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Redirection of lipid flux toward phospholipids in yeast increases fatty acid turnover and secretion

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Bio-based production of fatty acids and fatty acid-derived products can enable sustainable substitution of petroleum-derived fuels and chemicals. However, developing new microbial cell factories for producing high levels of fatty acids requires extensive engineering of lipid metabolism, a complex and tightly regulated metabolic network. Here we generated a Saccharomyces cerevisiae platform strain with a simplified lipid metabolism network with high-level production of free fatty acids (FFAs) due to redirected fatty acid metabolism and reduced feedback regulation. Deletion of the main fatty acid activation genes (the first step in β-oxidation), main storage lipid formation genes, and phosphatidate phosphatase genes resulted in a constrained lipid metabolic network in which fatty acid flux was directed to a large extent toward phospholipids. This resulted in simultaneous increases of phospholipids by up to 2.8-fold and of FFAs by up to 40-fold compared with wild-type levels. Further deletion of phospholipase genes PLA1 and PLA2 resulted in a 46% decrease in FFA levels and 105% increase in phospholipid levels, suggesting that phospholipid hydrolysis plays an important role in FFA production when phospholipid levels are increased. The multiple deletion mutant generated allowed for a study of fatty acid dynamics in lipid metabolism and represents a platform strain with interesting properties that provide insight into the future development of lipid-related cell factories.

phospholipids | free fatty acids | Saccharomyces cerevisiae | metabolic engineering | CRISPR

Free fatty acids (FFAs) are versatile chemicals that can readily be converted into a broad range of attractive industrial compounds. FFAs are traditionally extracted from plant oils; however, current engineering efforts aimed at creating a more sustainable process are focusing on the development of microbial cell factories capable of efficiently converting sugars into FFAs (1). With a strong resistance to fermentation inhibitors and an ability to survive at low pH levels, Saccharomyces cerevisiae is the preferred cell factory for industrial production of biochemicals (2, 3). It is an extensively well-studied organism with many available genetic tools, such as the CRISPR/Cas9 technology for fast and robust genome editing and pathway interference (4–7). Although the capabilities of S. cerevisiae for fatty acid synthesis are limited by strong intrinsic regulations, several attempts have been made to produce FFAs using this organism as a host (8). This increases the importance of understanding the dynamics of lipid metabolism surrounding FFA biosynthesis.

In S. cerevisiae, fatty acids are synthesized by the fatty acid synthetase complex encoded by FAS1 and FAS2, in which acetyl-CoA and malonyl-CoA are iteratively processed to produce acyl-CoA. Acyl-CoA formation is tightly regulated through allosteric feedback inhibition of the acetyl-CoA carboxylase Acc1, responsible for malonyl-CoA production (9, 10). Acyl-CoAs from fatty acid biosynthesis are esterified with a hydroxyl-containing molecule (e.g., glycerol or sterol) to form major lipid classes, such as triacylglycerols (TAGs), sterol esters (SEs), or phospholipids. These three lipid classes represent the major pool of fatty acyl chains in S. cerevisiae, accounting for most of the total fatty acid-containing lipids in the cell, while other lipid classes are detected only at very low levels (11, 12). In a wild-type cell, FFAs are directly converted to acyl-CoA by fatty acyl-CoA synthetases (13), a process that maintains low FFA levels in the cytoplasm. Previous engineering strategies have shown that deletion of the two dominant acyl-CoA synthetase genes, FAA1 and FAA4, results in deregulation of fatty acid biosynthesis and overaccumulation of FFAs by orders of magnitude above that of a wild-type strain (14–16). Characterization of these strains has indicated that the FFAs originate from complex lipids (14), and as such, are potentially a mere intermediary in lipid hydrolysis and remodeling processes. This effect has been further explored by combining the Δfaa1 Δfaa4 genotype with overexpression of the pathway for accumulation and hydrolysis of TAGs (17).

While the foregoing mechanisms have been described and consistently applied in FFA production, to date no studies have properly explored the dynamics of FFA production in the resulting strains in terms of the specific pathways and lipid species involved in FFA formation. Furthermore, during the development of fatty acid-overproducing cell factories, combinatorial effects of modifications regarding fatty acid regulation might come into play, which often are not being properly described or understood in the resulting strains.

Here we developed several strains to study FFA biosynthesis by constraining the lipid metabolic network toward its essential components, i.e., fatty acid and membrane lipid biosynthesis. Analysis of the resulting strains allowed us to understand the role of different lipid pools in FFA formation, as well as the effects of up-regulating phospholipid biosynthesis and the existing relationship between phospholipid and FFA levels in the cell.

Significance

Replacement of nonrenewable petrochemicals and liquid fuels requires sustainable production of oleochemicals. Free fatty acids (FFAs) are versatile molecules that can be produced by microbial fermentation and are used as precursors for production of these oleochemicals. In the past few years, we have seen major advancements in improving the yeast Saccharomyces cerevisiae for FFA production. Despite these successes, lipid metabolism is highly complex, and the pathways and metabolites involved in the formation of FFAs in yeast remain incompletely understood. In this work, we make important advancements in understanding the dynamics of FFA formation in the cell and explore the role of phospholipids in this process.

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The authors declare no conflict of interest.

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Results
Deletion of Main Acyl-CoA Synthetases and Storage-Lipid Formation Pathways Reveals That FFA Formation Is Independent of Storage Lipids. To construct a strain for studying fatty acid dynamics and fluxes, we removed the two dominant fatty acyl-CoA synthetase-encoding genes, FAA1 and FAA4 (Fig. 1), which has been previously shown to be sufficient to abolish FFA reactivation, disrupt the FFA feedback regulation loop, and deregulate fatty acid biosynthesis (13, 18). Removal of these two genes resulted in the RP01 strain that produced 35 mg·gDCW⁻¹ of FFAs in batch culture (Fig. 2), more than 10-fold compared with the control strain. To prevent FFA and acyl-CoA degradation and provide a more constrained lipid metabolism network, we deleted the fatty acyl-CoA oxidase encoded by POX1, which catalyzes the first step of the β-oxidation pathway located in the peroxisomes and is nonessential for growth on glucose (19, 20). POX1 deletion from the RP01 strain, resulting in progressive strain RP02, led to a 58% increase in produced FFAs, to 53 mg·gDCW⁻¹ (Fig. 2).

To investigate the role of neutral lipids in FFA formation, we removed fatty acid accumulation in the form of storage lipids by deleting the two main genes responsible for TAG formation—DGA1, encoding diacylglycerol acyltransferase, and LRO1, encoding phospholipid:diacylglycerol acyltransferase—and the two main genes responsible for sterol-ester formation—ARE1 and ARE2, encoding acyl-CoA:sterol acyltransferases (Fig. 1). Progressive deletion of the major genes responsible for storage lipid formation resulted in the strain RP09 (Δfaa1 Δfaa4 Δpox1 Δdga1 Δtro1 Δare1 Δare2), which did not show significant differences in FFA production levels compared with RP02 (53 mg·gDCW⁻¹ and 57 mg·gDCW⁻¹, respectively; P > 0.05).

These results suggest that even though storage lipids may have a role in FFA formation, these are not key intermediaries, since RP09 is devoid of TAGs and SES (Fig. S1) and does not present a major deficiency in FFA formation compared with RP02 (Fig. 2). Even though previous studies have shown that FFAs can effectively produced using storage lipids as an intermediary (17), our results show that there is an alternative native pathway capable of carrying high fluxes of FFA formation that does not rely on storage lipid synthesis.

Deletion of Phosphatidate Phosphatases Increases the Levels of FFAs and Phospholipids. Use of the strain RP09 allowed for further study of the fatty acid dynamics and pathways involved in FFA formation, since it is devoid of neutral lipids, and thus these could be excluded from the fatty acid pools existing in the cell. According to previous studies, the removal of TAG formation through DGA1 and LRO1 deletion leads to an accumulation of diacylglycerols (DAGs) (21). DAGs are formed mainly through dephosphorylation of phosphatidic acid (PA), which is an essential precursor for formation of other phospholipid species. PA has also been characterized as an important signaling molecule for regulation of lipid metabolism. High levels of PA lead to reduced translocation of the transcriptional regulator Op1 to the nucleus (22), preventing its binding to the transcription factor Ino2. Since Ino2 is an activator of many fatty acid and phospholipid biosynthesis genes, an increase in PA levels indirectly causes an up-regulation of the fatty acid biosynthesis machinery (Fig. 3B) (23–25).

To evaluate the effects of up-regulating fatty acid and lipid biosynthesis in this strain and further constrain the network of fatty acid fluxes, we aimed to remove dephosphorylation of PA to DAG (Fig. 3A), which would allow for PA accumulation and therefore interfere with Op1-mediated regulation. PA dephosphorylation is catalyzed by phosphatidate phosphatases encoded mainly by PAH1, LPP1, and DPP1, and as such, these genes were deleted from RP09, resulting in the strain MLM1.0. Deletion of PAH1, LPP1 and DPP1 led to an increase in total FFAs to 102 mg·gDCW⁻¹, a 98% increase compared with the previous strain RP09 (Fig. 3 C and E). While levels of both saturated and unsaturated FFAs were increased in MLM1.0, analysis of the FFA composition showed that the percentage of unsaturated (C16:1 and C18:1) compared with saturated (C16:0 and C18:0) FFAs decreased from 62% in RP09 to 56% in MLM1.0. Removal of the phosphatidate phosphatases resulted in exclusive use of PA for phospholipid biosynthesis (Fig. 3A) by removing the conversion of PA to DAG. As a result of this flux constraint and the deregulation through PA signaling, phospholipid levels were increased to 30 mg·gDCW⁻¹, an

![Fig. 1. Simplified schematic representation of key lipid metabolism fluxes and genes in S. cerevisiae. Highlighted in red are genes targeted for deletion in the RP09 strain: fatty acyl-CoA synthetases FAA1 and FAA4, fatty acyl-CoA oxidase POX1, diacylglycerol acyltransferase DGA1, phospholipid diacylglycerol acyltransferase LRO1, and acyl-CoA:sterol acyltransferases ARE1 and ARE2. The lipid species quantified experimentally are in dark blue. PA: phosphatidic acid; PC: phosphatidylcholine; PE: phosphatidylethanolamine; PI: phosphatidylinositol; PLP: phospholipids; PS: phosphatidylserine; SE: sterol esters; TAG: triacylglycerols.](image)

![Fig. 2. Effect of disrupting lipid droplet formation on total FFA levels. Total (intracellular and extracellular) FFAs were quantified for the different knockout strains lacking fatty acid reactivation (FAA1 and FAA4 deletions), β-oxidation (POX1 deletion), and storage lipid formation (DGA1, LRO1, ARE1, and ARE2 deletion). Strains were grown for 72 h in minimal medium containing 2% glucose, and FFAs were quantified from biological triplicates. **P < 0.005 (Student’s t test, two-tailed, two-sample equal variance). Differences between RP03-09 and RP02 were statistically nonsignificant (P > 0.05).](image)
eightfold increase compared with RP09 (Fig. 3D). The phospholipid fraction was composed mostly of phosphatidylcholine, which accounted for 54% of the total phospholipids, and a similar percentage of phosphatidylethanolamine (PE) and phosphatidylcholine (PC), which accounted for 22% and 21%, respectively. Phosphatidylinositol and cardiolipin were present only in low percentages, respectively 2% and 1% of total phospholipids (Fig. 3D and E).

Since deregulation of Opi1-controlled phospholipid biosynthesis genes has been previously associated with an expansion of the ER (28), we performed transmission electron microscopy (TEM) on the main strains, and observed formation of large membrane structures in MLM1.0 that were not observed in the RP02 or RP09 strains (Fig. S2). Furthermore, several stacked membrane sheets were observed in the enlarged ER structure. For strain MLM1.0, aggregates were observed in the medium supernatant after separation of cells by centrifugation. These aggregates could be stained by BODIPY and Nile Red, both fluorescent dyes specific toward lipids (Fig. S3). We speculated that accumulation of phospholipids disturbs membrane formation, ultimately leading to the formation of lipid aggregates in the extracellular medium; however, due to difficulties in isolating these aggregates, extensive characterization could not be carried out. In addition, while not detected in RP09, generation of the strain MLM1.0 resulted in a recurrence of SEs up to 11 mg gCDW⁻¹ in MLM1.0 (Fig. S1).
through deletion of the genes encoding the two major phospholipases, PLB1 and PLB2 (Fig. 4). This deletion resulted in a decreased level of the total FFA levels by 46%, to 55 mg gDCW⁻¹ (Fig. 4B). The percentage of unsaturated FFAs between MLM1.0 and MLM1.0 Δplb1 Δplb2 decreased from 62% to 40% (Fig. 4C), indicating that removal of PLB1 and PLB2 hindered the flux of unsaturated FFA formation more than saturated FFA formation. At the same time, these deletions led to an accumulation of total phospholipids up to 61 mg gDCW⁻¹, a twofold increase compared with MLM1.0 (Fig. 4D).

Phospholipid composition changed slightly on deletion of PLB1 and PLB2 from MLM1.0; phosphatidylcholine was reduced to 47%, while PI and PS increased to 26% and 4%, respectively. The PE percentage remained the same at 22% of total phospholipids (Fig. 4E). This might point toward a higher preference of Plb1 and Plb2 toward PI and PS hydrolysis. The simultaneous decrease in FFA levels and increase in phospholipid levels on deletion of the phospholipase genes PLB1 and PLB2 strongly indicates that a significant portion of FFAs in MLM1.0 is generated through phospholipid hydrolysis. Furthermore, previous studies have shown that phospholipids in S. cerevisiae mostly incorporate unsaturated fatty acids in both the sn-1 and sn-2 positions (11), and as such, the decrease in unsaturated FFA supports the hypothesis of FFA formation through phospholipid hydrolysis. Analysis of total fatty acid content in the cell suspension through derivatization of free and bound fatty acids shows similar values of 175 mg gDCW⁻¹ and 180 mg gDCW⁻¹ between MLM1.0 and MLM1.0 Δplb1 Δplb2, respectively, with similar distributions regarding chain length and saturation levels (Fig. S4). These similarities in fatty acid composition while the lipid class distribution is significantly changed (Fig. S1) highly suggest that the fatty acid synthesis machinery is not altered when PLB1 and PLB2 are deleted. Here we speculate that it is instead the conversion of phospholipids into other lipid classes, namely FFAs, that is affected. TEM of MLM1.0 Δplb1 Δplb2 did not show a major difference in accumulation of enlarged intracellular membrane structures; however, we observed that several cells showed a more compact and stacked morphology of these enlarged structures, as opposed to the single large-volume structures observed in MLM1.0 (Fig. S2). This is possibly due to changes in phospholipid composition, such as the change of ratio in saturated to unsaturated fatty acids, which might affect the capability of membrane bending and lead to the formation of particular structures (31, 32).

To investigate whereas Plb1 and Plb2 also play roles in FFA formation in a strain, in which storage lipids and lipid droplets are fully present, and the deregulation of Opi1-controlled phospholipid biosynthesis gene is not applied, we deleted PLB1 and PLB2 also in the RP02 strain and evaluated the levels of FFAs in that strain. Deletion of both genes resulted in 33 g gDCW⁻¹, a 20% decrease in FFA levels compared with RP02. This confirms the relevance of phospholipases B in FFA formation and phospholipids as a main precursor for generated FFAs (Fig. S5).

F AA Levels Are Not Affected by Phospholipase Overexpression. Because phospholipid accumulates in MLM1.0 even when high production of FFAs is observed, we wanted to further investigate if FFA formation from phospholipids was limited by phospholipase activity. As such, we individually overexpressed three different phospholipase B genes: PLB1, PLB2, and PLB3 (Fig. 3F). For this, we used a multicopy plasmid expressing the genes under the control of a strong constitutive TEF1 promoter. Surprisingly, overexpression of these phospholipases resulted in a maximum increase in total FFA production of only 16% for PLB2 (P values for PLB1, PLB2, and PLB3 > 0.05) (Fig. 5B). This result suggests that phospholipase activity might not be a major limiting step in FFA production. To further investigate if flux-limiting steps exist elsewhere downstream of acyl-CoA biosynthesis, we expressed a truncated version of the Escherichia coli thioesterase gene tesA (tes4). Thioesterase overexpression in this strain allows for FFA production directly through the hydrolysis of fatty acyl-CoA. This creates a pathway alternative to the phospholipid-mediated

![Graph](image)

**Fig. 4.** Effect of deleting phospholipase genes PLB1 and PLB2 on FFA production. (A) Role of phospholipases B in FFA formation in MLM1.0. Signaled in red are the genes PLB1 and PLB2 that were deleted to validate this hypothesis. (B) Total FFA quantification of MLM1.0 with PLB1 and PLB2 being deleted compared with strains MLM1.0 and RP09. (C) Distribution of saturated and unsaturated C16 and C18 FFAs in the three strains. (D) Phospholipid levels in strains RP09, MLM1.0 and MLM1.0 Δplb1 Δplb2. PC, phosphatidylcholine; PE, phosphatidylethanolamine; PI, phosphatidylinositol; PS, phosphatidylserine. (E) Phospholipid classes distribution in strains RP09, MLM1.0 and MLM1.0 Δplb1 Δplb2. Strains were grown for 72 h in minimal medium containing 2% glucose, FFA and total lipid quantifications were performed with five biological replicates. **P < 0.005 (Student’s t-test: two-tailed, two-sample equal variance).
Fig. 5. Effect of overexpressing phospholipase genes PLB1, PLB2, PLB3 and a thioesterase gene ‘tesA’ in strain MLM1.0. (A) Representation of the most significant fatty acid metabolism fluxes in MLM1.0. The genes over-expressed in this experiment are highlighted in green. (B) Total FFA quantification of MLM1.0 expressing either phospholipase genes PLB1, PLB2, PLB3, or a thioesterase gene ‘tesA’ from a 2 μl multicopy plasmid under control of the TDH3 (GPD) promoter. The empty plasmid served as a control. Strains were grown for 72 h in minimal medium containing 2% glucose, and FFA quantification was performed with biological triplicates. *P < 0.05 Student’s t test; two-tailed, two-sample equal variance). Differences between MLM1.0–PLB1/2/3 and MLM1.0 (empty plasmid) were statistically nonsignificant (P > 0.05).

pathway, thereby allowing for production of FFAs independent of the phospholipid production/hydrosis cycle (Fig. 5/4). Expression of the thioesterase ‘TesA’ improved FFA production by 26% up to 129 mg gDCW\(^{-1}\) (Fig. 5B) with statistical significance (P < 0.05). This significant increase in FFA from expression of a thioesterase with expression of phospholipases not resulting in the same increase points toward a flux control at the level of the phospholipid biosynthesis pathway from acyl-CoA rather than a limitation in fatty acid biosynthesis capabilities.

Discussion
Regulating levels of different lipid pools is an evolutionary advantage for a yeast cell, providing the capacity to adapt to different environments or carbon source availability and resistance to stresses. However, as a chassis for cell factory development and metabolic engineering, a complex metabolic network entwined with tight regulation mechanisms can offer many challenges and difficulties for progress. Here we have engineered the lipid metabolism of S. cerevisiae by removing many reactions involved in the production of storage lipids, fatty acid oxidation, and conversion of FFAs to fatty acyl-CoA. The combined deletions not only were relevant in reducing the complexity of the metabolic network, but also disrupted several lipid regulation mechanisms. Strains resulting from constraining the lipid metabolic network and redirecting fatty acid fluxes toward phospholipids showed increases in both FFAs and phospholipids, suggesting a strong correlation between the two. Characterization of the resulting strain MLM1.0 points strongly toward phospholipid hydrolysis as a major pathway for FFA formation, identifying phospholipases B as the key players in this process.

The process of deregulating fatty acid and phospholipid biosynthesis genes led to significant changes in the distribution and total levels of most analyzed lipid classes. It also caused severe morphological changes in the internal membrane structure. Taken together, our results highlight the complexity of entwined metabolic and regulatory networks found in S. cerevisiae lipid metabolism. By identifying the different factors that come into play and how the lipid species are redistributed in the cell on the redirection of lipid metabolism, we believe to have contributed important information toward understanding the effects that emerge when S. cerevisiae is engineered at the level of lipid metabolism.

While the strains developed in this study strongly contribute to fundamental insights into FFAs dynamics, they can also serve as a tool for the development of S. cerevisiae as a cell factory for the production of FFAs and fatty-acid derived products. One of the favorable traits of S. cerevisiae is its capacity to accumulate high levels of precursors without the tight feedback regulation mechanisms present in lipid metabolism and the lack of major competing reactions for acyl-CoA. Furthermore, the strain was developed through scarless deletions, without the use of resistance markers or other common genetic elements. This offers major advantages for further studies, because it does not limit genome editing options. Along with the advantages of reduced complexity of the lipid metabolic network with improved regulation, the strain also has the benefit of not carrying any over-expression, which means that the strain is not suffering from a protein burden. On the contrary, through the deletion of several genes, it may be possible to allocate proteome mass for the expression of heterologous pathways that further convert the FFAs to other valuable products, such as fatty alcohols, olefins, or alkanes.

Methods
Plasmid Construction and Strain Construction. The plasmids, strains, and primers used in this study are listed in Dataset S1. Oligonucleotides were ordered from Eurofins. All fragments obtained by PCR were gel- or column-purified (GeneJet Gel and PCR Clean-up columns) before cloning, and the resulting plasmids were verified by sequencing (Eurofins). Yeast transformation were performed using lithium acetate and PEG3350 (33). Gene deletions were performed in the strain IMX581 using CRISP/Cas9 as described previously (34). The diagnostic primers and the repair fragments (listed in Dataset S1) were designed using the Yeastriction tool (yeastriction.tudelft.nl), with the exception of the PLB1 and PLB2 deletions, which were carried out with a single gRNA promiscuously targeting both PLB1 and PLB2, as described previously (35).

Media and Culture Conditions. S. cerevisiae strains with auxotrophies were grown on YPD plates containing 20 g L\(^{-1}\) glucose, 10 g L\(^{-1}\) yeast extract, 20 g L\(^{-1}\) peptone from casein, and 20 g L\(^{-1}\) agar. Plasmid-carrying strains were grown on selective growth medium containing 6.9 g L\(^{-1}\) yeast nitrogen base without amino acids (Formedium), 0.77 g L\(^{-1}\) complete supplement mixture without uracil (Formedium), 20 g L\(^{-1}\) glucose, and 20 g L\(^{-1}\) agar. Shake flask cultivations were performed in minimal medium containing 20 g L\(^{-1}\) glucose, 5 g L\(^{-1}\) (NH\(_4\))\(_2\)SO\(_4\), 14.4 g L\(^{-1}\) KH\(_2\)PO\(_4\), and 0.5 g L\(^{-1}\) MgSO\(_4\)\(_7\)H\(_2\)O. After sterilization, 2 mL L\(^{-1}\) of trace element solution and 1 mL L\(^{-1}\) of vitamin solution were added. The compositions of the trace element and vitamin solution have been reported previously (36). All experiments were performed with strains cultivated as biological replicates, that is, five independent transformants were used to start the precultures.

Quantification of Lipids. Samples for lipid analysis were obtained as 10–15 mL of culture at the end of the shake flask cultivations, after 72 h. Subsequently, the samples were centrifuged at 3,000 × g for 5 min, after which the supernatant was discarded. The pellets were kept at –20 °C for 5 min and then freeze-dried in a Christ Alpha 2–4LC laboratory freeze drier (Christ Gefriertrocknungsanlagen). The samples were analyzed using 10 mg of dried cell biomass as described previously (32).

Quantification of FFAs. FFAs were simultaneously extracted and methylated by dichloromethane containing methyl iodide as a methyl donor (37). In brief, 100-μL aliquots of whole cell culture (cells + supernatant) were placed into glass vials and diluted with 100 μL of water. Then 10 μL 40%
tetramethylammonium hydroxide (base catalyst) was added, followed immediately by the addition of 200 μL of dichloromethane containing 200 μM of methyl donor and 100 mg·L⁻¹ pentadecanoic acid as an internal standard. The mixtures were shaken for 30 min at 1,400 rpm using a vortex mixer, and then centrifuged at 5,000 × g to promote phase separation. A 160-μL dichloromethane layer was transferred into a gas chromatography (GC) vial with a glass insert, and then evaporated for 30 min to dryness. The extracted methyl esters were resuspended in 200 μL of hexane and then analyzed by GC (Focus GC; Thermo Fisher Scientific) equipped with a Zebron ZB-5MS capillary column with GUARDIAN (30 μ × 0.25 mm × 0.25 μm; Phenomenex) and an FID using the GC program as described previously (38). Final quantification was performed using Xcalibur software.

Quantification of Total Fatty Acid Content. Samples for total (bound and free, intracellular and extracellular) fatty acid analysis were taken as 1 mL of culture at the end of 72-h shake flask cultivations. The 1-mL culture volume was kept at −80 °C for 5 min and then freeze-dried in a Christ Alpha 2–4 LSC for 3 d. The total lipophilized culture was processed for fatty acid extraction and derivatization to methyl esters as described previously (38). Samples were resuspended in 1 mL of hexane and analyzed with a gas chromatograph (Focus GC; Thermo Fisher Scientific) equipped with a Zebron ZB-5MS capillary column with GUARDIAN (30 μ × 0.25 mm × 0.25 μm; Phenomenex) and derivatization to methyl esters as described previously (38). Samples for total (bound and free, intracellular and extracellular) fatty acid analysis were taken as 1 mL of culture at the end of 72-h shake flask cultivations. The 1-mL culture volume was kept at −80 °C for 5 min and then freeze-dried in a Christ Alpha 2–4LSC for 3 d. The total lipophilized culture was processed for fatty acid extraction and derivatization to methyl esters as described previously (38).

Lipid Staining. After 72 h of shake flask culture, 100 μL of cell culture was transferred to a 1.5-mL Eppendorf tube, centrifuged, and washed with 1 mL of deionized water. Cells were then centrifuged at 3,000 × g for 5 min and resuspended in 100 μL of PBS. Resuspended cells were treated with 1 μL of BODIPY 493/503 solution (1 mg·mL⁻¹ in ethanol; Thermo Fisher Scientific), 3 μL Nile Red (1 mg·mL⁻¹ in DMSO), or 0.5 μL Calcofluor White M2R (1 mg·mL⁻¹ stock solution; Sigma-Aldrich) and kept at 4 °C in the dark for 10 min. Fluorescent microscope pictures were analyzed using a Leica DMI4000B inverted microscope and processed with Leica Application Suite (LAS) software.

TEM. The samples were prepared as described previously 39. In brief, after fixation with glutaraldehyde 2% in cacodylate buffer for 45 min at 4 °C, the yeast cells were treated with Lytice (Sigma-Aldrich) for 30 min at 37 °C and postfixed with osmium tetroxide 2%, to gently soften the cell wall. Then the samples were dehydrated and embedded in epoxy resin (Agar 100). Ultra-thin sections (70 nm) were imaged with a Leo Gemini transmission electron microscope (Zeiss).

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