Journal of Agriculture and Food Research 27 (2026) 102861

Contents lists available at ScienceDirect

Journal of Agriculture and Food Research

journal homepage: www.sciencedirect.com/journal/journal-of-agriculture-and-food-research

# Biochemical pesticides and soil changes resulting from biosolarization with cover crop amendments

Shayne Morrissey [a,b], Carolina Conte [a], Natalie Chueh [a], Vanessa Kalip [a], Christopher Simmons [a,*]

[a] Department of Food Science and Technology, University of California Davis, One Shields Avenue, Davis, CA, 95616, USA
[b] Agricultural and Environmental Chemistry Graduate Group, University of California Davis, One Shields Avenue, Davis, CA, 95616, USA

ARTICLE INFO

*Keywords:*
Cover cropping
Biosolarization
Biofumigation
Biochemical pesticides
Integrated pest management
Soil remediation

ABSTRACT

Biosolarization is an agricultural pest management technique that uses organic matter amendments and plastic mulch to control soil pests in a variety of cropping systems without harmful chemical fumigants. Biosolarization involves amending soil with organic matter, irrigating, and covering the surface with a clear tarp to suppress pests, combining stressors from increased temperature, reduced oxygen, and biochemical pesticide production. In particular, cover crops present a convenient, *in situ* amendment source for biosolarization. However, research is needed to evaluate the effectiveness of cover crops as soil amendments for biosolarization prior to large-scale applications. This study uses *Brassica juncea* (brown mustard) cover crop residues as biosolarization amendments to determine changes in soil properties contributing to pest suppression and crop growth. Specifically, a field trial was employed to quantify soil organic acid biochemical pesticide production, residual phytotoxicity, moisture content, pH, and electrical conductivity with *B. juncea* monoculture and *B. juncea-Vicia villosa* (hairy vetch) mixture cover crop amendments. Additionally, a simulated laboratory study evaluated changes in soil isothiocyanate biochemical pesticide concentrations during biosolarization with *B. juncea* cover crop amendments. Treatments were compared to non-biosolarized soil amended with cover crop residues, non-amended soil treated with solarization, and non-amended, unheated soil controls. Results show biosolarization with cover crop residues increase the cumulative exposure to fermentative organic acid and plant-derived isothiocyanate compounds in soil by 314% and 28%, respectively, without causing persistent phytotoxicity. These findings highlight the potential for biosolarization with cover crop amendments to control soil pests without negatively impacting the growth of subsequent crops.

## 1. Introduction

Chemical fumigants have historically been used to control a variety of agricultural soil pests and diseases. However, concerns over human health and environmental degradation have increased the regulation of fumigant use, including prohibition of certain compounds [1–5]. In response, alternative pest management techniques are increasingly necessary to manage soil pests [6,7]. Biosolarization is one such method of soil pest control that utilizes organic matter soil amendments and solarization techniques to replace chemical fumigation [8–12]. Instead, biosolarization practices involve amending the soil with organic matter, tarping the soil surface, and irrigation. Biosolarization practices have been successfully applied in a variety of cropping systems, including eggplants, tomatoes, lettuce, and strawberries, among others [10, 13–15]. While solarization controls pests primarily through thermal stress in the soil, biosolarization can enhance pest suppression through the production of biochemical pesticides generated during the degradation of organic matter amendments, culminating in an inhospitable soil environment for pests [9,10,16]. Using these techniques, biosolarization has been evaluated as a sustainable alternative to conventional chemical fumigation, with studies showing comparable suppression of soilborne pests, such as nematodes, fungi, and weeds [9, 10,15,17,18]. Additionally, biosolarization treatments are particularly relevant for soil pest control in farming systems where the use of conventional fumigants are largely prohibited, such as organic farming.

Previous research in biosolarization has examined the use of various

* Corresponding author.
*E-mail addresses:* sgmorrissey@ucdavis.edu (S. Morrissey), crconte@ucdavis.edu (C. Conte), nchueh@ucdavis.edu (N. Chueh), vwkalip@ucdavis.edu (V. Kalip), cwsimmons@ucdavis.edu (C. Simmons).

https://doi.org/10.1016/j.jafr.2026.102861
Received 29 January 2026; Received in revised form 16 March 2026; Accepted 18 March 2026
Available online 19 March 2026

organic amendments from food processing waste, such as rice bran, mustard meal, and almond hulls [19–22]. The organic matter amendments are typically mixed into the top layers of the soil at the beginning of the biosolarization process and are a key factor contributing to pest suppression. During biosolarization, degradation of organic matter in the soil releases biochemical pesticides, which are naturally produced compounds that suppress or deter pests. While amendments from food processing waste have shown efficacy for biosolarization, they require transportation from the processing facility to the field, which can limit the practicality of the treatment. Additionally, a previous life cycle assessment identified the transportation of amendment material as one of the main factors contributing to the primary energy demand and global warming potential of the biosolarization process [23]. As an alternative, cover crops present an *in situ* amendment source for biosolarization. Instead of transporting an amendment to the field, cover crops may be grown directly in the soils to be treated, avoiding the energy demand and global warming potential of amendment hauling, while decreasing reliance on outsourced amendments for biosolarization.

Cover crops have been used for soil pest suppression in the past, mainly through a technique characterized as biofumigation, which involves incorporating *Brassica* rotation or green manure crops into the soil. The focus on *Brassica* cover crop species for managing soil pests arises from their unique production of innate biochemical pesticides. When *Brassica* tissues are disrupted, a secondary plant metabolite, glucosinolate, is released and hydrolyzed by myrosinase enzymes to produce biocidal compounds, largely isothiocyanates [24,25]. In particular, *Brassica juncea* is a commonly applied cover crop in biofumigation due to its high glucosinolate content and biomass production, two major factors contributing to pest suppression [26]. Previous research has shown the potential for isothiocyanates to suppress several common soil pest species, including *Fusarium oxysporum*, nematodes, and weeds [27–30]. However, in practice, biofumigation often yields variable levels of soil pest control, with poor results often attributed to insufficient glucosinolate hydrolysis and isothiocyanate formation in the soil [26,31]. As a result, researchers have hypothesized that higher soil temperatures and increased water availability during biofumigation may enhance isothiocyanate production [26,32]. Since these conditions are consistent with the environmental factors present during biosolarization, it is reasonable to evaluate *Brassica* cover crops as an amendment source. However, the impact of biosolarization conditions on isothiocyanate formation is unknown and in need of investigation.

Beyond isothiocyanate compounds, biosolarization can also induce the production of other biochemical pesticides from biodegradation of the amendment material. Specifically, fermentation of the amendment during biosolarization often produces a range of organic acid compounds, common biochemical pesticides quantified in previous research with broad toxicity to a variety of soil pests [21,33,34]. However, the extent to which cover crop residues produce organic acids during biosolarization remains largely unknown. In addition to isothiocyanate production, the combination of organic acid formation, elevated soil temperature, and reduced oxygen availability during biosolarization may provide a multi-modal approach to soil pest control, increasing the consistency of results. Previous research has revealed significant suppression of soil pests, including weeds and fungal crown rot, as well as increased eggplant growth and yield following biosolarization with *B. juncea* monoculture and *B. juncea-V. villosa* mixture cover crop residue amendments [13]. These results suggest biosolarization with cover crop residue amendments may serve as an alternative to chemical fumigation, while providing health benefits to subsequent cash crops. However, more research is necessary to identify the soil properties, including soil biochemical pesticide exposure, contributing to the observed pest control.

In addition to pest control, the time needed to remediate the soil before planting subsequent cash crops is an important consideration when employing biosolarization. Extended periods of soil phytotoxicity, or the presence of compounds in the soil that inhibit plant growth, can interfere with planting schedules and reduce overall crop productivity. Following biosolarization, residual biochemical pesticides in the soil can have phytotoxic effects in subsequent crops by decreasing their growth and yield. Previous research in biosolarization with food by-product amendments have shown a typical remediation period of 1-5 weeks, resulting in a treatment duration largely matching that of common fumigants [10,35]. Additionally, after cover crop incorporation, it is typically suggested to wait 2-3 weeks before planting subsequent crops to avoid negative effects [36]. However, the remediation time required to prevent residual soil phytotoxicity from negatively impacting subsequent crop production for biosolarization with cover crop residue amendments is currently unknown.

The present study aims to characterize the aforementioned gaps in knowledge through a combination of field and laboratory-simulated biosolarization trials using cover crop residue amendments. The field trial quantifies organic acid biochemical pesticide production in the soil, the remediation time required to alleviate soil phytotoxicity, as well as soil moisture content, pH, and electrical conductivity following biosolarization with *B. juncea* monoculture and *B. juncea-V. villosa* mixture cover crop residue amendments. Meanwhile, the simulated biosolarization trial focuses on characterizing the effects of biosolarization conditions, including elevated temperature, reduced oxygen availability, and increased moisture, on soil isothiocyanate biochemical pesticide production from *B. juncea* cover crop residue amendments. Through the combination of the field and laboratory biosolarization trials, this study advances knowledge on the efficacy of using cover crop residues instead of food processing waste as amendment sources for biosolarization and provides context to previously observed increases in crop yield and soil pest suppression. This was performed by quantifying previously unexplored soil properties, including soil organic acid and isothiocyanate biochemical pesticide production, the remediation time required to effectively alleviate residual soil phytotoxicity, and changes in soil moisture content, pH, and electrical conductivity.

## 2. Methods

### 2.1. Biosolarization field trial

A test site was selected from the UC Davis Plant Pathology Research Farm (Davis, CA, USA; 38.5194239, −121.7684936). The soil was previously classified into the “fine-silty, mixed, superactive, thermic Fluventic Haploxerepts” family [37]. Within the sampling horizon (0-20 cm), the soil pH was 6.64, the electrical conductivity (EC) was 307.8 μS $cm^{-1}$, the volatile solids (VS) content was 4.86%, and the soil was composed of 54% gravel, 36% sand, and 10% silt/clay. The site history included a weedy fallow for two years immediately before the trial, a year of strawberry cultivation three years prior, and another period of weedy fallow in the fourth year preceding the trial.

Cover crops were sown in the test site on November 30, 2023 in three 15 × 15 m adjacent fields, creating a *B. juncea* monoculture field, a *B. juncea-V. villosa* mixture field, and a fallow field. *B. juncea* seeds were sown at a rate of 44 kg $ha^{-1}$ in the monoculture and mixture field. *V. villosa* seeds were exclusively sown in the mixture field at a rate of 100 kg $ha^{-1}$. No seeds were sown in the fallow field. The conditions across all fields were verified and did not differ, including soil texture, pH, electrical conductivity, VS content, shading, and management history. All cover crop seeds were purchased from True Leaf Market (Salt Lake City, UT, USA).

On May 9, 2024, prior to the onset of the biosolarization period, above and below-ground plant mass was extracted from the field to estimate the amendment rate. All plants were harvested from areas measuring one square meter within each field, in triplicate. The fresh mass of the plants, grouped by cover crop species and weeds, was measured. To determine the dry mass amendment rate, a subsample of the plants was retained and dried at 105 °C for four days. The resulting

values were used to calculate the moisture content and applied to estimate the dry mass amendment rate of each plant group and the total amendment rate within each field (Table 1).

A ten-day biosolarization trial began on May 10, 2024. All field areas were mowed and tilled at a depth of 20 cm. The fields were then divided into eight plots each and drip irrigation lines were laid across each plot. Four of the plots within each field were randomly selected and covered with a clear, low-density polyethylene tarp (TriCal Inc., 1 mil, TIF), creating *B. juncea* monoculture-biosolarized (MONO-B), *B. juncea-V. villosa* mixture-biosolarized (MIX-B), and unamended-solarized (U-S) treatments within the monoculture field, mixture field, and fallow field, respectively. For the controls, the remaining four plots within each field were left uncovered, creating the *B. juncea* monoculture-control (MONO-C), *B. juncea-V. villosa* mixture-control (MIX-C), and unamended-control (U-C) within the monoculture, mixture, and fallow fields, respectively. This field setup created a total of six soil treatments, two cover crop-amended biosolarization treatments (MONO-B and MIX-B), two cover crop-amended controls (MONO-C and MIX-C), one solarization treatment (U-S), and one untreated control (U-C), using a randomized block design and four replicate plots for each treatment. After tarping was complete, all plots were irrigated for 24 h until visually saturated. Throughout the biosolarization period, daily high and low ambient air temperatures ranged between 27.2-35.0 °C and 11.1-13.9 °C, respectively. High and low soil temperatures in covered plots ranged between 29.5-47.8 °C and 18.5-29.1 °C, respectively, and high and low soil temperatures in uncovered plots ranged between 25.9-34.4 °C and 15.8-21.6 °C, respectively. Soil temperatures were measured by burying temperature loggers 5 cm below the soil surface (Dwyer-Omega, OM-CP-MicroTemp, CT, USA). The tarps were removed from all covered plots on May 20, 2024 at the end of the biosolarization period.

Soil pH and electrical conductivity were measured in soil sampled from the top 0-20 cm of the soil horizon on days 0, 1, 2, 3, 6, and 10 during the biosolarization period, as well as days 18, 25, and 39 during the remediation period. For the measurements, wet soil was combined with deionized water in a 1:1 dilution in 15-mL centrifuge tubes (Falcon, NY, USA). The soil-water mixtures were vortexed for 30 s and centrifuged at 8000 rpm for 10 min (Eppendorf Centrifuge 5810R 15 Amp, Hamburg, Germany). Soil pH and electrical conductivity were measured in the resulting supernatant (Mettler Toledo InLab Expert NTC30 and Mettler Toledo InLab 731-2m, OH, USA). Two technical replicates were made for each soil sample. The samples were then immediately used for subsequent organic acid extraction and quantification.

Organic acid concentrations in the soil were determined using previously reported methods, with some modifications [21]. The supernatant produced from the pH and electrical conductivity measurements was filtered through a PTFE syringe filter with a 0.22 μm pore size (Thermo Fisher Scientific Inc., MA, USA). Then, 1.5 mL of the filtrate was dispensed into 2-mL HPLC vials (Agilent Technologies Inc., CA, USA). The organic acid extracts were then analyzed by high-performance liquid chromatography (HPLC, model 1260 Infinity II, Agilent Technologies Inc.) to determine concentrations of citric, malic, formic, succinic, acetic, lactic, propionic, butyric, and isobutyric acids with a Hi-Plex H column (300 × 7.7 mm, 8 μm, Agilent Technologies Inc.) and guard column (5 × 3 mm, 8 μm, Agilent Technologies Inc.). The mobile phase was 5 mM sulfuric acid in Milli-Q water with an isocratic flow rate of 0.6 mL $min^{-1}$. The run time was 30 min with an injection volume of 20 μL for each sample and the column temperature was 60 °C. The absorbance of all acids was detected at 210 nm, with the exception of succinic acid, which was detected at 195 nm (1260 Infinity II, Agilent Technologies Inc.). Absorbance values in the samples were compared to standard solutions ranging from 32.5 to 2000 mg $L^{-1}$. Measured organic acid concentrations were normalized to the moisture content of each sample to yield the concentration per unit dry weight of soil. The concentrations of individual organic acid species were summed to determine the total organic acid concentration in each sample. Cumulative soil organic acid exposure was defined by summing measured soil organic acid concentrations across sequential sampling timepoints over the course of the treatment period.

Residual phytotoxicity in the soil was determined in samples taken at the end of the biosolarization period, as well as 8, 15, and 29 days following the end of biosolarization. Phytotoxicity was quantified by measuring the Germination Index (GI) of radish seeds in the soil from each treatment and control. Forty-five grams soil from each treatment and control were added to Petri dishes (FisherScientific, MA, USA), as in previous studies [35,38,39]. Ten radish seeds (var. *longipinnatus*, *Raphanus sativus*, True Leaf Market, UT, USA) were added to each petri dish and incubated in the dark at 24 °C for 72 h. Following incubation, the total number of germinated seeds ($N$) and the average root length of each germinated seed ($L_{avg}$) were recorded for all plates. The GI was calculated by comparing these values to the mean number of germinated seeds ($N_0$) and mean root length ($L_0$) of non-amended, untreated controls according to Eq. (1). Root lengths of 0.5 cm or greater were considered positive germinations and soils with a mean GI of less than 80% were considered phytotoxic, as previously defined [40,41]. Two petri dishes were prepared for each sample as technical replicates.

$$GI = 100\%\left(\frac{L_{avg}N}{L_0N_0}\right) \quad (1)$$

Soil moisture content was measured in soil sampled on days 0, 1, 2, 3, 6, and 10 during the biosolarization period, as well as days 18, 25, and 39 during the remediation period. The soil moisture content was measured as the percent of mass lost after drying the soil at 105 °C for 24 h.

### 2.2. Laboratory-simulated biosolarization trial

To evaluate the impact of biosolarization conditions on isothiocyanate production from entire *B. juncea* plant tissues in soil, a complementary laboratory-simulated biosolarization experiment was conducted with a greater amendment incorporation rate than is generally used in the field to allow for more dynamic range in isothiocyanate measurements and increase the likelihood of detecting differences among soil treatments. This approach allowed for quantifiable detection of isothiocyanates to support a mechanistic understanding of their production during biosolarization. Similar laboratory-simulated conditions have been used in previous studies which also seek to understand underlying mechanisms for pest control during biosolarization [21,42–44]. For this experiment, aliquots of *B. juncea* tissues and soil were harvested from the field trial prior to incorporation. The moisture content of the *B. juncea* tissues and soil was measured using the same method as in the field trial to determine the mass of each necessary to achieve a 25% dry-basis amendment rate. The maximum water holding capacity following free drainage under gravity of the *B. juncea* tissues and soil was measured to determine the volume of water necessary to achieve 80% of the maximum value for the mixtures. *B. juncea* tissues were then blended to simulate the disruption caused by field mowing

**Table 1**
Estimated cover crop amendment rates for each field.

| Cover crop treatment | Total and plant type-specific amendment rates (kg $ha^{-1}$)[a,b] | | | |
|---|---|---|---|---|
| | Total | *B. juncea* | *V. villosa* | Weeds |
| *B. juncea* sole crop | 5697.9 ± 335.2 | 5614.0 ± 335.0 | N/A | 83.9 ± 12.0 |
| *B. juncea-V. villosa* mixture crop | 4941.7 ± 1589.0 | 2946.4 ± 1263.1 | 1858.9 ± 960.5 | 136.4 ± 82.7 |
| Control | 144.3 ± 31.8 | N/A | N/A | 144.3 ± 31.8 |

[a] Values represent mean dry amendment rate.
[b] Values are presented as mean ± 1 standard deviation and n = 3 for all groups.

(Waring Commercial Laboratory Blender).

To simulate biosolarization conditions, the blended *B. juncea* tissues were thoroughly mixed with soil and water at the given rates, then placed in microcosm bioreactors (50 mL). The bioreactors were maximally filled with 65 g of the soil-*B. juncea* mixture, then immediately sealed and incubated at 45 °C (Thermo Scientific Heratherm Oven) to simulate the high temperature and low oxygen conditions present during biosolarization. Additional bioreactors were filled with the soil-*B. juncea* mixture, but left unsealed and incubated at 25 °C to simulate conventional biofumigation practices. Bioreactors were also filled with unamended soil and either (1) sealed and incubated at 45 °C to simulate solarization conditions, or (2) left unsealed and incubated at 25 °C as untreated controls. Soil was extracted from the bioreactors 0.5, 2, 4, 8, 24, and 48 h after the onset of the simulated biosolarization experiment to test isothiocyanate concentrations for each treatment. These timepoints were selected to capture the typical residence time of isothiocyanate in soil based on previous research [45]. Separate bioreactors were constructed for each soil extraction timepoint to minimize losses to volatilization. Incubators and bioreactor seals were checked daily to ensure consistent temperatures and anaerobic conditions throughout the experimental period. Bioreactors were constructed in triplicate for each treatment and timepoint, and two technical replicate isothiocyanate extractions were taken from each bioreactor.

Soil isothiocyanate extractions were performed using previously reported methods, with some modifications [45]. Ten grams of soil were extracted from the bioreactors and mixed with methanol to create a 1:1 dilution in 50-mL centrifuge tubes (Falcon, NY, USA). The samples were mixed by vortex (Fisher Analog Vortex, 120V) for 1 min, then centrifuged for 5 min at 8000 rpm (Eppendorf Centrifuge 5810R 15 Amp, Hamburg, Germany). A 2 mL aliquot was removed from the supernatant using a 3-mL polypropylene syringe and filtered through a 0.22-μm syringe filter (Thermo Fisher Scientific Inc., MA, USA). Isothiocyanates in the extracts were then quantified by HPLC-UV/Vis using an established method [45]. A 600 μL aliquot of the extracted filtrate was mixed with 600 μL of 100 mM $K_2HPO_4$ buffer and 200 μL of 35 mM 1,2-benzenedithiol/1% mercaptoethanol solution in 2-mL HPLC vials [45]. Isothiocyanates react with the 1,2-benzenedithiol solution to quantitatively yield the derivation product 1,3-benzodithiole-2-thione which is amenable to ultraviolet spectrometric detection at 365 nm [46]. The vials were capped and inverted several times to mix, then incubated at 65 °C for 1 h. The resulting samples were analyzed by HPLC (Model 1260 Infinity II, Agilent Technologies Inc.) equipped a ZORBAX Eclipse Plus C18 column (4.6 × 150 mm, 3.5 μm, Agilent Technologies Inc.) and guard column (4.6 × 12.5 mm, 5 μm, Agilent Technologies Inc.). The mobile phase was 90% methanol (Millipore Sigma, MA, USA) in Milli-Q water with an isocratic flow rate of 1 mL $min^{-1}$. The run time was 15 min with an injection volume of 70 μL for each sample. The column temperature was set to 35 °C. Absorbance values were detected at 365 nm (1260 Infinity II, Agilent Technologies Inc.) and compared to standard solutions ranging from 0.49 to 250 mg $L^{-1}$ prepared using derivatized phenethyl isothiocyanate as an external standard. Measured concentrations were normalized to the extract concentration in the HPLC vials and moisture content of each sample to yield the concentration per unit dry weight of soil. Cumulative soil isothiocyanate exposure was calculated by summing measured concentrations across sequential sampling timepoints over the course of the treatment period.

### 2.3. Data analysis

Analysis of variance (ANOVA) with post-hoc Tukey's Honest Significant Difference (HSD) tests were used to compare cumulative total soil organic acid exposure, cumulative individual organic acid exposures, residual phytotoxicity, moisture content, pH, and electrical conductivity values across treatments. Analysis of covariance (ANCOVA) tests were used to compare the effects of soil treatment and time on cumulative soil organic acid exposure, phytotoxicity, moisture content, pH, and electrical conductivity. Additional analyses were performed using ANOVA with Tukey's HSD by aggregating the cover crop amended-biosolarized treatment (MONO-B and MIX-B) values and the cover crop amended-control (MONO-C and MIX-C) values. These combinations were used to assess how cover cropping with and without the biosolarization treatment impact cumulative organic acid exposure in the soil, regardless of cover crop species. These aggregations were conducted by grouping the data from the given treatments together, then performing the relevant statistical tests on the group as a whole. When necessary, Yeo-Johnson transformations were applied prior to ANOVA to comply with the homoscedastic assumptions of the test. Yeo-Johnson transformations were performed in R-Studio (version 2023.12.1 + 402, RStudio). Nonparametric Wilcoxon rank-sum tests were used to compare cumulative and timepoint-specific soil isothiocyanate exposures and concentrations between the *B. juncea* amended-biosolarized treatment and the *B. juncea* amended-control. The family-wise error rate equaled 0.05 for all statistical analyses and post-hoc tests. Statistical analyses were performed using JMP-pro software (version 18, JPM Statistical Discovery).

## 3. Results

### 3.1. Soil organic acid production

Soil treatment and biosolarization time showed significant main effects on total cumulative organic acid exposure in the soil ($P < 0.0001$ for all). However, the interaction effect of these factors was not significant ($P = 0.1103$; Fig. 1). Significant differences in cumulative soil organic acid exposures were detected on days 0, 2, 3, 6, and 10 ($P = 0.0324$, 0.0248, 0.0030, <0.0001, 0.0009, respectively), with no significance detected on day 1 ($P = 0.1556$). On days 0 and 2, the cumulative soil organic acid exposure in MONO-B was significantly greater than U-S (day 0 mean = 147.4 and 0.0 μg·h $g^{-1}$ dry soil, and day 2 mean = 163.1 and 5.3 μg·h $g^{-1}$ dry soil, respectively). On day 3, cumulative soil organic acid exposure in MONO-B, MIX-B, and MONO-C was significantly greater than U-S (mean = 225.2, 162.9, 94.15, and 5.3 μg·h $g^{-1}$ dry soil, respectively). On day 6, cumulative soil organic acid exposure was significantly greater in MONO-B, MIX-B, MONO-C, and MIX-C compared to U-S (mean = 549.3, 488.3, 112.6, 190.7, and 5.3 μg·h $g^{-1}$ dry soil, respectively). Additionally, on day 6, organic acid exposure in MONO-B and MIX-B was significantly greater than in U-C (mean = 20.9 μg·h $g^{-1}$ dry soil for U-C). On day 10, cumulative soil organic acid exposure was significantly greater in MONO-B and MIX-B compared to U-S and U-C (mean = 809.6, 687.9, 25.6, and 60.2 μg·h $g^{-1}$ dry soil, respectively). Additionally, when the data were grouped for comparison of biosolarization versus non-biosolarization, soil in the cover crop amended-biosolarized group (MONO-B and MIX-B) resulted in significantly higher organic acid exposures than the cover crop amended-control group (MONO-C and MIX-C) on day 6 of the biosolarization period ($P = 0.0143$, mean = 518.8 and 146.1 μg·h $g^{-1}$ dry soil for the cover crop amended-biosolarized group and cover crop amended-control group, respectively). This result also remained consistent on day 10 of the biosolarization period ($P = 0.0025$, mean = 748.7 and 172.4 μg·h $g^{-1}$ dry soil for the cover crop amended-biosolarized group and cover crop amended-control group, respectively). Organic acids were not detected in any soil treatment or control throughout the remediation period.

Soil treatment also resulted in a significant effect on final cumulative acetic acid, butyric acid, and citric acid exposure in the soil ($P < 0.0001$, $P = 0.0004$ and 0.0025, respectively; Fig. 2A and B). Both biosolarized treatments (MONO-B and MIX-B) resulted in significantly greater final cumulative soil acetic acid exposure compared to all other treatments and controls (mean = 493.5, 505.9, 9.7, 20.7, 16.1, and 0.0 μg·h $g^{-1}$ dry soil for MONO-B, MIX-B, MONO-C, MIX-C, U-S, and U-C, respectively). Final cumulative soil butyric acid exposure was significantly greater in MONO-B compared to all other treatments and controls (mean = 75.4

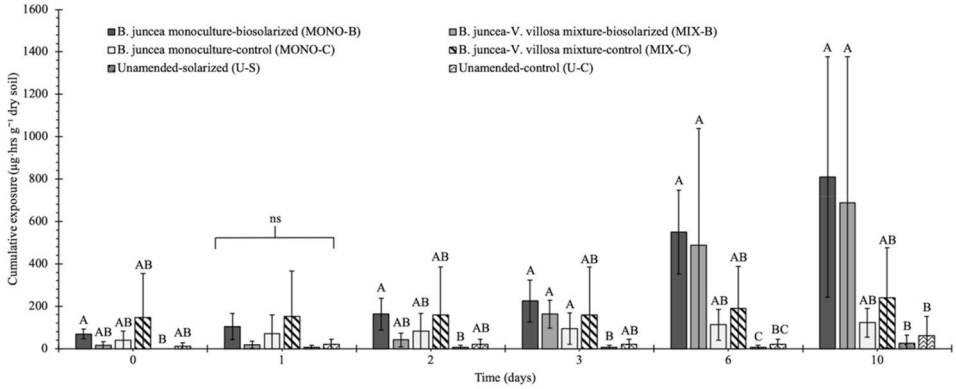


**Fig. 1.** Total cumulative organic acid exposure in soil during biosolarization with cover crop residue amendments. Organic acid concentrations were detected on days 0, 1, 2, 3, 6, and 10 of the biosolarization period. Different letters indicate significant differences within timepoint groups according to ANOVA with Tukey's HSD following a Yeo-Johnson transformation ($P < 0.05$; $\lambda = 0.101$), and ns indicates no significant differences were detected across the group. Error bars indicate ±1 standard deviation and $n = 4$.

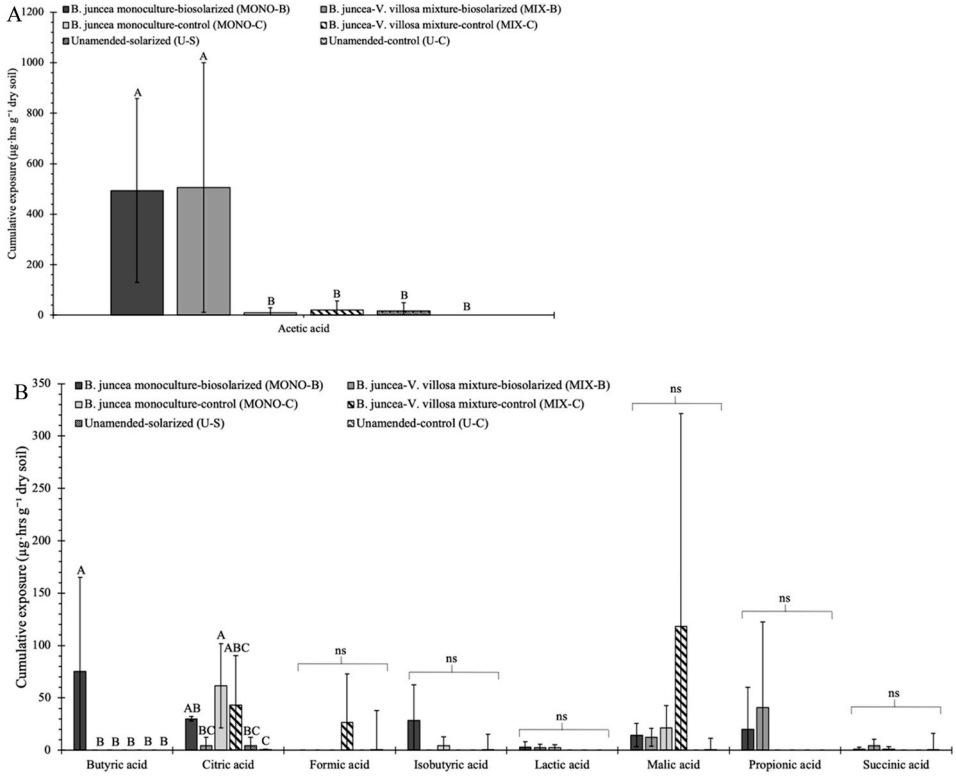


**Fig. 2.** Final cumulative individual organic acid exposure in soil following biosolarization with cover crop residue amendments. (A) Acetic acid and (B) butyric, citric, formic, isobutyric, lactic, malic, propionic, and succinic acids are shown. Different letters indicate significant differences among treatments for each individual organic acid according to ANOVA with Tukey's HSD following a Yeo-Johnson transformation ($P < 0.05$; $\lambda = -0.112$, $-1.803$, $-0.051$, $-2.634$, $-1.238$, $-2.255$, $-0.193$, $-2.420$, and $-1.368$ for acetic, butyric, citric, formic, isobutyric, lactic, malic, propionic, and succinic, respectively), and ns indicates no significant differences were detected. Error bars indicate ±1 standard deviation and $n = 4$ for all.

µg·h $g^{-1}$ dry soil for MONO-B and 0.0 µg·h $g^{-1}$ dry soil for all other treatments and controls). Final cumulative soil citric acid exposure was significantly greater in MONO-B and MONO-C compared to U-C (mean = 30.0, 61.4, and 0.4 µg·h $g^{-1}$ dry soil, respectively). Additionally, MONO-C resulted in a significantly higher cumulative soil citric acid exposure compared to MIX-B and U-S at the end of the biosolarization period (mean = 4.2 and 4.1 for MIX-B and U-S, respectively). No significant differences in final cumulative soil formic, isobutyric, lactic, malic, propionic, and succinic acid exposure were detected across the treatments ($P = 0.4566$, 0.2578, 0.2652, 0.0751, 0.6046, and 0.4615, respectively).

Soil treatment showed a significant main effect on total soil organic acid concentration during the biosolarization period ($P < 0.0001$), while biosolarization time did not show a significant effect ($P = 0.6017$; Fig. 3A). Significant differences in total soil organic acid concentrations were detected on days 0, 2, and 3 ($P = 0.0334$, 0.0099, and $< 0.0001$, respectively), with no differences detected on days 1, 6, and 10 ($P = 0.9003$, 0.1230, and 0.6598, respectively). On day 0, the total soil organic acid concentration in MONO-B was significantly higher compared to U-S (mean = 68.9 and 0.0 µg $g^{-1}$ dry soil, respectively). On day 2, MONO-B resulted in a significantly higher total soil organic acid concentration compared to U-S and U-C (mean = 60.2 µg $g^{-1}$ dry soil for

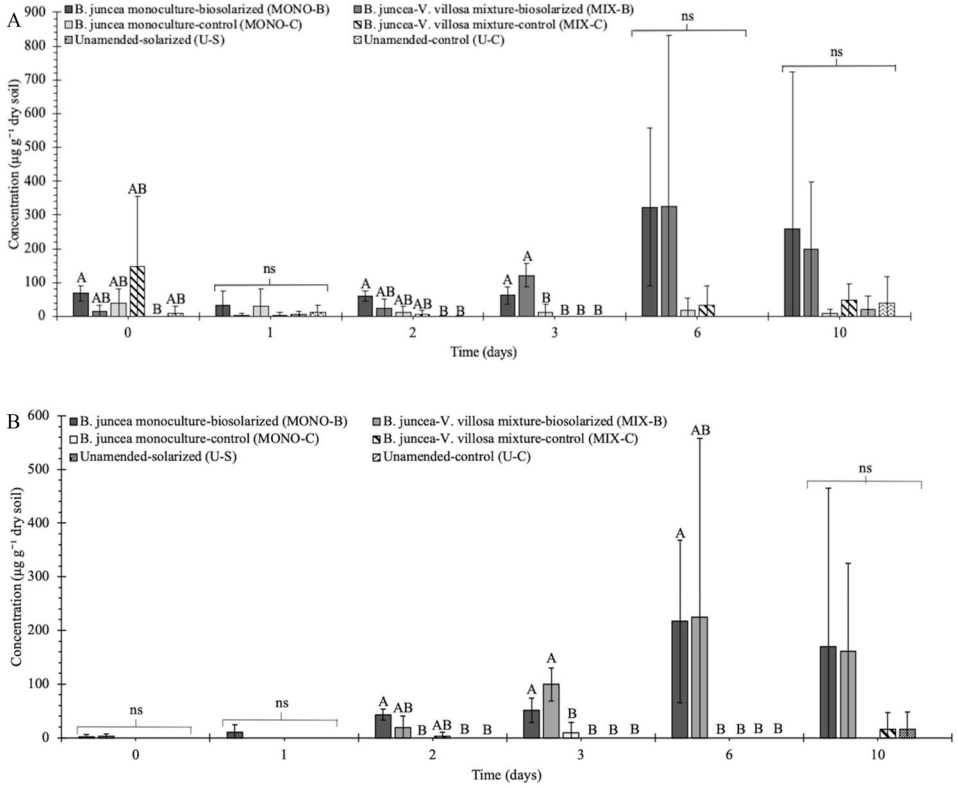


**Fig. 3.** Soil organic acid concentration during biosolarization with cover crop residue amendments. (A) Total organic acid concentration and (B) acetic acid concentration in the soil. Concentrations were detected on days 0, 1, 2, 3, 6, and 10 of the biosolarization period. Different letters indicate significant differences within timepoint groups according to ANOVA with Tukey's HSD following a Yeo-Johnson transformation ($P < 0.05$, $\lambda = -0.296$ and $-0.914$ for total organic acid concentration and acetic acid concentration, respectively), and ns indicates no significant differences were detected. Error bars indicate ±1 standard deviation and $n = 4$ for all.

MONO-B and 0.0 μg $g^{-1}$ dry soil for U-S and U-C). On day 3, the total soil organic acid concentration in MONO-B and MIX-B was significantly greater than in all other treatments and controls (mean = 62.2 and 121.3 μg $g^{-1}$ dry soil for MONO-B and MIX-B and mean = 0.0-11.6 μg $g^{-1}$ dry soil for all other treatments and controls).

Soil treatment and biosolarization time also showed significant main effects on soil acetic acid concentration during the biosolarization period ($P < 0.0001$ and $P = 0.0370$, respectively). However, the interaction effect of these factors was not significant ($P = 0.3873$; Fig. 3B). Significant differences in soil acetic acid concentration were detected on days 2, 3, and 6 ($P = 0.0011$, $< 0.0001$, and 0.0114, respectively), with no differences detected on days 0, 1, and 10 ($P = 0.6046$, 0.0504, and 0.1572, respectively). On day 2, the soil acetic acid concentration in MONO-B was significantly higher than in MONO-C, U-S, and U-C (mean = 43.1 μg $g^{-1}$ dry soil for MONO-B and 0.0 μg $g^{-1}$ dry soil for MONO-C, U-S, and U-C). On day 3, MONO-B and MIX-B both resulted in significantly higher soil acetic acid concentrations compared to all other treatments and controls (mean = 51.4 and 99.4 μg $g^{-1}$ dry soil for MONO-B and MIX-B, respectively and mean = 0.0-9.7 μg $g^{-1}$ dry soil for all other treatments and controls). On day 6, the concentration of acetic acid in the soil was significantly greater in MONO-B compared to MONO-C, MIX-C, U-S, and U-C (mean = 216.8 μg $g^{-1}$ dry soil for MONO-B and 0.0 μg $g^{-1}$ dry soil for MONO-C, MIX-C, U-S, and U-C).

### 3.2. Residual phytotoxicity

Soil treatment and remediation time showed significant main effects on residual phytotoxicity ($P = 0.0234$ and $< 0.0001$, respectively). The interaction effect for these factors was also significant ($P = 0.0018$). Generally, the germination index (GI) of all treatments increased between 0 and 29 days of remediation, indicating phytotoxicity was alleviated with time (Fig. 4). Immediately following the biosolarization period, the GI of both biosolarized treatments (MONO-B and MIX-B) and U-S was less than 80%, indicating soil phytotoxicity (mean = 55.7%, 69.0%, and 76.7%, respectively). Additionally, the GI of soil in MONO-B was significantly lower than soil in MONO-C at the beginning of the remediation period ($P = 0.0344$, mean = 110.18% for MONO-C). After 8

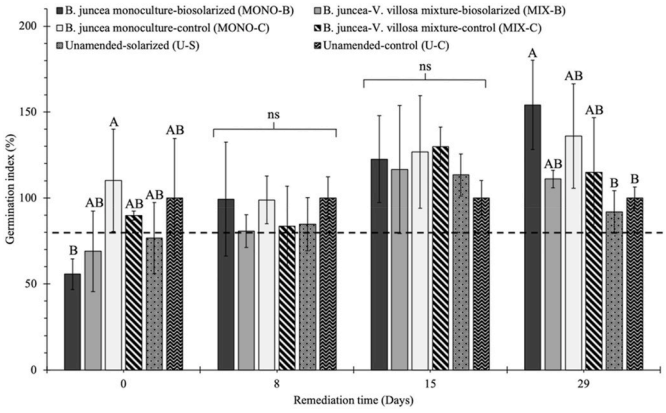


**Fig. 4.** Germination index of radish seeds in soil during the remediation period following biosolarization with cover crop residue amendments. Remediation time ranged from 0 to 29 days following the end of the biosolarization period. Data were normalized to the GI values of the unamended-control (U-C) soil. Different letters indicate significant differences within timepoint groups according to Tukey's HSD, and ns indicates no significant differences were found across the group ($P < 0.05$). Soil treatments with mean values below the horizontal dashed line are considered phytotoxic (GI < 80%). Error bars indicate ±1 standard deviation and $n = 4$.

days of remediation, soil from MONO-B, MIX-B, and U-S was no longer considered phytotoxic (mean = 99.3%, 80.8%, and 84.8%, respectively), and this result persisted through the remainder of the remediation period. Additionally, the GI of soil in MONO-B was significantly higher than soil from U-S and U-C after 29 days of remediation, demonstrating a potential fertilization effect in the soil (P = 0.0082 and 0.0248, respectively, and mean = 91.88% and 100.00% for U-S and U-C, respectively). Soil from both of the cover crop controls (MONO-C and MIX-C) was not considered phytotoxic throughout the studied remediation period.

### 3.3. Soil moisture content

Soil treatment and time showed significant main effects on soil moisture content (P = 0.022 and < 0.0001, respectively). However, the interaction of these factors was not significant (P = 0.1968). Generally, the soil moisture content increased between days 0 and 2 for all treatments and controls, then decreased throughout the remainder of the biosolarization and remediation periods (Fig. 5). Significant differences in moisture content among treatments were observed on days 10, 25, and 39 after the start of the biosolarization period (P = 0.0057, 0.0157, and 0.0281, respectively). On day 10, the moisture content of MONO-B and U-S was significantly higher than U-C (mean = 18.1%, 17.8%, and 14.6% for MONO-B, U-S, and U-C, respectively). Additionally, on day 25, the moisture content of MONO-B and MIX-B was significantly higher than U-C (mean = 14.6% for both MONO-B and MIX-B, and 11.9% for U-C). However, on day 39, the Tukey's HSD test lacked sufficient sensitivity to distinguish differences among treatments.

### 3.4. Soil pH and electrical conductivity

Soil treatment did not show a significant effect on soil pH, while biosolarization and remediation time did show a significant main effect (P = 0.8661 and 0.0059, respectively). Generally, soil pH across all treatments decreased from day 0 to day 2, then increased from day 2 to day 6, followed by a gradual decrease from day 6 through day 39 (Fig. 6A). Additionally, significant differences in pH were detected within individual timepoints across treatments. During the biosolarization period, these differences were detected on days 2 and 3 (P = 0.0216 and 0.0131, respectively). On day 2, the pH in MONO-C was significantly lower than in U-C (mean = 6.5588 and 7.0763, respectively). Whereas, on day 3, the pH of MONO-B and MIX-B was significantly lower than U-S (mean = 6.6650, 6.6813, and 7.1500, respectively). Additionally, during the remediation period, the pH of MONO-B was significantly lower than in MIX-C and U-C on day 39 (P = 0.0140 and mean = 6.9125, 7.0850, and 7.1013, respectively).

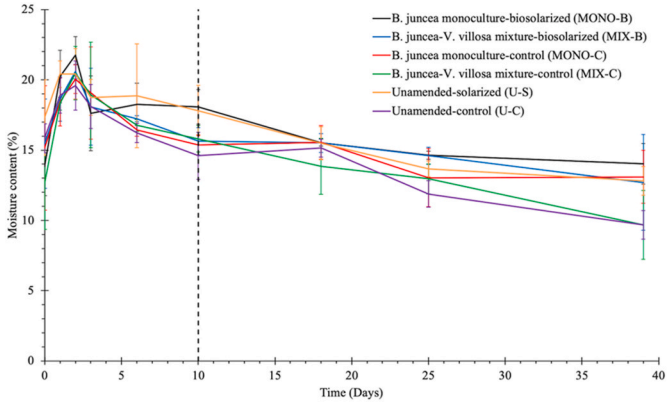


**Fig. 5.** Soil moisture content during and after biosolarization with cover crop residue amendments. The moisture content is reported on a g $g^{-1}$ wet weight basis. The vertical dashed line delineates the end of the biosolarization period (days 0-10) and beginning of the remediation period (days 10-39). Error bars indicate ±1 standard deviation and n = 4.

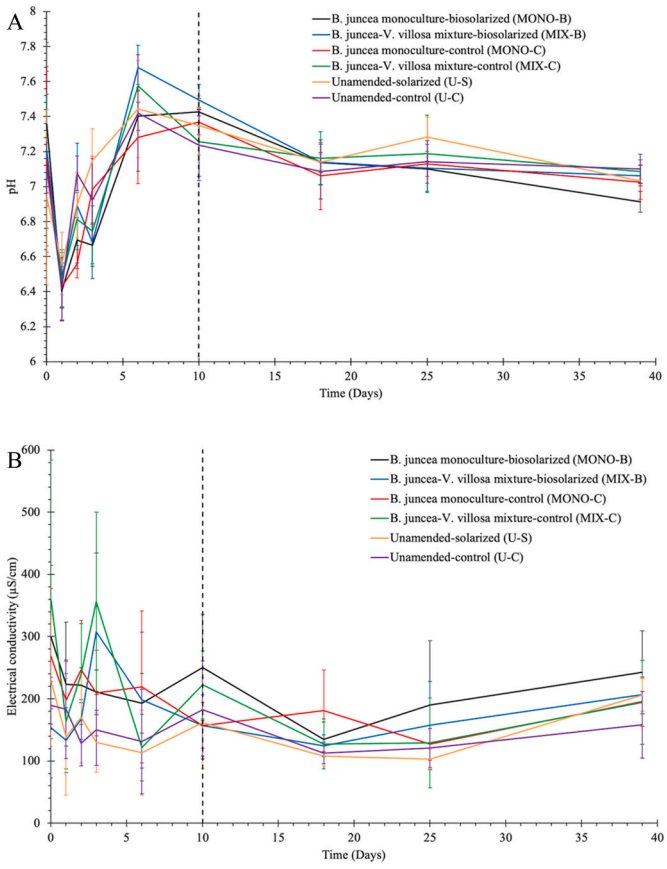


**Fig. 6.** Soil (A) pH and (B) electrical conductivity during the biosolarization and remediation periods with cover crop residue amendments. The vertical dashed line delineates the end of the biosolarization period (days 0-10) and beginning of the remediation period (days 10-39). Error bars indicate ±1 standard deviation and n = 4.

Soil treatment showed a significant effect on soil electrical conductivity (P < 0.0001). However, biosolarization and remediation time did not show a significant effect (P = 0.0981). Generally, the electrical conductivity of soils amended with cover crops (MONO-B, MIX-B, MONO-C, and MIX-C) was marginally higher than the unamended soils (U-S and U-C) during the biosolarization period (day 0-10; Fig. 6B). During the remediation period (day 10-39), the electrical conductivity of all treatments did not differ. Within individual timepoints, significant differences in electrical conductivity were detected across treatments on days 2 and 3 of the biosolarization period (P = 0.0493 and 0.0086, respectively). On day 2, the Tukey's HSD test lacked sufficient sensitivity to distinguish differences among treatments. However, on day 3, the electrical conductivity of soil from MIX-B and MIX-C was significantly higher than U-S (mean = 2.4600, 2.5294, and 2.0897 µS $cm^{-1}$, respectively). Additionally, the electrical conductivity in MIX-C was significantly higher than in U-C on day 3 (mean = 2.1515 µS $cm^{-1}$ for U-C).

### 3.5. Soil isothiocyanate production

Soil treatment and incubation time showed significant main effects on soil isothiocyanate exposure (P < 0.0001 for both). Additionally, the interaction effect for these factors was also significant (P < 0.0001). Isothiocyanates were exclusively detected in soil from the *B. juncea*-biosolarized treatment and the *B. juncea*-control throughout the test period, with zero detection in the unamended-solarized treatment and the unamended-control. In the *B. juncea*-biosolarized treatment, mean soil isothiocyanate concentrations peaked after 4 h of incubation, while

concentrations in the *B. juncea*-control peaked after 8 h of incubation (Fig. 7A). After 48 h of incubation, mean isothiocyanate concentrations were significantly higher in the *B. juncea*-biosolarized treatment compared to the *B. juncea*-control (P = 0.0318, mean = 9.85 and 0.00 μg $g^{-1}$ dry soil, respectively). Additionally, cumulative isothiocyanate exposure was generally higher in the *B. juncea*-biosolarized treatment compared to the *B. juncea*-control across the test period (Fig. 7B). This trend was reflected in the final cumulative isothiocyanate exposure values, where the *B. juncea*-biosolarized treatment was significantly higher than the *B. juncea*-control (P = 0.0404, mean = 539.3 and 421.3 μg·h $g^{-1}$ dry soil for *B. juncea*-biosolarized and *B. juncea*-control, respectively).

## 4. Discussion

This study demonstrated the effect of biosolarization with cover crop residue amendments on the production of common biochemical pesticides in the soil, in addition to quantifying residual soil phytotoxicity and soil properties. Specifically, *B. juncea* cover crops were grown as monocultures and in mixtures with *V. villosa* as *in situ* amendment sources for a field-scale biosolarization trial to evaluate their impact on soil organic acid biochemical pesticide production, post-treatment residual phytotoxicity, moisture content, pH, and electrical conductivity. Additionally, this study evaluated the effect of simulated-biosolarization conditions on soil isothiocyanate biochemical pesticides from *B. juncea* cover crop amendments to provide a mechanistic understanding of their production during biosolarization. The results highlight measurable increases in biochemical pesticide production in the soil from the cover crop amendments during biosolarization. Biosolarization with cover crop residue amendments significantly increased organic acid biochemical pesticide exposure in the soil compared to cover crop-amended, non-biosolarized soil and unamended soil controls. In particular, results suggest biosolarization stimulates the production of acetic acid from the cover crop amendments. Similarly, simulated-biosolarization conditions resulted in significantly increased isothiocyanate biochemical pesticide exposure in the soil compared to cover crop amended, non-biosolarized soil and unamended soil controls. Additionally, this study demonstrated the potential for a relatively rapid alleviation of residual phytotoxicity following biosolarization with cover crop residue amendments, correlating to a short remediation period, as well as minor changes in soil moisture content, pH, and electrical conductivity during the biosolarization and remediation periods.

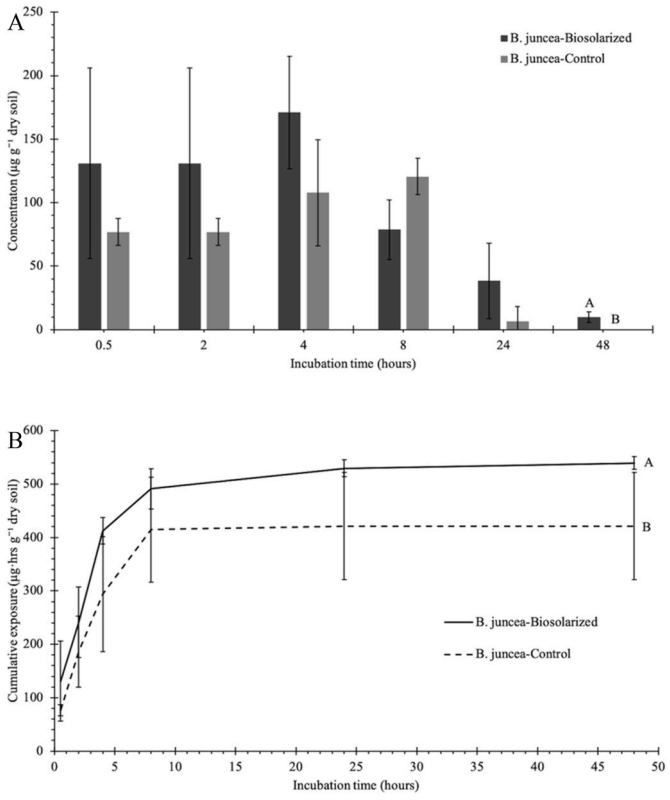


**Fig. 7.** Soil isothiocyanate concentrations detected in the soil following simulated-biosolarization with *B. juncea* cover crop residue amendments. (A) Soil isothiocyanate concentrations within individuals incubation times, including 0.5, 2, 4, 8, 24, and 48 h. (B) Cumulative soil isothiocyanate exposure across the test period. Isothiocyanates were not detected in samples from the solarized and untreated controls and, therefore, are not included. Different letters indicate significant differences in isothiocyanate concentrations within individual timepoints or final cumulative isothiocyanate exposure values at the end of the test period between the *B. juncea*-biosolarized treatments and the *B. juncea*-control according to Wilcoxon rank-sum test (P < 0.05). Error bars represent ±1 standard deviation and n = 3 for all.

The observed increase in soil organic acid biochemical pesticide exposure in the biosolarized treatments can likely be attributed to multiple mechanisms and correlate to enhanced soil pest suppression. Generally, the increased temperature, high moisture, and low oxygen conditions present during biosolarization likely created environmental conditions which favor the production of organic acids. These findings are consistent with previous research which has shown the potential for elevated temperatures and reduced oxygen availability to increase organic acid concentrations in soil [34,47]. Specifically, elevated organic acid concentrations during biosolarization are often associated with relative increases in fermentative microbial populations. Under the anaerobic and elevated temperature conditions created during biosolarization, microbial communities shift toward organisms capable of metabolizing labile carbon substrates from decomposing amendment material. Research has shown biosolarization conditions favor the proliferation of several bacterial phyla, including Firmicutes, Actinobacteria, and Proteobacteria, which are known to effectively contribute to organic matter decomposition and the production of organic acids, including those found in this study [9,34,48]. In particular, Firmicutes include many obligate or facultative anaerobes that are well adapted to oxygen-limited environments and are capable of producing a range of short-chain organic acids. Additionally, the tarp cover over the soil surface during biosolarization likely increased the residence time of organic acids in the soil by preventing volatilization to the atmosphere. Previous research has also shown organic acid biochemical pesticides contribute to the suppression of a variety of soil pests during biosolarization, including weeds and phytoparasitic *Fusarium oxysporum* and nematode species [18,21,49]. In the present study, the organic acid concentration profile was composed predominantly of acetic acid in the biosolarized treatments. Acetic acid has specifically been characterized as particularly effective for suppressing populations of soil pests [50]. Additionally, a previous study focusing on biosolarization with cover crop residue amendments cited improved weed suppression in the biosolarized treatments [13]. Therefore, it is likely the observed increase in soil organic acid biochemical pesticide exposure is capable of contributing to soil pest control during biosolarization with cover crop residue amendments. Additionally, the organic acid concentrations detected in this study were comparable to previous biosolarization research with food processing residue amendments using similar amendment rates. In this study, total soil organic acid concentrations at the end of the biosolarization period were approximately 350 μg $g^{-1}$ dry soil, which is relatively similar to the concentrations found in biosolarization experiments using almond, wheat bran, and green waste residue amendments, ranging between 200 and 500 μg $g^{-1}$ dry soil [21,33,34]. No differences were detected between the *B. juncea* monoculture and *B. juncea-V. villosa* mixture, indicating the elevated organic acid concentrations can be obtained from the cover crop monoculture or mixture.

In addition to organic acids, this study indicates biosolarization

conditions also increase the exposure of isothiocyanate biochemical pesticides in soil amended with *B. juncea* cover crop residues. Samples exposed to simulated biosolarization conditions resulted in significantly higher cumulative isothiocyanate exposure compared to the cover crop-amended control, suggesting biosolarization increases the production of these biochemical pesticides in the soil compared to conventional biofumigation practices. Additionally, after 48 h, isothiocyanate concentrations in the biosolarized treatment were significantly higher than in the cover crop-amended control, suggesting biosolarization conditions may also increase isothiocyanate retention or prolong their formation period in the soil compared to conventional biofumigation. The observed increase in cumulative soil isothiocyanate biochemical pesticide exposure is likely attributed to the elevated temperature present during biosolarization, increasing the conversion rate of glucosinolates in the plant residue. While not directly measured, previous biofumigation research has discussed this potential for increases in ambient temperatures to enhance isothiocyanate formation, as observed in the present study [26,51]. Additionally, studies have discussed the potential for soil covers, such as tarps, to increase retention of isothiocyanates in the soil by reducing volatilization [26]. Therefore, the observed increase in isothiocyanate concentrations in the samples exposed to biosolarization conditions following 48 h of incubation can likely be attributed to the soil surface being sealed, thereby preventing volatilization to the surrounding atmosphere. In combination, the increase in cumulative exposure and concentration in isothiocyanate biochemical pesticides during simulated biosolarization implicate the potential for improved soil pest control. Previous research has characterized isothiocyanates as a potent biochemical pesticide, capable of suppressing a variety of soil pests [28–30]. However, conventional biofumigation practices have displayed variable soil pest control levels, and poor results are often associated with insufficient isothiocyanate formation [26,31,32]. Therefore, the combination of biosolarization treatments and biofumigation practices may offer improved consistency in soil pest control by increasing isothiocyanate biochemical pesticide concentrations and exposure in the soil. Additionally, the production of organic acid biochemical pesticides and the induction of high temperature and low oxygen environments during biosolarization provide complementary modes of pest suppression in the soil beyond conventional biofumigation practices. These implications are supported by a study which characterized improved weed control in soil biosolarized with cover crop residue amendments in comparison to conventional biofumigation practices [13]. Overall, the addition of biosolarization to biofumigation practices may lead to more consistent soil pest control through the combined effects of multiple stress mechanisms.

The results of this study also indicate residual soil phytotoxicity is present immediately following the biosolarization treatment, with a relatively short remediation period necessary for alleviation. Immediately following the field biosolarization trial, soil in both the *B. juncea* monoculture and *B. juncea-V. villosa* mixture biosolarization treatments was considered phytotoxic. However, after 8 days of remediation, these treatments were no longer considered phytotoxic, correlating to a relatively short period for phytotoxicity alleviation. Following biosolarization treatments, previous studies have shown soils can remain phytotoxic for extended periods, sometimes spanning 4 weeks or more with almond processing residues [15,35]. Extended remediation intervals are disadvantageous as they may disrupt planting timelines and jeopardize early crop development due to residual phytotoxicity. In comparison, the shorter remediation period found in this study may enhance the feasibility and timing of biosolarization within cropping schedules by limiting interruptions prior to cash crop establishment. Additionally, the fertilization effect observed in the *B. juncea* monoculture biosolarization treatment highlights the potential for increased plant growth following the biosolarization treatment. This result is consistent with a previous study which found biosolarization with *B. juncea* cover crop residue amendments resulted in increased eggplant growth and yield [13]. However, additional research conducted across a broader range of environmental and soil conditions is necessary to verify the observed short remediation period prior to larger adoption in agricultural systems.

Minor differences were detected in soil moisture content, pH, and electrical conductivity among the treatments over the course of this study. During the remediation period, the results suggest biosolarized soil amended with both *B. juncea* monoculture and *B. juncea-V. villosa* mixture cover crops may improve moisture retention. This finding can partially be attributed to the soil surface being covered with a tarp, thereby preventing evaporation during the first 10 days of the study period. However, since the solarized treatment did not exhibit the same change, this result may also be attributed to an increase in soil organic matter from the cover crop residue amendments. Soils with relatively high organic matter contents are well known to retain moisture more effectively than low organic matter soils [52]. Additionally, previous research has specifically shown incorporated cover crop material can increase water retention in upper soil layers [53]. The differences in soil moisture content observed in the present study are relatively small. However, future research may focus on determining whether biosolarization with cover crop residues also increases soil moisture retention following subsequent irrigation during cash crop cultivation. Meanwhile, this study also suggests biosolarization with cover crop amendments may result in minor soil acidification, particularly from *B. juncea* monoculture residues. Previous research has characterized soil acidification following biosolarization treatments, often due to fermentation of the amendment material [21,43]. However, since the biosolarized soils were no longer considered phytotoxic following 8 days of remediation in this study, soil acidification from biosolarization with cover crop residue amendments is likely not a major concern for subsequent crop growth. However, this result should be taken into consideration if these treatments are applied to cropping systems involving species highly sensitive to changes in soil pH. Additionally, the results show increases in electrical conductivity in soil amended with cover crops both with and without biosolarization. Cover crops and other organic amendments have exhibited similar increases in electrical conductivity due to the release of salts and other ionic compounds during degradation of the amendment material [54,55]. In this study, these differences in electrical conductivity were no longer present throughout the remediation period and are, therefore, unlikely to impact soil health following biosolarization. However, more research focusing on repeated treatments over several years is necessary to determine the extent of the effects of biosolarization with cover crop residue amendments on soil moisture retention, pH, and electrical conductivity.

While the results of this study evaluated underlying mechanisms contributing to observed soil pest suppression from biosolarization with cover crop residue amendments, further investigation is necessary to better understand the range of cover crop species and environmental conditions in which these treatments can be successfully applied. Different agricultural systems utilize a wide range of cover crop species, highlighting the need for future studies to evaluate the potential of additional species as amendment sources for biosolarization. Additionally, changes in soil texture and microbial communities may impact biochemical pesticide production, motivating future studies examining biosolarization with cover crop amendments under variable soil conditions. Results of these trials would help provide additional context regarding the extent to which biosolarization with cover crop residue amendments can be applied to reduce dependence on synthetic fumigants, while continuing to control soil pests in agriculture.

## 5. Conclusions

This study investigated biochemical pesticide production, residual phytotoxicity, and changes in soil properties following biosolarization with cover crop residue amendments. The results indicate biosolarization treatments increase the exposure of organic acid and isothiocyanate biochemical pesticides in the soil compared to conventional

cover cropping and biofumigation practices, increasing the potential for more consistent soil pest control. Meanwhile, the relatively short remediation period required to alleviate soil phytotoxicity helps minimize operational interruptions within the cropping system. This work also contributes to the theoretical understanding of alternative soil pest management by clarifying the biochemical mechanisms through which biosolarization with cover crop amendments contributes to pest control. Specifically, the findings demonstrate how soil heating interacts with the decomposition of cover crop residues to enhance the production and persistence of biologically-derived pesticidal compounds in the soil. These results highlight the capacity of biosolarization with cover crop residue amendments to be implemented in cropping systems to mitigate soil pests and reduce reliance on chemical fumigants. This approach also offers growers a potentially effective strategy to integrate pest suppression with soil health management. These treatments may be incorporated into Integrated Pest Management (IPM) practices and are particularly relevant for soil pest control in organic farming systems. Consequently, further investigations are warranted to examine whether these results are sustained with different cover crop species and under alternative environmental conditions.

## CRediT authorship contribution statement

**Shayne Morrissey:** Writing – review & editing, Writing – original draft, Visualization, Validation, Project administration, Methodology, Investigation, Formal analysis, Data curation, Conceptualization. **Carolina Conte:** Writing – review & editing, Visualization, Methodology. **Natalie Chueh:** Writing – review & editing, Investigation, Formal analysis, Data curation. **Vanessa Kalip:** Writing – review & editing, Investigation, Formal analysis, Data curation. **Christopher Simmons:** Writing – review & editing, Supervision, Project administration, Methodology, Funding acquisition, Conceptualization.

## Funding

This work was supported by the National Institute of Food and Agriculture [grant #20217000635315], the National Institute for Occupational Safety and Health [grant #5U54OH007550], and the USDA NIFA Multistate Research Project: The Science and Engineering for a Biobased Industry and Economy [grant #S1075].

## 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.

## Acknowledgements

We thank Cerruti R.R. Hooks, University of Maryland, for input regarding integrated pest control techniques at the interface of cover cropping and biosolarization. We also thank Bryan Pellissier and Alexa Sommers, UC Davis, for providing the field site and assistance in setting up the field trial. Finally, we thank Stephanie Chen, Shruti Parikh, Tate Lone, Zeyu Zhu, Adan Gallo Lopez, Brooke Stehle, and Abigail Nagano, UC Davis, for their help in setting up the field trial and soil sampling.

## Appendix A. Supplementary data

Supplementary data to this article can be found online at https://doi.org/10.1016/j.jafr.2026.102861.

## Data availability

Data will be made available on request.

## References

[1] L.W. Baker, D.L. Fitzell, J.N. Seiber, T.R. Parker, T. Shibamoto, M.W. Poore, K. E. Longley, R.P. Tomlin, R. Propper, D.W. Duncan, Ambient air concentrations of pesticides in California, Environ. Sci. Technol. 30 (1996) 1365–1368, https://doi.org/10.1021/es950608l.

[2] M. Oriel, S. Edmiston, S. Beauvais, T. Barry, M. O'Malley, Illnesses associated with chloropicrin use in California agriculture, 1992 – 2003, in: D.M. Whitacre (Ed.), Reviews of Environmental Contamination and Toxicology, vol.200, Springer US, Boston, MA, 2009, pp. 1–31, https://doi.org/10.1007/978-1-4419-0028-9_1.

[3] C.H. Bell, Fumigation in the 21st century, Crop Prot. 19 (2000) 563–569, https://doi.org/10.1016/S0261-2194(00)00073-9.

[4] O. US EPA, Methyl bromide. https://www.epa.gov/ods-phaseout/methyl-bromide, 2015. (Accessed 29 July 2025).

[5] O. Us Epa, The Plain English guide to the Clean air act. https://www.epa.gov/clean-air-act-overview/plain-english-guide-clean-air-act, 2015. (Accessed 29 July 2025).

[6] California Council on Science and Technology, Fumigant Use in California and an Assessment of Available Alternatives, California Council on Science and Technology, 2025. Sacramento, CA, https://www.cdpr.ca.gov/wp-content/uploads/2025/03/ccst_fumigants_study.pdf. (Accessed 26 June 2025).

[7] E. Rosskopf, F.D. Gioia, I. Vincent, J. Hong, X. Zhao, Impacts of the ban on the soil-applied fumigant Methyl bromide, Phytopathology® 114 (2024) 1161–1175, https://doi.org/10.1094/PHYTO-09-23-0345-IA.

[8] P. Domínguez, L. Miranda, C. Soria, B. de los Santos, M. Chamorro, F. Romero, O. Daugovish, J.M. López-Aranda, J.J. Medina, Soil biosolarization for sustainable strawberry production, Agron. Sustain. Dev. 34 (2014) 821–829, https://doi.org/10.1007/s13593-014-0211-z.

[9] E.A. Shea, J.D. Fernández-Bayo, A.K. Hodson, A.E. Parr, E. Lopez, Y. Achmon, J. Toniato, J. Milkereit, R. Crowley, J.J. Stapleton, J.S. VanderGheynst, C. W. Simmons, Biosolarization restructures soil bacterial communities and decreases parasitic nematode populations, Appl. Soil Ecol. 172 (2022) 104343, https://doi.org/10.1016/j.apsoil.2021.104343.

[10] A.M. Pastrana, E.A. Shea, J.D. Fernandez-Bayo, B. Allison, D.C. Watson, J. Toniato, T.R. Gordon, C.W. Simmons, Impact of biosolarization with almond hull and shell amendments for the control of Fusarium oxysporum f. sp. lactucae in a lettuce/tomato cropping system, Crop Prot. 152 (2022) 105856, https://doi.org/10.1016/j.cropro.2021.105856.

[11] C.W. Simmons, H. Guo, J.T. Claypool, M.N. Marshall, K.M. Perano, J.J. Stapleton, J.S. VanderGheynst, Managing compost stability and amendment to soil to enhance soil heating during soil solarization, Waste Manag. 33 (2013) 1090–1096, https://doi.org/10.1016/j.wasman.2013.01.015.

[12] C.R. Conte, A. Nagano, M.C. Gentry, C.W. Simmons, Assessment of spent mushroom substrate recycling via soil biosolarization, Waste Manag. Bull. 3 (2025) 100264, https://doi.org/10.1016/j.wmb.2025.100264.

[13] S. Morrissey, N. Chueh, C. Conte, C. Simmons, Effect of cover cropping and biosolarization on eggplant growth, soil pests, and soil nitrogen, J. Agric. Food Res. 23 (2025) 102160, https://doi.org/10.1016/j.jafr.2025.102160.

[14] M. Ros, C. Garcia, M.T. Hernandez, A. Lacasa, P. Fernandez, J.A. Pascual, Effects of biosolarization as methyl bromide alternative for Meloidogyne incognita control on quality of soil under pepper, Biol. Fertil. Soils 45 (2008) 37–44, https://doi.org/10.1007/s00374-008-0307-1.

[15] Z. Haber, M. del M.R. Wilhelmi, J.D. Fernández-Bayo, D.R. Harrold, J.J. Stapleton, D. Toubiana, J.S. VanderGheynst, E. Blumwald, C.W. Simmons, N. Sade, Y. Achmon, The effect of circular soil biosolarization treatment on the physiology, metabolomics, and microbiome of tomato plants under certain abiotic stresses, Front. Plant Sci. 13 (2022), https://doi.org/10.3389/fpls.2022.1009956.

[16] M. Horowitz, Y. Regev, G. Herzlinger, Solarization for weed control, Weed Sci. 31 (1983) 170–179, https://doi.org/10.1017/S0043174500068788.

[17] M. Talavera-Rubia, M.D. Vela-Delgado, S. Verdejo-Lucas, A cost-benefit analysis of soil disinfestation methods against root-knot nematodes in mediterranean intensive horticulture, Plants 11 (2022), https://doi.org/10.3390/plants11202774.

[18] Y. Achmon, J.D. Fernández-Bayo, K. Hernandez, D.G. McCurry, D.R. Harrold, J. Su, R.M. Dahlquist-Willard, J.J. Stapleton, J.S. VanderGheynst, C.W. Simmons, Weed seed inactivation in soil mesocosms via biosolarization with mature compost and tomato processing waste amendments, Pest Manag. Sci. 73 (2017) 862–873, https://doi.org/10.1002/ps.4354.

[19] B.D. Aljawasim, C. Johnson, M. Manchester, J.B. Samtani, Evaluating soil solarization and mustard seed meal as preplant treatments for weed control in annual hill plasticulture strawberry production, Weed Technol. 39 (2025) e7, https://doi.org/10.1017/wet.2024.90.

[20] J. Muramoto, C. Shennan, M. Zavatta, G. Baird, L. Toyama, M. Mazzola, Effect of anaerobic soil disinfestation and mustard seed meal for control of charcoal rot in California strawberries, Int. J. Fruit Sci. 16 (2016) 59–70, https://doi.org/10.1080/15538362.2016.1199993.

[21] J.D. Fernandez-Bayo, E.A. Shea, A.E. Parr, Y. Achmon, J.J. Stapleton, J. S. VanderGheynst, A.K. Hodson, C.W. Simmons, Almond processing residues as a source of organic acid biopesticides during biosolarization, Waste Manag. 101 (2020) 74–82, https://doi.org/10.1016/j.wasman.2019.09.028.

[22] Z. Song, D. Yan, W. Fang, D. Zhang, X. Jin, Y. Li, Q. Wang, G. Wang, Q. Li, A. Cao, Response of strawberry fruit yield, soil chemical and microbial properties to anaerobic soil disinfestation with biochar and rice bran, Agriculture 13 (2023) 1466, https://doi.org/10.3390/agriculture13071466.

[23] T.L. Oldfield, Y. Achmon, K.M. Perano, R.M. Dahlquist-Willard, J. S. VanderGheynst, J.J. Stapleton, C.W. Simmons, N.M. Holden, A life cycle assessment of biosolarization as a valorization pathway for tomato pomace

utilization in California, J. Clean. Prod. 141 (2017) 146–156, https://doi.org/10.1016/j.jclepro.2016.09.051.
[24] A.L. Gimsing, J.A. Kirkegaard, Glucosinolates and biofumigation: fate of glucosinolates and their hydrolysis products in soil, Phytochem. Rev. 8 (2009) 299–310, https://doi.org/10.1007/s11101-008-9105-5.
[25] G.R. De Nicola, M. Bagatta, E. Pagnotta, D. Angelino, L. Gennari, P. Ninfali, P. Rollin, R. Iori, Comparison of bioactive phytochemical content and release of isothiocyanates in selected brassica sprouts, Food Chem. 141 (2013) 297–303, https://doi.org/10.1016/j.foodchem.2013.02.102.
[26] J.A. Kirkegaard, J. Matthiessen, Developing and refining the biofumigation concept, Agroindustria 3 (2004) 233–239, https://doi.org/10.1016/S0960-9822(97)70976-X.
[27] P. Lu, G. Gilardi, M.L. Gullino, A. Garibaldi, Biofumigation with Brassica plants and its effect on the inoculum potential of fusarium yellows of brassica crops, Eur. J. Plant Pathol. 126 (2010) 387–402, https://doi.org/10.1007/s10658-009-9543-y.
[28] U. Smolinska, M.J. Morra, G.R. Knudsen, R.L. James, Isothiocyanates produced by Brassicaceae species as inhibitors of Fusarium oxysporum, Plant Dis. 87 (2003) 407–412, https://doi.org/10.1094/PDIS.2003.87.4.407.
[29] N. Ntalli, P. Caboni, A review of isothiocyanates biofumigation activity on plant parasitic nematodes, Phytochem. Rev. 16 (2017) 827–834, https://doi.org/10.1007/s11101-017-9491-7.
[30] B. De Cauwer, J. Vanbesien, S. De Ryck, D. Reheul, Impact of Brassica juncea biofumigation on viability of propagules of pernicious weed species, Weed Res. 59 (2019) 209–221, https://doi.org/10.1111/wre.12358.
[31] S. Ashiq, S. Edwards, A. Watson, M. Back, Biofumigation for the management of Fusarium graminearum in a wheat-maize rotation, Pathogens 11 (2022) 1427, https://doi.org/10.3390/pathogens11121427.
[32] J.N. Matthiessen, J.A. Kirkegaard, Biofumigation and enhanced biodegradation: opportunity and challenge in soilborne Pest and disease management, Crit. Rev. Plant Sci. 25 (2006) 235–265, https://doi.org/10.1080/07352680600611543.
[33] K.V. Hestmark, J.D. Fernández-Bayo, D.R. Harrold, T.E. Randall, Y. Achmon, J. J. Stapleton, C.W. Simmons, J.S. VanderGheynst, Compost induces the accumulation of biopesticidal organic acids during soil biosolarization, Resour. Conserv. Recycl. 143 (2019) 27–35, https://doi.org/10.1016/j.resconrec.2018.12.009.
[34] C.W. Simmons, B. Higgins, S. Staley, L.D. Joh, B.A. Simmons, S.W. Singer, J. J. Stapleton, J.S. VanderGheynst, The role of organic matter amendment level on soil heating, organic acid accumulation, and development of bacterial communities in solarized soil, Appl. Soil Ecol. 106 (2016) 37–46, https://doi.org/10.1016/j.apsoil.2016.04.018.
[35] E. Shea, Z. Wang, B. Allison, C. Simmons, Alleviating phytotoxicity of soils biosolarized with almond processing residues, Environ. Technol. Innov. 23 (2021) 101662, https://doi.org/10.1016/j.eti.2021.101662.
[36] J. Acharya, M.G. Bakker, T.B. Moorman, T.C. Kaspar, A.W. Lenssen, A. E. Robertson, Time Interval between cover crop termination and planting influences Corn seedling disease, plant growth, and yield, Plant Dis. 101 (2017) 591–600, https://doi.org/10.1094/PDIS-07-16-0975-RE.
[37] USDA, Web soil Survey, Davis, CA. https://casoilresource.lawr.ucdavis.edu/gmap/, 2005. (Accessed 20 June 2025).
[38] A.C. Mitelut, M.E. Popa, Seed germination bioassay for toxicity evaluation of different composting biodegradable materials, Romanian Biotechnological Letters 16 (2011).
[39] D.N. Aslam, J.S. VanderGheynst, Predicting phytotoxicity of compost-amended soil from compost stability measurements, Environ. Eng. Sci. 25 (2008) 72–81, https://doi.org/10.1089/ees.2006.0284.
[40] S.M. Tiquia, N.F.Y. Tam, I.J. Hodgkiss, Effects of composting on phytotoxicity of spent pig-manure sawdust litter, Environ. Pollut. 93 (1996) 249–256, https://doi.org/10.1016/S0269-7491(96)00052-8.
[41] H.J. Ko, K.Y. Kim, H.T. Kim, C.N. Kim, M. Umeda, Evaluation of maturity parameters and heavy metal contents in composts made from animal manure, Waste Manag. 28 (2008) 813–820, https://doi.org/10.1016/j.wasman.2007.05.010.
[42] C. Li, P. Yu, Y. Zeng, B. Qiu, M. Chen, D. Cai, Z. Zhao, Z. Chen, S. Yaron, Y. Achmon, Impacts of soil biosolarization with eggshell waste on soil health, microbial communities, and gas emissions, Front. Sustain. Food Syst. 9 (2025), https://doi.org/10.3389/fsufs.2025.1543216.
[43] Y. Achmon, D.R. Harrold, J.T. Claypool, J.J. Stapleton, J.S. VanderGheynst, C. W. Simmons, Assessment of tomato and wine processing solid wastes as soil amendments for biosolarization, Waste Manag. 48 (2016) 156–164, https://doi.org/10.1016/j.wasman.2015.10.022.
[44] C.R. Conte, S. Morrissey, B.D. Hanson, C.W. Simmons, Biosolarization as an alternative method to inhibit parasitic Phelipanche ramosa germination, Front. Agron. 8 (2026), https://doi.org/10.3389/fagro.2026.1717415.
[45] M.J. Morra, J.A. Kirkegaard, Isothiocyanate release from soil-incorporated Brassica tissues, Soil Biol. Biochem. 34 (2002) 1683–1690, https://doi.org/10.1016/S0038-0717(02)00153-0.
[46] Y. Zhang, C.-G. Cho, G.H. Posner, P. Talalay, Spectroscopic quantitation of organic isothiocyanates by cyclocondensation with vicinal dithiols, Anal. Biochem. 205 (1992) 100–107, https://doi.org/10.1016/0003-2697(92)90585-U.
[47] J. Jiang, Y. Zhang, K. Li, Q. Wang, C. Gong, M. Li, Volatile fatty acids production from food waste: effects of pH, temperature, and organic loading rate, Bioresour. Technol. 143 (2013) 525–530, https://doi.org/10.1016/j.biortech.2013.06.025.
[48] Y. Achmon, J.T. Claypool, J.D. Fernández-Bayo, K. Hernandez, D.G. McCurry, D. R. Harrold, J. Su, B.A. Simmons, S.W. Singer, R.M. Dahlquist-Willard, J. J. Stapleton, J.S. VanderGheynst, C.W. Simmons, Structural changes in bacterial and fungal soil microbiome components during biosolarization as related to volatile fatty acid accumulation, Appl. Soil Ecol. 153 (2020) 103602, https://doi.org/10.1016/j.apsoil.2020.103602.
[49] J.D. Fernández-Bayo, T.E. Randall, D.R. Harrold, Y. Achmon, K.V. Hestmark, J. Su, R.M. Dahlquist-Willard, T.R. Gordon, J.J. Stapleton, J.S. VanderGheynst, C. W. Simmons, Effect of management of organic wastes on inactivation of Brassica nigra and Fusarium oxysporum f.sp. lactucae using soil biosolarization, Pest Manag. Sci. 74 (2018) 1892–1902, https://doi.org/10.1002/ps.4891.
[50] N. Momma, Biological Soil Disinfestation (BSD) of soilborne pathogens and its possible mechanisms, Japan Agricultural Research Quarterly: JARQ 42 (2008) 7–12, https://doi.org/10.6090/jarq.42.7.
[51] A.L. Gimsing, J.A. Kirkegaard, Glucosinolate and isothiocyanate concentration in soil following incorporation of Brassica biofumigants, Soil Biol. Biochem. 38 (2006) 2255–2264, https://doi.org/10.1016/j.soilbio.2006.01.024.
[52] R. Lal, Soil organic matter and water retention, Agron. J. 112 (2020) 3265–3277, https://doi.org/10.1002/agj2.20282.
[53] J. Wang, S. Zhang, U.M. Sainju, R. Ghimire, F. Zhao, A meta-analysis on cover crop impact on soil water storage, succeeding crop yield, and water-use efficiency, Agric. Water Manag. 256 (2021) 107085, https://doi.org/10.1016/j.agwat.2021.107085.
[54] R.A. Eigenberg, J.W. Doran, J.A. Nienaber, R.B. Ferguson, B.L. Woodbury, Electrical conductivity monitoring of soil condition and available N with animal manure and a cover crop, Agric. Ecosyst. Environ. 88 (2002) 183–193, https://doi.org/10.1016/S0167-8809(01)00256-0.
[55] B. Eghball, D. Ginting, J.E. Gilley, Residual effects of manure and compost applications on corn production and soil properties, Agron. J. 96 (2004) 442–447, https://doi.org/10.2134/agronj2004.4420.