Uptake and Phytotransformation of *o,p′*-DDT and *p,p′*-DDT by Axenically Cultivated Aquatic Plants

Jianping Gao,† A. Wayne Garrison,*,† Christopher Hoehamer,‡ C. S. Mazur,† and N. Lee Wolfe†

National Exposure Research Laboratory, U.S. Environmental Protection Agency, 960 College Station Road, Athens, Georgia 30605, and Department of Biochemistry and Molecular Biology, University of Georgia, Athens, Georgia 30605

The uptake and phytotransformation of *o,p′*-DDT and *p,p′*-DDT were investigated in vitro using three axenically cultivated aquatic plants: parrot feather (*Mariophyllum aquaticum*), duckweed (*Spirodela oligorrhiza*), and elodea (*Elodea canadensis*). The decay profile of DDT from the aqueous culture medium followed first-order kinetics for all three plants. During the 6-day incubation period, almost all of the DDT was removed from the medium, and most of it accumulated in or was transformed by these plants. Duckweed demonstrated the greatest potential to transform both DDT isomers; 50−66% was degraded or bound in a nonextractable manner with the plant material after the 6-day incubation. Therefore, duckweed also incorporated less extractable DDT (32−49%) after 6 days than did the other plants. The capacity for phytotransformation/binding by elodea is between that of duckweed and parrot feather; ∼31−48% of the spiked DDT was degraded or bound to the elodea plant material. *o,p′*-DDD and *p,p′*-DDD are the major metabolites in these plants; small amounts of *p,p′*-DDE were also found in duckweed (7.9%) and elodea (4.6%) after 6 days. Apparently, reduction of the aliphatic chlorine atoms of DDT is the major pathway for this transformation. This study, which provides new information on plant biochemistry as related to pollutant accumulation and phytotransformation, should advance the development of phytoremediation processes.

Keywords: *Phytotransformation; phytoremediation; plant accumulation; elodea; DDT*

INTRODUCTION

The organochlorine (OC) pesticide DDT and its metabolites are highly persistent, and residues may remain in the environment for a very long time (half-life ranges from 20 to 30 years) (Aigner et al., 1998; Szeto and Proce, 1991; Dimond and Owen, 1996). This is because OC pesticides contain chlorine substituents as well as novel functional groups rarely found in nature that increase recalcitrance and binding to natural organic matter. Such persistence results in long-range migration, threats to the survival of beneficial insects and various wildlife species, and suspected impacts on human beings. DDT-bound residues (DDTr) are lost from the environment mainly by microbial action (Mohn and Tiedje, 1992), which can release DDTr gradually and increase the load in the environment. Pretreatment, such as with surfactants, can also release DDTr from the bound state to the labile form (You et al., 1996). These released residues become bioavailable for uptake and degradation by biota, for example, plants. In addition, DDD, the first dechlorination product and the major end product of DDT transformation, is also a chlorinated pesticide and a priority pollutant with properties similar to those of DDT. Accumulation of DDD under anaerobic conditions and its stability under aerobic conditions have greatly hampered biodegradation processes using microorganisms. Thus, phytoremediation, which relies on extraction from the polluted medium by plants and subsequent degradation or transformation by plant protein and enzymes, may be a cost-effective and innovative alternative remediation technique (Schnoor et al., 1995; Medina and McCutcheon, 1996).

Aquatic plants have a great potential to function as in-situ and on-site biosinks and biofilters of aquatic pollutants because of their abundance and limited mobility. These plants possess a large surface area that is covered by a lipid-rich cuticle and thus have the potential to take up lipophilic OC pesticides such as DDT (Ockenden et al., 1998). Some whole plants have been used successfully in a number of pesticide metabolism studies as model systems for bioremediation (Hughes et al., 1997; Burken and Schnoor, 1997; Nzengung et al., 1996, 1999). Most of these studies were conducted in the plant root zone (the rhizosphere), which is rich in microbes and microbial diversity and where contaminants may be degraded more rapidly because of root exudates supplied by the plants (Mohn and Tiedje, 1992; Boersma et al., 1988). One recent study, however (Garrison et al., 2000), reported phytodegradation of *p,p′*-DDT and the enantiomers of *o,p′*-DDT, with half-lives ranging from 1 to 3 days, by the aquatic plant elodea and the terrestrial plant kudzu in radiation-sterilized aquatic systems. Nevertheless, in screening plants for in-situ and on-site phytoremediation, few data have been obtained using axenic plants, plant tissues, or cell cultures in the absence of microorganisms. Therefore, the ability of plants to transform OC compounds without the participation of associated microbes remains arguable (Hughes et al., 1997).

* Author to whom correspondence should be addressed [telephone (706) 355-8219; fax (706) 355-8202; e-mail garrison.wayne@epa.gov].

† U.S. Environmental Protection Agency.

‡ University of Georgia.

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In our previous study (Gao et al., 2000), three aquatic plants showed a considerable potential for accumulation/degradation of organophosphorus pesticides in axenic systems. In this paper, axenic cultures of the same aquatic plants—parrot feather (*Myriophyllum aquaticum*), duckweed (*Spirodela oligorrhiza L.*), and elodea (*Elodea canadensis*)—were further investigated in vitro for accumulation and transformation of *o,p*′-DDT and *p,p*′-DDT. These aquatic plants are widespread, free-floating aquatic macrophytes, a source of food for waterfowl, and a shelter for small aquatic invertebrates. They grow quickly and reproduce more rapidly than other vascular plants (Zhang and Jin, 1997). It has been discovered that several enzymes in these plants can degrade pesticides. These include nitroreductases that reduce nitro moieties to amines and dehalogenases that replace halogen atoms with hydrogen (Nzengung et al., 1996, 1999; Komossa et al., 1995). This study is part of a research program directed toward the discovery of new plant species with the capability for phytoremediation of various pesticides.

MATERIALS AND METHODS

Reagents. Analytical grade (>99% purity) *o,p*′-DDT [1,1,1-trichloro-2- (*o*-chlorophenyl)-2-(*p*-chlorophenyl)ethane], *o,p*′-DDD [1,1-dichloro-2-(*o*-chlorophenyl)-2-(*p*-chlorophenyl)ethane], *o,p*′-DDE [1,1-dichloro-2-(*o*-chlorophenyl)-2-(*p*-chlorophenyl)-ethylene], *p,p*′-DDT [1,1,1-trichloro-2,2-bis(*p*-chlorophenyl)-ethane], *p,p*′-DDD [1,1-dichloro-2,2-bis(*p*-chlorophenyl)ethane], and *p,p*′-DDE [1,1-dichloro-2,2-bis(*p*-chlorophenyl)ethylene] were obtained from Chem Service (West Chester, PA). Deionized water (Nanopure II; Boston MA) was used, and all other chemicals were of reagent grade or better. OC compound solutions were prepared by diluting with methanol the appropriate volumes from a 1000 mg/L stock solution in methanol. Such stock solutions were the sources for all experiments.

Plants and Tissue Culture. Axenic parrot feather was supplied by the Department of Biochemistry, University of Georgia (Boyajian and Carreira, 1997). These plants were propagated vegetatively on NH_4^+-free Murashige Skoog (MS) medium (Sigma Plant Culture, St. Louis, MO) supplemented with agar. Axenic plants were maintained in plastic boxes at room temperature under light from cool white and broad-spectrum bulbs for a 12-h photoperiod at a light intensity of 100 μeinstein/m^2 (Hughes et al., 1997).

Duckweed was taken from ponds and wetlands in the Athens, GA, vicinity (35° 55.919′ N and 83° 22.233′ W). After the plants were cleaned thoroughly under gentle running water to remove adhering algae and insect larvae, they were transferred to and cultivated in 75-L plastic containers with half-strength Hoagland's culture solution (Sigma Plant Culture) at pH 7.0. All of the containers were maintained at 25 °C and at an irradiance of 400 μmol of photon flux density m^{-2} s^{-1}. After cultivation for 3 weeks, duckweed was rinsed with flowing tap water for ~10 min and then transferred into sterilized water containing sodium hypochlorite at 1% of the final concentration. This solution was stirred with a magnetic bar for 15 min, and then the plants were rinsed with sterilized water.

Elodea plants were collected from Lake Herrick in Athens, GA. The procedures of cultivation and sterilization were similar to those used for duckweed as described above except the concentration of sodium hypochlorite was 5%. These plants were cultivated for 1 week in the laboratory for the experiments.

Phytotransformation Studies. Stems and the feathery leaves totaling 2 g (fresh weight) of axenic plants were placed in a 50-mL Erlenmeyer flask fitted with a Styrofoam stopper to which 20 mL of sterile hydroponic culture medium (Harland nutrient solution adjusted to pH 7.0 with 1.0 N NaOH, Sigma) containing 1 mg/L *o,p*′-DDT or *p,p*′-DDT was added. These were incubated in a rotating incubator (50 rpm; Inweave 4230, Edison, NJ) at 22 °C with fluorescent lights. The aqueous culture medium was sampled (0.5 mL) at time intervals of 0, 4, 8, 16, and 24 h and each day from 2 to 6 days and extracted twice with 0.5 mL of *n*-hexane. The hexane layers were combined, and the compounds were analyzed by GC-ECD as described below. Four controls were carried out in parallel: water solution with DDT, culture medium solution with DDT, autoclaved plant with culture medium and DDT (added after autoclaving), and plant with culture medium but without DDT. During experiments, aliquots of culture medium were randomly collected and sent to the Department of Biochemistry, University of Georgia, for a total bacterial colony count in which three media (basic blood agar, cooked meat broth, and thioglycollate broth) were used. The total bacterial count was always <50 colony-forming units/mL. All experiments were conducted in a fume hood (Labconco, Kansas City, MO) and performed in triplicate.

Analysis of Plant Tissue. At the end of the incubation (sixth day), the plants were rinsed thoroughly with water, blotted dry, finely chopped with pruning shears, and ground up using a mortar and pestle in liquid nitrogen. Then the plant materials were sonicated twice in an ultrasonic bath (Branson 5120, Danbury, CT) at 100 rpm and 30 °C, in a 10 mL mixture of acetonitrile and hexane (v/v = 1:1) for 20 min. The extracts containing DDT and its metabolites were then cleaned up by passage through a C-18 column (0.5 g; Supelco, Bellefonte, PA). The eluant was gently evaporated with nitrogen, and residues were redissolved in 1 mL of hexane for GC-ECD.

Extracts of the culture medium and plant tissues were analyzed by an HP 5890 series II GC equipped with a ^{63}Ni electron capture detector and a capillary column (30 m, 0.53 mm i.d.) coated with 95% dimethyl−5% diphenyl polysiloxane at 0.5 μm film thickness (RTS-5, Restek Corp., Bellefonte, PA). The column oven was programmed from an initial temperature of 120 °C for 2 min to 220 °C at a rate of 20 °C/min, held for 1 min, and then ramped at a rate of 4 °C/min to 240 °C with a final hold time of 10 min. The detector and injector were maintained at 300 and 250 °C, respectively. Helium was the carrier gas at 30 cm/s, and nitrogen was the makeup gas at 30 mL/min. Injection volume was 1 μL in *n*-hexane; injection was in the split mode at 1:40. To determine analytical recovery, pesticides were spiked into the aqueous growth medium and the ground plant materials. Recoveries ranged from 95 ± 5.3% to 103 ± 5.9% (RSD).

Data Analysis. A first-order one-compartment model ($C_t/C_0 = e^{-kt}$) was used to estimate the kinetic parameters of DDT uptake, where C_t is the concentration of the pollutant at time t, C_0 is its initial concentration, and k is the rate constant. The half-life, $t_{1/2}$, corresponds to the period of time during which the pesticide concentration is reduced to half of the initial concentration, given by $t_{1/2} = \ln(2)/k$. By plotting the log(percent residual pesticide in the culture medium) versus time, straight lines were obtained. This first-order constant (k) was taken as the absolute value of the slope of the line obtained by least-squares analysis of the kinetic data. Mean values and standard deviations were calculated for each test group on the basis of the values obtained for each individual plant tissue. These values were compared by analysis of variance (ANOVA). All statistical analyses were performed on an IBM computer using the Excel program. The significance level was set at 0.05.

The bioconcentration factor (BCF) was calculated as follows:

$$\mathrm{BCF} = \frac{\text{DDT extracted from plant tissue (mg/kg) at harvest}}{\text{at harvest DDT in the culture media (mg/L)}}$$

RESULTS AND DISCUSSION

Uptake and Phytotransformation by Parrot Feather. Nearly all of the DDT spiked in this culture could be extracted by hexane from the culture medium at zero time. This initial value is taken as 100%. With

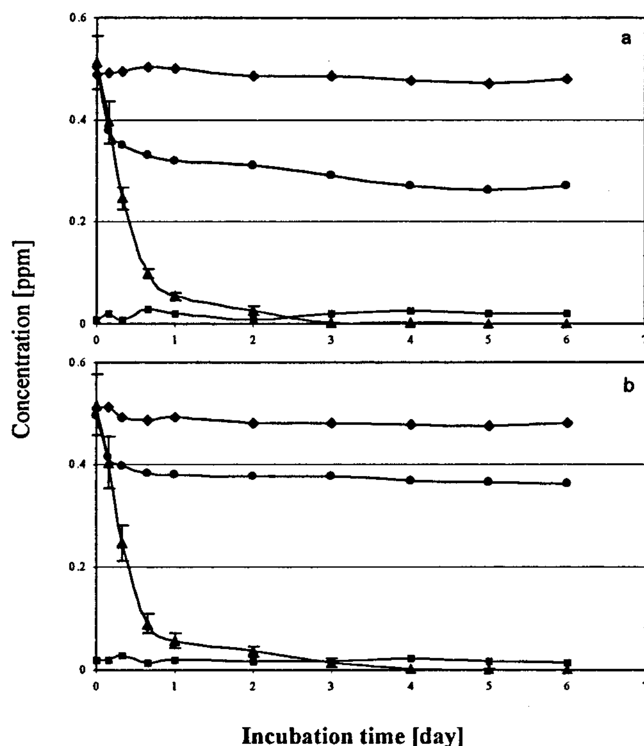

Figure 1. Uptake of *o,p′*-DDT (a) and *p,p′*-DDT (b) exposed to axenically cultivated parrot feather (*M. aquaticum*) under controlled conditions: control (◆); autoclaved parrot feather (●); parrot feather (▲); total metabolites (*o,p′*-DDD + *o,p′*-DDE, or *p,p′*-DDD + *p,p′*-DDE) (■). Concentrations are of the residual substrate in the liquid culture medium as determined by GC-ECD analysis; $C_0 = 0.5$ ppm. Error bars represent ±1 standard deviation.

16 h of incubation, ~80% of *o,p′*-DDT and *p,p′*-DDT had disappeared from the parrot feather culture medium. After that, the concentration in the culture medium decreased slowly; at the end of incubation (6 days) no significant *o,p′*-DDT and *p,p′*-DDT was detected in the medium (Figure 1). An initial decrease in DDT concentration in the aqueous medium was also observed with autoclaved plants; this was attributed to sorption on the surface of autoclaved plants. Thereafter, the concentration stabilized and autoclaved controls remained constant. Over the whole incubation period, DDT metabolites *o,p′*-DDD, *o,p′*-DDE, *p,p′*-DDD, and *p,p′*-DDE, which were absent at zero time in the medium, remained stable at a low level.

The concentration of DDT followed a logarithmic decay, with a first-order decay constant of 0.09−0.11 h^{-1}(Table 1). This 6.3−7.7-h half-life was achieved with a ratio of plant to medium of about 0.1 (2 g of parrot feather to 20 mL of medium). Mass recovery studies after 6 days showed that about 24 and 18% of *o,p′*-DDT and *p,p′*-DDT, respectively, were degraded/bound in a nonextractable manner with plant material. In contrast, about 76 and 82%, respectively, are extractable from the plant (Figure 2). Both *o,p′*-DDE and *p,p′*-DDE are found in the plant at a low levels (1.3 and 1.9%), whereas 7.9% of *o,p′*-DDD and 11.5% of *p,p′*-DDD are detected at the end of the 6-day incubation (Figure 3).

Before undergoing any transformation, OC compounds must be taken up by the plants, which is a complex process that may be compound specific (Bromilow and Charmberlain, 1995). In the parrot feather, accumulation of DDT appears to be a passive process because autoclaved plants also sorbed DDT. It is not clear whether the transformation of the DDT compounds occurs on the surface or after transport into the plant tissue (Medina and McCutcheon, 1996; Hughes et al., 1997; Burken and Schnoor, 1997). In any case, these results show that the loss of DDT involves two processes: the uptake of DDT by the plant and the transformation/binding of DDT by the plant. The rate of uptake by plants is unlikely to be limiting as DDT is extremely hydrophobic; rather, the rate at which the chemical is transformed or bound should be the rate-limiting step. The metabolic pathways and transformation mechanisms of DDT in plants are not well-known, although its transformation has been plausibly elucidated in microorganisms and animals (Mohn and Tiedje, 1992; Buser and Muller, 1995). Three main reactions may be involved in the degradation of DDT in plants: (1) Reductive dehalogenation of an alkyl chlorine atom via a free radical mechanism is the critical step to transform DDT to DDD. This reaction requires the enzyme DDT-dehydrochlorinase and a reductant (e.g., an iron porphyrin), which, in a natural system, may be a product of biological activity (Garrison et al., 2000). Thus, sterilization could destroy the source of the reductant instead of the enzymatic catalyst (Aislable et al., 1997). (2) Direct dehalogenation of the aromatic ring with concomitant introduction of a hydroxyl substituent has been reported by some authors. Two pathways were suggested for this reaction; they involve the nonspecific action of an oxygenase or the action of a halidohydrolase. The second pathway is the most efficient and is generally part of a catalytic mechanism (Masse et al., 1989). (3) The third major DDT degradation pathway involves the formation of DDE from DDT through dehydrodehalogenation (simultaneous removal of both halogen and H) (Aislable et al., 1997).

Several points should be noted: (1) The *p*-chloro isomer is metabolized in these plants at a similar rate to the *o*-chloro isomer. In mammals, on the other hand, the *o*-chloro ring is extensively metabolized, whereas the *p*-chloro ring remains intact (Buser and Muller, 1995). (2) We observed no significant acclimation periods in the disappearance of DDT from the medium, whereas a long lag time was reported in reductive dehalogenation by microorganisms (Mohn and Tiedje, 1992). However, it is not clear whether any lag time occurs in the actual transformation step after DDT is taken up by the plants. A transformation lag time might have been expected in our experiments because the plant tissues were not previously acclimatized to the chlorinated substrates. (3) With plant tissue cultures, the greater proportion of the applied pesticide has often been reported in the nonextractable residue fraction of the plant (Mumma and Davidonis, 1983). This was also observed in our study with duckweed but not with parrot feather or elodea. (4) In contrast to multicellular animals in which transformation of the substrate facilitates excretion, compartmentalization and immobilization tend to be the ultimate fate of a toxicant in plants (Sandermann, 1992). During the 6-day incubation period, only trace amounts of DDD and DDE were observed in the medium; larger amounts of DDD and DDE were measured in the plants. This indicates that these compounds are not likely produced in the medium and that they do not tend to be released into the medium. (5) The observation of only trace concentrations of DDE indicates that the plants may be deficient in the mixed-function oxidase (MFO) that can dehydrochlorinate DDT

Table 1. Disappearance Rate Constants (k), Half-Lives ($t_{1/2}$), and BCF of DDT Exposed to Axenically Cultivated Plants

	o,p′-DDT					*p,p′*-DDT				
plant	C_0 (mg/L)	k (h^{-1})	r^2	$t_{1/2}$ (h)	BCF (L/kg)	C_0 (mg/L)	k (h^{-1})	r^2	$t_{1/2}$ (h)	BCF (L/kg)
parrot feather	0.5	0.09	0.89	7.7	842	0.5	0.11	0.91	6.3	1040
duckweed	0.5	0.14	0.91	4.9	14.4	0.5	0.15	0.88	4.6	40.7
elodea	0.5	0.15	0.90	4.6	74.1	0.5	0.16	0.92	4.3	62.2

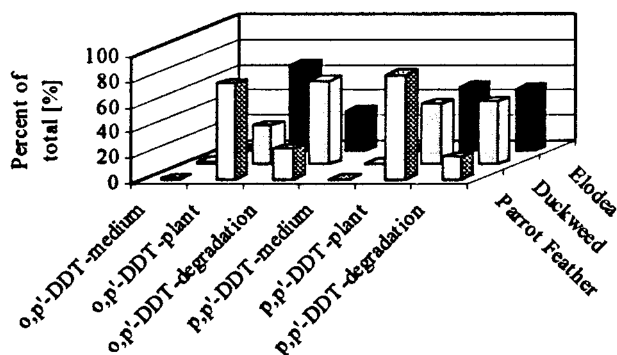

Figure 2. Mass balance of DDT in the plant tissue and culture medium at the end of incubation (6 days). DDT in both culture medium and plant was extracted and determined by GC-ECD analysis. The amounts are reported as percentages of the initial DDT concentration in the medium. "Plant" is the amount extracted from the plant at the end of incubation, whereas "degradation", calculated as the difference in the initial spiked concentration and the sum of concentrations recovered from medium and plant, refers to the amount of nonextractable DDT, an indication of that metabolized and/or irreversibly bound.

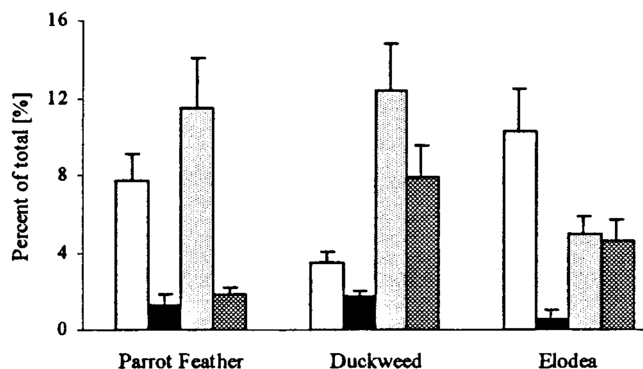

Figure 3. Metabolites of DDT in the extracts of plant tissues. Detection was performed by GC-ECD at the end of 6 days of incubation: bars represent *o,p′*-DDD, *o,p′*-DDE, *p,p′*-DDD, and *p,p′*-DDE from left to right in each grouping. Metabolite levels are calculated as the percentage of the initial spiked DDT. Error bars represent ±1 standard deviation.

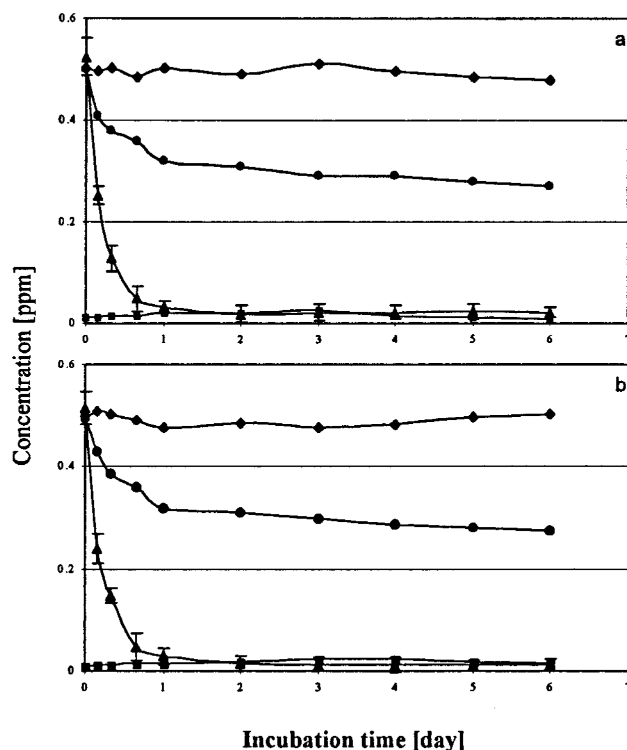

Figure 4. Uptake of *o,p′*-DDT (a) and *p,p′*-DDT (b) exposed to axenically cultivated duckweed (*S. oligorrhiza*) under controlled conditions: control (◆); autoclaved duckweed (●); duckweed (▲); total metabolites (*o,p′*-DDD + *o,p′*-DDE, or *p,p′*-DDD + *p,p′*-DDE) (■). Concentrations are of the residual substrate in the liquid culture medium as determined by GC-ECD analysis; C_0 = 0.5 ppm. Error bars represent ±1 standard deviation.

to DDE in mammals or that DDT resists the attack by MFO, which is known to be inhibited by electron-transfer agents (Wang and Simpson, 1996; Pereira et al., 1996).

Although no attempt was made to carry out a mass balance using radiolabled DDT, the high compound recovery from the autoclaved plants confirmed that neither abiotic losses (e.g., reaction with the medium or volatilization) or losses in sampling can explain the disappearance of DDT observed in viable plants; reversible sorption, phytodegradation and/or irreversible binding must be the main mechanisms.

Uptake and Phytotransformation by Duckweed. The most extensive degradation of DDT in this study occurred with duckweed, in which 66 and 50% of *o,p′*-DDT and *p,p′*-DDT, respectively, are converted to their metabolites or bound with plant materials (Figure 2) and <0.1% remained as DDT in the culture medium after 6 days of incubation (Figure 4). Both DDT isomers accumulate (are extractable) in duckweed much less than in parrot feather, 32 and 49%, respectively (Figure 2). Also, the half-lives show that DDT disappears more quickly from the duckweed culture medium than from the parrot feather medium (Table 1). The distribution patterns of DDT metabolites observed in duckweed are shown in Figure 3. *o,p′*-DDD and *p,p′*-DDD are major metabolites, as with parrot feather; however, considerably more *p,p′*-DDE and less *o,p′*-DDD were produced by the duckweed. This may be caused by differences in cell characteristics of the two plants.

The metabolism of DDT was examined in cell suspension cultures of terrestrial plants such as parsley and soybean (Scheel and Sandermann, 1977). In that study, only 0.6−2.2% of the applied DDT was metabolized after 44−48 h of incubation; DDE was identified as the major metabolite. Generally, the metabolism of DDT in organisms is accomplished through dehydrodechlorination to DDE, although the transformation to DDD and other metabolites may occur to a lesser extent in some species (Strandberg et al., 1998). In this study, DDT accumulates in duckweed and can be transformed as well. In contrast to terrestrial plants, DDD, which occurs generally by abiotic processes (Mohn and Tiedje, 1992), is the main transformation product of DDT in these

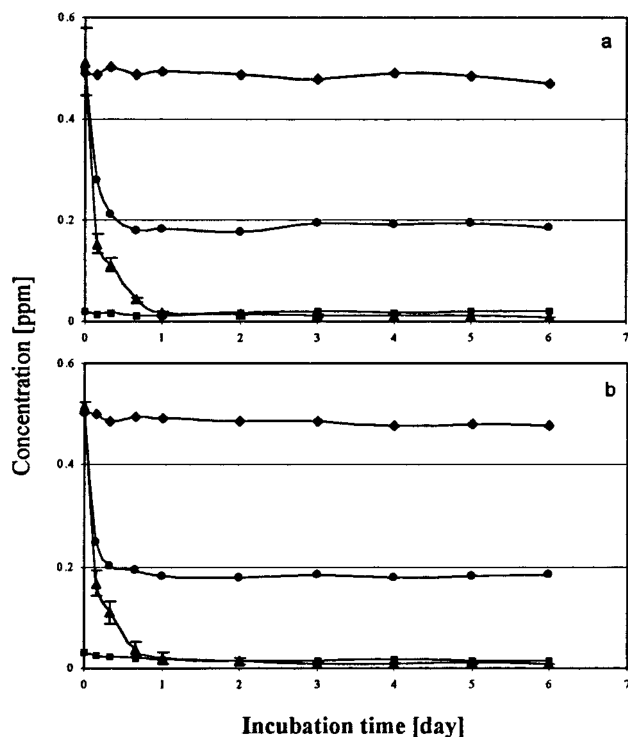

Figure 5. Uptake of *o,p′*-DDT (a) and *p,p′*-DDT (b) exposed to axenically cultivated elodea (*E. canadensis*) under controlled conditions: control (◆); autoclaved elodea (●); elodea (▲); total metabolites (*o,p′*-DDD + *o,p′*-DDE or *p,p′*-DDD + *p,p′*-DDE) (■). Concentrations are of the residual substrate in the liquid culture medium as determined by GC-ECD analysis; $C_0 = 0.5$ ppm. Error bars represent ±1 standard deviation.

aquatic plants. The greater effectiveness of transformation of DDT by duckweed is not clearly understood; this may be related to its large surface, less wax, and rapidly inducible enzymes (Morita et al., 1996).

Uptake and Phytotransformation by Elodea. The ability of elodea to degrade/bind DDT is between that of parrot feather and duckweed (Figures 2 and 5). At the end of the incubation period, the amounts of *o,p′*-DDT and *p,p′*-DDT accumulated reach 67 and 50%, respectively, whereas the amounts degraded/bound reach 31 and 48% (Figure 2). Subtraction of the 10% of *p,p′*-DDD plus *p,p′*-DDE metabolites produced (Figure 3) leaves 38% nonextractable and presumably bound *p,p′*-DDT material. This compares with the 22% nonextractable material produced during another study of DDT degradation by elodea (Garrison et al., 2000). Disappearance rates (k) and half-lives ($t_{1/2}$) are comparable to that of duckweed (Table 1). None of the observed DDT metabolites are formed by the heat-inactivated (autoclaved) plants (15 min, 121 °C). *o,p′*-DDD and *p,p′*-DDD are the main metabolites in this plant, 10.3 and 5.0%, respectively (Figure 3). However, the concentration of *p,p′*-DDE is comparable with that of *p,p′*-DDD, which is formed to a lesser extent than by parrot feather and duckweed. These results are somewhat higher than those from a study using cell suspension cultures of wheat and soybean, in which polar and nonpolar metabolites of DDT were formed in yields of only 1−2.5% and 5.3%, respectively; in that study, >80% of DDT was recovered after the end of incubation (Arjmand and Sandermann, 1985).

In the environment, DDT is sorbed to organic matter and clay materials. The resulting low bioavailability of DDT is one of the reasons for its persistence. On the other hand, microorganisms play a role as a mobile colloid to transport hydrophobic compounds, including DDT, to other compartments such as groundwater (Fujimura and Katayama, 1997). At any rate, DDT remains in the environment, and its remediation may be accomplished by the introduction of certain aquatic plants. As shown in this study, aquatic plants are able to accumulate high amounts of DDT and subsequently partly irreversibly bind it or degrade it to DDD and DDE. Although these products are also persistent, recent studies have demonstrated that they can be further converted to polar and less toxic compounds such as 1-chloro-2,2-bis(*p*-chlorophenyl)ethylene (DDMU) and 2,2-bis(*p*-chlorophenyl)acetic acid (DDA) (Fox et al., 1998).

Bioaccumulation of DDT. The BCF values as calculated here (see Materials and Methods) show that both *o,p′*-DDT and *p,p′*-DDT accumulate in parrot feather to a much greater extent than in elodea or duckweed (Table 1). These BCF values are related to the octanol−water partition coefficients (K_{ow}) of the compounds and to the metabolic potentials of the accumulating organisms. The former parameter affects the uptake rate of compounds into plants, whereas the latter influences the disappearance from plants. Different organisms gave various BCF values for DDT, ranging from 6.53×10^5 to 3.43×10^7 in leaf/air systems (Polder et al., 1998) and from 3.06×10^4 to 9.10×10^4 in fish/water systems (Haitzer et al., 1998; Kannan et al., 1993). The reasons for these differences are probably due to the fact that different amounts of biomass and different DDT concentrations were employed. No BCF data have been previously reported for DDT and its metabolites in the plants tested here. In this study, the K_{ow} affects the BCF values less than do the plant properties (Table 1); whereas the K_{ow} values (and BCF) for the two DDT isomers are similar, there are large differences between the BCF for parrot feather and the other two plants for both DDT compounds. However, all three plants showed similar rates of uptake from the culture medium. In this work, the "metabolic potential" is indicated by both the formation of identified metabolites (Figure 3), and the amount of nonextractable DDT material present after incubation (Figure 2, "degradation"). Thus, the large differences in BCF values between plants should be attributed to their metabolic potentials as well as to the accumulation of extractable DDT. Nevertheless, the BCF values of both *p,p′*-DDT and *o,p′*-DDT are high in all three plants, indicating the importance of these pesticides in the food-chain web. This importance seems to decrease at the higher trophic levels of the food chain because DDT is effectively metabolized to DDE and DDD at these levels (Muir et al., 1994). However, as DDD and DDE are still highly persistent, their fate in the environment remains a critical concern in phytoremediation processes.

Conclusions. The patterns of *o,p′*-DDT and *p,p′*-DDT uptake and metabolism in the three axenically cultivated plants studied here are similar; that is, these plants are able to accumulate almost all of the DDT from an aqueous medium and degrade from 1 to 13% of it to DDD and DDE. The phytodegradation ability is in the order duckweed > elodea ≅ parrot feather for both DDT isomers. This plant-mediated metabolism/degradation of DDT may be at least partly attributed to dehalogenation and dehydrochlorination reactions, which appear to involve a general plant dehalogenase. In

addition, even more of the DDT is apparently irreversibly bound to the plant tissue; that is, it is not extractable. The removal of DDT from aqueous solution by aquatic plants as demonstrated here may be the basis for a new phytoremediation technology.

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