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Differences in *p,p′*-DDE bioaccumulation from compost and soil by the plants *Cucurbita pepo* and *Cucurbita maxima* and the earthworms *Eisenia fetida* and *Lumbricus terrestris*

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Availability of p,p′-DDE to earthworms and plants was dramatically different in soil and compost.

Abstract

Two plant species, *Cucurbita pepo* and *Cucurbita maxima*, and two earthworm species, *Eisenia fetida* and *Lumbricus terrestris*, were exposed to soil and compost with equivalent *p,p′*-DDE contamination. Pollutant bioconcentration was equal in plant roots in both media, but translocation was higher in *C. pepo*. Bioaccumulation by *E. fetida* was approximately 6- and 3-fold higher than that by *L. terrestris* in the soil and compost, respectively. For all species, *p,p′*-DDE uptake was significantly greater from soil than from compost; 7- to 8-fold higher for plant roots and 3- to 7-fold higher for worms. Abiotic desorption from soil was approximately twice that from the compost. When all the data are normalized for organic-carbon content of the media, the contaminant is more tightly bound by soil than compost. Although the risk associated with *p,p′*-DDE is higher in soil than compost, important mechanistic differences exist in contaminant binding to organic carbon in the two media.
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Keywords: Compost; Earthworm; *Cucurbita*; Bioavailability; Bioaccumulation; *p,p′*-DDE

1. Introduction

Biological availability of soil pollutants is important because it controls the extent to which toxic substances can adversely affect organisms and ecosystems. Many variables affect bioavailability, including compound residence time in soil, pollutant chemistry, soil and environmental conditions, and species differences and interactions. A number of studies have demonstrated that aging of pollutants in soil can lead to reduced uptake by organisms (Nash and Woolson, 1967; Steinberg et al., 1987; Scribner et al., 1992; Erickson et al., 1993; Hatzinger and Alexander, 1995; Kelsey et al., 1997; Riaz et al., 2004; Swindell and Reid, 2006). This decreasing bioavailability occurs even as the total contaminant soil concentration remains constant. In addition to residence time, soil organic matter content, cation exchange capacity, and soil surface area have also been shown to influence the bioavailability of organic compounds (Chung and Alexander, 2002). In a similar study, the extractability of phenanthrene, pyrene, and benzo(*a*)pyrene from soil by non-rigorous methods differed as a function of soil organic matter and clay content (Swindell and Reid, 2006). Soil moisture content and wetting/drying cycles have also been shown to alter the uptake and extractability of contaminants (Kottler et al., 2001; Ruggiero et al., 2002). Pollutant properties such as hydrophobicity, charge/polarity, and other chemical characteristics affect availability as well (Alexander, 2000; Ruggiero et al., 2002). Finally, biological factors such as species differences and interactions appear to exert a substantial influence on the

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uptake of soil pollutants. We have previously shown that *p*, *p*′-DDE bioaccumulation by the earthworm *Eisenia fetida* was approximately an order of magnitude higher than that seen in *Lumbricus terrestris* and *Aporrectodea caliginosa* (Kelsey et al., 2005). Bioconcentration and translocation of the same compound have also been shown to be much higher in *Cucurbita pepo* than in other plants (White, 2001, 2002; Mattina et al., 2000). Furthermore, we reported that certain interactions between plants (either *C. pepo* or *Cucurbita maxima*) and earthworms (either *E. fetida* or *L. terrestris*) could have profound effects on bioaccumulation by both organisms (Kelsey and White, 2005). Current investigations are addressing these species-specific interactions.

Although much is known about the bioavailability of organic pollutants in soil, relatively little attention has been devoted to variables controlling the bioaccumulation of such compounds from compost. Many studies have documented the presence of organic pollutants (Strom, 2000; Lee et al., 2003) and heavy metals (Stilwell, 1993) in residential and commercial compost. Organic compounds detected include organochlorine pesticides such as chlordane, and DDT and its metabolites (Webber and Wang, 1995; Heberg et al., 1991; Meijer et al., 2001; Lee et al., 2003; Brandli et al., 2005). Because of their resistance to degradation, persistent organic pollutants (POPs) like those listed above are likely to remain in compost for extended periods of time. Moreover, due to the decomposition of biodegradable organic matter, the mass of compost can be reduced by 40–60%; as a result, the relative concentration of POPs may increase with time (Gronauer et al., 1997; Schleiss, 2003; Brandli et al., 2005). Despite evidence of wide-spread occurrence of low-to-moderate POP concentrations in compost, very little is known about the bioavailability of such compounds. Wågman et al. (1999) did report that bioaccumulation factors for chlorobenzenes and DDT metabolites in *E. fetida* were found to vary in different composts (Wågman et al., 1999). To our knowledge, no study designed to evaluate the uptake of DDE from compost by different species has been carried out. Furthermore, the POP bioavailability in compost as compared to soils remains poorly understood.

Given the wide-spread use of compost in both commercial and residential agriculture, data on factors influencing the bioaccumulation of organic pollutants in it are important. In addition, since the nutrient content of compost tends to be low, large amounts of it are often required to sufficiently fertilize soil (Herter et al., 2003; Timmermann et al., 2003). Thus, the use of compost contaminated with POPs presents exposure concerns to human and non-human receptors. To improve our understanding of the variables controlling the organic pollutant bioavailability in compost, a series of investigations were conducted comparing *p*,*p*′-DDE bioaccumulation from compost and soil with equivalent levels of contamination. As compost differs markedly in its structure and chemistry relative to soil, it was hypothesized that contaminant uptake by earthworms and plants in compost would not follow trends seen in soil. The species chosen for this study were the plants *C. pepo* and *C. maxima* and the earthworms *E. fetida* and *L. terrestris*. These organisms have previously been shown to accumulate *p*,*p*′-DDE in species-specific amounts (White, 2001, 2002; Kelsey et al., 2005; Kelsey and White, 2005).

2. Materials and methods

2.1. Soil

Soil (56% sand, 36% silt, 8.0% clay, 1.4% organic carbon, and bulk density 1.14 g/cm^3) containing *p*,*p*′-DDE residues as a result of historical application of DDT was collected from the Connecticut Agricultural Experiment Station's experimental farm and stored in covered plastic bins at 22 ± 2 °C. The soil was air dried for 48 h and hand sieved to <2.0 mm to provide homogenous sampling and to remove non-soil debris. Compost aged approximately 6 months was collected from Lehigh County, PA, USA and then stored in covered plastic bins at 22 ± 2 °C. It contained 37.0% organic carbon (for both soil and compost, this was determined by loss on ignition, LOI, at 360 °C) and had a bulk density of 0.61 g/cm^3. The compost also contained *p*,*p*′-DDE residues. Presumably, the plants that were composted had accumulated residual *p*,*p*′-DDE from the air or soil which had received historical applications of DDT. The compost was air dried for 7 d and hand sieved to <2.0 mm to provide homogenous sampling and to remove large debris. Replicate 5-g samples of each treatment were dried in an oven at 100 °C for 24 h to determine moisture content of the soil and compost. Additional 3-g samples of the soil and compost were transferred to 40-ml amber vials and were amended with 10 ml of *n*-hexanes and 1 μg of *o*,*p*′-DDE (Chem Service, Inc., West Chester, PA, USA) in 100 μL hexanes as an internal standard. The vials were sealed and placed in an oven at 65 °C for 5.5 h. After the samples were removed and allowed to cool for 10 min, a 1.5-ml aliquot of the supernatant was filtered with a 0.2-μm glass microfiber filter (Laboratory Science Inc., Sparks, NV, USA) for particulate removal prior to analysis (described in Section 2.5). Six replicate samples of the soil and compost were analyzed separately. The soil contained 474 ± 59.3 ng *p*,*p*′-DDE/g of soil and the compost contained 539 ± 79.8 ng *p*,*p*′-DDE/g compost (dry-weight basis for both media). These two values for *p*,*p*′-DDE concentration are statistically indistinguishable ($p < 0.05$, Kruskal–Wallis one-way analysis of variance and a comparison of mean ranks). The extraction method used to determine soil concentration was previously validated through comparison with microwave assisted extraction (MAE) of soil contaminated with *p*,*p*′-DDE (White, 2002).

2.2. Plants

The pots of soil and compost were prepared in the same manner. Three hundred and fifty grams of each dried, sieved medium were mixed with 30 g of Perlite (The Scotts Co., Marysville, OH, USA) and then added to 16 square plastic pots. The pots were 12.5 cm deep and 10.5 cm wide (Hummert International, Earth City, MO, USA). Seventy-six milliliters of tap water were then added to bring the medium to 20% moisture content by weight. As described below, four separate treatments were prepared. All treatments consisted of seven replicate pots as well as a control that contained no organisms. *C. pepo* (Baby Bear) and *C. maxima* (Prizewinner) (Johnny's Selected Seeds, Winslow, ME, USA) seeds were sown separately in seed germination medium (Frank's Nursery and Crafts, Inc., Troy, MI, USA) and maintained in the dark at 22 ± 2 °C for 7 d. Three seedlings (either *C. pepo* or *C. maxima*) were then transferred to each of the replicate soil or compost pots, and a 4-ml aliquot of NH_4NO_3 (100 mg/l aqueous solution) was added with each seedling. The pots were then grown under four 40-watt plant and aquarium grow lamps (Philips Lighting Co., Somerset, NJ, USA) at 22 ± 2 °C. They were exposed to 10 h of this artificial light during each 24-h period (plus 3–4 h of natural light) and watered twice daily to maintain approximately 20% moisture. After 7 d of growth, one of the plants was removed from each pot. The remaining plants were grown for an additional 21 d. After the 28-d growth period, the plants from each pot were destructively harvested and divided into roots, stems, and leaves. All vegetation was washed with tap water to remove attached medium particles and patted dry with a paper towel. The roots, stems, and leaves

from each pot were weighed individually to obtain biomass per pot. The replicate plant tissues were then pooled and divided into approximately equal portions by mass. The number of replicate sub-samples from each treatment was as follows: *C. pepo* leaves in soil, 3; *C. pepo* stems in soil, 4; *C. pepo* roots in soil, 3; *C. pepo* leaves in compost, 4; *C. pepo* stems in compost, 5; *C. pepo* roots in compost, 3; *C. maxima* leaves in soil, 4; *C. maxima* stems in soil, 5; *C. maxima* roots in soil, 3; *C. maxima* leaves in compost, 6; *C. maxima* stems in compost, 8; and *C. maxima* roots in compost, 3.

A modified version of the extraction method of Pylypiw (1993) was used to recover *p,p′*-DDE from the vegetation. Replicate samples of roots, stems, or leaves were placed in 2-qt blender jars. To these were added 3 μg of *o,p′*-DDE (Chem Service, Inc., West Chester, PA, USA) in 100 μL hexanes as an internal standard and 30 ml of 2-propanol (20 ml for roots). After blending at high speed for 30 s, 60 ml of petroleum ether were added (40 ml for roots), and the sample was blended at high speed for an additional 4 min (using an explosion-resistant blender and shield). The solid plant material on the jar walls was then scraped into the liquid and blended for an additional 30 s. The blender jars were allowed to settle for 30 s, after which the extraction was decanted through a funnel packed with glass wool and collected in a 500-ml separatory funnel with Teflon stopcock. The extracts were allowed to settle for 15 min and then 100 ml of deionized water (66 ml for roots) and 10 ml of saturated sodium sulfate solution (7 ml for roots) were added to each funnel. The funnels were swirled for 8–10 s and allowed to settle for 20 min to allow the aqueous and non-aqueous phases to separate. The water layer was then drained from the funnel and discarded. The same rinsing procedure was performed two additional times (100 ml deionized water, 10 ml saturated sodium sulfate solution, swirling, and settling for 20 min). A final rinse of 30 ml deionized water (20 ml for roots) and 5 ml saturated sodium sulfate solution was then performed. After an additional 20 min, the water layer was removed from the funnel. The petroleum ether that remained was collected in a graduated cylinder containing 10 g of anhydrous sodium sulfate and allowed to settle for 90 min. Five milliliters of the extract were removed and stored in a freezer at −18 °C. A portion of the petroleum ether extracts was filtered through florisil. A pre-conditioned florisil cartridge (200 mg) (Alltech, Deerfield, IL) was amended with 1 ml of vegetation extract, followed by 6 ml of 6% diethyl ether in petroleum ether. The extract was collected in 8 ml glass vials and reduced to approximately 1 ml under nitrogen flow prior to analysis (described in Section 2.5).

2.3. Earthworms

The containers using soil and those using compost were prepared in the same manner. Three hundred and fifty grams of the dried, sieved medium were mixed with 30 g of Perlite (The Scotts Co., Marysville, OH, USA) and added to seven 1000-ml glass beakers. All containers were then brought to 20% moisture content by weight with 76 ml of tap water. All treatments consisted of three replicate pots as well as a no-worm control. Worms were obtained from uncontaminated soil and washed with tap water, and then approximately 9 g of earthworm biomass were added to replicate samples of soil and compost mixtures. Either 30 individual *E. fetida* (Carolina Biological Supply, Burlington, NC, USA) or four *L. terrestris* (Lehigh River Bait and Bow, Allentown, PA, USA) were added to separate beakers. The worms were added to the top of the soil or compost of all beakers and allowed to burrow underneath the surface. The containers were covered with ventilated foil and stored in the dark at 22 ± 2 °C. After 7 d, approximately 20 ml of tap water were added to each replicate. After 14 d, the worms were removed from the soil and compost, washed with tap water, and transferred to replicate Petri plates for depuration. The plates were maintained in the dark at 22 ± 2 °C during depuration. After 48 h, the earthworms were washed with tap water, patted dry with paper towels, and weighed to obtain biomass per replicate. The earthworms were divided into approximately equal portions by mass and transferred to 40-ml amber vials. The numbers of replicate samples from each treatment were as follows: *E. fetida* in soil, 7; *E. fetida* in compost, 6; *L. terrestris* in soil, 5; and *L. terrestris* in compost, 6. The vials were then placed in a freezer at −18 °C for 1 h. Prior to extraction, 1 μg of *o,p′*-DDE (Chem Service, Inc., West Chester, PA, USA) in 100 μL of hexanes were added as an internal standard to each vial. Ten milliliters of *n*-hexanes were then added to each vial. The vials were sealed and placed upright in a test tube rack in a 65 °C oven for 5.5 h. After 10 min of cooling, an 8-ml sample of the supernatant from each vial was removed and stored in the freezer at −18 °C. A 1.5-ml aliquot was then passed through a glass microfiber filter (0.2 μm, Laboratory Science Inc., Sparks, NV, USA) for particulate removal prior to analysis (described in Section 2.5).

2.4. Abiotic desorption

Tests of *p,p′*-DDE desorption from the soil and compost were conducted in the same way. Four replicate 10-g samples of dried and sieved soil (or compost) were added separately to 125-ml ground-neck Erlenmeyer flasks. Sixty milliliters of tap water, and 1 μg of *o,p′*-DDE (Chem Service, Inc., West Chester, PA, USA) in 100 μL of hexanes were added to each flask. The *o,p′*-DDE acted as an internal standard. One Empore C18 (octadecyl) extraction disk (C18 disk, 3 M Filtration Products, St. Paul, MN, USA) was added to each flask, and the flasks were placed on a shaker at 120 rpm for 3 d. The disks were removed from solution and washed with tap water to remove soil (or compost) and transferred to 8-ml vials containing 5 ml of hexanes and approximately 1 g of $NaSO_4$ crystals. Finally, the vials were capped and heated at 70 °C for 2 h. A 1.5-ml aliquot of the extract was then passed through a glass microfiber filter (0.2 μm, Laboratory Science Inc., Sparks, NV, USA) for particulate removal prior to analysis (described in Section 2.5).

2.5. Chemical analysis

The *p,p′*-DDE content in all extracts was determined using an Agilent (Avondale, PA, USA) 6890 gas chromatograph (GC) with a ^{63}Ni micron-electron capture device (ECD). The column (30m × 0.53 mm × 0.5 μm) contained an SPB-1 film (Supelco, Bellefonte, PA, USA). The GC program used was 175 °C initial temperature ramped at 3.5 °C/min–225 °C then ramped at 25 °C/min–250 °C with a hold time of 4.71 min. The total run time was 20 min. A 2-μL splitless injection was used, and the injection port was maintained at 250 °C. The carrier gas was H_2 and the make-up gas was 5% CH_4 in Ar at 20 ml/min. The electron capture detector was maintained at 325 °C. The retention times of *o,p′*-DDE and *p,p′*-DDE were 9.9 and 10.3 min, respectively. Crystalline *p,p′*-DDE (EPA National Pesticide Standard Repository, Fort Meade, MD, USA) was added to *n*-hexanes for soil and worm standards or petroleum ether for vegetation standards at 1000 ng/ml. The *p,p′*-DDE standard solutions were then diluted to prepare calibration standards of 10, 25, 50, 100, 150, and 250 ng/ml. An internal standard of *o,p′*-DDE (EPA National Pesticide Standard Repository, Fort Meade, MD, USA) at 100 ng/ml was then added to each standard.

2.6. Statistical analysis

The mean values of reported *p,p′*-DDE concentration from replicate samples as expressed on a dry-weight basis of biomass, soil or compost were used in determining statistical significance. Statistical differences between replicates were then determined by a Kruskal–Wallis one-way Analysis of Variances followed by Tukey's Multiple Comparison Test or Student's *t*-test. Specific analyses used are cited in Section 3.

3. Results

3.1. Bioconcentration in plants and earthworms

All plants survived the experimental period and appeared in good health at harvest. Bioconcentration factors (BCF; the dry-weight ratio of contaminant concentration in plant tissues to that in the soil) of *p,p′*-DDE in the roots and stems of *C. pepo* grown in soil were nearly 7- and 5-fold higher, respectively, than those grown in compost (Fig. 1A). Translocation to the leaves of *C. pepo* was the same in soil and compost.

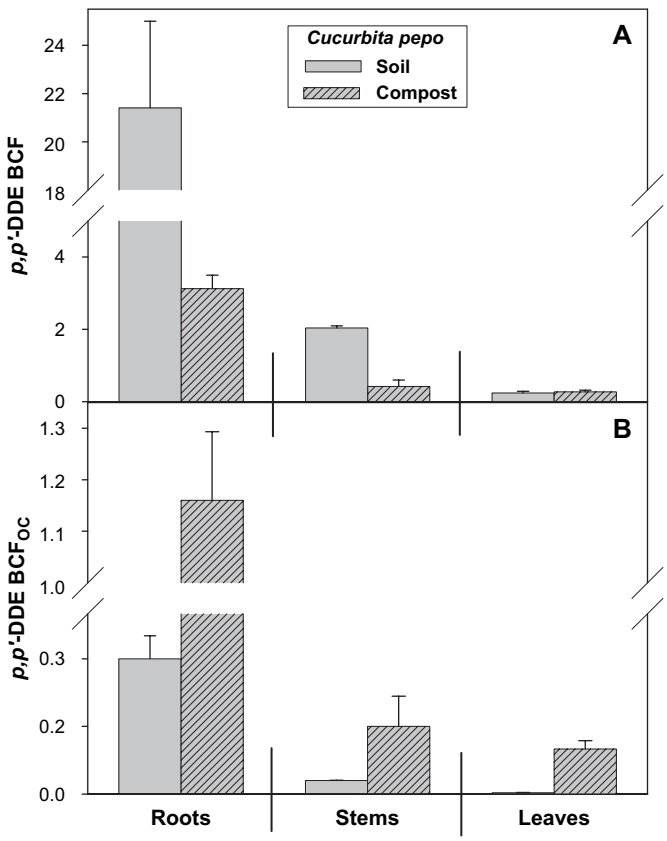

Fig. 1. Accumulation of DDE from soil and compost by *C. pepo.* Data are expressed as bioconcentration factors (BCFs) (A) or as organic-carbon normalized BCFs (B).

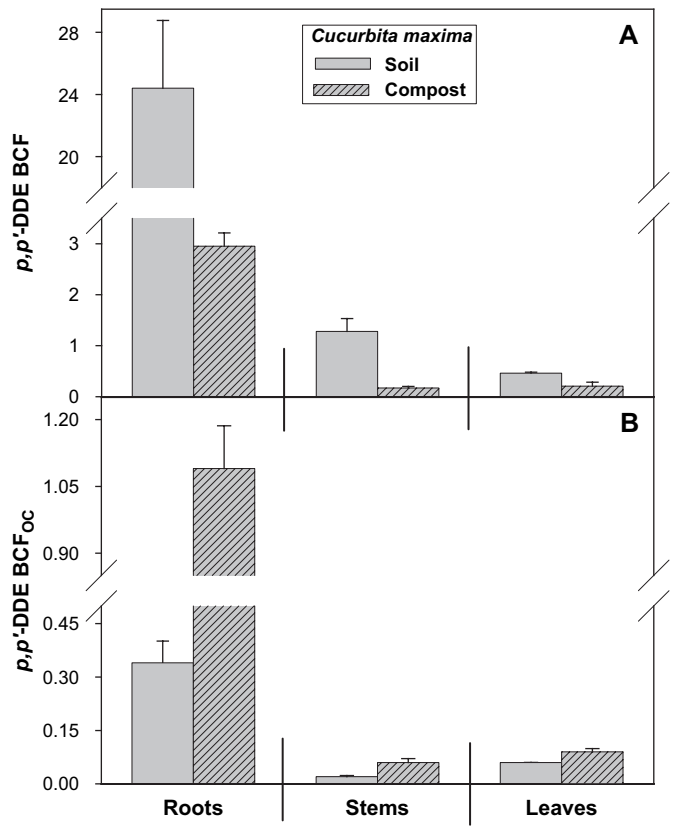

Fig. 2. Accumulation of DDE from soil and compost by *C. maxima.* Data are expressed as bioconcentration factors (BCFs) (A) or as organic-carbon normalized BCFs (B).

This trend was also seen in *C. maxima*. Bioconcentration factors from soil were approximately 8-, 7-, and 2-fold higher in the roots, stems, and leaves, respectively, of these plants relative to those from compost (Fig. 2A). A comparison of BCF values between species reveals that, in both soil and compost, uptake by the roots of the two plants was statistically indistinguishable. In soil, root BCFs were 21.40 ± 3.6 and 24.40 ± 4.37 for *C. pepo* and *C. maxima*, respectively. In compost, BCF in *C. pepo* roots was 3.13 ± 0.37, and in *C. maxima* roots it was 2.95 ± 0.26. However, in both media, translocation to stems was higher in *C. pepo* than in *C. maxima*. Translocation to leaves was minimal in all cases. The two plants showed similar leaf BCF values in compost, but those in soil were slightly higher in *C. maxima*. When total *p,p′*-DDE uptake in the two media is compared, substantial differences can be seen as well. As shown in Table 1, bioconcentration of the compound by *C. pepo* was approximately 4-fold higher in soil than it was in compost. For *C. maxima*, uptake was nearly 6-fold higher from the soil than from the compost. These differences are evident even as more biomass was produced by plants in compost than those in soil.

As can be seen in Table 2, uptake of *p,p′*-DDE by the two earthworm species was dramatically different in both media. The bioaccumulation factors (BAF; the dry-weight ratio of contaminant concentration in worm tissues to that in the soil) for *p,p′*-DDE seen in *E. fetida* were approximately 6-fold higher in soil and 3-fold higher in compost than those seen in *L. terrestris*. Whether the worms were grown in soil or compost exerted a substantial effect as well. The bioaccumulation factors in soil-cultivated *E. fetida* were over 7 times greater than the BAF in the same organisms grown in compost. Similarly, BAF increased by 3.4-fold for *L. terrestris* when the worms were grown in soil.

Figs. 1 and 2 and Table 2 also present an alternative way to express bioconcentration in plants and earthworms. The organic-carbon normalized bioconcentration factor (BCF_{oc}) can be derived from the ratio of the concentration of *p,p′*-DDE in tissues to the ratio of the compound in the organic carbon (organic C) fraction of the soil or compost. Given the

Table 1
Total biomass and uptake of *p,p′*-DDE in the plants *Cucurbita pepo* and *Cucurbita maxima* in compost and soil

Species	Treatment	Dry biomass[a,*] (g/pot)	Total DDE uptake[a,*] (ng/pot)
C. pepo	Soil	1.12 ± 0.15A	4919.5 ± 428.8A
C. pepo	Compost	1.93 ± 0.10B	1216.1 ± 85.7B
C. maxima	Soil	1.18 ± 0.10A	5395.1 ± 436.6C
C. maxima	Compost	2.80 ± 0.29C	904.81 ± 94.9B

*Within columns, values followed by different letters are significantly different ($p < 0.05$, Kruskal–Wallis one-way analysis of variance followed by a Tukey's Multiple Comparison Test).
[a] Mean ± standard deviation of replicates.

Table 2
Total biomass and bioaccumulation of *p,p′*-DDE in the earthworms *Eisenia fetida* and *Lumbricus terrestris* in compost and soil

Species	Medium	Dry biomass[a,*] (g/beaker)	Tissue conc[a,*] (ng/g)	BAF[a,b,*]	BAFoc[a,c,*]
E. fetida	Soil	1.29 ± 0.01A	3230.0 ± 348.9A	6.82 ± 0.74A	0.09 ± 0.009A
E. fetida	Compost	0.98 ± 0.61A	479.9 ± 173.1B	0.89 ± 0.32B	0.33 ± 0.118B
L. terrestris	Soil	1.24 ± 0.11A	516.2 ± 127.7A	1.09 ± 0.27 A	0.01 ± 0.002A
L. terrestris	Compost	0.99 ± 0.03A	173.0 ± 38.6B	0.32 ± 0.07B	0.12 ± 0.026B

*Within columns, values followed by different letters are significantly different ($p < 0.05$, Kruskal–Wallis one-way Analysis of Variance followed by a Tukey's Multiple Comparison Test).
[a] Mean ± standard deviation of replicate samples.
[b] BAF, Bioaccumulation Factor = tissue concentration/soil concentration.
[c] BAF_{oc}, *p,p′*-DDE concentration in tissues/*p,p′*-DDE concentration in organic carbon in soil or compost. See text for details.

values for organic C (1.4 and 37.0% for soil and compost, respectively; see Section 2.1) and pollutant concentration (434 and 539 ng DDE/g of medium for soil and compost, respectively; see Section 2.1), BCF_{oc} was calculated based on 33,857 ng *p,p′*-DDE/g of organic C in soil and 1455 ng *p,p′*-DDE/g of organic C in compost. When expressing DDE accumulation normalized to organic carbon, BCF_{oc} values were consistently higher for organisms in compost relative to those from soil. For example, BCF_{oc} in the roots of both *C. pepo* and *C. maxima* were more than 4-fold higher when the plants were grown in compost than when they were grown in soil (Figs. 1B and 2B). Similarly, BCF_{oc} values were 3.7- and 12-times higher for *E. fetida* and *L. terrestris*, respectively, in compost than in soil (Table 2).

3.2. Abiotic desorption

The desorption of DDE from soil was nearly twice that from compost. The amounts released in aqueous solution were 77.3 ± 3.56 (soil) and 38.3 ± 29.31 ng/g DDE (compost), a difference that is statistically significant ($p < 0.05$ by Student's *t*-test). These values equate to a 16.3 and 7.12% release of the contaminant from soil and compost, respectively.

4. Discussion

As described above, the biological availability of *p,p′*-DDE was species-specific and differed markedly in soil and compost containing equal levels of contamination. That accumulation of the compound varied among the organisms tested was not surprising and was consistent with previous work conducted by us and others. Edwards and Coulson (1992) noted different responses among species in their review of literature values of the effect of organic pollutants on earthworm mortality rates. In a recent study, Kelsey et al. (2005) reported substantially higher rates of bioaccumulation by *E. fetida* than *L. terrestris* or *Apporectodea caliginosa.* With regard to plant species, White (2001, 2002), Mattina et al. (2002), and White et al. (2005) observed that *C. pepo* concentrated substantially higher levels of *p,p′*-DDE and chlordane than did other plant species, including those within the same species. Other researchers have reported dramatic differences in pollutant bioaccumulation among different organisms. For example, the uptake of atrazine and phenanthrene was 10- and 3-fold higher, respectively, by bacteria than by earthworms (Kelsey et al., 1997). The availability of sorbed polyaromatic hydrocarbons differed between two species of bacteria as well (Guerin and Boyd, 1992; Tang et al., 1998). Reid et al. (2000) described some of the important factors affecting the prediction of bioavailability among different organisms. Our current data support those suggesting the need for exposure models to take into account species differences in the prediction of the risk posed by toxic compounds in soil.

In addition to the effect of variations among species in their ability to take up soil pollutants, differences in soil and compost appear to exert a substantial influence on the bioaccumulation of *p,p′*-DDE. The five assays used in this study – uptake by two earthworm species and two plant species and abiotic desorption – suggest that, despite the same level of contamination, *p,p′*-DDE is significantly more available in soil than in compost. As expressed as BAF (earthworms) or BCF (plants), the differences in availability of the compound ranged from approximately 2- to over 8-fold. Data from the abiotic desorption experiments suggest an approximately 2-fold increase in desorption from the soil relative to that from the compost. Since the soil and compost differed markedly in their chemical and physical properties (most notably organic-carbon content and quality) one would expect dissimilar levels of uptake of an organic pollutant from the two media. A number of studies have indicated that soil properties will affect the bioavailability of soil pollutants (Alexander, 2000; Chung and Alexander, 2002; Swindell and Reid, 2006). More work will be needed to fully describe the mechanisms responsible for the observed differences, though.

Interestingly, when bioconcentration factors are normalized for organic carbon content (BCF_{oc}, described in Section 3.1), bioavailability is significantly higher in compost than it is in soil. This trend is the opposite of that noted with total BCF. Clearly, risk assessments of contaminated sites are concerned with the overall contaminant bioconcentration (i.e., total BCF) and exposure of organic pollutants and, therefore, are likely to disregard the role of organic carbon on bioavailability. Bioconcentration factors are of utmost importance because they affect overall exposure of an organism to toxins. However, as organic carbon is the principle component responsible for the sequestration of hydrophobic pollutants (Karickhoff et al., 1979; Luthy et al., 1997), factors affecting its chemical and physical properties will influence the ultimate fate of compounds like *p,p′*-DDE. The observed differences in BCF_{oc} in soil and compost suggest important mechanistic considerations in the

sequestration of an organic pollutant such as *p,p′*-DDE. Our organic-C-normalized BCF values indicate that *p,p′*-DDE is not as tightly bound – in other words, less sequestered – in compost than it is in soil. These data are not surprising when one considers the literature on soil organic matter structure, formation, and diagenesis. In a series of studies by Weber et al. (1992) and Huang et al. (1997), it was noted that older and more condensed organic matter would form stronger and more irreversible associations with organic pollutants than would younger, amorphous, soft carbon. These types of differences are perhaps exaggerated in the current study, where the compost used was aged for 6 months as compared to the organic carbon associated with an agricultural soil which is likely orders of magnitude older.

5. Conclusion

Uptake of *p,p′*-DDE from soil and compost varies, and no single species can accurately model the bioaccumulation of all organisms. For all assays used in this study (biological and abiotic) the contaminant was more available from the soil than from compost. These findings are important because such differences could affect the evaluation of exposure and risk of organic pollutants released into natural systems. However, the data also indicate that differences in the quality and age of organic carbon can affect the availability of hydrophobic pollutants. Additional research will be necessary to fully elucidate the mechanistic aspects of sequestration of organic pollutants in soil and compost.

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