Occurrence and biological activity of palmitoleic acid isomers in phagocytic cells

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Abstract Recent studies have highlighted the role of palmitoleic acid [16:1n-7 (cis-9-hexadecenoic acid)] as a lipid hormone that coordinates cross-talk between liver and adipose tissue and exerts anti-inflammatory protective effects on hepatic steatosis and insulin signaling in murine models of metabolic disease. More recently, a 16:1n-7 isomer, cis-7-hexadecenoic acid (16:1n-9), that also possesses marked anti-inflammatory effects, has been described in human circulating monocytes and monocyte-derived macrophages. By using gas chromatographic/mass spectrometric analyses of dimethyl disulfide derivatives of fatty acyl methyl esters, we describe in this study the presence of a third 16:1 isomer, sapienic acid [16:1n-10 (6-cis-hexadecenoic acid)], in phagocytic cells. Cellular levels of 16:1n-10 appear to depend not only on the cellular content of linoleic acid, but also on the expression level of fatty acid desaturase 2, thus revealing a more complex regulation both at the enzyme level, via fatty acid substrate competition, and directly at the gene level. However, unlike 16:1n-7 and 16:1n-9, 16:1n-10 levels are not regulated by the activation state of the cell. Moreover, while 16:1n-7 and 16:1n-9 manifest strong anti-inflammatory activity when added to the cells at low concentrations (10 μM), notably higher concentrations of 16:1n-10 are required to observe a comparable effect. Collectively, these results suggest the presence in phagocytic cells of an unexpected variety of 16:1 isomers, which can be distinguished on the basis of their biological activity and cellular regulation.—Astudillo, A. M., C. Meana, C. Guijas, L. Pereira, P. Lebrero, M. A. Balboa, and J. Balsinde. Occurrence and biological activity of palmitoleic acid isomers in phagocytic cells. J. Lipid Res. 2018. 59: 237–249.

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Recent studies have revealed that in animal models of metabolic disease, the adipose tissue releases the monounsaturated fatty acid, palmitoleic acid [16:1n-7 (cis-9-hexadecenoic acid)], which suppresses hepatic steatosis and improves insulin sensitivity in the whole body (1, 2). Thus, this fatty acid has taken the spotlight as a promising anti-inflammatory lipid that may help to ameliorate metabolic disorders (1, 2). There is a large body of literature suggesting that monounsaturated fatty acids are beneficial to human health (3). The ratio of saturated to unsaturated fatty acids constitutes an important property of the phospholipid composition of biological membranes. Changes to this proportion are thought to exert deleterious effects on cells and, in particular, in the number of unsaturated fatty acids in membranes may contribute to the development of a number of pathophysiological states, such as cardiovascular disease, diabetes, and cancer (3). From a biophysical point of view, monounsaturated fatty acids are “good” to biological membranes because, being liquid at body temperature yet not easily oxidized, they help to maintain membrane fluidity within the appropriate limits.

However, aside from, or in addition to, these biophysical effects, there appears to be something else to 16:1n-7 that makes it unique in terms of biological activity, leading to the concept of this fatty acid serving as a lipid hormone or “lipokine” that coordinates metabolic responses between tissues (1, 2). Work in murine models generally supports a role for 16:1n-7 as a novel anti-inflammatory mediator and a positive correlate of insulin sensitivity (4). Accordingly, in

Abbreviations: 16:1n-5, palmitvaccenic acid; 16:1n-7, palmitoleic acid (cis-9-hexadecenoic acid); 16:1n-9, cis-7-hexadecenoic acid; 16:1n-10, sapienic acid (cis-6-hexadecenoic acid); DMDS, dimethyl disulfide; FADS2, fatty acid desaturase 2 (Δ5 desaturase); LPS, bacterial lipopolysaccharide; PC, choline glycerophospholipid; PE, ethanolamine glycerophospholipid; PI, phosphatidylinositol; PS, phosphatidylserine; qPCR, quantitative PCR.

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cells of murine origin, circulating 16:1 \( n \)-7 increases basal glucose uptake and activates insulin signaling, enhances \( \beta \)-cell function, improves glycemic control, increases glucose transport into skeletal muscle, and prevents palmitate-induced endoplasmic reticulum stress and apoptosis (1, 2, 4–7). Studies in humans, however, have suggested different outcomes (8, 9). Circulating levels of 16:1 \( n \)-7 in humans have been reported to correlate positively with the degree of hepatic steatosis, as well as the adiposity that promotes fat deposition in hepatocytes (10–12). A recent study has found a positive correlation between circulating 16:1 \( n \)-7 levels and markers of inflammation in young healthy Canadians (13).

Thus, although a unifying mechanistic hypothesis that may help reconcile the variety of effects that 16:1 \( n \)-7 appears to serve has yet to emerge, the recent finding that innate immune cells contain an isomer of 16:1 \( n \)-7, namely \( \text{cis-7-hexadecenoic acid} \) \( 16:1 \text{\( n \)-9} \) that also displays potent anti-inflammatory activity (14), has raised the challenging possibility that this multiplicity of actions and the compartmentalized manner in which they occur could be due, at least in part, to the overlapping actions of such isomers being present at the same or neighboring locations. The 16:1 \( n \)-9 is synthesized by human phagocytic cells via \( \beta \)-oxidation of oleic acid and its levels are elevated in the neutral lipids of lipid droplet-laden monocytes, suggesting the possibility that detection of increased levels of this fatty acid in monocytes may constitute a biomarker for foamy cell formation (14). When added exogenously, 16:1 \( n \)-9 exhibits potent anti-inflammatory actions in vitro and in vivo that are distinguishable from those of 16:1 \( n \)-7 and comparable in magnitude to those exerted by \( n \)-3 fatty acids (14).

In the current study, we report that human phagocytic cells also express low levels of a third 16:1 isomer, sapienic acid \( [16:1 \text{\( n \)-10 (cis-6-hexadecenoic acid)}] \). Despite the common assumption that 16:1 \( n \)-10 is restricted to humans (15), we found relatively high levels of this fatty acid in murine macrophages and in murine macrophage-like cell lines also. In these cells, 16:1 \( n \)-10 levels appear to be controlled by complex mechanisms of regulation, including substrate competition and gene expression. Of note, 16:1 \( n \)-10 shows anti-inflammatory activity when added to cells, but the activity is observed at higher concentrations (>25 \( \mu \)M) than those required for 16:1 \( n \)-7 and 16:1 \( n \)-9 to exert similar effects.

**MATERIALS AND METHODS**

**Cell isolation and culture conditions**

Human monocytes were isolated from buffy coats of healthy volunteer donors obtained from the Centro de Hemoterapia y Hemodonación de Castilla y León. Written informed consent was obtained from each donor. In brief, blood cells were diluted 1:1 with PBS, layered over a cushion of Ficoll-Paque, and centrifuged at 750 \( g \) for 30 min. The mononuclear cellular layer was recovered and washed three times with PBS, resuspended in RPMI 1640 medium supplemented with 40 \( \mu \)g/ml gentamicin, and allowed to adhere in sterile dishes for 2 h at 37°C in a humidified atmosphere of \( \text{CO}_2/\text{air} \) (1:19). Nonadherent cells were removed by washing extensively with PBS and the remaining attached monocytes were used the following day (14, 16, 17). Human macrophages were obtained by incubating plastic-adhered monocytes in RPMI with heat-inactivated 5% human serum and 40 \( \mu \)g/ml gentamicin for 2 weeks in the absence of exogenous cytokine mixtures (16). All

**Fig. 1.** GC/MS analyses of 16:1 isomers. Authentic 16:1 fatty acid standards were methylated and transformed into DMDS adducts as described in the Materials and Methods. A: Chromatographic separation of 16:1 fatty acid isomers. Mass spectra corresponding to the \( n \)-10 (B), \( n \)-9 (C), \( n \)-7 (D), and \( n \)-5 (E) isomers showing the molecular ion (M+) and the three diagnostic peaks (fragment corresponding to the terminal methyl part of the molecule, fragment containing the carbonyl group, and the latter with the loss of methanol).
procedures involving human samples were undertaken in accordance with the Spanish National Research Council Committee on Bioethics, under the guidelines established by the Spanish Ministry of Health and the European Union.

Murine resident peritoneal macrophages from Swiss mice (University of Valladolid Animal House; 10–12 weeks old) were obtained by peritoneal lavage using 5 ml of cold PBS, as described elsewhere (18, 19). The cells were plated at $2 \times 10^6$ per well (6-well plates) in 2 ml of RPMI 1640 medium with 10% heat-inactivated fetal bovine serum, 100 U/ml penicillin, and 100 mg/ml streptomycin, and allowed to adhere for 20 h in a humidified atmosphere of 5% CO$_2$ at 37°C. Wells were extensively washed with PBS to

Fig. 2. Analysis of 16:1 isomers in human cells. After sample collection and lipid extraction, total lipids were transmethylated. The resulting fatty acid methyl esters were derivatized with DMDS and DMDS adducts of 16:1 isomers measured by GC/MS. Chromatographic region showing the main 16:1 fatty acid isomers present in human monocytes (A), human monocyte-derived macrophages (B), and human serum (C). The mass spectra corresponding to the n-10 (D, H), n-9 (E, I), n-7 (F, J), and n-5 (G, K) isomers, indicating the molecular ion (M$^+$) and the three diagnostic peaks (fragment corresponding to the terminal methyl part of the molecule, fragment containing the carboxyl group, and the latter with the loss of methanol) are also shown.
remove nonadherent cells. Adherent macrophages were then used for experimentation. All the protocols and procedures were approved by the Institutional Animal Care and Usage Committee of the University of Valladolid and are in accordance with the Spanish and European Union guidelines for the use of experimental animals. The macrophage-like cell lines, RAW264.7, P388D1, and THP-1, were maintained in culture and handled exactly as described elsewhere (20–27).

For experiments, the cells were incubated in fresh serum-free medium for 1 h before addition of the various stimuli for different times. Unstimulated cells received vehicle. Opsonized zymosan was prepared exactly as described (28). No endogenous phospholipase A₂ activity was detected in the zymosan batches used in this study, as assessed by assay in vitro. Total cellular protein was measured utilizing a commercial kit (Bio-Rad Protein Assay). Protein levels did not significantly change over the course of the experiments.

**GC/MS of fatty acid methyl esters**

The samples, corresponding approximately to 10⁷ cells, were washed with PBS, lysed with water, and sonicated in a tip homogenizer twice for 15 s. Afterward, 10 nmol of 1,2-diheptadecanoyl-sn-glycero-3-phosphocholine and 4 nmol of 1,2,3-trihexadecanoylgllycerol were added and total lipids were extracted according to Bligh and Dyer (29). Lipid classes were separated by thin-layer chromatography. For neutral lipid separation, a solvent according to Bligh and Dyer (29) was used. Lipid subclasses, the plates were predeveloped with chloroform/methanol (1:1, v/v) and, after being air-dried, they were impregnated with 1.5% boric acid in ethanol/water (1:1, v/v). Phospholipids were separated twice with chloroform/methanol/28% ammonium hydroxide (60:37.5:4, v/v/v) as the mobile phase (31). The various lipid classes were scraped and extracted from the silica with 1 ml of chloroform/methanol (1:2, v/v) followed by 1 ml chloroform/methanol (2:1, v/v). The lipid extracts thus obtained were treated with 500 µl of 0.5 M KOH in methanol for 60 min at 37°C to obtain fatty acid methyl esters. To neutralize, 500 µl of 0.5 M HCl were added. Extraction of the fatty acid methyl esters was carried out with 2 ml n-hexane and 1 ml µl was subjected to GC/MS analysis, as previously described (14, 16, 32), using an Agilent 7890A gas chromatograph coupled to an Agilent 5975C mass-selective detector operated in electron impact mode (EI, 70 eV), equipped with an Agilent 7693 autosampler and an Agilent DB25 column (60 m length × 0.25 mm internal diameter × 0.15 µm film thickness). Data analysis was carried out with the Agilent G1701EA MSD Productivity Chemstation software, revision E.02.00.

**Dimethyl disulfide derivatization of fatty acid methyl esters**

Derivatization of fatty acid methyl esters to dimethyl disulfide (DMSD) adducts was carried out according to the procedure described by Sansone et al. (33), with slight modification. Briefly, the sample containing the fatty acid methyl esters in n-hexane (70–100 µl) was placed in a screw amber cap vial with Teflon-lined cap. DMSD (100 µl) (Sigma-Aldrich) was added, together with 50 µl of a 6% solution of iodine in diethyl ether. The vial was capped under nitrogen. The reaction was stirred at 40°C overnight. Afterward, 1 ml of n-hexane and 1 ml of 5% sodium thiosulfate solution were added. After mixing vigorously, the sample was centrifuged and the organic phase was collected, dried with anhydrous sodium sulfate, concentrated, and subjected to GC/MS analysis. DMSD derivatives were analyzed with the same protocol as fatty acid methyl esters, with slight modifications of the oven conditions. The final ramp temperature was set at 10°C/min up to 250°C and this temperature was held for 20 min to allow the DMSD derivatives of 16:1 isomers to elute. Identification of 16:1 isomers was carried out by elution time of the different DMSD adducts and by specific fragments of the mass spectra generated by cleavage of the C–C bond that originally contained the double bond (ω-fragment, Δ-fragment, and Δ-fragment with the loss of methanol).

**LC/MS of 16:1-containing phospholipid**

A cell extract corresponding to 5 × 10⁵ cells was used for these analyses. The following internal standards were added: 600 pmol each of 1,2-dipentadecanoyl-sn-glycero-3-phosphocholine, 1,2-dilauroyl-sn-glycero-3-phosphoethanolamine, 1,2-dipalmityl-sn-glycero-3-phosphoinositol, and 1,2-dimyristoyl-sn-glycero-3-phosphoserine, before lipid extraction according to the method of Bligh and Dyer (29). After evaporation of the organic solvent under vacuum, lipids were redissolved in 100 µl methanol/water (9:1, v/v) and injected into a high-performance liquid chromatograph equipped with a binary pump, Hitachi LaChrom Elite L-2130 and a Hitachi Autosampler L-2200 (Merck). The column

![Fig. 3. Analysis of 16:1 fatty acids in activated cells.](image-url)
was a Supelcosil LC-18 (5 μm particle size, 250 × 2.1 mm) (Sigma-Aldrich) protected with a Supelguard LC-18 (20 × 2.1 mm) guard cartridge (Sigma-Aldrich). Mobile phase was a gradient of solvent A (methanol/water/n-hexane/32% ammonium hydroxide, 87.5:10.5:1.5:0.5, v/v/v/v) and solvent B (methanol/n-hexane/32% ammonium hydroxide, 87.5:12:0.5, v/v/v). The gradient was started at 100% solvent A; it was decreased linearly to 65% solvent A and 35% solvent B in 20 min, to 10% solvent A and 90% solvent B in 5 min, and to 0% solvent A and 100% solvent B in an additional 5 min. The flow rate was 0.5 ml/min and 80 μl of the lipid extract was injected. The LC system was coupled online to a Bruker esquire6000 ion-trap mass spectrometer (Bruker Daltonics, Bremen, Germany). The total flow rate into the column was split and 0.2 ml/min entered into the electrospray interface of the mass spectrometer. Nebulizer gas was set to 30 psi, dry gas to 7 l/min, and dry temperature to 325°C. Ethanolamine glycerophospholipid (PE), phosphatidylinositol (PI), and phosphatidylserine (PS) were detected in negative ion mode with the capillary current.
set at +3,500 V over the initial 25 min as \([\text{M} - \text{H}]^-\) ions. Choline glycerophospholipid (PC) species were detected over the elution interval from 25 to 35 min in positive ion mode, as \([\text{M} + \text{H}]^+\) ions, with the capillary current set at −3,500 V. The 16:1-containing PE, PI, and PS species were identified by comparison with previously published data (34–39). The cut off parameter was set at \(m/z\) 150 and fragmentation amplitude at 1 arbitrary unit. Because of the lability of vinyl ether linkages in acid media, plasmanyl (1-alkyl) and plasmenyl (1-alk-1′-enyl) glycerophospholipids were distinguished by acidifying the samples before lipid extraction. For the identification of acyl chains of 16:1-containing PC species, ionization was carried out in negative mode with post-column addition of acetic acid at a flow rate of 100 \(\mu\)l/h as \([\text{M} + \text{CH}_3\text{CO}_2]^-\) adducts, and acyl chains were identified by MS\(^3\) experiments (34–39).

**Real-time PCR**

Total RNA was extracted using Trizol reagent (Ambion). The cDNA templates were synthesized using Verso cDNA synthesis kit (Thermo Scientific), following the manufacturer’s instructions. Quantitative PCR (qPCR) was carried out with an ABI 7500 machine (Applied Biosystems, Carlsbad, CA) using Brilliant III Ultra-Fast SYBR Green qPCR Master Mix (Agilent Technologies, Santa Clara, CA), as previously described (40).

**Immunoblot analysis**

After the different treatments, the cells were lysed for 30 min in ice-cold buffer containing 20 mM Tris-HCl (pH 7.4), 150 mM NaCl, 0.5% Triton X-100, 100 mM Na\(_3\)VO\(_4\), 1 mM phenyl methyl sulfonyl fluoride, and protease inhibitor cocktail (Sigma). Total protein (50 \(\mu\)g) was resolved on 10–12% SDS-PAGE gels and transferred to a PVDF membrane. After transfer, nonspecific binding sites were blocked with 5% BSA in PBS containing 0.1% Tween-20 at room temperature for 1 h. The membranes were then probed with the corresponding antibodies followed by HRP-conjugated secondary antibodies in blocking solution. β-Actin was used as a load control. The immunoblots were visualized using enhanced luminescence. Densitometry was performed on scanned images using Quantity One\textsuperscript{®} software (Bio-Rad Laboratories) and values were normalized for the corresponding controls of each experiment.

**Statistical analysis**

All experiments were carried out at least three times with incubations in duplicate or triplicate, and the data are expressed as mean ± SE. Statistical analysis was carried out by Student’s \(t\)-test, with \(P < 0.05\) taken as statistically significant.

**RESULTS**

We have previously shown that human monocytes and macrophages contain significant amounts of 16:1n-9, an unusual isomer of 16:1n-7 (14). We began the current study by investigating the possible occurrence of other unidentified 16:1 isomers in human phagocytic cells. For these experiments, we prepared fatty acid methyl esters of cellular fatty acids, which were then converted into DMDS derivatives. DMDS derivatization provides unambiguous analytical resolution of positional isomers because methyl sulfide substituents add to the two carbon atoms of the double bond, thus preventing isomerization (41). Figure 1 shows a GC/MS analysis of a mixture of four commercial 16:1 isomers, namely 16:1n-10, 16:1n-9, 16:1n-7, and palmitovaccenic acid (16:1n-5). All four isomers resolved well in GC and each of them produced specific fragments with electron-impact ionization MS, because fragmentation occurred at the level of the C–C bond, where the sulfide groups are attached. In addition, a third fragment was
detected, which corresponded to the loss of 32 mass units (methanol) from the fragments containing the methyl ester (Fig. 1). Collectively, these data indicate that analysis of fatty acyl DMDS derivatives by GC (retention time) coupled to electron impact MS (detection of diagnostic peaks) allows unambiguous identification of the various isomers.

Application of this technique to analyze the occurrence of 16:1 isomers in human peripheral blood monocytes confirmed the predominant presence of 16:1n-7 and 16:1n-9, in agreement with our previous estimates (Fig. 2) (14). Interestingly, significant amounts of 16:1n-10 could also be detected, albeit at lower amounts than those of the other isomers (Fig. 2A). Remarkably, the analysis of human monocyte-derived macrophages showed a different pattern of expression; while 16:1n-7 and 16:1n-9 were readily detected, the levels of 16:1n-10 were much lower in comparison. Traces of 16:1n-5 were also detected in macrophages (Fig. 2B). In human serum, 16:1n-7 was, by far, the major isomer detected. Lower amounts of 16:1n-9 and even lower amounts of 16:1n-10 were also detected. No 16:1n-5 was found in serum (Fig. 2C). Collectively, these results suggest that production of 16:1n-10 by human cells could be cell-type specific, being detectable at significant amounts only in monocytes. On the other hand, because 16:1n-5 was found only in macrophages and at trace levels, this isomer was not considered further.

The occurrence of 16:1 isomers was also studied in monocytes activated with different stimuli. In the first place, we used arachidonic acid, which can be secreted by damaged endothelium and possesses monocyte-activating properties (42). We have recently shown that human monocytes exposed to this fatty acid (10 μM, 2 h) acquire a foamy phenotype, which is due to the accumulation of cytoplasmic lipid droplets (14, 16). Lipid loading of the monocytes under these conditions is due to activation of

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Fig. 6. Fatty acid composition of human and murine phagocytic cells. The total fatty acid profile cells were determined by GC/MS after converting the fatty acid glyceryl esters into fatty acid methyl esters. Approximately 10^7 cells were utilized for these determinations. Results are shown as the mean ± SE of three independent experiments with incubations in duplicate.
the de novo pathway of lipid biosynthesis by arachidonic acid and ultimately results in the accumulation of 16:1\textit{n-9} and 16:1\textit{n-7} in various lipid fractions (14, 16). In keeping with our previous observations, monocytes activated by arachidonic acid demonstrated a time-dependent increase in the levels of both 16:1\textit{n-9} and 16:1\textit{n-7} (Fig. 3A). In contrast, the cellular levels of 16:1\textit{n-10} did not significantly change. Other monocyte stimulants, such as bacterial lipopolysaccharide (LPS) (Fig. 3B), yeast-derived zymosan (Fig. 3C), or the Ca\textsuperscript{2+} ionophore, A23187 (Fig. 3D), all failed to increase the levels of any of the three 16:1 isomers. Thus, while there are activation conditions that result in upregulation of 16:1\textit{n-9} and 16:1\textit{n-7} levels (i.e., those leading to the formation of lipid-laden monocytes), 16:1\textit{n-10} levels appear not to be regulated upon any state of activation of the cells. Note as well that differentiation of monocytes to macrophages also did not upregulate 16:1\textit{n-10} levels (compare Fig. 2A and Fig. 2B).

Importantly, 16:1\textit{n-10} was found at quite significant levels in cells of murine origin, such as resident peritoneal macrophages (Fig. 4A). Moreover, when we analyzed RAW264.7 cells, a murine cell line that is widely used as a paradigm for macrophages (43), 16:1\textit{n-10} was detected at levels comparable to those of 16:1\textit{n-9} and much higher than those of 16:1\textit{n-7} (Fig. 4B). High levels of 16:1\textit{n-10} were also observed in another murine-macrophage-like cell line, P388D1 (Fig. 4C). When the study was extended to a human cell line, THP-1, 16:1\textit{n-10} was also found at high levels (Fig. 4D).

The distribution of 16:1 fatty acids among phospholipids of RAW264.7 cells was measured by LC/MS. The 16:1-containing phospholipids were identified by determining the formation of a \textit{m/z} 253 fragment in MS\textsuperscript{n} experiments, corresponding to a 16:1 acyl chain. Structural identification of the glycerophospholipids was achieved by looking at the fragments and/or neutral losses obtained in MS\textsuperscript{n} experiments for PE, PI, and PS, or MS\textsuperscript{3} experiments for PC (34–39). Fatty chains within phospholipids are designated by their number of carbons:double bonds. A designation of O—before the first fatty chain indicates that the \textit{sN-1} position is ether-linked, while a P—designation indicates a plasmalogen form (\textit{sN-1} vinyl ether linkage) (44).

A total of 15 glycerophospholipid species were found to contain 16:1 acyl chains, with PC species being, by far, the most enriched in these fatty acids. Remarkably, a single PC species, PC(16:0/16:1), comprised almost half of total cellular 16:1. By class, PC contained \(\sim 70\%\) of total 16:1, while crude lipids were transmethylated and fatty acid methyl esters were measured by GC/MS. D: Expression of the FADS2 gene in different cell types. Results are shown as the mean \(\pm\) SE of three independent experiments with incubations in duplicate (A, C) or triplicate (B, D).
incubation of the cells with the selective Δ⁶ desaturase inhibitor, SC-26196 (46, 47), strongly inhibited 16:1n-10 levels, while leaving 16:1n-9 and 16:1n-7 levels unchanged (Fig. 7A).

Because FADS2 also converts linoleic into γ-linolenic acid within the n-6 fatty acid synthesis pathway, we reasoned that high levels of 16:1n-10 in cultured cells could reflect, at least in part, an increased access of Δ⁶ desaturase to palmitic acid due to the relative scarcity of linoleic acid. In keeping with this observation, we noted that the palmitic acid (16:0) to linoleic acid (18:2n-6) molar ratio in cultured cell lines was several fold higher than that of primary cells (Fig. 7B). Further, long-term incubation of the cells with exogenous linoleic acid led to a significant nanomolar reduction of 16:1n-10 levels, which corresponded with a similar nanomolar increase of n-6 products in cells (Fig. 7C). Importantly, analyses of FADS2 expression by qPCR clearly demonstrated higher expression levels of the gene in cultured cell lines with respect to primary cells (Fig. 7D). Thus, high expression levels of FADS2, together with higher 16:0/18:2n-6 ratios (i.e., low 18:2n-6 levels), would explain why cell lines produce more 16:1n-10 than primary cells. Thus, increased 16:1n-10 levels in cultured cells are not only a consequence of favored desaturation of palmitic acid over linoleic acid due to reduced levels of the latter, but also to increased expression of the Δ⁶ desaturase.

We have recently shown that both 16:1n-7 and 16:1n-9 possess potent anti-inflammatory activity both in vivo and in vitro (14). To extend these results to 16:1n-10, we prepared cells enriched in the various isomers by incubating them with a low concentration (10 μM) of the corresponding fatty acid for 14 h in serum-free medium (14). This procedure results in the cells taking up the fatty acids and accumulating them in various cellular lipids, mainly phospholipids (Fig. 8A). In keeping with our previous data (14), subtle differences were found between 16:1n-7 and 16:1n-9 with regard to their incorporation into cellular lipid classes; the latter manifesting a greater tendency to accumulate into neutral lipids than the former. Significant amounts of 16:1n-10 were also found in neutral lipids (Fig. 8A). Further analyses of 16:1 incorporation into phospholipid classes revealed that, in keeping with the distribution of the endogenous fatty acids, most of the exogenous fatty acids incorporated into PC, with lesser amounts being found in PE. Exogenous 16:1n-7 was found to be a little more enriched in PE than in 16:1n-10 or 16:1n-9; the relative distribution of the latter two being very similar (Fig. 8B).

After the incubations with the fatty acids, the cells were stimulated with bacterial LPS and the effect on the expression of a number of pro-inflammatory genes was investigated. In agreement with our previous data (14), cells enriched with 10 μM 16:1n-7 or 16:1n-9 showed significant inhibitory effects on the expression level of a number of inflammatory genes. Remarkably, these effects were not observed in the 16:1n-10-enriched cells at this concentration (Fig. 9A–C); significantly higher concentrations of 16:1n-10 (>25 μM) were necessary to observe a similar anti-inflammatory effect, as shown in dose-response studies (Fig. 9D). At doses at which 16:1n-10 was ineffective (<25 μM), it did not alter the anti-inflammatory effect of 16:1n-9 (Fig. 9E). These results show that, although all 16:1 isomers present in macrophages may manifest anti-inflammatory activity, there is a marked difference between 16:1n-7 and 16:1n-9 on the one hand, and 16:1n-10 on the other, with regard to the range of concentrations at which an effect is demonstrated. Interestingly, when yeast-derived zymosan was used instead of LPS to stimulate the cells, none of the fatty acids exerted significant effects on pro-inflammatory gene expression (Fig. 9F). Thus the anti-inflammatory effects of 16:1 fatty acids may be specific for LPS stimulatory conditions, suggesting an interference with signals arising from TLR4 engagement. In this regard, a hallmark of LPS activation of macrophages is the rapid phosphorylation activation of the extracellularly regulated mitogen-activated protein kinases p42 and p44 (48). Figure 10 shows that 10 μM 16:1n-9 blunted the

![Figure 10](image-url)
LPS-induced phosphorylation of the two kinases in a time-dependent manner, while 16:1n-10 was ineffective.

DISCUSSION

In this study, we show that human and murine phagocytic cells contain, in addition to 16:1n-7 and 16:1n-9, a third 16:1 isomer, namely 16:1n-10. Originally described in human sebum and later in hairs and nails, 16:1n-10 was commonly thought not to be present anywhere else in the human body (15, 49, 50). However, 16:1n-10 has recently been identified in erythrocyte membranes (41). Thus, our identification in circulating human monocytes was certainly no surprise, although the low levels at which the fatty acid presents in monocytes and macrophages make it difficult to envision a biological role for it in innate immunity. It was more unexpected, however, to identify 16:1n-10 at relatively high levels in mouse peritoneal macrophages and, especially, cultured macrophage cell lines, because this fatty acid is conventionally considered to be unique to humans (hence its trivial name, sapienic acid, deriving from the root sapiens).

While the double bond present in 16:1n-7 and 16:1n-9 is introduced by a Δ⁹ desaturase enzyme (stearoyl-CoA desaturase), 16:1n-10 is produced by the action of a different enzyme, Δ⁶ desaturase. Our results indicate that, consistent with these different metabolic origins, cellular regulation of 16:1n-10 levels differs from that of 16:1n-7 and 16:1n-9 in several key respects. The levels of the latter two fatty acids may be regulated during lipid loading conditions, owing to stimulus-induced activation of de novo fatty acid biosynthesis (14, 16, 51, 52). In contrast, the levels of 16:1n-10 appear not to depend on the activation state of the cells, but rather on complex, possibly interacting, mechanisms involving both substrate competition reactions and increased expression of FADS2.

It has been long thought that desaturation at carbon 6 is greatly favored when the fatty acid substrate already contains a double bond at position 9, as is the case of linoleic acid (53). Accordingly, in cells, desaturation of linoleic acid to γ-linolenic acid would be preferred over desaturation of palmitic acid to 16:1n-10, which would easily explain why cells containing high endogenous levels of linoleic acid produce 16:1n-10 at very low amounts and vice versa. While this idea is fully consistent with our results using human phagocytic cells and cultured cell lines, it is worth noting that mouse peritoneal macrophages, which contain large amounts of linoleic acid and, in general, of n-6 fatty acids, amounting to approximately 25% of total cellular fatty acids (54, 55), also contain relatively high levels of 16:1n-10. Moreover, our studies using macrophage cell lines, which contain low levels of linoleic acid, show much greater expression of FADS2 relative to primary cells, suggesting that not only the palmitic acid to linoleic acid molar ratio but also the amount of enzyme present in the cell constitute another key factor regulating 16:1n-10 levels. Experiments are currently in progress to explore questions such as whether FADS2 operates constitutively at different efficiency depending on cell type or whether there are
cell-specific cofactors that may stimulate FADS2’s preference for palmitic acid utilization. It is also possible that, depending upon cell type, linoleic acid is diverted to other reactions, i.e., serving as substrate of lipoxygenases for oxylipin formation, thus enabling the enzyme to use palmitic acid to a larger extent.

A striking feature of this work is that, unlike 16:1 n-7 and 16:1 n-9, 16:1 n-10 is devoid of anti-inflammatory activity when used at low concentrations (up to 25 μM), as judged by its inability to prevent the increased expression of pro-inflammatory cytokines by LPS-stimulated macrophages. Higher concentrations of 16:1 n-10 are required to observe an effect. It is worth mentioning, in this context, that high concentrations of 16:1 n-10 (100 μM and above) have been demonstrated to possess antimicrobial activity in skin by a mechanism involving partitioning of the fatty acid into the cell membrane (56, 57). While a change in cellular lipid composition due to the incorporation of the various 16:1 isomers may itself have an effect on receptor-mediated responses, our initial studies on the mechanism of regulation of LPS signaling by 16:1 n-9 show a selectively reduced phosphorylation of the extracellular signal-regulated kinases p44 and p42 by LPS in the presence of the fatty acid, thus suggesting genuine interaction of the fatty acid with receptor-activated signaling. It is also striking that 16:1 fatty acids blunt LPS-mediated signals only and not those from other innate immune receptors, such as TLR2, dectin-1, or mannose receptors, which recognize yeast-derived zymosan (58), emphasizing selective receptor-mediated effects. This situation is reminiscent of recent work by Snodgrass et al. (59) on the inhibitory effect of docosahexaenoic acid, an n-3 fatty acid, on TLR-mediated responses in monocytes. In this study, the fatty acid was found to impair receptor dimerization, blunting subsequent signaling in this way (59). In our previous studies, we showed that docosahexaenoic acid is as effective as 16:1 fatty acids in inhibiting LPS-mediated gene expression responses (14).

Our results also raise provocative questions as to the cellular utilization of the various lipid pools that may function as reservoirs for anti-inflammatory 16:1 fatty acids. In preliminary work, we noted that the anti-inflammatory effects of both 16:1 n-7 and 16:1 n-9 can be abrogated if the incorporation of the fatty acids into cellular lipids is prevented by the presence of the general acyl-CoA synthetase inhibitor, triacsin C (C. Meana and J. Balsinde, unpublished observations), suggesting that formation of an ester of the corresponding 16:1 fatty acid is necessary for biological activity. Hence, the initial lipid localization where the fatty acid is esterified after incubating it with the cells may determine its subsequent biological activity. It is remarkable, in this regard, that 70–80% of total 16:1 in untreated cells is found in PC and that almost half of it is present in just a single phospholipid species, namely PC(16:0/16:1). These observations become even more relevant in the light of the data showing that, regardless of isomer, the bulk of exogenous fatty acid added to the cells gets incorporated into PC. Given that recent work has demonstrated that certain PC species with defined fatty acyl ester composition have biological activity on their own (60, 61), it will be of interest to determine whether pure phospholipid species containing either 16:1 n-7, 16:1 n-9, or 16:1 n-10 as one of the acyl substituents are also biologically active. While it is clear that much work needs to be carried out to unravel the effects of 16:1 fatty acids on innate immune reactions, the elucidation of intracellular metabolic routes utilizing these fatty acids might provide interesting opportunities of intervention to ameliorate the inflammatory response.
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REFERENCES

1. Cao, H., K. Gerhold, J. R. Mayers, M. M. Wiest, S. M. Watkins, and G. S. Hotamisligil. 2008. Identification of a lipokine, a lipid hormone linking adipose tissue to systemic metabolism. Cell. 134: 933–944.

2. Erbay, E., V. R. Babaev, J. R. Mayers, L. Makowski, K. N. Charles, M. E. Snitow, S. M. Wiest, S. M. Watkins, M. F. Linton, et al. 2009. Reducing endoplasmic reticulum stress through a macrophage lipid chaperone alleviates atherosclerosis. Nat. Med. 15: 1383–1391.

3. Schwingshackl, L., and G. Hoffmann. 2012. Monounsaturated fatty acids and risk of cardiovascular disease: synopsis of the evidence available from systematic reviews and meta-analyses. Nutrients. 4: 1899–2007.

4. Çimen, I., B. Kocaturk, S. Kovuncu, O. Tufanli, U. I. Onat, A. D. Yildirim, O. Apaydın, S. Demirsoy, Z. G. Aykut, U. T. Nguyen, et al. 2016. Prevention of atherosclerosis by bioactive palmitoleate through suppression of ornithine synthase and lipopolysaccharide activation. Sci. Transl. Med. 8: 358ra126.

5. Tsuchiya, H., H. Hatakeyama, N. Emoto, F. Wagatsuma, S. Matsushita, and M. Kanzaki. 2010. Palmitoleate-induced down-regulation of sorbin and impaired GLUT4 trafficking in C2C12 myotubes. J. Biol. Chem. 285: 34371–34381.

6. Akazawa, Y., S. Cazanave, J. L. Mott, N. Elmi, S. F. Bronk, S. Kohno, M. R. Charlton, and G. J. Gores. 2010. Palmitoleate attenuates palmitate-induced RIM and PUMA up-regulation and heptocyte lipoprotein-apo-poptosis. J. Hepatol. 52: 586–593.

7. Yang, Z. H., C. Deng, and A. Hatanaka. 2011. Chronic administration of palmitoleic acid reduces insulin resistance and hepatic lipid accumulation in KK-Ay mice with genetic type 2 diabetes. Lipids Health Dis. 10: 120.

8. Hodson, L., and F. Karpe. 2013. Is there something special about palmitoleate? Curr. Opin. Clin. Nutr. Metab. Care. 16: 225–231.

9. De Fabiani, E. 2011. The true story of palmitoleic acid: between myth and reality. Eur. J. Lipid Sci. Technol. 113: 809–811.

10. Mozaffarian, D., H. Cao, I. B. King, R. N. Lemaire, X. Song, D. S. Siscovick, and G. S. Hotamisligil. 2010. Circulating palmitoleic acid and risk of metabolic abnormalities and new-onset diabetes. Am. J. Clin. Nutr. 92: 1350–1358.

11. Fabbriini, E., F. Magkos, X. Xu, N. A. Abumrad, N. Nejedly, C. C. Cougill, A. L. Okunade, B. W. Patterson, and S. Klein. 2011. Insulin sensitivity is not associated with palmitoleate availability in obese humans. J. Lipid Res. 52: 808–812.

12. Gong, J. H., C. E. Chambers, S. McGarvey, Z. Wu, R. Goldberg, and A. Baylin. 2011. Adipose tissue palmitoleic acid and obesity in humans: does it behave as a lipokine? Am. J. Clin. Nutr. 93: 186–191.

13. Perreault, M., K. Roke, A. Badawi, D. E. Nielsen, S. A. Abdelmagid, A. El-Sohemy, D. W. L. Ma, and D. M. Mutch. 2014. Plasma levels of 14:0, 16:0, 16:1n-7, and 20:3n-6 are positively associated, but 18:0 acid (16:1n-9), a possible biomarker for early detection of cardio-vascular disease. J. Nutr. Biochem. 25: 252–263.

14. Guijas, C., C. Meana, A. M. Astudillo, M. A. Balboa, and J. Balsinde. 2016. Foamy monocytes are enriched in cis-7-hexadecenoic fatty acid (16:1n-9), a possible biomarker for early detection of cardiovascular disease. Cell Chem. Biol. 23: 689–699.

15. Ge, L., J. S. Gordon, C. Hsuan, K. Stenn, and S. M. Prouty. 2003. Identification of the delta-6-desaturase of human sebaceous glands: expression and enzyme activity. J. Invest. Dermatol. 120: 707–714.

16. Guijas, C., G. Pérez-Chacón, A. M. Astudillo, J. M. Rubio, L. Gil-de-Gómez, M. A. Balboa, and J. Balsinde. 2012. Simultaneous activation of p38 and JNK by arachidonic acid stimulates the cytosolic phospholipase A2-mediated oleic acid mobilization in RAW 264.7 macrophages: comparison with arachidonic acid. Biochim. Biophys. Acta. 1821: 1386–1393.

17. Pérez-Chacón, G., A. M. Astudillo, V. Ruíz-Pérez, M. A. Balboa, and J. Balsinde. 2010. Signaling role for lysophosphatidylcholine acyltransferase 3 in receptor-regulated arachidonic acid reacylation reactions in human monocytes. J. Immunol. 184: 1071–1078.

18. Balsinde, J., B. Fernández, and E. Diez. 1990. Regulation of arachidonic acid release in mouse peritoneal macrophages. The role of extracellular calcium and protein kinase C. J. Immunol. 144: 4298–4304.
40. Valdearcos, M., E. Esquinas, C. Meana, L. Peña, L. Gil-de-Gómez, J. M. Rubio, J. P. Rodríguez, L. Gil-de-Gómez, C. Guijas, M. A. Ferreri, C., A. Masi, A. Sansone, G. Giacometti, A. V. Larocca, G. Dong, C., R. J. Davis, and R. A. Flavell. 2002. MAP kinases in the immunological response. J. Immunol. 169: 3927–3935.

41. Østerud, B., and E. Bjørklid. 2003. Role of monocytes in atherogenesis. J. Immunol. 171: 567–575.

42. Norris, P. C., D. Reichart, D. S. Dumlao, C. K. Glass, and E. A. Dennis. 2011. Specificity of eicosanoid production depends on the TLR4-stimulated macrophage phenotype. J. Leukoc. Biol. 89: 6026–6042.

43. Astudillo, A. M., D. Balgoma, M. A. Balboa, and J. Balsinde. 2012. Dynamics of arachidonic acid mobilization by inflammatory cells. Biochim. Biophys. Acta. 1821: 249–256.

44. Harmon, S. D., T. L. Kaduce, T. D. Manuel, and A. A. Spector. 2003. Effect of the Δ6 desaturase inhibitor SC-26196 on PUFA metabolism in human cells. Lipids. 38: 469–476.

45. Obukowicz, M. G., D. J. Welsch, W. J. Salsgiver, C. L. Martin-Berger, K. S. Chinn, K. L. Duffin, A. Raz, and P. Needleman. 1998. Novel, selective Δ9 or Δ6 fatty acid desaturase inhibitors as anti-inflammatory agents in mice. J. Pharmacol. Exp. Ther. 287: 157–166.

46. Dong, C., R. J. Davis, and R. A. Flavell. 2002. MAP kinases in the immune response. Annu. Rev. Immunol. 20: 55–72.

47. Nicolaides, N. 1974. Skin lipids: their biochemical uniqueness. Science. 186: 19–26.

48. Destaillats, F., M. Guitard, and C. Cruz-Hernández. 2011. Identification of Δ6-monounsaturated fatty acids in human hair and nail samples by gas-chromatography–mass-spectrometry using ionic-liquid coated capillary column. J. Chromatogr. A. 1218: 9384–9389.

49. Gubern, A., J. Casas, M. Barceló, D. Barneda, X. de la Rosa, R. Masgrau, F. Picatoste, J. Balsinde, M. A. Balboa, and E. Claro. 2008. Group IVA phospholipase A2 is necessary for the biogenesis of lipid droplets. J. Biol. Chem. 283: 27360–27362.

50. Gujias, C., J. P. Rodríguez, J. M. Rubio, M. A. Balboa, and J. Balsinde. 2014. Phospholipase A2 regulation of lipid droplet formation. Biochim. Biophys. Acta. 1841: 1661–1671.

51. Brenner, R. R. 1971. The desaturation step in the animal biosynthesis of polyunsaturated fatty acids. Lipids. 6: 567–575.

52. Casas, J., M. A. Gijón, A. G. Vigo, M. S. Crespo, J. Balsinde, and M. A. Balboa. 2006. Phosphatidylinositol 4,5-bisphosphate anchors cytosolic group IVA phospholipase A2 to perinuclear membranes and decreases its calcium requirement for translocation in live cells. Mol. Biol. Cell. 17: 155–162.

53. Rouzer, C. A., P. T. Ivanova, M. O. Byrne, H. A. Brown, and L. J. Marnett. 2007. Lipid profiling reveals glycerophospholipid remodeling in zymosan-stimulated macrophages. Biochemistry. 46: 6026–6042.

54. Wille, J. J., and A. Kydoneus. 2003. Palmitoleic acid isomer in human skin sebum is effective against gram-positive bacteria. Skin Pharmacol. Appl. Skin Physiol. 16: 176–187.

55. Drake, D. R., K. A. Brogden, D. V. Dawson, and P. W. Wertz. 2008. Skin lipids. Antimicrobial lipids at the skin surface. J. Lipid Res. 49: 4–11.

56. Underhill, D. M. 2003. Macrophage recognition of zymosan particles. J. Endotoxin Res. 9: 176–180.

57. Snoongrass, R. G., S. Huang, I. W. Choi, J. C. Rutledge, and D. H. Hwang. 2013. Inflammamsonemediated secretion of IL-1β in human monocytes through TLR2 activation; modulation by dietary fatty acids. J. Immunol. 191: 4337–4347.

58. Chakravarthy, M. V., I. J. Lodhi, L. Yin, R. R. Malapaka, H. E. Xu, J. Turk, and C. F. Semenkovich. 2009. Identification of a physiologically relevant endogenous ligand for PPARs in liver. Cell. 138: 476–488.

59. Lee, J. M., Y. K. Lee, J. L. Mamrosh, S. A. Busby, P. R. Griffin, M. C. Pathak, E. A. Ortlund, and D. D. Moore. 2011. A nuclear-receptor-dependent phosphatidylycholine pathway with antidiabetic effects. Nature. 474: 506–510.