Multiple Modes of Action of Diphenyl Ethers

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Diphenyl ethers are the most versatile phytotoxic compounds known so far with respect to their targets in the plant cell. According to their substituents, the following modes of action can be found: (a) inhibition of photosynthetic electron transport effective at two binding sites, (b) peroxidative activity (degradation of cellular constituents), (c) inhibition of carotenogenesis, (d) decrease of ATP-synthase (CF_1) activity, (e) inhibition of respiratory electron transport, (f) stress response. This paper presents a condensed overview.

Research on biochemistry of diphenyl ethers, some of which are commercial herbicides [1, 2], has made considerable progress within the last few years. Fig. 1 demonstrates possible modes of action which have been found so far. The concentration range for all of the six effects expands over more than three orders of magnitude, indicating that not all modes of action have a bearing to herbicidal effects.

Of course, substituents at the diphenyl-ether skeleton are responsible for the effects, as is briefly outlined in Fig. 2. This overview intends to emphasize the importance of investigating the modes of action, since knowledge of the target sites will allow for reliable research on structure/activity relationships necessary for chemical design. It appears that DPEs eventually applicable as herbicides and having sufficient selectivity may combine several modes of action all exhibiting moderate activity.

Phenoxy-phenoxy type DPEs are not considered here (compare *e.g.* [3]).

1. Photosynthetic electron transport

The I_{50} values of DPEs like nitrofen or oxyfluorfen are $\geqq 10^{-5}$ M ([4], see Fig. 1). It is unlikely

Chemical names, abbreviations: 1. Herbicides: acifluorfen-methyl, methyl 5-[2-chloro-4-(trifluoromethyl)phenoxy]-2-nitrobenzoate; bentazon, 3-isopropyl-(1 H)-2,1,3-benzothiadiazin-4(3 H)-one-2,2-dioxide; bifenox, methyl 5-(2,4-dichlorophenoxy)-2-nitrobenzoate; CME-127, 2-chloro-3-amino-4-nitrophenyl ether; DCMU (diuron), 3-(3,4-dichlorophenyl)-1,1-dimethylurea; difunon, 5-(dimethylaminomethylene)-2-oxo-4-phenyl-2,5-dihydrofurane-carbonitrile-(3); DNP-INT (KNJ-724), 2,4-dinitrophenyl-2′-iodo-4′-nitro-6′-isopropylphenyl ether; fluridone, 1-methyl-3-phenyl-5-[3-(trifluoromethyl)phenyl]-4(1 H)-pyridinone; fomesafen, 5-[2-chloro-4-(trifluoromethyl)phenoxy]-N-methanesulfonyl-2-nitrobenzamide; glyphosate, N-(phosphonomethyl)glycine; KC 1331, ethyl α-[5-(2-chloro-4-trifluoromethylphenoxy)-2-nitrophenylthio]acetate; nitrofen, 2,4-dichlorophenyl-4′-nitrophenyl ether; nitrofluorfen, 2-chloro-4-(trifluoromethyl)phenyl-4′-nitrophenyl ether; norflurazon, 4-chloro-5-methylamino-2-(3-trifluoromethylphenyl)pyridazin-3(2 H)one; oxyfluorfen, 2-chloro-4-(trifluoromethyl)phenyl-3′-ethoxy-4′-nitrophenyl ether; MV (methylviologen, paraquat), N,N′-dimethyl-4,4′-bipyridylium dichloride; trifluralin, N,N-dipropyl-2,6-dinitro-4-(trifluoromethyl)aniline. 2. Other compounds: DPE's, diphenyl ethers; DQH_2, 2,3,5,6-tetramethyl-1,4-benzoquinone (reduced); ethoxyquin, 1,2-dihydro-6-ethoxy-2,2,4-trimethylquinoline.

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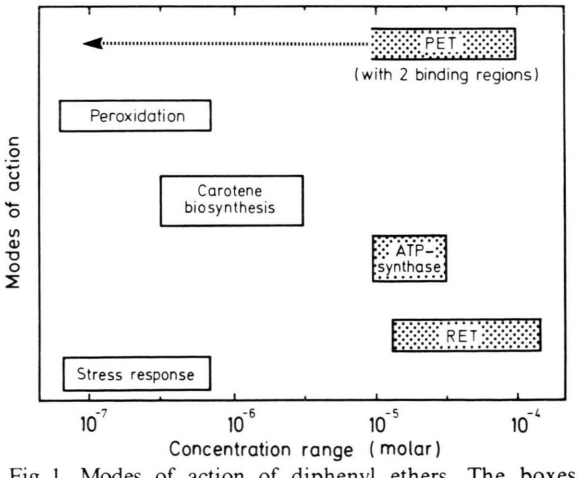


Fig. 1. Modes of action of diphenyl ethers. The boxes indicate the ranges of I_{50} values belonging to the six effects. The I_{50} may extend to higher concentrations, in the case of PET inhibition some compounds are effective down to 10^{-7} M. PET, photosynthetic electron transport; ATP-synthase, plastidic coupling factor CF_1; RET, respiratory electron transport (plant mitochondria).

that this activity is responsible when nitrofen or oxyfluorfen are used as herbicides [5]. However, complex substitution may well yield potent Hill-reaction inhibitors [6, 7], which have been shown to inhibit oxidation of plastoquinone (Fig. 2, No. 1 b). The ESR signal of the reduced Rieske iron-sulfur center of the isolated cytochrome $b_6 - f$ complex is altered by DNP-INT [8]. Inhibition located after the DCMU site is found with the majority of DPEs having higher I_{50}-values as indicated in Fig. 3 (see [7, 9], even with phenoxy-phenoxy types. Noteworthy, also trifluralin exhibits a binding affinity similar to diphenyl ethers [10]. Apparently, both the DCMU site and a site at or after the plastoquinone pool can be used for (inhibitory) binding, depending on the concentration (see *e.g.* for diphenyl ether No. 2).

We do not yet understand which chemical modifications will shift the affinity from one inhibition site to the other. There is some evidence that substitution at 2′- and 3′-position may be influential in this respect (see formula in Fig. 3 for numbering).

2. Peroxidation

By measurement of light-induced hydrocarbon-gas production [11], membrane leakage [12], or determining loss of ^{35}S-labeled sulfolipid [13] evidence was presented that herbicidal DPEs like bifenox, oxyfluorfen or acifluorfen(-methyl) induce peroxidative degradation processes. Polyunsaturated fatty acids are primary substrates leading to formation of hydrocarbons with definite chain lengths. Their carbon number is $\omega - 1$ [14, 15]; ω denotes the position of that fatty-acid double bond most distant from the carboxyl group. Peroxidatively generated short-chain hydrocarbon gases are alkanes (> 90%, some alkenes; for the peroxidative pathway see [16]). Substantial production of ethylene as observed with higher plants is due to stress response (see K. J. Kunert, this issue, and Chapter 6).

Peroxidation is light-dependent [11]. With green algae and isolated (spinach) chloroplasts white and red light (> 610 nm) can be used [17, 18]; red light is most effective. Production of short-chain hydro-

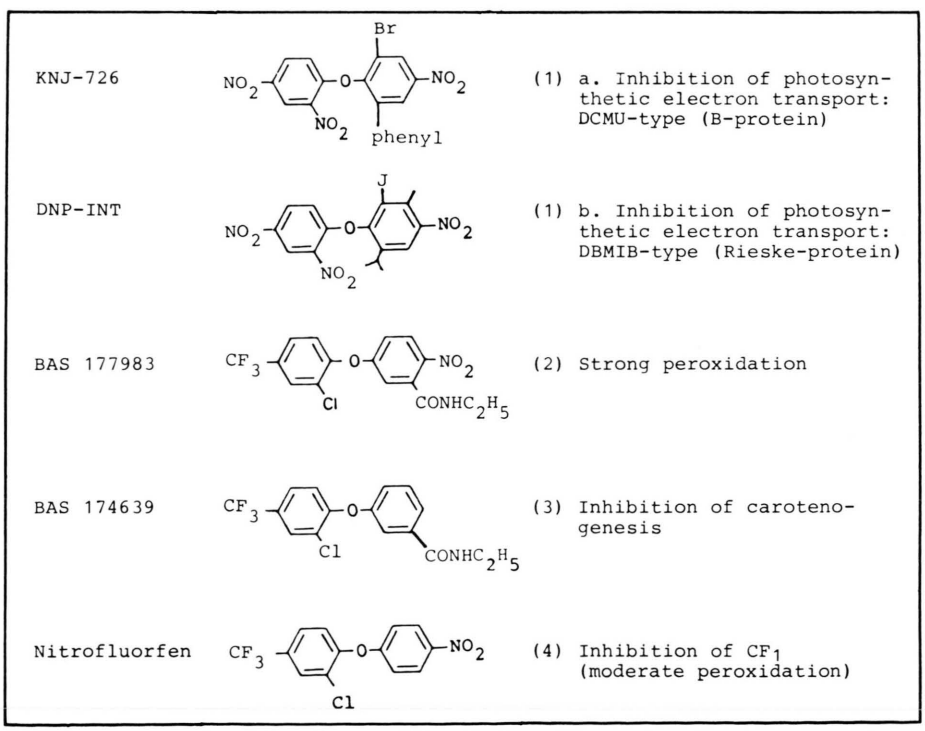


Fig. 2. Structure and activity of some diphenyl ethers. The modes of action change dramatically by substituents. The numbers denote the corresponding chapters (compiled in the given order from refs. [5, 7, 28, 31, 20]).

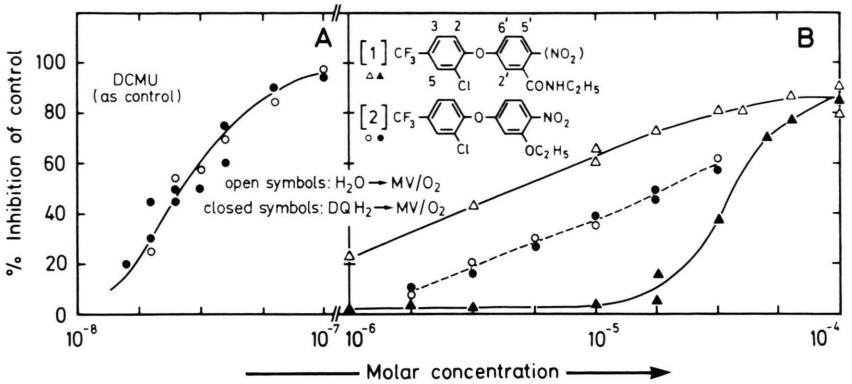


Fig. 3. Inhibition of photosynthetic electron transport by two diphenyl ethers with isolated chloroplast material (from *Bumilleriopsis filiformis*) to demonstrate two inhibition sites. Both assay systems are inhibited to the same extent by DCMU [part A] and by DPE No. (2) [part B]. The diphenyl ether No. (1) (with or without a *p*-NO_2 group) affects the system $H_2O \rightarrow MV/O_2$ (△—△), but is effective with DQH_2 as donor at concentrations above 5×10^{-5} M only (▲—▲). This donor feeds in electrons at plastoquinone. Apparently, compound No. (2) is attached after the DCMU-binding site and after the plastoquinone pool.

Control rates: 110–120 with water, 300 to 320 μmol O_2 uptake/mg chlorophyll × h with DQH_2 as donor (assay see [49]; DQH_2, 0.5 mM).

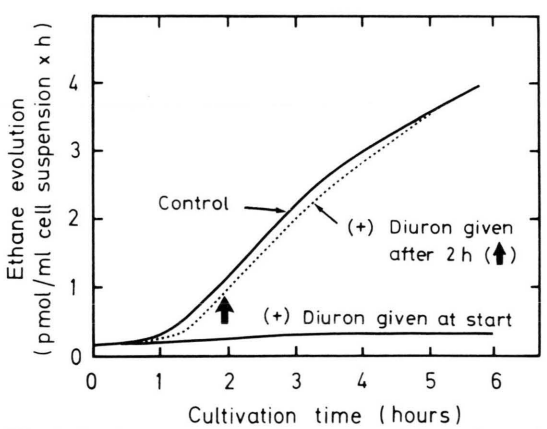


Fig. 4. Oxyfluorfen-induced formation of C_2 hydrocarbons by *Scenedesmus* in the light under cultivation conditions: DCMU effect (1 μM for both herbicides; after [11]).

carbons is inhibited by DCMU (Fig. 4). The herbicidal symptoms of soybeans and certain broad-leaf weeds (with acifluorfen) and cereals (with KC 1331) become less obvious with concurrent bentazon application. As was shown with soybeans, deterioration of photosynthesis by these DPEs is delayed for several hours until bentazon has been metabolized [19], allowing again photosynthetic electron transport, while in plants wihout bentazon present peroxidation DPEs are active after a short lag phase (B. Würzer and G. Retzlaff of BASF, Limburgerhof, pers. communication). Oxyfluorfen (1 μM) is not effective with illuminated spinach-cell cultures which, although green, do not grow autotrophically (K. J. Kunert, unpublished). Consequently, for green tissue with an intact photosynthetic apparatus, photosynthetic electron transport is assigned the decisive role to "activate" peroxidative DPEs. It still remains to be shown whether this occurs by a transient (radicalic) 1-electron reduction of the DPE itself or whether an unknown endogenous radical reaction (in the cell or membrane) is enhanced or stabilized by DPEs. This in turn will start peroxidative reactions. Oxygen must be present to form a free peroxy radical with fatty acids. Little oxygen is needed, since no light-induced O_2 uptake can be measured with, *e.g.*, 1 μM oxyfluorfen present, which is sufficient for peroxidation and subsequent cell death. Furthermore, superoxide dismutase has no influence in a cell-free chloroplast system using oxyfluorfen [11], which evidences that O_2^- does not play a major role in peroxidation (as in the case of paraquat). It was reported that light-induced O_2 uptake of isolated chloroplasts was enhanced by higher fomesafen concentrations (3 to 5×10^{-5} M) [20]. It remains unclear whether this effect is related to a possible peroxidative activity and has a bearing to the fomesafen concentration used in weed control.

Using cucumber cotyledons, carotenoids were proposed as sensitizers for DPE activation [21], as

was claimed long ago by Matsunaka ([1] for rice seedlings), since the effect of acifluorfen-methyl (membrane leakage) was abolished by preventing carotenoid biosynthesis with fluridone present. Also the stimulation of light-induced anthocyanin synthesis by acifluorfen in etiolated mustard seedlings was alleviated when carotenoids were absent [22]. Lowering carotenoids in *Scenedesmus*, however, no decrease of peroxidation by oxyfluorfen is observed provided electron transport remains operative [23].

A report on flavin-mediated activation of acifluorfen-methyl (enhancing phototropism) appears doubtful [24]. Photosynthetic studies with intact chloroplasts give some evidence that CO_2 assimilation itself may become affected by acifluorfen [25].

The chemical nature of radicals starting peroxidation and how these first radicals are originated is still unknown. Both, photosynthetic electron transport and carotenoids may activate peroxidizing DPEs, though the first apparently is dominant in fully developed photosynthetic cells. Noteworthy, strong light (pigment-absorbing radiation of $> 125\ W/m^2$) applied to photosynthetically "incompetent" tissue [21] with low chlorophyll content may cause (oxidative) photochemistry at pigments leading to radicals. In theory, a single radicalic molecule can "autocatalytically" induce peroxidation, when radical quenchers are absent. Consequently, the problem of radical initiation has to be related to endogenous antioxidants.

α-Tocopherol [17, 21] or ethoxyquin [26] effectively suppress peroxidation in intact tissue or cells. In oxyfluorfen-treated mustard seedlings, the level of the antioxidant vitamin C decreases inversely with increasing ethane formation [27]. The plants indicated in Fig. 5 exhibit selectivity against oxyfluorfen during the experimental time. Tolerance of the three species tested is inversely related to the ascorbic-acid content originally present in the target plant. After a 2-h treatment with oxyfluorfen, when photosynthetic electron transport is already damaged (90%), DCMU does not stop peroxidation, although light is still required for oxyfluorfen activity [26] (Fig. 4). This can be explained by exhaustion of endogenous antioxidants (which are low anyway in *Scenedesmus* as compared to higher plants; own findings) during this 2-h period. Then, a small number of radicals (originated by light through carotenoids?) may be sufficient to maintain deteriorating effects.

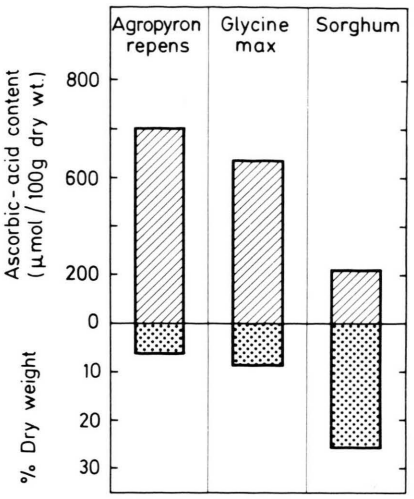
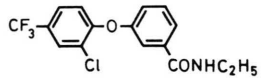
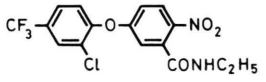
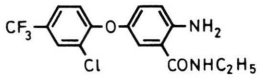
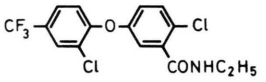


Fig. 5. Peroxidative activity of oxyfluorfen referred to the level of ascorbic acid present in three higher-plant species before herbicide treatment. Increase of dry weight of the plants per fresh weight (= water loss) is used as a marker for herbicidal activity, as given by the lower ordinate = % dry wt. of treated samples minus % dry wt. of control. Plants (about 2 weeks old) were sprayed with oxyfluorfen (equivalent to 6.6 kg a.i./ha), darkened for 12 h, and subsequently exposed to white light ($170\ W/m^2$, day/night regime, 22 °C) in a growth chamber for 48 h. The comparatively high herbicide dosis ensured rapid development of physiological effects (K. J. Kunert and B. Finckh, this laboratory, unpubl. results).

Since peroxidation is counteracted by high endogenous levels and adequate regeneration of cellular radical quenchers, conceivably DPEs of low peroxidative activity are promising candidates for *selective* weed control. CME 127, acifluorfen or fomesafen should be mentioned in this respect. Of course (degradative) metabolism of these xenobiotics is another important factor for selectivity, which is not considered here.

Investigations on structure/activity relationships have shown that *para* (4′)-nitro DPEs are best peroxidative agents provided the neighboring 3′-substituents are $-OCH_3$, $-OC_2H_5$, $-NHC_2H_5$, $-COOCH_3$ or $-CONHCH_3$ (the latter two substituents may have C_2H_5 instead of CH_3). The low peroxidizing activity of acifluorfen-sodium salt is thought to be caused by its polarity (low lipophilicity); see [5] for more information. The *p*-nitro group, however, is not essential for peroxidation as is demonstrated by Table I and [28, 29]. Using *Scenedesmus*, peroxidation is again abolished by DCMU. Recently, trapped ESR signals were mea-

Table I. Example for peroxidizing non-nitro diphenyl ethers.

	C_2-hydrocarbon formation (nmol/ml packed cell volume over 15 h) [a]	
Control	0.4	
1. 	1.5	
2. 	14	no ethane formation with 10 µM diuron present
3. 	6	
4. 	11	

[a] In the light, with autotrophic *Scenedesmus acutus*. Concentration of DPEs 10 µM; see [28].

sured with isolated spinach chloroplasts to show radical formation in the presence of oxyfluorfen (a *para*-nitro type DPE) or a *para*-Cl type DPE (= No. 4 of Table I). The signals with both were identical, and were suppressed by DCMU (see R. Lambert *et al.*, this issue).

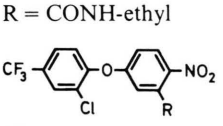
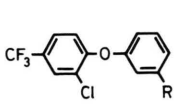
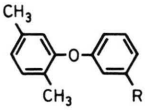
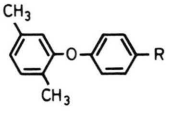
3. Inhibition of carotenogenesis

Peroxidation degrades pigments [11], but phytoene never accumulates [30]. *m*-Phenoxybenzamides like Nos. (2) or (3) of Table II, however, exhibit strong bleaching, *i.e.* stalling formation of carotenoids [28, 31], while phytoene is enriched. Their effect is similar to norflurazon or difunon [32]. These DPEs operate in the dark, as can be reliably demonstrated with a green algal species that forms its (green) chloroplast in the light as well as in the dark (Table II, control). Exclusively using autotrophic cells, the particular effect of this DPE-type will be overlooked, since in the *light* both the non-nitro and nitro compounds decrease carotenoids and chlorophyll content (Nos. 1, 2). However, their modes of action are different as seen with the *dark* culture. Here, No. (2) is active and effects carotenoids only. No. (1) has no effect; its nitro group counteracts inhibitory activity on carotenogenesis. The same is true with fomesafen. In the light, of course, both DPEs are active: No. (2) through lowering the carotenoid level (which concurrently decreases chlorophyll content), No. (1) by pigment degradation *via* peroxidation. As seen further (Nos. 3, 4) the *m*-position of the $-CONHC_2H_5$ group is essential.

The finding of Table II with intact cells was confirmed using a cell-free carotenogenic system recently developed from *Aphanocapsa* [33] (see, *e.g.*, Table II of G. Sandmann *et al.*, this issue). Some data are given in Table III. The ^{14}C-ratio of phytoene *vs.* β-carotene can be used as an index to reliably describe the inhibitory activity of the compounds.

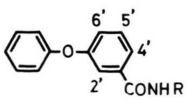
We do not yet know all structural prerequisites. As shown in the cell-free assay, R at the 3′-CONHR element is effective as alkyl up to C_4, but branching is unfavorable; the "left" phenyl ring contributes by substituents which increase lipophilicity. Also the

Table II. Effect of *m*-phenoxybenzamides on carotenoid and chlorophyll level.

Culture condition, Pigments		R = CONH-ethyl 			
	Control	(1)	(2)	(3)	(4)
Autotrophic (light)					
Carotenoids	1.24	0.35	0.45	0.50	1.2
Chlorophyll	14.0	6.0	8.0	10.8	14.0
Heterotrophic (dark)					
Carotenoids	0.45	0.45	0.23	0.25	0.45
Chlorophyll	5.9	6.0	6.0	5.9	5.8

With intact *Scenedesmus acutus*, cultivated for 24 h in the presence of 1 µM of the diphenyl ethers indicated. Pigments are expressed as mg/ml packed cell volume. See [31] for experimental details.

Table III. Inhibition of carotenogenesis by *m*-phenoxybenzamides (10 μM) in a cell-free system [a].



	[^{14}C]phytoene/ [^{14}C-β]carotene
Control	0.06
R = $-CH_3$	0.11
$-C_2H_5$	0.36
$-C_3H_7$	0.98
n-butyl	1.20
s-butyl	0.84
t-butyl	0.17
4'-CONH-*n*-butyl, 3'-H	0.20

[a] From *Aphanocapsa* 6714 according to [33]; incorporation of [^{14}C]geranylgeranyl pyrophosphate into phytoene and β-carotene [48].

N-ethyl-3-(trifluoromethyl)benzamide is very active. Furthermore, we have presented some evidence that such an *m*-phenoxybenzamide must in part resemble the phytoene structure, possibly to block phytoene desaturase (G. Sandmann *et al.*, this issue).

4, 5. Inhibition of ATP-synthase and respiratory electron transport

As reported in a classical paper [34], *p*-nitro-DPEs may inhibit both photosynthetic electron transport and photophosphorylation. Later on, it was shown with nitrofen [35], and some other chlorinated *p*-nitro DPEs [5, 20] that the ultimate (enzymic) step of ATP formation is inhibited. At concentrations too low to block electron transport, nitrofen (or nitrofluorfen) compete with ADP [35, 20]. This "energy-transfer" inhibition affects nucleotide exchange at the plastidic ATP-synthase (CF_1) both at its "loose" and "tight" ADP-binding sites [36, 37].

The effect of nitrofen on photophosphorylation can be demonstrated by its interference with "photosynthetic control" (Table IV). The basal electron-transport rate is enhanced by ADP (line 1), which is not observed with a CF_1-inhibitor present (line 2). Uncouplers (like NH_4Cl) restore a rate identical with the uncoupled control, indicative of electron transport not being impaired by the nitrofen concentration used here. In corn mitochondria, the ADP-enhanced electron transport is inhibited alike. In contrast to chloroplasts, an uncoupler has no alleviating effect. Inhibition remains of the same extent (%) whether ADP or ADP + gramicidin are present (line 4). Apparently, in mitochondria only electron transport itself is essentially affected.

I_{50} values for mitochondrial electron-transport inhibition are about 5×10^{-5} M [38]. This is far too high to account for herbicidal activity.

6. Stress response

As an early response to peroxidizing nitro-DPEs higher plants evolve ethylene [39]. This is evident when using low light intensity, thus limiting phytotoxic effects [40] with little (peroxidative) ethane formation (K. J. Kunert, this issue). Application of a phytotoxic dosis (≈ 10 μM) of acifluorfen (or oxyfluorfen) induces rapid ethylene production [41] in the light (after less than 3 h), later followed by ethane evolution (15–20 h) together with necrosis as a typical herbicidal symptom. Aminoethoxyvinylglycine [42] inhibits formation of ethylene, but not of ethane [41]. Therefore, we are dealing with a typical stress response.

Ethylene is thought to be an inducer of phenylalanine-ammonia lyase (PAL). Accordingly – as was demonstrated by application of phytotoxic

Table IV. Nitrofen inhibition on plastidic and mitochondrial electron transport.

System	(−)ADP	(+)ADP	(+)NH_4Cl, ADP
A. Spinach chloroplasts			
1. Control	48	120	162
2. (+)Nitrofen, 5 μM	48	76	160
B. Corn mitochondria (seedlings)			(+)Gramicidin D
3. Control	47	145	177
4. (+)Nitrofen, 50 μM	49	78 [59%]	104 [59%]

Data are rates of oxygen uptake expressed as μmol O_2/mg chlorophyll × h in *A* (system $H_2O \rightarrow MV/O_2$; for details see [35]), and as nmol O_2/mg protein × min in *B* (system NADH → O_2; NADH, 0.5 mM; ADP, 10 μM; gramicidin 10 μM). Figures in brackets: percent inhibition *vs.* control of line 3. Inhibition of respiration is under investigation (R. Lambert, this laboratory, unpubl. results).

doses of acifluorfen to spinach leaves – the PAL level increases 25-fold within 20 h in the light (not in the dark) and declines when typical herbicide-induced leaf injuries become visible [43]. By PAL induction phenylpropanoid biosynthesis is turned on leading to isoflavonoid phytoalexins in several higher-plant species [41]. Such a formation of "stress metabolities" is known from other abiotic elicitors like toxic concentrations of Cu(II) or sodium chlorate (comp. [44]). No increase of PAL activity is generally observed with herbicides, except for paraquat and glyphosate [45]. The action of the latter is known not to reflect a stress response.

Phytoalexins formed (after infection by *Phytophthora megasperma*) remain localized close to the infection site [46]. Phytoalexins exert phytotoxicity also upon the host, apparently most effectively when they are induced all over the tissue by DPEs. It is tempting to speculate that such an effect may contribute to DPE phytotoxicity. Noteworthy, phytoalexins, like rishitin, affect permeability of membranes (*cf.* [47], see refs. therein).

It is unknown how DPEs act as elicitors and whether peroxidative properties are obligatory.

Acknowledgement

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[1] S. Matsunaka, Residue Rev. **25,** 45–58 (1969).
[2] R. Y. Yih and C. Swithenbank, J. Agric. Food Chem. **23,** 592–593 (1975).
[3] R. Handte, M. Koch, H. Bieringer, and G. Hörlein, Z. Naturforsch. **37b,** 912–922 (1982).
[4] G. Van den Berg and J. Tipker, Pestic. Sci. **13,** 29–38 (1982).
[5] R. Lambert, G. Sandmann, and P. Böger, Pest. Biochem. Physiol. **19,** 309–320 (1983).
[6] A. Trebst, H. Wietoska, W. Draber, and H. J. Knops, Z. Naturforsch. **33c,** 919–927 (1978).
[7] W. Draber, H. J. Knops, and A. Trebst, Z. Naturforsch. **36c,** 848–852 (1981).
[8] R. Malkin, Biochemistry **21,** 2945–2950 (1982).
[9] M. W. Bugg, J. Whitmarsh, C. E. Rieck, and W. S. Cohen, Plant Physiol. **65,** 47–50 (1980).
[10] S. J. Robinson, C. F. Yocum, H. Ikuma, and F. Hayashi, Plant Physiol. **60,** 840–844 (1977).
[11] K. J. Kunert and P. Böger, Weed Sci. **29,** 169–172 (1981).
[12] G. L. Orr and F. D. Hess, Pest. Biochem. Physiol. **16,** 171–178 (1981).
[13] G. Sandmann and P. Böger, Plant Sci. Lett. **24,** 347–352 (1982).
[14] G. Sandmann and P. Böger, Lipids **17,** 35–41 (1982).
[15] G. Sandmann and P. Böger, Lipids **18,** 37–41 (1983).
[16] G. Sandmann and P. Böger, in: Biochemical Responses Induced by Herbicides (D. E. Moreland, J. B. St. John and F. D. Hess, eds.) ACS Symposium Series no. 181, pp. 111–130, Amer. Chem. Society, Washington D. C., 1982.
[17] R. Lambert, G. Sandmann, and P. Böger, in: Pesticide Chemistry – Human Welfare and the Environment (J. Miyamoto and P. C. Kearney, eds.), **Vol. 3,** pp. 97–102, Pergamon Press, Oxford-New York-Toronto-Sydney-Paris-Frankfurt 1983.
[18] R. Lambert and P. Böger, Z. Pflanzenkrankh. Pflanzenschutz, special issue, **no. 9,** 147–152 (1981).
[19] G. Retzlaff and R. Hamm, Weed Res. **16,** 263–266 (1976).
[20] S. M. Ridley, Plant Physiol. **72,** 461–468 (1983).
[21] G. L. Orr and F. D. Hess, Plant Physiol. **69,** 502–507 (1982); in: Biochemical Responses induced by Herbicides (D. E. Moreland, J. B. St. John, and F. D. Hess, eds.), ACS Symposium Series no. 181, pp. 131–152, Amer. Chem. Society, Washington D. C., 1982.
[22] H. Mohr and H. Drumm-Herrel, Physiol. Plant. **58,** 408–414 (1983).
[23] R. Lambert and P. Böger, J. Agric. Food Chem. (1984), in press.
[24] T.-Y. Leong and W. R. Briggs, Plant Physiol. **70,** 875–881 (1982).
[25] R. Alscher-Herman and C. A. Michrina, Pest. Biochem. Physiol. (1984), in press.
[26] K. J. Kunert and P. Böger, J. Agric. Food Chem. (1984), in press.
[27] K. J. Kunert, in: Oxygen Radicals in Chemistry and Biology (W. Bors *et al.*, eds.), de Gruyter, Berlin-New York 1984, in press.
[28] R. Lambert and P. Böger, Proc. 6th Int. Congr. Photosynth., Brussels 1983, in press.
[29] M. P. Ensminger, F. D. Hess, and J. T. Bahr, Pest. Biochem. Physiol. (1984), submitted.
[30] G. Sandmann and P. Böger, Weed Sci. **31,** 338–341 (1983).
[31] R. Lambert and P. Böger, Pest. Biochem. Physiol. **20,** 183–187 (1983).
[32] K. J. Kunert and P. Böger, Weed Sci. **26,** 292–296 (1978); Z. Naturforsch. **34c,** 1047–1051 (1979).
[33] I. E. Clarke, G. Sandmann, P. M. Bramley, and P. Böger, FEBS Lett. **140,** 203–206 (1982).
[34] D. E. Moreland, W. J. Blackman, H. G. Todd, and F. S. Farmer, Weed Sci. **18,** 636–642 (1970).
[35] R. Lambert, K.-J. Kunert, and P. Böger, Pest. Biochem. Physiol. **11,** 267–274 (1979).
[36] B. Huchzermeyer, Z. Naturforsch. **37c,** 787–792 (1982).
[37] B. Huchzermeyer and A. Loehr, Biochim. Biophys. Acta **724,** 224–229 (1983).
[38] J. A. Bohn, Quantitative structural relationship to physiological activity of diphenylether herbicides. Thesis, University of Kentucky, Lexington 1980.

[39] S. F. Gorske and H. J. Hopen, Weed Sci. **26,** 585–588 (1978).
[40] D. E. Vanstone and E. H. Stobbe, Weed Sci. **25,** 352–354 (1977).
[41] T. Kömives and J. E. Casida, J. Agric. Food Chem. **31,** 751–755 (1983).
[42] T. Boller, R. C. Herner, and H. Kende, Planta **145,** 293–303 (1979).
[43] T. Kömives and J. E. Casida, Pest. Biochem. Physiol. **18,** 191–196 (1982).
[44] F. B. Abeles, Ethylene in Biology, Academic Press, New York 1983.
[45] R. E. Hoagland and S. O. Duke, Weed Sci. **29,** 433–439 (1981); **31,** 845–852 (1983).
[46] H. Grisebach, H. Börner, and P. Moesta, Ber. Dtsch. Bot. Ges. **95,** 619–642 (1982).
[47] D. A. Smith, in: "Phytoalexins" (J. A. Bailey and J. W. Mansfield, eds.), pp. 289–318, Wiley, New York 1982.
[48] I. E. Clarke, G. Sandmann, P. Bramley, and P. Böger, Pest. Biochem. Physiol. (1984), submitted.
[49] P. Böger, G. Sandmann, and R. Miller, Photosynth. Res. **2,** 61–74 (1981).