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PP2A<sub>Rts<sub>1</sub></sub> is a master regulator of pathways that control cell size

Jessica Zapata,¹ Noah Dephoure,² Tracy MacDonough,¹ Yaxin Yu,³ Emily J. Parnell,³ Meghan Mooring,¹ Steven P. Gygi,² David J. Stillman,³ and Douglas R. Kellogg¹

Introduction

Cell size checkpoints ensure that passage through G1 and mitosis occurs only when sufficient growth has occurred. The mechanisms by which these checkpoints work are largely unknown. PP2A associated with the Rts1 regulatory subunit (PP2A<sub>Rts<sub>1</sub></sub>) is required for cell size control in budding yeast, but the relevant targets are unknown. In this paper, we used quantitative proteome-wide mass spectrometry to identify proteins controlled by PP2A<sub>Rts<sub>1</sub></sub>. This revealed that PP2A<sub>Rts<sub>1</sub></sub> controls the two key checkpoint pathways thought to regulate the cell cycle in response to cell growth. To investigate the role of PP2A<sub>Rts<sub>1</sub></sub> in these pathways, we focused on the Ace2 transcription factor, which is thought to delay cell cycle entry by repressing transcription of the G1 cyclin CLN3. Diverse experiments suggest that PP2A<sub>Rts<sub>1</sub></sub> promotes cell cycle entry by inhibiting the repressor functions of Ace2. We hypothesize that control of Ace2 by PP2A<sub>Rts<sub>1</sub></sub> plays a role in mechanisms that link G1 cyclin accumulation to cell growth.

Cell size checkpoints operate at entry into the cell cycle in G1 and again at mitosis. The mitotic checkpoint works through the Wee1 kinase, which delays mitosis via inhibitory phosphorylation of Cdk1 (Nurse, 1975; Gould and Nurse, 1989). Wee1 is a dose-dependent regulator of cell size. Thus, loss of Wee1 in fission yeast causes premature entry into mitosis at a reduced cell size, whereas increased activity of Wee1 causes delayed entry into mitosis and increased cell size (Nurse, 1975; Russell and Nurse, 1987). The signals that connect Cdk1 inhibitory phosphorylation to cell growth are poorly understood.

The G1 cell size checkpoint is best understood in budding yeast. Cell division in budding yeast is asymmetric, yielding a large mother cell and a small daughter cell. The observation that the small daughter cell spends more time undergoing growth in G1 provided early evidence for the existence of cell size checkpoints (Hartwell and Unger, 1977; Johnston et al., 1977). It also focused attention on the mechanisms that control cell cycle entry and how they might be linked to cell size. The key molecular event that drives cell cycle entry is activation of Cdk1 by G1 cyclins (Richardson et al., 1989; Cross, 1990). There are three G1 cyclins that bind and activate Cdk1 in budding yeast, called Cln1, Cln2, and Cln3 (Hadwiger et al., 1989; Richardson et al., 1989). Transcription of CLN3 is initiated in early G1, and the Cln3–Cdk1 complex helps trigger transcription of the late G1 cyclins CLN1 and CLN2 (Dirick and Nasmyth, 1991). Cln1/2 drive growth of a new daughter cell, which marks commitment to a new round of cell division (Richardson et al., 1989; Cross, 1990; McCusker et al., 2007).

Abbreviations used in this paper: ACN, acetonitrile; ChIP, chromatin immunoprecipitation; FA, formic acid; FDR, false discovery rate; LC, liquid chromatography; MS, mass spectrometry; m/z, mass per charge; qRT-PCR, quantitative RT-PCR; SCX, strong cation exchange.

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Early evidence pointed to Cln3 as playing a critical role in cell size control. Loss of CLN3 causes a prolonged delay in entry into the cell cycle. Cell growth continues during the delay, leading to increased cell size (Cross, 1988). Conversely, overexpression of CLN3 causes premature entry into the cell cycle at a reduced cell size (Cross, 1988; Nash et al., 1988). Together, these observations suggested that Cln3, like Wee1, is a critical dose-dependent regulator of cell size (Cross, 1988; Nash et al., 1988). In this view, cell size in G1 phase could be controlled by mechanisms that link production of active Cln3/Cdk1 to attenuation of a critical cell size. Several observations, however, indicate that this kind of model is too simplistic. First, cln3Δ cells still show size-dependent entry into the cell cycle (Di Talia et al., 2009; Ferrezuelo et al., 2012). Thus, although cln3Δ cells are significantly larger than wild-type cells, small unbudded cln3Δ cells spend more time undergoing growth in G1 than larger unbudded cells. In addition, cln3Δ cells undergo normal nutrient modulation of cell size, in which cells reduce their size in response to poor nutrients (Jorgensen et al., 2004). Together, these observations indicate that modulation of Cln3 alone is insufficient to explain cell size control in G1.

Although Wee1 and G1 cyclins clearly play roles in cell size control, it is unlikely that they are involved in the mechanisms that determine size. Both are capable of accelerating or delaying the cell cycle in a dose-dependent manner, which suggests that they respond to checkpoint signals that determine the duration of growth at specific phases of the cell cycle. Thus, they appear to be downstream effectors of a global mechanism of cell size control. The nature of this global mechanism has remained deeply mysterious.

We recently discovered that a specific form of PP2A (protein phosphatase 2A) is required for cell size control (Artiles et al., 2009). Canonical PP2A is a trimeric complex composed of a catalytic subunit, a scaffolding subunit, and a regulatory subunit (Zhao et al., 1997; Janssens and Goris, 2001). In budding yeast, there are two regulatory subunits, referred to as Rts1 and Cdc55, that form two distinct complexes: PP2A Rts1 and PP2A Cdc55 (Zhao et al., 1997). We previously discovered that rts1Δ cells cause a prolonged delay in transcription of the G1 cyclin Cln2, a prolonged delay in mitosis, and defects in regulatory phosphorylation of Wee1 (Artiles et al., 2009; Harvey et al., 2011). Together, these observations suggest that PP2A Rts1 functions in both G1 and mitotic cell size checkpoints. However, the targets of PP2A Rts1 that mediate these functions were unknown. Here, we used proteome-wide mass spectrometry (MS) to identify targets of PP2A Rts1. This revealed that PP2A Rts1 controls key elements of both cell size checkpoints, which suggests that it functions in the mysterious cell size control mechanisms that send signals to G1 cyclins and Wee1. We further discovered that PP2A Rts1 controls the transcription factor Ace2, which likely contributes to mechanisms that link CLN3 transcription to cell growth.

### Results

**A proteomic screen for targets of PP2A Rts1**

To identify targets of PP2A Rts1, we used quantitative phosphoproteomics to search for proteins that become hyperphosphorylated in rts1Δ cells. Because we previously found that PP2A Rts1 is required for control of G1 cyclin transcription, we were particularly interested in G1 targets of PP2A Rts1 (Artiles et al., 2009). We therefore synchronized wild-type and rts1Δ cells and collected samples for MS 10 min before the G1 cyclin Cln2 appeared, which is when the decision to initiate G1 cyclin transcription is made. Proteolytic peptides from each strain were covalently modified by reductive dimethylation to generate light (wild type) and heavy (rts1Δ) stable isotope-labeled pools. After combining, phosphopeptides were enriched by strong cation exchange (SCX) followed by TiO2 affinity chromatography and identified via liquid chromatography (LC)–MS/MS (Fig. 1 A; Villén and Gygi, 2008; Kettenbach and Gerber, 2011).

The heavy to light ratios of phosphorylated peptides in rts1Δ cells versus wild-type cells were log_{2} transformed. Thus, positive values indicate increased phosphorylation in rts1Δ, whereas negative values indicate decreased phosphorylation. A parallel analysis of sample-matched unphosphorylated peptides was used to generate protein abundance ratios that were used to correct for differences in protein abundance between the two samples (Table S1). Three biological replicates of the experiment were performed, which allowed calculation of average log_{2} ratios and SDs for most peptides. The complete dataset appears in Tables S1–S3. Table S1 lists all protein quantification data, Table S2 lists all identified phosphorylation sites along with quantitative data, and Table S3 provides detailed information for each of the 78,204 phosphopeptides that were detected.

A total of 10,807 sites were identified on 2,066 proteins. Of these, 9,255 sites on 1,937 proteins could be quantified. We focused on sites that were quantified in at least two of three biological replicates. This high-quality set includes 5,159 sites on 1,544 proteins (Fig. 1 B). Note that the analysis is not comprehensive; many peptides are not detected as a result of poor ionization, loss during chromatography, or low abundance.

Relative peptide abundances were calculated as the ratio of corresponding heavy and light peptide pairs as determined from their extracted ion chromatograms. A visual representation of this is shown for a single phosphopeptide in Fig. S1. For each phosphorylation site, we calculated an average ratio from all quantified peptides harboring each site. We used a twofold SD from the mean, representing a ~2.5-fold change in either direction, to define significant changes in phosphorylation (Fig. 1 C). At this threshold, we identified 241 sites on 156 proteins that were hyperphosphorylated in rts1Δ cells (Table S4). We observed fewer sites whose phosphorylation decreased: 59 sites on 45 proteins (Table S5).

**PP2A Rts1** is required for normal regulation of key effectors of cell size control

Table S4 lists proteins that underwent significant hyperphosphorylation in rts1Δ cells. It is likely that additional regulated sites whose ratios fell below our cutoff exist in the data. Several of the regulated proteins are linked to known roles of PP2A Rts1. For example, PP2A Rts1 controls Kin4 in the spindle orientation checkpoint (Chan and Amon, 2009). A site in Kin4, serine 351, was up-regulated nearly threefold in rts1Δ cells. PP2A Rts1 also controls chromosome cohesion (Yu and Koshland, 2007). Here,
we identified two proteins involved in chromosome cohesion as new targets of PP2ARts1-dependent regulation: Pds1 and Ulp2.

We focused on targets of PP2ARts1 that could provide clues to its role in cell size control. The analysis identified multiple proteins involved in cell size control in G1, including three factors that control G1 cyclin transcription: Swi4, Swi5, and Ace2. Swi4 is a transcriptional activator of the late G1 cyclins CLN1 and CLN2 (Nasmyth and Dirick, 1991; Ogas et al., 1991). Swi5 and Ace2 are related factors that control transcription of genes expressed in late mitosis and early G1 (Dohrmann et al., 1992; Doolin et al., 2001). Ace2 is a repressor of CLN3 transcription, whereas Swi5 controls transcription of the G1 cyclin Pcl2, which activates Pho85 and acts redundantly with Cln1/2 to promote bud emergence (Aerne et al., 1998; Laabs et al., 2003; Moffat and Andrews, 2004; Di Talia et al., 2009). Bck2, an upstream regulator of CLN1/2 transcription, was hyperphosphorylated in one of the biological replicates (Table S2; Di Como et al., 1995; Bastajian et al., 2013). Thus, PP2ARts1 appears to control transcription of all of the key G1 cyclins. The analysis also identified Ydj1, which is thought to control Cln3 localization and stability (Yaglom et al., 1996; Vergés et al., 2007). Loss of Ace2, Swi4, or Ydj1 causes defects in cell size control (Breeden and Mikesyell, 1991; Caplan and Douglas, 1991; Dohrmann et al., 1992; Di Talia et al., 2009; Ferrezuelo et al., 2012).

The analysis also identified proteins involved in cell size control during mitosis. For example, the inhibitory site on Cdk1 that is phosphorylated by Wee1 showed one of the most dramatic increases, being up-regulated >25-fold in rts1Δ cells. Moreover, Swe1, the budding yeast homologue of Wee1, was hyperphosphorylated on multiple sites that were previously found to be required for its activation (Harvey et al., 2005, 2011). This is consistent with our previous finding that rts1Δ causes Swe1 to accumulate in a hyperphosphorylated active form, which is the likely cause of a prolonged mitotic delay (Harvey et al., 2011). We also identified three related kinases that are required for Swe1 inactivation: Hsl1, Gin4, and Kcc4 (Ma et al., 1996; Barral et al., 1999; McMillan et al., 1999; Longtine et al., 2000). Loss of these kinases can cause delayed entry into mitosis and severe cell size defects (Altman and Kellogg, 1997; Barral et al., 1999). 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again at 50 min as cells entered the next mitosis. Ace2 was dramatically hyperphosphorylated in \(rts1\) cells relative to wild-type cells (Fig. 2C). In addition, Ace2 dephosphorylation and destruction of Clb2 were delayed by \(\sim 20\) min in \(rts1\) cells, which indicated that PP2A\(Rts1\) is required for normal mitotic inhibitory phosphorylation of Cdk1. Thus, PP2A\(Rts1\) may be a component of global cell size control mechanisms. Here, we focused on the Ace2 transcription factor. Table 1 shows data for all identified Ace2 phosphorylation sites, which includes four significantly regulated sites: S122, S253, S709, and T713. Ace2 is asymmetrically segregated into the nuclei of small daughter cells, where it is thought to delay cell cycle entry via inhibition of \(CLN3\) transcription (Laabs et al., 2003; Di Talia et al., 2009). However, regulation of Ace2 has not been linked to signals that relay information about cell growth or size. Ace2 also functions as a transcriptional activator for genes involved in septation (Dohrmann et al., 1992).

### Table 1. Ace2 phosphorylation sites

<table>
<thead>
<tr>
<th>Site</th>
<th>Local sequence</th>
<th>No. identified peptides</th>
<th>No. quantified peptides</th>
<th>No. replicates quantified</th>
<th>Average ratio</th>
<th>SD</th>
</tr>
</thead>
<tbody>
<tr>
<td>S122</td>
<td>SHKRGLSGTAIFG</td>
<td>2</td>
<td>2</td>
<td>2</td>
<td>2.51</td>
<td>0.72</td>
</tr>
<tr>
<td>T135</td>
<td>FLGHNKTLSSSL</td>
<td>1</td>
<td>1</td>
<td>1</td>
<td>2.41</td>
<td>ND</td>
</tr>
<tr>
<td>S137</td>
<td>GHNKTLSSSLQQ</td>
<td>2</td>
<td>0</td>
<td>0</td>
<td>ND</td>
<td>ND</td>
</tr>
<tr>
<td>S140</td>
<td>KTLSSSLQQSIL</td>
<td>3</td>
<td>0</td>
<td>0</td>
<td>ND</td>
<td>ND</td>
</tr>
<tr>
<td>T245</td>
<td>KLVSGATNSNSKP</td>
<td>6</td>
<td>2</td>
<td>1</td>
<td>2.44</td>
<td>ND</td>
</tr>
<tr>
<td>S249</td>
<td>CATNSNSPKCPSPV</td>
<td>6</td>
<td>5</td>
<td>2</td>
<td>0.93</td>
<td>0.02</td>
</tr>
<tr>
<td>S253</td>
<td>SNKPGSPVILKT</td>
<td>8</td>
<td>7</td>
<td>2</td>
<td>1.43</td>
<td>0.73</td>
</tr>
<tr>
<td>S709</td>
<td>KKSLLDSPHDTSP</td>
<td>9</td>
<td>9</td>
<td>3</td>
<td>1.54</td>
<td>0.29</td>
</tr>
<tr>
<td>T713</td>
<td>LDSPHTSPKVETI</td>
<td>9</td>
<td>7</td>
<td>2</td>
<td>1.62</td>
<td>0.24</td>
</tr>
<tr>
<td>S714</td>
<td>DSPHTSPKVETI</td>
<td>4</td>
<td>4</td>
<td>2</td>
<td>1.27</td>
<td>0.05</td>
</tr>
</tbody>
</table>

The table shows all identified and quantified phosphosites in Ace2. The phosphorylated residue is shown in bold in the context of its flanking sequence. Six sites were identified in two out of three biological replicates, and out of those six, four showed increased phosphorylation above the log\(_2\) threshold: S122, S253, S709, and T713.

The table shows all identified and quantified phosphosites in Ace2. The phosphorylated residue is shown in bold in the context of its flanking sequence. Six sites were identified in two out of three biological replicates, and out of those six, four showed increased phosphorylation above the log\(_2\) threshold: S122, S253, S709, and T713.

Loss of PP2A\(Rts1\) causes defects in phosphorylation of the Ace2 transcription factor

To extend the MS data, we assayed Ace2 phosphorylation in synchronized wild-type and \(rts1\) cells. Phosphorylation of Ace2 causes an electrophoretic mobility shift that can be assayed by Western blotting (Sbia et al., 2008; Mazanka and Weiss, 2010). We first assayed Ace2 after release from a G1 arrest imposed by mating pheromone. The mitotic cyclin Clb2 was assayed in the same samples as a marker for cell cycle progression. In wild-type cells, Ace2 was present at low levels early in the cell cycle and began to accumulate and undergo extensive hyperphosphorylation as cells entered mitosis, consistent with previous studies that Ace2 is phosphorylated by mitotic Cdk1 (Fig. 2A; O’Conalláin et al., 1999; Sbia et al., 2008; Mazanka and Weiss, 2010). In \(rts1\) cells, the mitotic hyperphosphorylation of Ace2 was delayed by \(\sim 20\) min, consistent with a previously reported G1 delay in \(rts1\) cells (Artiles et al., 2009). To compare differences in Ace2 phosphorylation during G1, we loaded the initial time points from Fig. 2A in an intercalated manner (Fig. 2B). This revealed that a fraction of Ace2 was hyperphosphorylated in \(rts1\) cells during G1.

We also assayed Ace2 phosphorylation and Clb2 levels after release from a metaphase arrest. Ace2 was phosphorylated in metaphase-arrested wild-type cells and was dephosphorylated as cells exited mitosis (Fig. 2C). Ace2 underwent a transient phosphorylation at 30 min and became phosphorylated again at 50 min as cells entered the next mitosis. Ace2 was dramatically hyperphosphorylated in \(rts1\) cells relative to wild-type cells (Fig. 2C). In addition, Ace2 dephosphorylation and destruction of Clb2 were delayed by \(\sim 20\) min in \(rts1\) cells, which indicated that PP2A\(Rts1\) is required for normal mitotic

![Figure 2. The Ace2 transcription factor is hyperphosphorylated in \(rts1\) cells.](image)

(A) Cells were released from a G1 arrest, and the behavior of Ace2 and Clb2 was assayed by Western blotting. (B) The same samples shown in Fig. 2B were loaded in an intercalated manner to visualize differences between wild-type and \(rts1\) cells. (C) Cells were released from a metaphase arrest, and the behavior of Ace2 and Clb2 was assayed by Western blotting.
exit. Because we did not observe a drop in Ace2 levels as cells traversed G1 after release from a metaphase arrest (Fig. 2 C), it is likely that low levels of Ace2 after release from a G1 arrest (Fig. 2 A) is caused by the prolonged arrest, rather than by a mechanism that degrades Ace2 during G1 in every cell cycle.

Cbk1 contributes to hyperphosphorylation of Ace2 in rts1Δ cells

We next searched for the kinase that hyperphosphorylates Ace2 in rts1Δ cells. Cdk1 is thought to phosphorylate Ace2 during mitosis to block its nuclear import (O’Conalláin et al., 1999; Sbia et al., 2008; Mazanka and Weiss, 2010). If Cdk1 and PP2AΔn sensitive allele of Cbk1 to test

Figure 3. Multiple kinases contribute to hyperphosphorylation of Ace2 in rts1Δ cells.

(A) Samples were taken at the indicated times after addition of 1NM-PP1 to log phase cells, and phosphorylation of Ace2 was assayed by Western blotting. (B) Ace2-3xHA was immunoprecipitated from wild-type, rts1Δ, or cdk1Δ cells and probed with anti-Ace2 antibody or with an antibody that recognizes phosphorylated S122. Ace2-3xHA was immunoprecipitated from the cdk1Δ cells as a control to demonstrate that the phosphospecific antibody recognizes a site phosphorylated by Cbk1. (C) Kinase reactions containing affinity purified Ace2-3xHA, Cbk1-3xHA, and Cdk1/Cbk1-3xHA in the indicated combinations were initiated by addition of ATP. Ace2-3xHA phosphorylation was assayed by Western blotting with the anti-HA mouse monoclonal antibody. (D) Cells were grown to log phase at room temperature, and Ace2 phosphorylation was assayed by Western blotting. The arrow indicates a hyperphosphorylated form of Ace2.

family that plays roles in bud growth and mitotic exit (Mazanka et al., 2008). During late mitosis, Cbk1 is asymmetrically localized to the daughter nucleus, where it phosphorylates Ace2 on several sites that inhibit nuclear export (Colman-Lerner et al., 2001; Weiss et al., 2002; Mazanka et al., 2008). Cbk1 could therefore inhibit nuclear export of Ace2 to delay CLN3 transcription in newborn daughter cells. One of the high confidence Ace2 sites corresponds to a Cbk1 consensus site (S122) that is phosphorylated in vitro and in vivo in a Cbk1-dependent manner (Mazanka et al., 2008). Western blotting with a phosphospecific antibody (Mazanka et al., 2010) demonstrated that this site is hyperphosphorylated in rts1Δ cells (Fig. 3 B). Thus, Cbk1 is likely responsible for hyperphosphorylating at least one site on Ace2 in rts1Δ cells. However, most of the sites that were hyperphosphorylated in rts1Δ cells do not correspond to Cbk1 consensus sites, which suggests that multiple kinases may be involved.

To further test the roles of Cdk1 and Cbk1, we reconstituted phosphorylation of Ace2 in vitro. Cdk1 caused a shift in the electrophoretic mobility of Ace2 that was similar to the Cdk1-dependent shift observed in vivo (Fig. 3, A and C). Cbk1 also shifted the electrophoretic mobility of Ace2, but the extent of the shift appeared to be less than the shift caused by rts1Δ in vivo (Fig. 3 C). We considered the possibility that efficient phosphorylation of Ace2 by Cbk1 requires priming by Cdk1; however, Cdk1 did not appear to enhance Cbk1 phosphorylation of Ace2 in vitro (Fig. 3 C).

Because there was the possibility that the reconstituted reactions lacked key factors necessary for efficient phosphorylation of Ace2 by Cbk1, we also tested the role of Cdk1 in vivo. We attempted to use an analogue-sensitive allele of Cbk1 to test whether hyperphosphorylation of Ace2 after release from a mitotic
transcription. We first used genetics to test this model. If PP2A
\textit{Rts1} is an inhibitor of Ace2, ace2 should rescue temperature-dependent
growth defects caused by rts1. To test this, we assayed rate of colony formation in wild-type, rts1, ace2, and rts1 ace2 cells at 30 and 37°C. We found that ace2 partially rescued the temperature-dependent growth defect caused by rts1 (Fig. 4 A).

Because ace2 causes a cell separation defect, colonies could appear larger because they start from a clump of cells rather than a single cell. We therefore used a Bioscreen apparatus to measure rates of growth of each strain. This confirmed that ace2 partially rescued the slow growth phenotype of rts1 cells at both 30 and 34°C (Fig. 4 B).

We also discovered that overexpression of Ace2 from the GAL1 promoter was lethal in rts1 cells, consistent with the idea that PP2A\textit{Rts1} inhibits transcriptional repressor functions of Ace2 (Fig. 4 C). The lethality of \textit{ACE2} overexpression in rts1 suggests that hyperactive Ace2 must have targets in addition to \textit{CLN3}.

\textbf{PP2A\textit{Rts1} is likely a negative regulator of Ace2}

Because \textit{rts1} causes a prolonged G1 delay, we hypothesized that PP2A\textit{Rts1} inhibits repressor functions of Ace2. In this model, inactivation of PP2A\textit{Rts1} causes Ace2 to become hyperphosphorylated, which makes it hyperactive as a repressor of \textit{CLN3} transcription. We first used genetics to test this model. If PP2A\textit{Rts1} is an inhibitor of Ace2, ace2 could rescue temperature-dependent growth defects caused by rts1. To test this, we assayed rate of colony formation in wild-type, rts1, ace2, and rts1 ace2 cells at 30 and 37°C. We found that ace2 partially rescued the temperature-dependent growth defect caused by rts1 (Fig. 4 A). Because ace2 causes a cell separation defect, colonies could appear larger because they start from a clump of cells rather than a single cell. We therefore used a Bioscreen apparatus to measure rates of growth of each strain. This confirmed that ace2 partially rescued the slow growth phenotype of rts1 cells at both 30 and 34°C (Fig. 4 B).

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Figure 4. Genetic analysis suggests that PP2A\textit{Rts1} is a negative regulator of Ace2. (A) A series of fivefold dilutions of cells were grown on YPD media at 30 or 37°C. (B) Eight independent colonies grown overnight at 23°C were diluted into fresh medium and grown in a Bioscreen C apparatus. The average growth of the eight cultures was plotted, with SDs shown. (C) A series of fivefold dilutions of cells were grown at 30°C on YEP media containing either dextrose or galactose.
Overexpression of \textit{CLN3} partially rescues cell size defects caused by \textit{rts1Δ}

Loss of \textit{RTS1} or \textit{CLN3} causes cells to become abnormally large (Cross, 1988; Artiles et al., 2009). We hypothesized that hyperactive Ace2 in \textit{rts1Δ} cells causes failure to produce normal levels of Cln3, leading to increased cell size. To test this, we overexpressed \textit{CLN3} from the \textit{GAL1} promoter in \textit{rts1Δ} cells. This reduced the size of \textit{rts1Δ} cells to nearly the same size as wild-type cells, consistent with the hypothesis (Fig. 5). A previous study found that \textit{CLN3} overexpression causes cells to become significantly smaller than wild-type cells (Fig. 5; Tyers et al., 1992). Thus, \textit{GAL1-CLN3} does not cause the same size reduction in wild-type and \textit{rts1Δ} cells, which indicates that cell size defects caused by \textit{rts1Δ} are not caused solely by a failure to produce normal levels of Cln3. This is consistent with the discovery that \textit{PP2A\textsc{Rts1}} controls diverse pathways required for cell size control.

\textbf{PP2A\textsc{Rts1} is required for normal control of Cln3 protein and mRNA levels}

To further test the hypothesis that \textit{PP2A\textsc{Rts1}} controls production of Cln3 via Ace2, we assayed \textit{CLN3} mRNA accumulation in wild-type, \textit{rts1Δ}, \textit{ace2Δ}, and \textit{rts1Δ ace2Δ} cells after release from a G1 arrest. \textit{CLN3} mRNA levels were assayed both by quantitative RT-PCR (qRT-PCR) and by Northern blotting, which gave similar results. Accumulation of Cln3-6×HA protein was assayed in identical time courses, and the mitotic cyclin Clb2 was assayed in the same samples to provide a marker for cell cycle progression.

In wild-type cells, \textit{CLN3} mRNA and protein peaked in G1 at 20–30 min (Figs. 6, A–C; and S2). There was a second peak of \textit{CLN3} mRNA and protein later in the cell cycle that appeared at the same time as peak levels of the mitotic cyclin Clb2, indicating that Cln3 is produced in mitosis (Fig. 6, A–D). In \textit{rts1Δ} cells, accumulation of \textit{CLN3} mRNA and protein was reduced and delayed (Figs. 6, A–C; and S2). The defect in \textit{CLN3} mRNA accumulation caused by \textit{rts1Δ} at 20 min showed a statistically significant rescue by \textit{ace2Δ} (Fig. 6 B). In addition, \textit{ace2Δ}
PP2A\textsuperscript{Rts1} is required for normal control of \textit{CLN3} mRNA and protein levels in cells released from a metaphase arrest. In wild-type cells, \textit{CLN3} mRNA and protein were present at the metaphase arrest and then increased as Clb2 levels declined, reaching a peak 20 min after release from the arrest (Fig. 7, A–C). Cln3 protein was present throughout most of G1, decreased before mitosis, and then accumulated again during the second mitosis. The decline in \textit{CLN3} mRNA and protein at 30 min was correlated with hyperphosphorylation of Ace2, consistent with a role for Ace2 hyperphosphorylation in repression of \textit{CLN3} transcription (Figs. 2 C and 7, A and B, compare wild-type samples). In \textit{rts1\Delta} cells, destruction of Clb2 was delayed, indicating a delay in exit from mitosis, and \textit{CLN3} mRNA and protein failed to accumulate to normal levels as cells exited mitosis (Fig. 7, A–C). Defects in \textit{CLN3} mRNA accumulation in \textit{rts1\Delta} cells were not rescued by \textit{ace2\Delta} (Fig. 7, A and B).

Together, these observations show that \textit{ace2\Delta} may rescue some, but not all, defects in \textit{CLN3} mRNA accumulation caused by \textit{rts1\Delta}. A possible explanation is that PP2A\textsuperscript{Rts1} controls an additional repressor of \textit{CLN3} transcription. The only other known repressor of \textit{CLN3} transcription is Yox1 (Pramila et al., 2002; Bastajian et al., 2013). The MS analysis identified a hyperphosphorylated Yox1 peptide in \textit{rts1\Delta} cells with high confidence in one of the biological replicates (Table S3). In addition, \textit{yox1\Delta} improved the growth rate of \textit{rts1\Delta} cells at 34°C (Fig. 7 D). Finally, \textit{rts1\Delta} increased the toxicity caused by expression of \textit{YOX1} from the \textit{GAL1} promoter (Fig. 7 E). PP2A\textsuperscript{Rts1} may therefore control multiple repressors of \textit{CLN3} transcription.
Rts1-dependent regulation, we used wild-type cells. It is thought that Ace2 causes a G1 delay in isolation, the cells were released into rich media, and samples to enrich for very small daughter cells, the cells were grown in we used centrifugal elutriation to isolate small daughter cells. To circumvent this problem, to study events that occur in small newborn daughter cells by repressing $\text{CLN3}$. Loss of $\text{Rts1}$ did not cause significant effects on binding of Ace2 to the $\text{CLN3}$ promoter; however, binding of Ash1 was significantly increased (Fig. 9, A and B). In addition, binding of Ace2 was dependent on Ash1, and binding of Ash1 was strongly dependent on Ace2 (Fig. 9, C and D). These findings suggest that hyperphosphorylation of Ace2 causes increased recruitment of Ash1, leading to transcriptional repression.

Ace2 is hyperphosphorylated in small unbudded daughter cells

The preceding experiments show that Ace2 is hyperphosphorylated in $\text{rts1} \Delta$ cells, which likely activates it to repress transcription of $\text{CLN3}$. We next investigated regulation of Ace2 in wild-type cells. It is thought that Ace2 causes a G1 delay in small daughter cells by repressing $\text{CLN3}$ transcription (Laabs et al., 2003; Di Talia et al., 2009). However, the signals that control the duration of the delay are unknown. The discovery that $\text{PP2A}^{\text{Rts1}}$ controls Ace2 suggested that it could play a role in determining the duration of the G1 delay. We reasoned that one way to test this would be to monitor Ace2 hyperphosphorylation, as well as levels of $\text{CLN3}$ mRNA and protein, during G1 in newborn daughter cells. If $\text{PP2A}^{\text{Rts1}}$ plays a role in enforcing a G1 delay, Ace2 should be hyperphosphorylated in newborn daughter cells and dephosphorylation of Ace2 should be correlated with accumulation of Cln3.

Cells synchronized via a cell cycle arrest cannot be used to study events that occur in small newborn daughter cells because cells grow during the arrest. To circumvent this problem, we used centrifugal elutriation to isolate small daughter cells. To enrich for very small daughter cells, the cells were grown in media containing a poor carbon source before elutriation. After isolation, the cells were released into rich media, and samples were taken to assay Ace2 phosphorylation and levels of $\text{CLN3}$ mRNA and protein as the cells underwent growth and entry into the cell cycle. We also measured cell size as a function of time to monitor cell growth and the fraction of cells undergoing bud emergence to determine when cells enter the cell division cycle. We used Northern blotting to assay $\text{CLN3}$ mRNA levels because qRT-PCR requires normalization to an internal standard RNA, which could undergo significant changes as cells grow. By using Northern blotting to probe the same fraction of total RNA at each time point, we could assay levels of $\text{CLN3}$ mRNA per cell.

The small newborn daughter cells underwent continuous growth and initiated bud emergence at 130 min (Fig. 10, A and B). Ace2 was hyperphosphorylated in the small unbudded cells and underwent gradual dephosphorylation (Fig. 10 C). Maximal dephosphorylation of Ace2 occurred at 70–80 min. Cln3 was first detectable at 10 min and then accumulated gradually, reaching peak levels around the time of maximal Ace2 dephosphorylation (Fig. 10 C). Thus, Ace2 dephosphorylation and Cln3 protein accumulation occurred gradually during growth and were correlated. $\text{CLN3}$ mRNA accumulated gradually during growth, similar to Cln3 protein (Fig. 10 D). We consistently observed a transient increase in $\text{CLN3}$ mRNA at 5 min, and Cln3 protein began to accumulate shortly thereafter. This burst of $\text{CLN3}$ mRNA was not correlated with Ace2 phosphorylation.

As expected, the mitotic cyclin Cib2 was not detectable, which indicates that phosphorylation of Ace2 in this context was not caused by mitotic Cdk1 activity. It was not possible to isolate unbudded $\text{rts1} \Delta$ cells because of their severe cell size defects: centrifugal elutriation yielded a mixture of budded and unbudded cells that were of similar size.

Discussion

Identification of $\text{PP2A}^{\text{Rts1}}$ targets by proteome-wide MS

To identify targets of $\text{PP2A}^{\text{Rts1}}$-dependent regulation, we used quantitative proteome-wide MS to search for proteins that undergo changes in phosphorylation in $\text{rts1} \Delta$ cells. Proteome-wide MS should prove to be a powerful tool for identifying phosphatase targets because one searches for proteins that undergo hyperphosphorylation, which is unlikely to be a result of indirect or toxic effects caused by inactivation of the phosphatase.

The analysis identified 156 proteins that undergo significant hyperphosphorylation when $\text{PP2A}^{\text{Rts1}}$ is inactivated. These likely include direct targets of $\text{PP2A}^{\text{Rts1}}$ but also appear to delineate entire pathways regulated by $\text{PP2A}^{\text{Rts1}}$. For example, several components of a pathway that regulates mitosis via Cdk1 inhibitory phosphorylation were identified. In this pathway, three related kinases called Gin4, Hsl1, and Kcc4 promote entry into mitosis by inactivating Swe1. Gin4 and Hsl1 are controlled by the septins, which were also identified as potential targets of $\text{PP2A}^{\text{Rts1}}$-dependent regulation (Table S2). Previous work found that $\text{rts1} \Delta$ causes a mitotic delay and defects in Swe1 phosphorylation; however, the underlying mechanisms were unknown. The MS data suggest that $\text{PP2A}^{\text{Rts1}}$ regulates mitosis via a pathway that includes the septins, Gin4/Hsl1/Kcc4, and Swe1.
mechanisms that signal to Swe1 and the G1 cyclins have proven remarkably difficult to discover. In previous work, we found that PP2A Rts1 is required for normal control of cell size as well as nutrient modulation of cell size (Artiles et al., 2009). Here, we discovered that PP2A Rts1 regulates pathways that control both Swe1 and G1 cyclins. Together, these observations suggest the possibility that PP2A Rts1 is a component of the enigmatic cell size control mechanisms that signal to Swe1 and G1 cyclins.

In G1 phase, PP2A Rts1 controls phosphorylation of two key transcription factors for G1 cyclins: Ace2 and Swi4. Ace2 is a repressor of \(CLN3\) transcription (Laabs et al., 2003; Di Talia et al., 2009), whereas Swi4 is a transcriptional activator for late G1 cyclins \(CLN1\) and \(CLN2\) (Nasmyth and Dirick, 1991; Ogas et al., 1991). Because Cln3 appears first and helps promote transcription of \(CLN1\) and \(CLN2\), it was initially thought that size control in G1 works through \(CLN3\). However, although \(cln3\Delta\) cells are abnormally large, they still show size-dependent entry into the cell cycle and nutrient modulation of cell size, which

In addition to identifying targets of PP2A Rts1-dependent regulation, the MS identified 10,807 phosphorylation sites on 2,066 proteins. These sites significantly expand phosphorylation site data in budding yeast. Nevertheless, it must be kept in mind that proteome-wide MS is not comprehensive. Thus, little can be inferred from the absence of proteins or sites in the data. In addition, the analysis generally does not provide sufficient sequence coverage to warrant mutagenesis of identified sites to test their functions. Rather, further site mapping must be performed using purified proteins to yield a more comprehensive identification of sites.

PP2A Rts1 as a master regulator of cell size control pathways

Swe1 and the G1 cyclins play important roles in mechanisms that control cell size. Both serve as downstream effectors of cell size control mechanisms that accelerate or delay the cell cycle to allow more or less time for cell growth. The cell size control mechanisms that signal to Swe1 and the G1 cyclins have proven remarkably difficult to discover.

In previous work, we found that PP2A Rts1 is required for normal control of cell size as well as nutrient modulation of cell size (Artiles et al., 2009). Here, we discovered that PP2A Rts1 regulates pathways that control both Swe1 and G1 cyclins. Together, these observations suggest the possibility that PP2A Rts1 is a component of the enigmatic cell size control mechanisms that signal to Swe1 and G1 cyclins.

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**Figure 9. Loss of PP2A Rts1 causes increased binding of Ash1 to the CLN3 promoter.** ChIP experiments were performed using Ace2-Myc, Ash1-Myc, or untagged control strains. Transcription factor binding was measured for two regions of the \(CLN3\) promoter, \(CLN3\)-A (−1,026 to −830) and \(CLN3\)-B (−853 to −642), in distance from the ATG. Data from at least three replicates were analyzed by an unpaired two-tailed t test to test for statistically significant differences. Error bars indicate SDs.
shows that cell size control in G1 cannot work solely through \textit{CLN3} (Nasmyth and Dirick, 1991; Jorgensen et al., 2004; Di Talia et al., 2009; Ferrezuelo et al., 2012). The discovery that PP2A<sup>Rts1</sup> controls both Ace2 and Swi4 suggests that a common mechanism could link transcription of both early and late G1 cyclins to cell size or growth. Thus, size dependent entry into the cell cycle in \textit{cln3Δ} cells could work through PP2A<sup>Rts1</sup> and Swi4-dependent control of \textit{CLN1} and \textit{CLN2} transcription.

In mitosis, PP2A<sup>Rts1</sup> regulates a pathway that controls inhibitory phosphorylation of Cdk1. Although much emphasis has been placed on control of cell size at G1 in budding yeast, it is likely that size is also controlled in mitosis. Loss of Swe1 or regulators of Swe1 causes defects in cell size (Ma et al., 1996; Shulewitz et al., 1999; Sreenivasan and Kellogg, 1999; Longtine et al., 2000; Jorgensen et al., 2002; Harvey and Kellogg, 2003; Kellogg, 2003; Harvey et al., 2005). Moreover, nutrients almost certainly modulate cell size at both G1 and mitosis. Classic experiments performed over 30 years ago discovered that daughter cells exit mitosis at a smaller size in poor nutrients (Johnston et al., 1977). More recent work has confirmed that the smallest cells in a population of cells growing in poor nutrients are smaller than the smallest cells in rich nutrients, which can only occur if daughter cells exit mitosis at a smaller size (see, for example, Fig. 3 A in Jorgensen et al. [2004] or Fig. 11 A in Artiles...
et al. [2009]). This suggests the existence of a mechanism that drives progression through mitosis at a smaller bud size when cells are growing in poor nutrients. Because PP2A<sup>Rts1</sup> is required for nutrient modulation of cell size and regulates mitosis, it may be an essential component of this mechanism. PP2A<sup>Rts1</sup> appears to control mitosis via the related kinases Gin4, Hsl1, and Ack4. Fission yeast homologues of these kinases (Cdr1 and Cdr2) are required for nutrient modulation of cell size at entry into mitosis, which suggests a conserved mechanism (Young and Fantes, 1987).

**PP2A<sup>Rts1</sup> controls G1 cyclin transcription via the Ace2 transcription factor**

We focused on Ace2 as a starting point for characterizing proteins controlled by PP2A<sup>Rts1</sup>. Ace2 is thought to delay cell cycle entry in small daughter cells via repression of CLN3 transcription (Colman-Lerner et al., 2001; Laabs et al., 2003). The signals that control the length of the Ace2-dependent delay could control cell size but have been largely unknown. Our analysis suggests that PP2A<sup>Rts1</sup> controls Ace2 and likely influences the duration of G1 and cell size.

Diverse experiments support a model in which hyperphosphorylated Ace2 is active as a repressor of CLN3 transcription and that PP2A<sup>Rts1</sup> dephosphorylates Ace2 to promote CLN3 transcription (Fig. 10 E). In rts1<sup>Δ</sup> cells, Ace2 was hyperphosphorylated, which correlated with decreased CLN3 mRNA and protein. In wild-type cells, there was a decrease in CLN3 mRNA and protein 30 min after release from metaphase arrest that correlated with Ace2 hyperphosphorylation. Finally, in small unbudded cells undergoing growth, dephosphorylation of Ace2 occurred gradually and was correlated with gradually increasing Cln3 protein levels.

Genetic analysis provided additional support for the model: overexpression of ACE2 was lethal in rts1<sup>Δ</sup> cells, and ace2<sup>Δ</sup> partially rescued the reduced growth rate of rts1<sup>Δ</sup> cells at elevated temperatures. In addition, ace2<sup>Δ</sup> rescued defects in CLN3 mRNA levels in rts1<sup>Δ</sup> cells early in the cell cycle. However, ace2<sup>Δ</sup> did not cause increased CLN3 mRNA levels in wild-type cells, and it did not rescue reduced levels of CLN3 mRNA in rts1<sup>Δ</sup> cells exiting mitosis. There are several potential explanations for these observations. First, previous work suggests that Ace2 represses CLN3 transcription only in small unbudded daughter cells; however, we were not able to test effects of rts1<sup>Δ</sup> and ace2<sup>Δ</sup> upon CLN3 transcription in small daughter cells for two reasons. First, small daughter cells grow during cell cycle arrests and are therefore lost in synchronized cells. Second, ace2<sup>Δ</sup> and rts1<sup>Δ</sup> cause cell clumping or size defects that preclude isolation of small daughter cells by centrifugal elutriation. Thus, we could not analyze the effects of rts1<sup>Δ</sup> and ace2<sup>Δ</sup> in the context most likely to show strong effects. Another consideration is that PP2A<sup>Rts1</sup> could also activate factors that promote CLN3 transcription, in which case inactivation of the repressor would not be sufficient to rescue CLN3 mRNA levels in rts1<sup>Δ</sup> cells. Finally, PP2A<sup>Rts1</sup> could regulate multiple repressors of CLN3 transcription. Consistent with this, we found evidence that PP2A<sup>Rts1</sup> controls Yox1, another repressor of CLN3 transcription (Pramila et al., 2002; Bastajian et al., 2013). The CLN3 gene has an unusually large 5′ untranslated region and is subject to complex regulation (Polymenis and Schmidt, 1997).

What is the kinase that hyperphosphorylates Ace2 in rts1<sup>Δ</sup> cells? Previous work found that Ace2 is phosphorylated by Cbk1, which drives asymmetric localization of Ace2 into daughter cell nuclei (Mazanka et al., 2008). There is also evidence that Cbk1 controls additional Ace2 functions (Mazanka et al., 2008). The MS identified a Cbk1 target site (S122), and Western blotting with a phosphospecific antibody confirmed that the site is hyperphosphorylated in rts1<sup>Δ</sup> cells. However, purified Cbk1 did not appear to be capable of phosphorylating Ace2 in vitro to the same extent observed in vivo in rts1<sup>Δ</sup> cells. In addition, the MS identified numerous sites that have not been attributed to Cbk1. Thus, it is likely that at least one additional kinase regulates Ace2. We found no clear evidence that hyperphosphorylation of Ace2 in rts1<sup>Δ</sup> cells is caused by Cbk1. Moreover, the rts1<sup>Δ</sup> phenotype is not consistent with hyperphosphorylation of Ace2 on mitotic Cdk1 sites because previous work suggests that this should lead to constitutive cytoplasmic localization of Ace2, where it could not repress CLN3 transcription (O’Conallain et al., 1999; Sbia et al., 2008).

PP2A<sup>Rts1</sup> could act directly on Ace2, or it could act further upstream to inhibit a kinase or activate a phosphatase that acts on Ace2. We found that purified PP2A<sup>Rts1</sup> was not able to dephosphorylate Ace2; however, we could not demonstrate that the PP2A<sup>Rts1</sup> was active, so the experiment was inconclusive.

**PP2A<sup>Rts1</sup> may contribute to gradual Cln3 accumulation during growth of small cells**

Cln3 has been difficult to detect by Western blotting so its behavior during the cell cycle has been little characterized. We used highly sensitive Western blotting techniques to gain an unprecedented view of Cln3 protein during the cell cycle. This revealed that Cln3 shows significant periodic oscillations. One peak of Cln3 occurs in G1, as expected for a G1 cyclin, and a second peak occurs as the mitotic cyclin Cib2 reaches peak levels. The functions of Cln3 during mitosis are unknown.

We also assayed Cln3 and Ace2 during growth of newborn daughter cells. An important hypothesis for cell size control suggests that Cln3 levels are proportional to cell growth or size and that cell cycle entry is triggered when Cln3 levels reach a threshold (Jorgensen and Tyers, 2004; Turner et al., 2012). However, Cln3 has never been assayed in growing newborn daughter cells to test this model. We found that Cln3 was initially absent in newborn daughter cells and then accumulated gradually during growth. Cln3 reached peak levels at ~90 min, and bud emergence began 40 min later. The striking correlation between Cln3 protein levels and cell growth is consistent with the Cln3 threshold model. The delay between peak Cln3 levels and bud emergence may correspond to a previously described size-independent delay in G1 that occurs before bud emergence (Di Talia et al., 2007, 2009). Alternatively, the delay may reflect additional size-dependent mechanisms that regulate the activity or localization of Cln3 (Vergès et al., 2007). Mechanisms that restrain the ability of CLN3 to trigger cell cycle entry during early daughter cell growth may play a role in setting a size threshold.
Ace2 was hyperphosphorylated in newborn daughter cells and underwent gradual dephosphorylation during cell growth, reaching maximal dephosphorylation at approximately the same time that Cln3 protein reached peak levels. This observation suggests the possibility that the extent of Ace2 dephosphorylation could help set the level of CLN3 transcription.

Cell size checkpoints must translate a parameter related to growth into a proportional checkpoint signal that can be read by downstream components to determine when sufficient growth has occurred. The nature of the proportional checkpoint signal is one of the central enigmas of cell size control. The discovery that Ace2 phosphorylation is proportional to growth suggests that it may respond to a proportional checkpoint signal. Moreover, the central role of PP2A^{Rts1} in control of both Ace2 phosphorylation and cell size suggests that it could be responsible for generating or relaying a proportional checkpoint signal to Ace2, thereby ensuring that Cln3 levels are proportional to growth. PP2A^{Rts1} could also play a role in setting the threshold.

In this case, poor nutrients could regulate PP2A^{Rts1} to lower the threshold, thereby allowing cells to go through the cell cycle at a reduced cell size. This kind of model could explain the puzzling observation that cells growing in poor nutrients enter the cell cycle at a reduced cell size. This kind of model could explain the puzzling observation that cells growing in poor nutrients enter the cell cycle at a reduced cell size.
cell cycle at a reduced size, despite having reduced levels of Cln3 (Hall et al., 1998; Newcomb et al., 2003). If poor nutrients reduce the threshold, lower levels of Cln3 would be required for cell cycle entry. Further analysis of the targets of PP2A<sup>Rts1</sup>, as well as the signals that control PP2A<sup>Rts1</sup>, will likely provide important new clues to how cell division is linked to cell growth.

**Materials and methods**

**Yeast strains, culture conditions, and plasmids**

All strains are in the W303 background [leu2-3,112 ade2-1 his3-11,15 trp1-1 GAL+ ssd1-d2]. The genotypes of the strains used for this study are listed in Table 2. Full-length CLN3 was expressed from the GAL1 promoter using the integrating plasmid pDK93A (GAL1-CLN3 URA3). One-step PCR-based gene replacement was used for construction of deletions and epitope tags at the endogenous locus. Strains that contain GAL1-CDCC20 strains were made by genetic crosses (Bhoite et al., 2001) or by using a PCR-based approach to integrate the GAL1 promoter in front of the endogenous CDCC20 gene in the appropriate background.

Cells were grown in YEPD media (1% yeast extract, 2% peptone, and 2% glucose) overnight at room temperature. The columns were washed with 6 ml of 1% FA, and the peptides were eluted with 1 ml of 70% acetonitrile (ACN) and 1% FA. Equal amounts of wild-type (light) and rts1Δ (heavy) peptides were combined and dried in a SpeedVac (Savant).

**Phosphopeptide enrichment by SCX/TiO<sub>2</sub>**

Phosphopeptides were enriched by using a modified version of the two-step, SCX-immobilized metal affinity chromatography/TiO<sub>2</sub> protocol using step elution from self-packed solid-phase extraction SCX chromatography cartridges as previously described with some changes (Villén and Gygi, 2008; Dephure and Gygi, 2011). Peptides were resuspended in 1 ml SCX buffer A (7 mM KH<sub>2</sub>PO<sub>4</sub>, pH 2.65, and 30% ACN) and loaded onto precolumn syringe-barrel columns packed with 500 µg of 20-µm, 300-A PolySULFOETYL A resin (PolyLC). Peptides were eluted by the sequential addition of 3 ml SCX buffer A containing increasing concentrations of KCl. 12 fractions were collected after elution with 0 (flow through), 10, 20, 30, 40, 50, 60, 70, 80, 90, 100, and 200 mM KCl. All fractions were frozen in liquid nitrogen, lyophilized, resuspended in 1 ml of 1% FA, and desalted on 50-µg SepPaks. Peptides were eluted with 500 µl of 70% ACN and 1% FA. 5% of each fraction was taken off for protein abundance analysis.

The remaining peptides were dried in a SpeedVac. TiO<sub>2</sub> enrichment was performed by either of two protocols. For replicate one, peptides from fractions 8–11 were pooled after desalting (fraction 12 was not used for phosphopeptide analysis). Dried peptides were resuspended in 50 µl of wash/blinding buffer (30% ACN, 1% FA, and 70 mM glutamic acid) and incubated with 500 µg of Titaneufor TiO<sub>2</sub> beads (GL Sciences) with vigorous shaking for 60 min at room temperature. The beads were washed three times with 200 µl of wash/blinding buffer and once with 1% FA. Phosphopeptides were eluted in two steps by sequential treatments with 50 µl of 0.5 M KH<sub>2</sub>PO<sub>4</sub>, pH 7.5. The eluates were acidified by the addition of FA to 1% final concentration, desalted on Stage tips (Rappsilber et al., 2003), and dried in a SpeedVac. Eight fractions corresponding to 0, 10, 20, 30, 40, 50, 60, 70, 80, 90, and 200 mM KCl. Fractions 9–11 were pooled before TiO<sub>2</sub> enrichment. TiO<sub>2</sub> phosphopeptide enrichment was performed using a modified protocol (Kettenbach and Gerber, 2011) using 2 mg TiO<sub>2</sub> resin for each fraction and a wash/blinding buffer composed of 50% ACN and 2 M lactic acid.

**MS**

Phosphopeptides samples were analyzed on a mass spectrometer (LTQ Orbitrap Velos; Thermo Fisher Scientific) equipped with a quadrupole pump (Accla 600; Thermo Fisher Scientific) and a microtrosamplar (Famos; LC Packings). Nanospray tips were hand pulled using 100-µm inner diameter fused-silica tubing and packed with 0.5 cm of Magic C4 resin (5 µm and 100 Å; Michrom BioResources) followed by 20 cm of Magic C18AQ resin (3 µm and 200 Å, Nest Group). Peptides were separated using a gradient of 3–28% ACN in 0.125% FA over 70 min with an in-column flow rate of ~300–500 nL/min.

Peptides were detected using a data-dependent top 20 MS2 method. For each cycle, one full MS scan of mass per charge (m/z) = 300–1,500 was acquired in the Orbitrap at a resolution of 60,000 at m/z = 400 with automatic gain control target = 10<sup>5</sup> and a maximum ion accumulation time of 500 ms. Each full scan was followed by the selection of the most intense ions, up to 20, for collision-induced dissociation and MS2 analysis in the LTQ. An automatic gain control target of 2 x 10<sup>4</sup> and maximum ion accumulation time of 150 ms were used for MS2 scans. Ions selected for MS2 analysis were excluded from analysis for 60 s. Precursor ions with charge = 1 or unassigned were excluded from selection for MS2 analysis. Lock mass, using atmospheric polydimethylsiloxane (m/z = 445.120025) as an internal standard, was used in all runs to calibrate Orbitrap MS precursor masses. For replicate 1, eight fractions were analyzed once each. For replicates 2 and 3, sufficient material was recovered to shoot samples in duplicate for MS2 scans. For replicate 2, all nine fractions were analyzed once each. For replicates 2 and 3, fractions 1–6 were analyzed in duplicate, whereas fractions 7–9 were analyzed once. For protein abundance analysis, 5% of each SCX fraction was removed before phosphopeptide enrichment, desalted on a Stage tip, resuspended in 5% FA, and analyzed in a single run for each fraction as described in the previous paragraphs for phosphopeptides but using a 90-min gradient of 3–25% buffer B and 75 s dynamic exclusion.
To synchronize cells at metaphase, cells containing GAL1-CDCC20 were grown overnight in YEP media containing 2% raffinose and 2% galactose. Cells were arrested by washing into media containing 2% raffinose and incubated at room temperature for 4 h. Cells were released from the metaphase arrest by adding 2% galactose and were then shifted to 30°C for Western blotting experiments or 25°C for CLN3 mRNA analysis.

For induced expression experiments, cells were grown overnight in YEP medium containing 2% glucose and 2% galactose and addition of genes from the GAL1 promoter was induced by addition of 2% galactose, and the cells were shifted to 30°C.

For time courses using analogue-sensitive alleles, cells were grown overnight in YEDP without adenine. The adenine analogue inhibitor 1NM-PP1 was added to log phase cells at a final concentration of 25 µM, and the cells were then shifted to 30°C.

To analyze log phase cells, cultures were grown in YEDP, YEPD + 2% galactose, or YEPD + 2% glycerol/ethanol overnight at room temperature. 1.6 ml of cells at an OD600 of 0.6 were collected and centrifuged at 13,000 rpm for 30 s, the supernatant was removed, and 250 µl of glass beads were added before freezing in liquid nitrogen.

Western blotting
To collect samples for Western blotting, 1.6-ml samples were collected at each time point and centrifuged at 13,000 rpm for 30 s. The supernatant was removed, and 250 µl of glass beads were added before freezing in liquid nitrogen. Cells were lysed using 140 µl of sample buffer (65 mM Tris-HCl, pH 6.8, 3% SDS, 10% glycerol, 50 mM NaF, 100 mM β-glycerophosphate, 5% 2-mercaptoethanol, and bromophenol blue). PMSF was added to the sample buffer to 2 mM immediately before use. Cells were lysed in a Multibea-8 at top speed for 2 min. The samples were removed and centrifuged for 15 s at 13,000 rpm in a microfuge and placed in boiling water for 5 min. After boiling, the samples were centrifuged for 5 min at 13,000 rpm and loaded on an SDS polyacrylamide gel.

To assay phosphorylation of Ace2 serine 122, cells were grown overnight in YEPD media at room temperature to OD600 = 0.6. Cells from 50 ml of culture were pelleted, resuspended in 1 ml of ice-cold 50 mM Hepes, pH 7.6, and pelleted in a wide-bottomed 1.6-ml screw tap tube. Cells were lysed by bead beating in 600 µl lysis buffer (50 mM Tris-HCl, pH 7.5, 150 mM NaCl, 1% Triton X-100, 10% glycerol, 120 mM β-glycerophosphate, and 2 mM sodium orthovanadate) containing 2 mM PMSF, 1 mM leupeptin, 1 mM chymostatin, 1 mM pepstatin, and 20 µM cantianin. Ace2-3×HA was immunoprecipitated with mouse monoclonal 12CA5 antibody. Western blots were probed with rabbit anti-phospho-S122 antibody (gift of E. Weiss, Northwestern University, Evanston, IL) as previously described, except that the antibody was used at a dilution of 1:3000 in TBST (10 mM Tris-Cs, pH 7.5, 100 mM NaCl, and 0.1% Tween 20) containing 4% BSA (Mazanek and Weiss, 2010).

SDS-PAGE was performed as previously described (Harvey et al., 2005). Gels were run at a constant current of 20 mA. For Ace2, Cib2, and Cln3, electrophoresis was performed on 10% polyacrylamide gels until a 29-kD prestained marker ran to the bottom of the gel. Protein was transferred to nitrocellulose membranes for 1.5 h at 800 mA at 4°C in a transfer tank (TE22; Hoeffer) in buffer containing 20 mM Tris base, 150 mM glycine, and 20% methanol. Blots were probed overnight at 4°C with affinity-purified rabbit polyclonal antibodies raised against Ace2, Cib2, or the HA peptide or mouse monoclonal antibody against the HA peptide. Cln3-6×HA blots were probed with the 12CA5 anti-HA monoclonal antibody. All blots were probed with an HRP-conjugated donkey anti-rabbit secondary antibody (GE Healthcare) or HRP-conjugated donkey anti-mouse antibody for 45–90 min at room temperature. Secondary antibodies were detected via chemiluminescence with Advansta ECL or Quantum reagents.

RNA analysis
qRT-PCR was used to measure CLN3 mRNA levels as described using RPR1 RNA as the internal control (CLN3 primers: 5′-CAGCGGATCAG-CGAATCAAATAA-3′ and 5′-TGATAATAAGACGCGAGAAGG-3′; RPR1 control primers: 5′-CAACTGCAGGGGCGGCTATCGA-3′ and 5′-CCATGG-CGGCAGACTCCGTG-3′; Usami et al., 2007). Northern blotting probes for CLN3 and ACT1 RNA were made using gel-purified PCR products (CLN3 oligonucleotides: 5′-GCCGGATCCGTGGCAGAGTATGTTCT-3′ and 5′-CGGGACCTCAGCCAGGTTTGCAGG-3′). Probes were labeled using the DNA labeling kit (Megaprime; GE Healthcare). RNA for Northern blotting was isolated as previously described (Cross and Tinkelenberg, 1991; Kellogg and Murray, 1994).
1995). In brief, 1.6-mL samples were collected at each time point and centrifuged for 30 s at 13,000 g. The supernatant was removed, and 200 \( \mu l \) of acid-washed beads was added before freezing in liquid nitrogen. 350 \( \mu l \) NETS buffer (0.3 M NaCl, 1 mM EDTA, 10 mM Tris-HCl, pH 7.5, and 0.2% SDS) and 350 \( \mu l \) phenol/chloroform were added to each cell pellet, and cells were lysed by shaking in a Multibeads-8 at full speed for 2 min. Samples were then centrifuged at 15,000 g for 5 min, and 300 \( \mu l \) of the aqueous phase was transferred to a new tube. RNA was ethanol precipitated and resuspended in TE buffer (10 mM Tris-HCl, pH 8.0, and 1 mM EDTA) containing 0.2% SDS followed by incubation at 65°C for 10 min. The samples were run in a 1% formaldehyde-agarose gel at 5 V/cm². The same fraction of total extracted RNA was loaded at each time point. This typically corresponded to 8 \( \mu g \) RNA at the 0 time point. CIN3 blots were stripped and reprobed for ACT1 to control for loading.

ChIP assays

ChIPs were performed as previously described (Yoth et al., 2007). Yeast cells were collected at an OD of 0.6–0.8 and cross-linked in 1% formaldehyde for 20 min at room temperature. Cross-linking was quenched with 0.125 M glycine for 5 min, and cells were washed twice with TBS. Cell pellets were resuspended in lysis buffer (0.1% deoxycholic acid, 1 mM EDTA, 100 mM NaCl, pH 7.5, 140 mM NaCl, and 1% Triton X-100 supplemented with 0.8 mM DTT and protease inhibitors) and were lysed with 0.5-mm zirconia beads (BioSpec) in a cell disruptor (Mini-Beadbeater; Bio-Spec). After crosslinking, the pellet was washed with lysis buffer and sonicated to a shearing size of <500 nucleotides using a bath sonicator (Biorupter Xi; Diagonode). The sonicated material was centrifuged, and the supernatant containing the chromatin was quantitated by Bradford assay. Immunoprecipitations were performed overnight at 4°C using 500–700 \( \mu g \) of chromatin, 4\( \times \) monoclonal antibody to the Myc epitope (EMD Millipore), and Pan–Mouse Dyna Beads IgG (Invitrogen). The beads were washed twice with lysis buffer, twice with high salt buffer (lysis buffer with 500 mM NaCl), twice with LCI buffer (0.5% deoxycholic acid, 1 mM EDTA, 250 mM UCl, 0.5% NP-40, and 10 mM Tris-HCl, pH 8.0), and once with TE. Cross-links were reversed overnight in elution buffer (10 mM EDTA, 1% SDS, and 50 mM Tris-HCl, pH 8.0) at 65°C. DNA was purified using the PCR purification kit (QiAquick; QIAGEN). Quantitative PCR reactions were performed using a detection system (LightCycler480 II; Roche). A standard curve representing a range of concentrations of input samples was used for quantitating the amount of product for each sample with each primer set. All ChIP samples were normalized to corresponding input control samples, to a genomic reference region on chromosome I, and to a genetically unrelated region on chromosome III. All ChIP samples were normalized to corresponding input control samples, to a genomic reference region on chromosome I, and to a genetically unrelated region on chromosome III. All ChIP samples were normalized to corresponding input control samples, to a genomic reference region on chromosome I, and to a genetically unrelated region on chromosome III.


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