was measured on 40 g dry weight sub-samples that were irradiated on a microwave for 5 min. The irradiated and non-irradiated samples were incubated for 7 days at 25°C and 60% water holding capacity. We then determined the CO$_2$ held by NAOH titrimetrically. An efficiency coefficient of 0.45 was used to convert the CO$_2$ difference between the irradiated and non-irradiated microbial biomass C. The results for carbon biomass were expressed as mg C per kg of soil and that of nitrogen as mg N per kg of soil. Microbial respiration was obtained from sieved soil sample (100 g) packed to a bulk density of 1 g/ml wetted to a soil moisture of 60% of field capacity and then incubated for 7 days at a temperature of 22°C (Parkin, Doran, and Franco-Vizcaíno 1996). Glass vials holding 10 ml of NaOH were used to trap CO$_2$ from the samples which was then determined titrimetrically. The results were expressed in mg CO$_2$ per kg of soil per minute. Microbial biomass is a measure of the total amount of living microbes in the soil while microbial respiration is the process that reflects the potential activity of the soil microbial population.

Each of the other sample set was air dried, ground, and then sieved through a 1 mm diameter sieve, followed by soil chemical analysis as described by Carter and Gregorich (2007). Total carbon and nitrogen were determined using an Elementar Vario Carbon Nitrogen Sulphur (CNS) analyzer manufactured by Elementar Analysensysteme GmbH (Germany). Here, the sample is passed through a heated copper catalyst which converts the various forms of nitrogen to N$_2$ that is then measured by a thermal conductivity detector. At the same time, the CO$_2$ produced from the sample is measured in an infrared detector cell. For New Zealand soils with a pH lower than 7, the free carbonate content is
negligible (Miller 1968) and therefore the total carbon content obtained was taken as the total carbon content of the soil. A filtrate of soil and 0.001 M sulfuric acid buffered to pH 3.0 with ammonium sulfate at a temperature of 20°C was analyzed for phosphates on a Flow Injection Analysis (FIStar™ 5000 Analyser).

Bulk density samples were oven dried at 105°C for 24 h and then bulk density calculated from the equation:

\[
\text{Bulk density} \left( \frac{g}{cm^3} \right) = \frac{\text{Oven dry weight (g)}}{\text{Core volume (cm}^3)}
\]  

We modified the equation to account for course fragments in each collected sample by the following:

The volumes of course fragments contained in each sample were calculated by submerging them under water and measuring the volume of the displaced water. Bulk volume density of these rock fragments was then calculated. The fine earth volume was obtained by subtracting the bulk volume of coarse fragments in each sample from sample volume. These measurements were used to calculate the percentage gravel by volume by converting gravel mass to bulk volume using coarse fragment bulk density. Finally, bulk density values accounting for gravel are obtained from Equation 1 below, as described by Vincent and Chadwick (1994). These values were then used to calculate nutrient levels per unit area (Mehlich 1972).

\[
\text{MT/}(V_{bk} \leq 2 + \sum V_{bkv} \geq 2)
\]

Where:

- \(MT\) = Total whole-soil mass (calculated by fine earth/percent total mass from fine earth)
- \(V_{bk} \leq 2\) = Bulk volume of fines, obtained by subtracting the bulk volume of coarse fragments in each sample from sample volume
- \(\sum V_{bkv} \geq 2\) = Sum for all sieve sizes \(\geq\) the volume associated with the gravel mass divided by measured rock fragment bulk density for each sample.

This bulk density was used to calculate soil nutrient amounts per unit volume of the soil sample measured, rather than per g of soil. First, we calculated the gravimetric nutrient value:

\[
\text{Nutrient} \left( \frac{\text{Weight}}{\text{g}} \right) = \frac{\text{Dry weight (g)}}{100}xg
\]

Where:

- \(\text{weight}\) Is the dry weight of soil sub-sample used in nutrient extraction
- \(\text{Dry weight}\) Is the weight of the extracted nutrient in g (the nutrients are normally given in grams hence the g in front of the equation)
The amount of nutrient in the field bulk density surface area and depth was calculated from the formula:

$$ Nutrient \left( \frac{kgNt}{M2} \right) = A \times \text{Sampling depth} \times \text{Bulk density} \times \text{Nutrientlab} $$(4)

where:

- $A$ is the base area of the sampling corer (m$^2$)
- Sampling depth – is the depth of sampled soil in this study it was 0.075 m
- NutrientLab – is the laboratory test result expressing the weight of nutrient extracted from a known weight of soil sample.

**Statistical analysis**

Differences in soil properties were analyzed using Generalised Linear Mixed Models in GenStat™ for Windows (release 16) statistical software. These GLM Models used the Residual Maximum Likelihood (REML) method. The REML used a fixed model that incorporated (i) Treatment (Organic versus Conventional), (ii) Time (Before verses After) and (iii) Landform (slope/crest/flat). An interaction between treatment and time was included when comparing the interaction effects. To account for the lack of independence and the hierarchical nature of the sampling, random effects were always nested as Pair/Farm/Paddock/SSL within the REML models. Preliminary models were constructed and residuals inspected to check for heteroscedasticity and to ensure that the residuals were distributed evenly around the predicted means. The significances of predictor variables were assessed by Wald’s tests.

For robust analysis, increasingly severe transformations (untransformed < square root < log$_e$ < log$_{10}$) were applied to response variables to find the simplest model with the best residuals and fit to model assumptions. Where transformed data produced the best residuals, predicted transformed data and confidence intervals were back transformed for reporting but the $p$-values reflect the tests done on the transformed data. For continuous measures rather than discrete counts (not normally distributed) such as earthworm biomass, we used arcsine transformations (Sokal and Rohlf 1981) to normalize variances within REML models by incorporating the usual blocking structure. Predicted arcsine means and confidence intervals were back-transformed, but the $p$-values reflect the tests performed with arcsine transformed data. To
determine whether earthworms and soil microbes can predict nutrient levels, we formulated additional models that incorporated earthworm and microbial measurements. Results are presented as means ±2× SE (standard error of differences).

**Results**

**Changes in soil bulk density after conversion**

Soil bulk density decreased by −3.5% over the 7 years since conversion to organic but increased by 9.8% on the farms that remained under conventional management. The two systems had similar bulk density before half the farms converted (Table 1; Figure 5). The noticeable shift in bulk density change and the highly significant interaction effect ($p = 0.006$) raises important questions especially in cases with huge bulk density differences: (a) is it most appropriate to measure the soil nutrient values according to soil volume (i.e. the nutrients contained in the top 7.5 cm of a metre square of paddock; or (b) should nutrients be expressed per weight of soil removed from the top 7.5 cm.

This change can come when soil becomes more structured thereby encapsulating more air and/or water between the soil particles. The soil structure and density potentially affect the pasture plants’ root envelope volume, which in turn affects the amount of nutrient available for uptake. This raises the issue of how best to express changes in soil quality after farm conversion. Using a nutrient per unit area of paddock (‘volumetric’) measure takes into account the added spaces for air and water within the soil profile, whereas expressing the nutrient weight per unit soil weight (the ‘gravimetric method’) excludes consideration of water and/or aeration changes. Here we used the volumetric method where air and water between the soil particles is taken into consideration).

**Effects of landform**

The GLMMs predicting soil property metrics from the experiment also included landform as an explanatory variable. This was necessary because stratification across landforms was built into the experimental design since the position of a SSL (on a hill crest, mid-slope or flat) is likely to influence soil properties. Our modeling confirmed this expectation (final column of Tables 1 and 5). However, our study was interested in important soil properties and biota triggered by conversion to organics. Therefore, we included landform as a fixed effect to minimize the unexplained variance in the whole model and thereby enable a more powerful test of farm system conversion.
<table>
<thead>
<tr>
<th>Soil property</th>
<th>Transformation used</th>
<th>Conventional Before</th>
<th>Conventional se</th>
<th>Conventional After</th>
<th>Conventional se</th>
<th>Organic Before</th>
<th>Organic se</th>
<th>Organic After</th>
<th>Organic se</th>
<th>System</th>
<th>Time</th>
<th>BACI interaction</th>
<th>Landform</th>
</tr>
</thead>
<tbody>
<tr>
<td>Total C (kg C m$^{-2}$)</td>
<td>Square root</td>
<td>4.62</td>
<td>0.21</td>
<td>5.00</td>
<td>0.23</td>
<td>4.97</td>
<td>0.22</td>
<td>4.83</td>
<td>0.24</td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Total N (kg N m$^{-2}$)</td>
<td>untransformed</td>
<td>0.46</td>
<td>0.02</td>
<td>0.49</td>
<td>0.01</td>
<td>0.46</td>
<td>0.02</td>
<td>0.45</td>
<td>0.02</td>
<td>**</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>P (mg P m$^{-2}$)</td>
<td>untransformed</td>
<td>2733</td>
<td>143</td>
<td>2287</td>
<td>141</td>
<td>2308</td>
<td>140</td>
<td>1339</td>
<td>138</td>
<td>***</td>
<td>**</td>
<td>***</td>
<td></td>
</tr>
<tr>
<td>Bulk density (g cm$^{-3}$)</td>
<td>Untransformed</td>
<td>0.685</td>
<td>0.023</td>
<td>0.753</td>
<td>0.015</td>
<td>0.703</td>
<td>0.031</td>
<td>0.677</td>
<td>0.013</td>
<td>***</td>
<td>***</td>
<td>***</td>
<td></td>
</tr>
</tbody>
</table>

Table 1. Soil total C, total N, P and bulk density values from the interaction model between system and time [Before – After – Control – Impact – (BACI)] showing differences and standard errors (SE) before and after half farms converted to organic farming methods. Soil property measurements in this analysis are based on nutrient per m$^2$ of soil. The carbon values were back transformed for reporting. Significant differences are: ***,$p < 0.001$; **,$p < 0.01$ and *,$p < 0.05$. 
Did conversion to organic cause changes in soil total C, N and P? (Hypothesis 1)

There was no evidence of changes in total C and total N in the before and after samples (Tables 1 and 3). Phosphorus was the only measure with a significant BACI interaction effect though the levels were already lower on organic farms before formal processes to organic conversion was decided. These levels fell to a greater degree in the organic farms in the after sample (Table 2; Figure 6). Phosphorus also exhibited a large shift through time even within the conventional farms where no systemic change in farming occurred (Table 2; Figure 6).

Did conversion to organics cause changes in earthworms? (Hypothesis 2)

Eight earthworm species were recorded before conversion; Aporrectodea longa (Ude, 1885), Aporrectodea calignosa (Savigny, 1826), Aporrectodea rosea (Savigny, 1826), Octolasion cyaneum (Savigny, 1826), Lumbricus rubellus (Hoffmeister, 1843), Amynthas diffringens (Baird, 1869), Lumbricus terrestris (Linnaeus, 1758) and a native species that was not identified. After conversion, the number of observed species reduced to 4, thus A. caliginosa, A. longa, O. cyaneum, and L. rubellus. This created uncertainty about the reliability of earthworm identifications in the before sample (the before and
Table 2. Soil property values and standard errors (SE) from the main effect of time (before verses after) and farming system (conventional verses organic) model. Soil property measurements in this analysis are based on nutrient per m$^2$ of soil. C values were back transformed for reporting.

<table>
<thead>
<tr>
<th>Soil property</th>
<th>Transformation used</th>
<th>Value from the main effect of time</th>
<th>Value from the main effect of farming system</th>
</tr>
</thead>
<tbody>
<tr>
<td></td>
<td></td>
<td>Before</td>
<td>se</td>
</tr>
<tr>
<td>Total C (kg C m$^{-2}$)</td>
<td>Square root</td>
<td>4.80</td>
<td>0.14</td>
</tr>
<tr>
<td>Tot N (kg N m$^{-2}$)</td>
<td>Untransformed</td>
<td>0.46</td>
<td>0.01</td>
</tr>
<tr>
<td>P (mg P m$^{-2}$)</td>
<td>Untransformed</td>
<td>2510</td>
<td>197</td>
</tr>
</tbody>
</table>

The $p$-values determine whether before values differed significantly from the after ones, or between the different farming systems. Significant differences are: $***$, $p < 0.001$. 
Table 3. Earthworm mean estimates, standard errors (SE) and \( p \)-values from the interaction model between system and time (Before – After – Control – Impact – (BACI)) showing the differences in earthworm measurements between organic and conventional systems before and after half of sampled farms converted to organic. Earthworm biomass analysis was done on arcsine transformed data which was back transformed for reporting.

<table>
<thead>
<tr>
<th>Earthworm measurement</th>
<th>Conventional</th>
<th>Organic</th>
<th>Predictor ( p )-value</th>
</tr>
</thead>
<tbody>
<tr>
<td></td>
<td>Before se</td>
<td>After se</td>
<td>Before se</td>
</tr>
<tr>
<td>Total EW density (earthworm m(^{-2}))</td>
<td>195 22 431 24</td>
<td>411 23 468 25</td>
<td>* *** **</td>
</tr>
<tr>
<td>Total EW biomass (g m(^{-2}))</td>
<td>148 17 198 18</td>
<td>207 22 198 19</td>
<td>* *** **</td>
</tr>
<tr>
<td>Anecic +endogeic (earthworms m(^{-2}))</td>
<td>219 39 399 43</td>
<td>269 41 412 52</td>
<td>***</td>
</tr>
<tr>
<td>Epigeic (earthworms m(^{-2}))</td>
<td>80 21 62 19</td>
<td>160 24 57 17</td>
<td>* *** **</td>
</tr>
</tbody>
</table>

Significant differences are: ***, \( p < 0.001 \); **, \( p < 0.01 \) and *, \( p < 0.05 \).
after samples were identified by different people, and we double checked the identifications in the after samples. We, therefore, based our analysis of earthworm data according to broad ecological functional groups, and pooled counts for anecic and endogeic earthworms to compare with epigeic species. Nevertheless, A. caliginosa was dominant and abundant in both farming systems before and after conversion.

Detection of mean differences in earthworms between the farming systems was statistically challenging because their density was variable, ranging from no individuals to 1,052 individuals m\(^{-2}\) in a single SSL. The maximum biomass recorded at a single SSL was 621.6 g/m\(^2\). There were statistically significant BACI interaction effects in total earthworm density and in the abundance of epigeic worms in particular (Table 3). This observation resulted from more earthworms in farms about to become organic in the ‘Before’ samples, and a rapid rise in earthworms (anecic and endogeics) on the conventional farms by the time of the after samples (Table 3). Epigeic worms were nearly twice as abundant on organic farms before formal conversion, but then decreased significantly by the time of the after sample. More earthworms were found on organic farms when main effects are considered, which is consistent with Hypothesis 2. However, the direction of the relative shifts in abundance after formal conversion to organics was in the opposite direction, which led to the rejection of Hypothesis 2 for earthworms.

There was no evidence of changes in microbial measures within the BACI treatments (Table 4), or when considering the main effect of conversion, so Hypothesis 2 had to be rejected as far as microbial measures are concerned.
Table 4. Soil microbe means, standard errors (SE) and p-values from the interaction model between system and time [Before – After – Control – Impact – (BACI)] showing the differences in microbial measurements between organic and conventional systems before and after half of sampled farms converted to organic. Microbial biomass C analysis was done on square root transformed data which was back transformed for reporting.

<table>
<thead>
<tr>
<th>Microbial measure</th>
<th>Conventional</th>
<th>Organic</th>
<th>Predictor p-values</th>
</tr>
</thead>
<tbody>
<tr>
<td></td>
<td>Before</td>
<td>se</td>
<td>After</td>
</tr>
<tr>
<td>Microbial Biomass C (mg C kg(^{-1}) soil)</td>
<td>1098</td>
<td>101</td>
<td>1088</td>
</tr>
<tr>
<td>Microbial Respiration (mg CO(_2) kg(^{-1}) soil min(^{-1}))</td>
<td>29.43</td>
<td>2.87</td>
<td>94.07</td>
</tr>
<tr>
<td>Microbial Biomass N (mg N kg(^{-1}) soil)</td>
<td>184.6</td>
<td>14.74</td>
<td>178.9</td>
</tr>
</tbody>
</table>

Significant differences are: ***, p < 0.001; and *, p < 0.05.
Is soil quality higher where there are higher earthworm and soil microbe measures? (Hypothesis 3)

More complex models were built to explore the same soil response variables to the BACI experiment but with earthworm and microbial variables as additional predictors. There was no evidence that soil nutrient levels could be predicted by earthworm measurements (Table 5). When the interaction between farming method and time since conversion were considered, total nitrogen was lower where microbial respiration was higher (Table 5). Because of the limited evidence of relationships between changes in soil biota and soil chemical properties, we concluded that changes in soil biota were insufficient to account for observable changes in soil chemical properties and therefore Hypothesis 3 was rejected.

Discussion and conclusion

Changes in soil nutrient levels following conversion to organic

Tests of Hypothesis 1 did not provide evidence of C and N changes while phosphorus reduced in converting farms. Only soil bulk density changed with conversion in the way predicted and therefore Hypothesis 1 had to be rejected. The lack of positive change in total C and N in this study is consistent with Parras-Alcántara et al. (2014) but contrasts with many other studies that record higher C and N levels in organically managed soils (Clark et al. 1998; Mäder et al. 2002; Melero et al. 2006; Stockdale et al. 2001). This observation may have been caused by our measuring only the top 7.5 cm where rapid adjustments of soil are added rather than deeper in the soil profile where nutrients accumulate and management induced nutrient changes occur (Lorenz and Lal 2005). A more comprehensive study of soil changes in successively deeper soil strata is needed. Similarly, the duration of our study (seven years) may have been too short for detectable changes. Alternatively, practices in New Zealand dairy farming may have prevented or slowed the emergence of soil C and N changes that are normally seen elsewhere (Hathaway-Jenkins et al. 2011; Parras-Alcántara, Díaz-Jaimes, and Lozano-García 2015). For example, New Zealand soils under long term pastoral land use may already be at equilibrium and therefore not possible to store more soil carbon (Tate et al. 1997).

Experimental effects of farm conversion in soil bulk density and P were detected. Phosphorus was already higher on conventional farms before conversion. The lower phosphorus found in organic farms is consistent with findings in other studies (Carey, Benge, and Haynes 2009; Løes and Øgaard 2001). Although nutrient budgets in organic systems show lower requirements for P (Nguyen, Haynes, and Goh 1995; Oehl et al. 2002), these drops may
Table 5. Coefficient values for earthworm and microbial explanatory variables predicting soil properties from the interaction of the GLM models that were used to predict soil property estimates to show the relationship between earthworm and microbial measurements and observed values. Soil property measurements in this analysis are based on nutrient per m² of soil.

<table>
<thead>
<tr>
<th>Soil Property</th>
<th>Conventional</th>
<th>Organic</th>
<th>BACI p-value</th>
<th>Epigeics</th>
<th>Anecic + Endogeic</th>
<th>EW Biomass</th>
<th>Microbial Biomass C</th>
<th>Microbial Respiration</th>
</tr>
</thead>
<tbody>
<tr>
<td>Total C</td>
<td>4.62</td>
<td>5.00</td>
<td>4.97</td>
<td>4.83</td>
<td>-0.00103</td>
<td>0.00012</td>
<td>0.02435</td>
<td>-0.00087</td>
</tr>
<tr>
<td>Total N</td>
<td>0.46</td>
<td>0.49</td>
<td>0.46</td>
<td>0.45</td>
<td>-0.00003</td>
<td>-0.00001</td>
<td>0.00089</td>
<td><strong>0.0017</strong>*</td>
</tr>
<tr>
<td>P</td>
<td>2733</td>
<td>2287</td>
<td>2308</td>
<td>1339</td>
<td><strong>1.00035</strong></td>
<td>-1.00001</td>
<td>-32.6100</td>
<td>0.36060</td>
</tr>
<tr>
<td>BD</td>
<td>0.70</td>
<td>0.74</td>
<td>0.70</td>
<td>0.68</td>
<td><strong>-0.00001</strong></td>
<td>-0.00005</td>
<td>0.00444</td>
<td>-0.00002</td>
</tr>
</tbody>
</table>

The significant relationship term is: *** p < 0.001; ** p < 0.01.
undermine sustainable production. Strict organic certification requirements that prohibit applications of superphosphate fertilizer may have caused this observation.

The reduction in bulk density in converted farms suggest that conversion to organics trigger a building of lighter more structured soil. This observation may have resulted from changes in the quality and quantity of organic inputs. In contrast, nitrogen fertilization may have caused increased bulk density on the conventional farms by reduced soil aggregation and binding SOM micro-aggregates (Mikha and Rice 2004). Reduced bulk density is likely to have far-reaching and potentially very important effects. It can offer the plant enhanced opportunities for nutrient absorption through increased water infiltration and retention, aeration and root growth. Water storage can enhance drought resistance and accelerate grass growth, which in turn might trigger many changes in rates of nutrient cycling and soil biota.

**Changes in soil biota following conversion to organics**

Organic farming has been shown to have a generally positive influence on soil biota abundance, biomass and diversity (Birkhofer et al. 2008; Hansen, Alrøe, and Kristensen 2001; Mäder et al. 2002). However, in this study, earthworms and soil microbes were not enhanced by conversion to organic. Earthworms were more abundant and reached higher biomass on organic farms before they formally set a strategy to go organic. Whatever caused the higher earthworm abundance on organic farms in the before sample may in some way be associated with organic farming practices. This might have been due to stocking rate, soil fertilization, grazing, pasture management and the overall farming philosophy being applied on the farms that are associated with a subsequent decision to formally decide to convert to organic.

Why then did the abundance of endogeic and anecic earthworms rise so rapidly on conventional farms between before and after samples, yet remain about the same in organic farms? And why did epigeic earthworms decline in the organic farms by the time of the after sample, despite being much more abundant before conversion? It may be that earthworm populations are generally more resilient on organic farms (Bengtsson, Ahnstrom, and Weibull 2005; Birkhofer et al. 2008; Cabell and Oelofse 2012; Paoletti 1999) i.e. some factors like drought or toxic shocks from fertilizers may periodically knock-down earthworms, but they resurge in abundance more rapidly in organic farms. This potential interpretation exposes the main weakness of this study – that only two samples of earthworms are available for analysis. We also lack a basic long term population dynamics study of fluctuations of earthworms in New Zealand and clear evidence of what affects their survival, reproduction, growth and community structure. We cannot expect overseas studies to necessarily apply in New Zealand because the species here are all introduced. Introduced
species may behave differently in New Zealand’s ecosystems than in their home country (Moller et al. 2005; Perley et al. 2001). Also, earthworm species diversity is extremely reduced in New Zealand compared to elsewhere (Curry et al. 2008; Manono and Moller 2015; Muldowney et al. 2003).

Although an increase in microbial biomass and respiration were expected in converting farms, these increases were not observed. This contrasted evidence from literature indicating increased microbial biomass and activity in organic systems (Carey, Benge, and Haynes 2009; Fließbach and Mäder 2000; Glover, Reganold, and Andrews 2000). The higher microbial respiration in the after samples on conventional farms suggest that any form of fertilizer addition to soil can affect microbial biomass and enhance their activity. This scenario has also been reported in other studies (Böhme and Böhme 2006; Shannon, Sen, and Johnson 2002). Therefore, the higher respiration rates observed on conventional farms may have been caused by higher metabolic rates of the microbial decomposer community.

**Relationships between soil quality and soil biota**

The lack of relationships between microbial measures and soil total carbon in the present study was unexpected. The lower total nitrogen levels where microbial respiration was higher may be attributed to microbial driven nitrogen losses from agricultural land. This may be the case since soil microbes regulate N cycling (de Vries and Bardgett 2012). Unlike soil microbes, there were no associations between soil properties and earthworms. This contrasted the generally accepted claim that earthworms drive soil structure and quality (Brussaard, de Ruiter, and Brown 2007; Fonte et al. 2007; Irmler 2010; Paoletti 1999), but is consistent with another study (Manono, Moller, and Morgan 2016).

The lack of positive correlations between earthworm and microbial measures and soil properties suggest that interactions between other soil biotic and abiotic components may have developed several feedback disruptions that limited these relationships. These factors may have influenced soil nutrient levels independent of the conversion progress. Earthworm and microbial presence in all treatments may have posed problems for detecting these relationships. Extreme treatments, such as experiments with and without earthworms/microbes or microcosm experiments containing a known number of individual worms or known biomasses would be ideal in such a comparison. However, we interpret the association found between microbial respiration and total nitrogen to indicate a loose and potentially indirect coupling of soil biota and soil abiotic properties. This post hoc interpretation requires follow-up research to better ascertain soil biota associations with soil chemical and physical property measurements, but in the meantime, Hypothesis 3 is formally rejected.
It is important to note that our statistical models did not prove or disprove causal linkage: they just tested covariance (association) between variables that might have come about by a variety of indirect and deeply correlated features of soil ecosystems. Perhaps the absence of these relationships in this study maybe partly related to the shallow depth at which soil for laboratory analysis was collected. Nutrient changes resulting from land management can occur deeper in the soil profile (Lorenz and Lal 2005).

In New Zealand, management impacts on earthworm communities at the field level are well documented (Fraser, Williams, and Haynes 1996; Manono 2014; Manono and Moller 2015; Schon et al. 2008). Their large size and limited movements make them easy to capture and sort and therefore attractive as potential tools for farmers to use as indicators of soil quality. The challenge is to determine how land use changes impact soil properties commensurate with earthworm and microbial community changes. If the effects of land-use change associated with soil properties and earthworm measurements are established, soil quality indicators can be developed that farmers can readily manage and monitor to assess and mitigate negative management impacts.

**Methodological considerations**

The results of this study should be treated with caution as individual farmers made personal decisions on whether to convert or not. The study experimental farms were scattered in a large geographical area of New Zealand covering several soil types. Although this was advantageous in increasing the study’s applicability and zone of inference to cover the full range in landforms, soils, climate, and regions where New Zealand dairying occurs, this spread potentially undermines the power to detect experimental effects. The role of farmer orientations, knowledge, and decisions concerning soil nutrient management are just as important in determining overall outcomes as the fine scale ecological and biophysical consequences of adopting organic inputs. Inter-annual fluctuations are obviously operating, potentially as a result of recent soil fertilization, soil temperature, rainfall, paddock or stock management that is independent of whether or not conversion to organics had occurred. The power to detect experimental effects will have been much reduced by this inter-annual variability, and more repeated samples are needed to fully control these variations.

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References


