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Effects of Biodynamic and Organic Garden Treatments on Crop Yield, Soil Properties, and Microarthropod Communities

Alexx Diera
Environmental Studies
The University of North Carolina Asheville
One University Heights
Asheville, North Carolina 28804 USA

Faculty Advisor: Dr. Kevin K. Moorhead

Abstract

Biodynamic agriculture is one organic farming system that has emerged as an alternative response to the high-chemical input farming systems commonly practiced today. Previous research has presented clear benefits of biodynamics on soil and biological health, but few comparisons have been drawn between biodynamic and low-impact organic systems. This study tested for differences between these two systems in a design that compared the growth of three crops (beans, carrots, and chard) in two adjacent garden beds. Crop yield was predicted to be higher in the biodynamic bed, and significant differences among vegetative, soil, and faunal indices were anticipated between the two treatments. The results of this study did not support either of these hypotheses, and low-impact organic farming demonstrated parallel competence to the improvement of soil quality and biological activity as much as the biodynamic treatment. Future investigations into effects on crop productivity, soil stabilization, and food web dynamics would enrich comparisons between biodynamic and organic agriculture.

1. Introduction

The widespread use of controversial synthetic fertilizers, herbicides, and pesticides in modern agriculture has driven growing demand for alternative farm management practices worldwide[1]. Such demand in the United States has prompted the National Organic Program to eliminate the use of synthetic chemicals on organically certified food products, but for many farmers such an initiative is insufficient towards improving food quality[2]. Proponents of more sustainable agricultural practices aim to establish systems integrating natural components of a farm while minimizing environmental degradation and eliminating the health risk of chemicals used in conventional agriculture[2].

Biodynamic (BD) agriculture is one of these alternative systems practiced around the world[3]. Biodynamics is a holistic approach to food production originally conceived by Austrian philosopher Rudolf Steiner in 1924 via a series of lectures entitled *Spiritual Foundations for the Renewal of Agriculture*[4]. Biodynamics incorporates cosmic elements of the universe with organic farming techniques[1]. Biodynamic practitioners perceive their farms as organisms[1-2], seeking to improve the nutritional and spiritual health of all its elements, with a primary focus on improving soil quality and plant life[4].

Steiner prescribed a collection of preparations to be added to soil, plants, or compost, which he claimed would enhance the life forces working within these entities. BD 500 preparation is applied directly to soil to enhance root growth and humus formation. The preparation is made from a fermented manure-filled cow horn that has been buried underground over the fall and winter months. The BD 501 preparation is applied as a foliar spray directly to plants to enhance and regulate their growth. Powdered quartz is stuffed into a cow horn and buried over spring and

summer to make the preparation. BD 502-507 preparations are used to enhance compost production, with each preparation made with specific combinations of plant and animal components[1].

Because biodynamics inherently links organic farming to metaphysical forces, the biodynamic and scientific communities are often at odds with one another[5-6]. Steiner's biodynamic processes were not scientifically tested when he initially delivered them, and even now, testing them is challenging because his descriptions were not precise[6]. In recent studies comparing biodynamic and conventional agriculture, biodynamic systems exhibited improvement among biological and physical indices, such as organic matter decomposition, microbial activity, phosphorous solubility, and soil pH[8-10]. Unlike conventional farms, biodynamic systems have been observed to be less reliant on chemical inputs, because they support the natural processes and interactions within biological communities[9-10]. However, few differences in soil chemistry have been observed in biodynamic systems[9,11-12].

Although many studies have focused on comparing biodynamics to conventional systems, few have drawn comparisons between biodynamics and organic systems[8-10,12]. The primary objective of this study was to compare the effects of biodynamic to low-input organic practices on crop yield of bush beans, carrots, and chard. The second objective was to explore differences between the crops, soil, and microarthropod communities of the two treatments. Total plant population, growth success of expected number of plants, dry root and shoot biomass, root to shoot ratio, bean plant nodules, soil pH, organic C, extractable P, and microarthropod assemblages were the indices used to observe these differences. Based on the literature of biodynamics' claim to stimulate plant growth, a greater crop yield was predicted in the biodynamic treatment[1]. Additionally, significant changes between the two treatments' vegetative, soil, and faunal indices were expected because biodynamics claims to improve soil and biological health of agricultural systems[1,4].

2. Methods

2.1. Experimental Design

2.1.1. garden bed preparation

In July 2014, two garden beds located at the Rhoades Property garden approximately 600 m southeast of the University of North Carolina Asheville campus were manually cleared of weeds and preexisted cover crop using a rake. Each bed measured 5.5 m x 0.9 m and was oriented north to south on a west-facing slope. Sticks were set up 0.3 m apart from one another along the perimeter of each bed. To establish a visual grid, two 5.5-m parallel strings were stretched lengthwise along the middle of each garden bed to indicate a total of six rows measuring 0.3 m x 0.9 m. Mulch was laid on the surface of the edges surrounding 5.5 m x 0.9 m area of each garden bed and in between individual crop beds as 0.3 m x 0.9 m plots.

2.1.2. biodynamic preparations

Approxmiately 36.2 g of BD "prepared" 500 horn manure plus #502-507 (per Podolinsky method)[14] and 4.92 g of 500(X) pre-potentized horn silica were purchased from the Josephine Porter Institute.

Approximately 11.4 L of Asheville tap water were collected in a plastic bucket and allowed to de-gas for 48 h. On the morning of July 25, 2014 (day 1), the 500+ preparation was poured into the bucket and stirred vigorously for 1 h with a bamboo stick. Direction of stirring was reversed immediately when the liquid formed a whirlpool. The 500+ preparation was strained through cheesecloth into another bucket. Approximately 8.9 L of the 500+ preparation were applied to the soil surface of the biodynamic garden bed using spray bottles and a watering can to ensure even and complete application before planting.

On day 28, approximately 9.5 L of Asheville tap water were collected in a plastic bucket and allowed to de-gas for 96 h. On the morning of day 26, the 501(X) preparation was poured into the bucket and stirred vigorously for 20 minutes with a bamboo stick. Direction of stirring was reversed immediately when the liquid formed a whirlpool. Approximately 2.5 L of the 500+ preparation were applied as a foliar spray to the beans, carrots, and chard growing in the biodynamic garden bed using spray bottles to ensure even and complete application. Approximately 2.5 L of water were applied as a foliar spray to the crops in the control garden bed.

2.2. Crop Production

2.2.1 bush beans

Cherokee wax bush bean (*Phaseolus vulgaris* L.) seeds were mixed in a single container to ensure randomization. On July 25, 2014, beans were designated to be planted in the northernmost side of both garden beds, each in plots measuring 1.8 m x 0.9 m. To identify individual 0.3 m x 0.3 m square plots, flag markers were laid down perpendicular to the string indicating the visual grid. In each square plot, nine pairs of seeds were each planted at a depth of approximately 2.5 cm, with approximately 10 cm separating each pair. In the biodynamic plot, single seeds were planted approximately 10 cm apart from one another in two square plots due to a shortage of seed supply. After all seeds were in the ground, soil was patted down on top of them. Both garden plots were watered twice to saturation.

Initial germination was observed 6 days after planting. Seven days later, beans were thinned to nine evenly spaced plants per 0.3 m x 0.3 m plot. Where two or more bean sprouts germinated, plants exhibiting the most damage by bean beetles (*Epilachna varivestis* Mulsant) were removed. The surviving, or total plant population counts were recorded. Equation (1) was used to determine the growth success of bean plants that survived the initial 13 days, where the expected plant population was derived from the number of seed pairs planted in the ground on day 1.

$$\% \text{ growth success} = 100 \text{ x } \frac{\text{Total plant population}}{\text{Expected plant population}} \quad (1)$$

To reflect sensible farming practices, additional beans were planted in the places where no beans germinated. Plastic markers were used to identify their location and avoid counting them in this study. As the bean beetle population persisted, 5.9 ml of neem oil were diluted with approximately 240 ml of water (1:40 ratio) and applied to both beds with a spray bottle. Upon review of potential negative impacts of neem oil to earthworm populations, two subsequent applications of the pesticide were used only on the control treatment[13]. To match moisture input, approximately 240 ml of water were sprayed on the biodynamic treatment after the neem oil application to the control.

Beans matured 48 days after sowing. On days 49 and 56, bean pods were harvested from both garden beds. To support further fruit production, bean pods selected for harvest must have had an approximate minimum length of 5 cm. In both harvests, pods from each treatment were collected in labeled paper bags. Beans were rinsed of debris using water and a sieve and allowed to dry. Number of pods from each treatment were recorded and used to determine bean yield. After drying the bean pods in a drying oven set at 60ºC for approximately three days, the total dry weight (g) of control and biodynamic pods were measured and recorded. The sum pod dry weights from each harvest were used to determine total sum dry weight of each treatment. The mean pods per plant and pod dry weight per plant were also calculated.

On both days 56 and 63, five bean plants were randomly selected from each treatment and excavated from the soil using a trowel. Each plant was gently rinsed with running water, and a knife was used to separate each root from its shoot. Root and shoot samples were allowed to dry and individually placed in labeled paper bags. Each root and shoot was dried in a drying oven set at 60ºC for approximately three days. The dry weights (g) of roots and shoots from both treatments were measured and recorded. Equation (2) was used to determine the root-shoot ratio of each sampled bean plant. The nodules of each bean plant root were counted and recorded. Unpaired two-tailed t-tests were used to analyze statistical significance between the means (± SD) of the root and shoot dry weights, root-shoot ratio, and nodule count between control and biodynamic treatments.

$$\text{Root-shoot ratio} = \frac{\text{Sample root dry weight (g)}}{\text{Sample shoot dry weight (g)}} \quad (2)$$

2.2.2. carrots

On July 25, 2014, carrots were designated to be planted in plots measuring 1.2 m x 0.9 m in the middle of each garden bed, with mulch separating them from the other two crops observed in the experiment. To identify individual 0.3 m x 0.3 m square plots, flag markers were laid down perpendicular to the string indicating the visual grid. In

each square plot, 16 groups of 2-4 seeds were each planted at a depth of approximately 1.3 cm, with approximately 7.6 cm separating each group. After all seeds were in the ground, soil was patted down on top of them. Both garden plots were watered twice to saturation.

Initial germination was observed 13 days after planting. Fifteen days later, carrots were thinned to 16 evenly spaced plants per 0.3 m x 0.3 m square plot. Where two more sprouts germinated, plants were randomly selected for removal. Population counts were recorded. Equation (1) was used to determine growth success of carrot plants that survived the initial 28 days.

On both days 69 and 77, ten carrots were randomly selected from each treatment for premature harvest and excavated using a trowel. Each plant was gently rinsed with running water, and a knife was used to separate each root from its shoot. Each root and shoot sample underwent the same process as the beans to determine their dry weights. The sum dry weights of sampled carrot roots were used to determine carrot yield. Equation (2) was also used to calculate root-shoot ratio of each carrot sample. Unpaired two-tailed t-tests were used to analyze statistical significance between means (± SD) of the root dry weights, shoot dry weights, and root-shoot ratio between control and biodynamic treatments.

2.2.3 swiss chard

Ruby red Swiss chard (*Beta vulgaris* L.) seeds were mixed in a single container to ensure randomization. On July 25, 2014, chard were designated to be planted in the southernmost side of both garden beds, also each in plot measuring 1.8 m x 0.9 m. To identify individual 0.3 m x 0.3 m square plots, flag markers were laid down perpendicular to the string indicating the visual grid. In each square plot, four pairs of seeds were each planted at a depth of approximately 1.3 cm, with approximately 15 cm separating each pair. After all seeds were in the ground, soil was patted down on top of them. Both garden plots were then watered twice to saturation.

Initial germination was observed 6 days after planting. Twenty-two days later, chard plants were thinned to four evenly spaced plants per 0.3 m x 0.3 m square plot. Where two or more sprouts germinated, plants were randomly selected for removal. Population counts were recorded. Equation (1) was used to determine growth success of chard plants that survived the initial 28 days. Both garden beds exhibited signs of suspected cutworm damage[15] where young chard leaves had been sheared off the stem. Additional seeds were planted in the places where seeds had either not germinated or experienced irreparable damage to reflect sensible farming practices. Plastic markers were used to identify their location and avoid counting them in this study.

On day 69, ten chard plants from each treatment were randomly selected for premature harvest and excavated using a trowel. Each plant was gently rinsed with running water, and a knife was used to separate each root from its shoot. All root and shoot samples underwent the same process as the beans to determine their dry weights. The sum dry weights of sampled chard shoots were used to determine carrot yield. Equation (2) was also used to calculate root-shoot ratio of each bean and carrot plant sample. Unpaired two-tailed t-tests were used to analyze statistical significance between the means (± SD) of the root dry weights, shoot dry weights, and root-shoot ratio between control and biodynamic treatments.

2.3. Soil Properties

On July 25, 2014, prior to planting, eight random soil samples were collected at a depth of approximately 20 cm from the surface of each crop plot. Samples from the biodynamic plots were collected after applying the 500+ plus preparation. Each collection of eight samples was consolidated into a single bag and labeled to represent each individual crop plot in both garden beds, for a total of 12 samples.

2.3.1. texture

To determine variation in the garden beds, soil texture was determined by the hydrometer method in each crop plot of the control and biodynamic beds[16]. Because texture is the particle size distribution of clay, silt, and sand in a soil, texture is an invariable characteristic of soil[17]. Thus, only samples collected on July 25, 2014 were observed in the analysis.

Each sample was weighed to approximately 50 g and poured into individual 400-ml beakers. Twenty milliliters of 10% H_2O_2 were added to each sample to oxidize the organic matter in the soil sample. After fizzing subsided, each sample received 100 ml of 5% Calgon solution and was vigorously stirred for five minutes using an overhead stirrer.

Each mixed slurry solution was poured into a 1000-ml graduated cylinder. One hundred ml of 5% Calgon solution were added to a seventh 1000-ml graduated cylinder and established as the control sample. Each graduated cylinder was filled to the 1000-ml mark with distilled water.

A thermometer was placed in the control sample, and the temperature was recorded. Two pieces of parafilm were stretched over each graduated cylinder, and each sample was shaken end-over-end until all soil particles were suspended. After two more shakes, each sample was set on the table, and after 30 s and 60 s, hydrometer readings were observed and recorded. Approximately 1.5 h and 24 h after the initial hydrometer readings, third and fourth hydrometer and thermometer readings were observed and recorded for each sample. All hydrometer readings were corrected for the control and temperature.

Percent sand was determined for each sample using equation (3) and the 30 s and 60 s hydrometer readings. The average between the two percent sand calculations was used for the final soil texture analysis.

$$\% \text{ Sand} = 100 - \left(\frac{\text{Hydrometer reading (g/cm}^3\text{) x 100}}{\text{Sample weight (g)}}\right) \quad (3)$$

Percent clay was determined for each sample using equation (4) and the 1.5 h and 24 h hydrometer readings. The average between the two percent clay calculations was used for the final soil texture analysis.

$$\% \text{ Clay} = \frac{\text{Hydrometer reading (g/cm}^3\text{) x 100}}{\text{Sample weight (g)}} \quad (4)$$

Percent silt was determined for each sample using equation (5).

$$\% \text{ Silt} = 100 - (\% \text{ Sand} + \% \text{ Clay}) \quad (5)$$

The percent sand, percent clay, and percent silt values of each soil sample were applied to a textural class triangle, determining the textural class of each soil analyzed.

2.3.2. pH

To observe changes in the soil chemistry of the control and biodynamic treatments, pH was determined for each crop plot on days 1 and 63 using standard methods[18]. Soil pH is a measurement of acidity that affects plant growth, nutrient reserves, and chemical and physical processes from the ground up, and thus, pH is a commonly used indicator of soil quality[17].

Soil samples were weighed to 5 g and mixed with 5 ml of distilled water. After the samples equilibrated for 30 minutes, pH was determined using a pH meter and standard electrode. Equation (6) was used to determine the percent change of pH between days 1 and 63.

$$\% \text{ change in pH} = \left(\frac{\text{Day 63 pH - Day 1 pH}}{\text{Day 1 pH}}\right) \times 100 \quad (6)$$

2.3.3. organic C

To observe changes in the soil chemistry of the control and biodynamic treatments, organic C (SOC) was determined by loss-on-ignition for each crop plot on days 1 and 63[19]. The availability of SOC is important to plant growth and microbial activity, thus SOC is also commonly used to indicate soil quality[19].

Soil samples were weighed to approximately 5 g and dried in a muffle furnace set at 105ºC for one hour. Samples were allowed to cool in a desiccator, weighed, and combusted for 2 h in the furnace set at 360ºC. The temperature was adjusted to 105ºC for one additional hour. The samples were cooled in a desiccator and weighed again. Percent SOC was calculated using equation (7), and percent change of percent SOC between days 1 and 63 was calculated using equation (8).

$$\% \text{ SOC} = \left(\frac{\text{Oven dry soil weight (g)} - \text{Soil weight after combustion (g)}}{\text{Oven dry soil weight (g)}}\right) \times 100 \quad (7)$$

$$\% \text{ change in \% SOC} = \left(\frac{\text{Day 63 \% SOC} - \text{Day 1 \% SOC}}{\text{Day 1 \% SOC}}\right) \times 100 \quad (8)$$

2.3.4. extractable P

To observe changes in the soil chemistry of the control and biodynamic treatments, extractable P was determined for each crop plot on days 1 and 63 using Mehlich I extraction[20]. Phosphorus is one essential nutrient for plants[7,17]. Phosphorus primarily occurs as immobilized phosphate in the soil and is, thus, inaccessible to plants. Because soluble P levels rise as a soil's mineral content decreases and organic matter increases, extractable P can be used to observe nutrient cycling in a soil system[17].

Mehlich I Extracting Solution was prepared with 0.05 N HCl and 0.025 N H_2SO_4. Reagent A was prepared by mixing 20 g of ammonium molybdate, 450 mL of 1 N H_2SO_4, and 100 mL of 0.5% antimony potassium tartrate in a 1-L volumetric flask and filled to volume with deionized water. Reagent B was prepared by dissolving 1.5 g of ascorbic acid in 100 ml of Reagent A. A 100-ppm P stock solution was used to prepare standards of 0, 5, 10, 15, and 20 ppm P. Five grams of each soil sample were weighed, placed in individual extracting bottles, and shaken on a reciprocating shaker for 5 minutes. Samples were filtered for Whatman No. 42 filter papers. Three milliliters of each standard and sample were poured into 50-ml Erlenmeyer flasks. Each flask received 5 ml of Reagent B and brought to 50 ml of deionized water. One hour later, absorbance of each standard and sample was measured at 660 nm and recorded. The ppm P and absorbance values of the standards were used to establish a standard curve and line of best fit in equation to determine ppm P values for the soil samples.

2.4. Microarthropods

To observe difference in a faunal community of the control and biodynamic treatments, microarthropods (Collembola and Acari) were examined. In soil food webs, microarthropods primarily contribute to decomposition and mineralization processes, increasing N flux (another essential nutrient for plant growth) in farming systems[7,16]. Thus, microarthropods can provide additional insight into biotic activity and nutrient cycling in both the control and biodynamic treatments of this study[7].

On day 42, 24 5-cm long PVC pipes with inside 4-cm diameter were prepared by fastening fiberglass screens to one end of each pipe with a rubber band. Four random samples were collected in each crop bed by embedding sampling pipes into the top 5 cm of the soil surface. Cores were gently dug out of the soil with a knife and transferred to empty soft drink cans with funnels glued to the bottom placed in extraction racks. Vials were filled with ethanol and attached at the bottom of the funnels. Five-watt Christmas tree bulbs were positioned above the sampling cores and covered with aluminum foil to retain heat. Light intensity was set to low and was gradually increased to maximum heat over the course of seven days to drive microarthropods down into the ethanol vials.

Microarthropods from each sample were identified under a dissecting microscope to taxon. Individuals from each sample were combined to determine sum and mean (± SD) of sample populations of Collembola, Acari, and other uncategorized taxa. An unpaired two-tailed t-test was used to analyze the statistical significance of mean sample populations between control and biodynamic treatments. Taxon composition of each treatment was calculated using equation (9).

$$\text{Composition (\%)} = \left(\frac{\text{Number of individuals in a sample}}{\text{Total number individuals}}\right) \times 100 \quad (9)$$

Menhinick's index (D) expressed in equation (10) was used to determine taxon richness in each treatment, where s is the number of taxa and the number of individuals of a sample is represented by N[21].

$$D = \frac{s}{\sqrt{N}} \quad (10)$$

Shannon index (H′) was used to calculate taxon diversity in each crop plot expressed in equation (11), where percent composition of a single taxon of each crop plot represented by p_1[21]. The sum of calculated H′ from each crop plot of each treatment was used to determine taxon diversity.

$$H' = \sum(p_1)|\ln p_1| \quad (11)$$

3. Results

3.1. Crop Production

3.1.1 bush beans

Of the expected 162 bush bean plants in each plot, 153 grew in the control and 120 grew in the biodynamic (Table 1). Approximately 20% more bush bean plants grew in the control plot than the biodynamic. The control plot had approximately 23% more bean pod yield than the biodynamic. However, biodynamic beans exhibited slightly higher values for average pods per plant and pod dry weight per plants. Overall, bush bean growth success, yield, and dry weight were greater in the control treatment, but the trends cannot be statistically supported.

Table 1. comparison of total plant population, survival rate, total pod yield, mean pods per plant, total dry weight of pods, and mean pod dry weight per plant from two harvests of bush beans from control vs. biodynamic treatments.

Treatment	Control	Biodynamic
Plant population	153	120
Growth success (%)	94	74
Total yield (pods)	662	539
Mean pods per plant	4.3	4.5
Sum pod dry weight (g)	166.0	147.9
Mean pod dry weight per plant (g/plant)	1.08	1.23

The mean dry weight of bean plant roots was higher in the control treatment, but the difference was statistically insignificant (p=0.80). Similarly, the differences between the mean dry weight of bean shoots and root-shoot ratios were also insignificant (p=0.76). Although the biodynamic treatment exhibited a greater mean number nodules per plant than the control, these differences were also not statistically significant (p=0.61).

Table 2. mean (± SD), probability value, and statistical significance of root and shoot dry weight, root-shoot ratio, and nodule count from samples of 20 bush bean plants from control vs. biodynamic treatments.

	Mean ± SD			
	Treatment			Statistical
	Control	Biodynamic	p-value	significance ($p<0.05$)
Root dry weight (g)	0.22±0.14	0.21±0.12	0.80	No
Shoot dry weight (g)	7.69±2.85	7.36±2.35	0.76	No
Root-shoot dry weight ratio	0.034±0.024	0.031±0.018	0.76	No
Nodules per plant	24±12	27±14	0.61	No

3.1.2. carrots

Of the 192 carrots expected to grow in each plot, 165 grew in the control and 163 grew in the biodynamic. The total yield of sampled carrots was 0.16 g greater in the control treatment (Table 3).

Table 3. comparison of total plant population, growth success, and total yield of carrots from control vs. biodynamic treatments.

Treatment	Control	Biodynamic
Total plant population	165	163
Growth success (%)	86	85
Total yield of 20 plants (g)	2.24	2.09

Mean dry weights of the carrot roots and shoots were both higher in the control treatments, but this difference was not found to be statistically significant ($p=0.15$) (Table 4). However, the dry weights of carrot shoots in the control and biodynamic treatments did vary significantly ($p<0.0001$). Yet the difference between mean root-shoot ratio in both treatments was found to be insignificant ($p=0.73$).

Table 4. mean (± SD), probability value, and statistical significance of root and shoot dry weight, and root-shoot ratio from 40 carrot plants from control vs. biodynamic treatments.

	Mean ± SD Treatment			Statistically significant
	Control	Biodynamic	p-value	($p<0.05$)
Root dry weight (g)	0.36±0.37	0.22±0.25	0.15	No
Shoot dry weight (g)	1.43±0.59	0.82±0.49	<0.0001	Yes
Root-shoot dry weight ratio	0.26±0.23	0.29±0.21	0.73	No

3.1.3 swiss chard

Of the 72 chard plants expected to grow in each treatment, 43 grew in the control and 51 grew in the biodynamic (Table 5). Approximately 8% more chard plants grew in the biodynamic plot, but the control had 3.4 g higher yield.

Table 5. comparison of total plant population, growth success, and total yield of chard from control vs. biodynamic treatments.

Treatment	Control	Biodynamic
Total plant population	43	51
Growth success (%)	45	53
Total yield of 10 plants (g)	33.7	30.3

Mean dry weight of the chard roots was higher in the biodynamic treatment, while mean dry weight of the chard shoots was higher in the control treatment (Table 6). Although both differences between mean dry weights of chard roots ($p=0.08$) and shoots ($p=0.84$) were not statistically significant. The mean root-shoot ratios ($p=0.07$) in both treatments were also statistically insignificant.

Table 6. mean (± SD), probability value, and statistical significance of root and shoot dry weight, and root-shoot ratio from 20 chard plants from control vs. biodynamic treatments.

	Mean ± SD Treatment			Statistically significant
	Control	Biodynamic	p-value	($p<0.05$)
Root dry weight (g)	0.23±0.12	0.34±0.15	0.08	No
Shoot dry weight (g)	3.37±1.50	3.03±1.28	0.84	No
Root-shoot dry weight ratio	0.08±0.05	0.14±0.08	0.07	No

3.2. Soil Properties

3.2.1 texture

The textural class of both control and biodynamic garden beds was determined to be sandy loam, exhibiting homogeneity across all crop plots (Table 7).

Table 7. particle size distribution and textural class of six soil samples collected from control and biodynamic garden beds.

Treatment	% Sand	% Silt	% Clay	Textural class
Control				
Beans	63.6	32.0	4.4	sandy loam
Carrots	62.9	32.0	5.1	sandy loam
Chard	63.0	32.2	4.8	sandy loam
Biodynamic				
Beans	61.9	33.1	5.0	sandy loam
Carrots	62.8	31.6	5.6	sandy loam
Chard	64.4	31.7	3.9	sandy loam

3.2.2. pH

All soil samples exhibited low acidity throughout the duration of the experiment. (Table 8).

Table 8. comparison of soil pH, percent SOC, and extractable P of soil samples collected from each crop bed in control and biodynamic treatments on days 1 and 63.

	Treatment			
	Control		Biodynamic	
Soil property	Day 1	Day 63	Day 1	Day 63
pH				
Beans	6.71	6.77	6.38	6.25
Carrots	6.29	5.80	6.32	6.57
Chard	6.00	5.78	6.19	6.33
SOC (%)				
Beans	7.2	7.1	5.8	6.3
Carrots	7.2	9.0	6.3	7.3
Chard	6.9	6.9	6.6	7.8
Extractable P (ppm)				
Beans	55.1	62.0	66.5	55.9
Carrots	58.6	61.8	60.3	60.1
Chard	51.8	56.4	48.9	55.7

Among all the soil samples collected, the control beans on day 63 had the highest pH, while the control chard on day 63 had the lowest pH. Over time, pH increased in the bean plots and decreased in the carrot and chard plots of the control treatment (Figure 2). The inverse trend was true in the biodynamic plots. Overall, the control plots exhibited greater variation in pH change than the biodynamic plots. Carrots in the control plot experienced the greatest flux in pH with an approximate 8% decrease, and control beans experienced the smallest flux of a 1% increase. Change in pH ranged between approximately 2% and 4% among biodynamic plots.

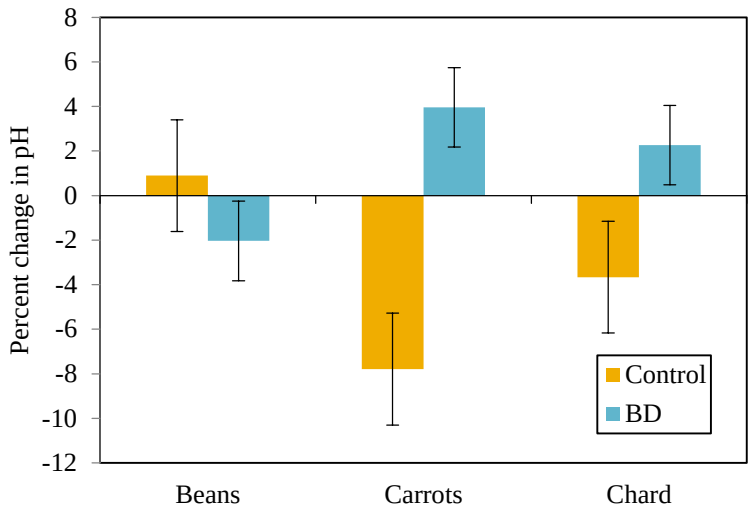

Figure 2. Percent change of soil pH ± standard error (SE) in control and biodynamic treatments between days 1 and 63.

3.2.3. organic C

The SOC of all soil samples ranged from 5.8% to 7.2% on day 1 and 6.3% to 9.0% on day 63 (Table 8). Control carrots exhibited the highest percent SOC on both days, along with control beans on day 1. Biodynamic beans had the lowest percent SOC on both days. Over time, SOC levels increased in all crop plots except in the control beans (Figure 3). Similarly to the pH results, SOC flux exhibited a steadier range in the biodynamic plots than the control. Biodynamic SOC flux ranged from approximately 9% to 18% increase, while the control SOC flux varied with a 1% decrease in the beans, no change in the chard, and a 25% increase in the carrots, the largest value of both treatments.

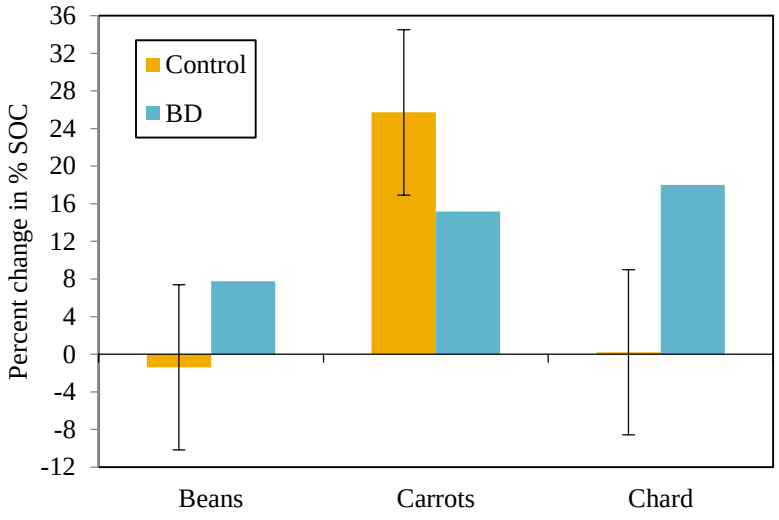

Figure 3. Percent change of %SOC ± SE in control and biodynamic treatments between days 1 and 63.

3.2.4. extractable P

The extractable P of all soil samples ranged from 48.9 ppm to 66.5 ppm on day 1 and 55.7 ppm 62.0 ppm on day 63 (Table 8). Biodynamic beans represented the highest levels of extractable P on day 1, while biodynamic chard had

the lowest amount on both days. Control beans had the highest extractable P levels on day 63 and exhibited an approximate 13% increase over the course of 9 weeks (Figure 3). Extractable P levels dropped the most in the biodynamic bean plots by nearly 16%. Interestingly, like the pH and % SOC trends exhibited across the control treatments, the biodynamic plots had more variation in extractable P flux amongst crops, with a miniscule decrease in the carrot plots and a near 14% increase in the chard plots. Despite the variability amongst extractable P flux, all plots had abundant levels of extractable P.

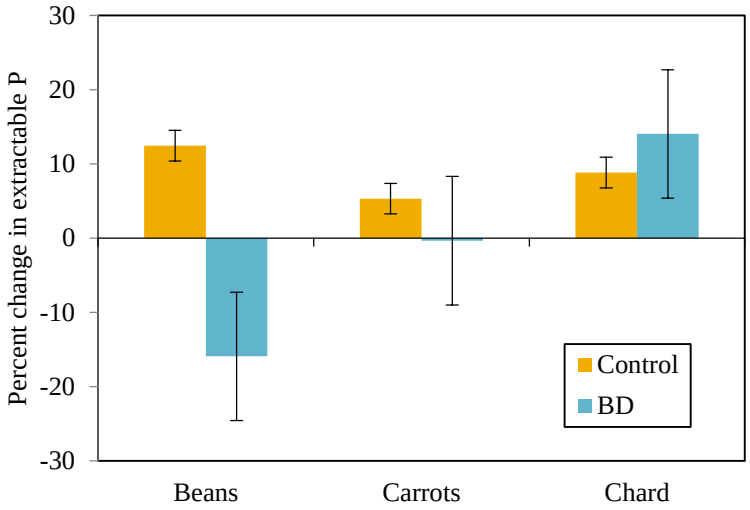

Figure 4. Percent change of extractable P ± SE in control and biodynamic treatments between days 1 and 63.

3.3. Microarthropods

The control treatment had a greater total sample population of 402 individuals, while the biodynamic treatment had nearly 25% less individuals (Table 8).

Table 8. Sum and mean (± SD) sample populations, composition, and Menhinick's richness index of microarthropod taxa of Collembola, Acari, and other uncategorized taxa from soil cores sampled from control and biodynamic treatments on day 42.

		Taxon		
	Total	Collembola	Acari	Other
Sample populations				
Sum				
Control	402	204	164	34
Biodynamic	303	113	157	33
Mean ± SD				
Control	-	17±13	14±8	3±3
Biodynamic	-	9±9	13±12	3±2
Composition (%)				
Control	-	50.7	40.8	8.5
Biodynamic	-	37.3	51.8	10.9
Richness				
Control	0.96	0.21	0.23	0.51
Biodynamic	1.04	0.28	0.24	0.52

Collembola had the highest sum and mean sample populations in the control treatment, while Acari exhibited the highest sum and mean sample populations in the biodynamic treatment. Taxa composition reflected greater Acari dominance of 51.8% in the biodynamic plots than the Collembola dominance of 50.1% in the control. However, the control treatment had a greater proportion of Acari to Collembola than the biodynamic. Richness was slightly larger

among all taxa in the biodynamic treatment. The biodynamic treatment exhibited a higher taxon diversity index of 1.49, while the control treatment had a diversity index of 1.38.

4. Discussion

Total yield of the biodynamic and control garden beds did not support the hypothesis that a biodynamic treatment would produce greater crop yield, but larger chard yield in the biodynamic plot rendered the conclusion indeterminate at the crop plot level. Thus, the chard anomaly was indicative of overall variability observed between the biodynamic and control treatments in this study.

The second hypothesis expecting significant change among vegetative, soil, and faunal indices was also not supported by results from this study. Although differences among between variables could not be statistically supported, some trends emerged that would be interesting to investigate in future studies. Crop plots with higher populations and growth success (control beans, carrots, and biodynamic chard) exhibited slightly higher root-shoot ratios, suggesting their soil conditions supported resource allocation towards vegetative and fruit production[22]. The temporal and spatial constraints of this experiment did not generate enough data to draw definitive conclusions about root response to biodynamic and control treatments, thus additional observations of multiple crop root systems over time would provide more insight into this relationship in these garden beds. While biodynamics did not ultimately produce greater yield, an exploration into biodynamics' influence on improving crop productivity seems like a logical expansion of this study. Biodynamic beans exhibited a larger pod size average, although the parameters here could not examine this variable further. Thus, further exploration of the root-shoot ratio and nutritional composition of plant tissue between biodynamic and organic systems could potentially draw interesting comparisons.

Variability of the soil properties analyzed in this study was nominal, although important trends also emerged between control and biodynamic treatments. All observed pH values fell within the ideal pH range for beans, carrots, and chard[23], while the extractable P levels were consistently high throughout both garden beds[24]. Percent SOC indicated consistent levels of microbial activity in the soil, with the exception the control carrots experiencing the greatest increase in SOC levels. SOC positively correlates to carrot quality, likely contributing to the control carrots' larger yield[25]. The biodynamic treatments exhibited more consistency in pH and SOC flux than the control, indicating a possible correlation between biodynamics and soil stabilization supported in a previous four-year study[8], but further research in these garden beds would be needed to draw any conclusions. Overall, the results from this study support previous comparisons drawn between soil chemical properties of biodynamic and organic treatments that also found more similarities than differences among these treatments[9,11-12].

The microarthropod assemblages observed in this study provide insight into faunal community dynamics between the control and biodynamic treatments[26]. Collembola are usually fungivorous, and their dominance in the control treatment may have indicated a higher amount of fungi in this garden bed that likely contributed to the increased plant growth and yield in the control bed[7,26]. Acari are important decomposers and represent multiple feeding groups, such as detritivores, predators, and fungivores, among trophic levels[26]. Further study of the feeding groups represented in this bed could explain food web dynamics and their influence on the productivity in the biodynamic garden plots. Interestingly, biodynamics did support higher taxa richness and diversity, although the difference could not be statistically analyzed within the parameters of this experiment. A long-term study on biodynamics observed an increase in natural predator populations to pest, vastly reducing the need for fertilizer and pesticide use[9]. The richness and diversity indices in this study, as well as the qualitative observations of reduced bean beetle damage in the biodynamic plots, suggest biodynamics could support such a possible increase in natural predator-prey interactions in the studied garden beds.

5. Conclusion

The results from this study supported neither hypotheses that the biodynamic treatment would produce greater yield or exhibit significant differences from the vegetative, soil, and faunal indices of the control treatment. Trends emerged that could warrant further investigation into biodynamics' effect on crop productivity, soil stabilization, and food web dynamics. Data collected between treatments were limited based on temporal and spatial constraints of the experimental design, but previous studies supported the similarities observed organic and biodynamic farming

systems[9-10]. While biodynamics has been shown to support better soil quality and biological activity than conventional agriculture, low-impact organic farming has demonstrated parallel competence to achieve these ends.

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