Antisense transcription-dependent chromatin signature modulates sense transcript dynamics

Thomas Brown†, Françoise S Howe‡, Struan C Murray†, Meredith Wouters, Philipp Lorenz‡, Emily Seward‡, Scott Rata, Andrew Angel* & Jane Mellor**

Abstract

Antisense transcription is widespread in genomes. Despite large differences in gene size and architecture, we find that yeast and human genes share a unique, antisense transcription-associated chromatin signature. We asked whether this signature is related to a biological function for antisense transcription. Using quantitative RNA-FISH, we observed changes in sense transcript distributions in nuclei and cytoplasm as antisense transcript levels were altered. To determine the mechanistic differences underlying these distributions, we developed a mathematical framework describing transcription from initiation to transcript degradation. At GAL1, high levels of antisense transcription alter sense transcription dynamics, reducing rates of transcript production and processing, while increasing transcript stability. This relationship with transcript stability is also observed as a genome-wide association. Establishing the antisense transcription-associated chromatin signature through disruption of the Set3C histone deacetylase activity is sufficient to similarly change these rates even in the absence of antisense transcription. Thus, antisense transcription alters sense transcription dynamics in a chromatin-dependent manner.

Keywords antisense transcription; chromatin; sense transcript dynamics; Set3C lysine deacetylase; stochastic model

Introduction

The transcription of genomes is not limited to the transcription of genes alone. Transcription is a universally pervasive and interleaved process, with transcription events initiating from regulatory sequences such as enhancers, divergently from gene promoters, and on the antisense strand of genes (Tisseur et al, 2011; Lam et al, 2014; Mellor et al, 2016; Murray & Mellor, 2016). Nascent transcripts at enhancers or around promoters can be used to recruit (Battaglia et al, 2017) and activate (Bose et al, 2017) epigenetic modifiers associated with chromatin and to activate neighbouring genes in a cell type-specific manner (Werner et al, 2017). This may explain why some chromatin modifications only appear after transcription has initiated (Howe et al, 2017). In addition to transcripts, co-transcriptional processes also influence chromatin modifications in genomes. Transcription of many non-coding transcripts uses a form of RNA polymerase II (RNAPII) that is depleted for conserved features normally associated with efficient transcription elongation including serine 2 phosphorylation of the C-terminal domain (CTD) on the largest RNAPII subunit, the H3K36 methyltransferase Set2 and the elongation factor Paf1 (Murray et al, 2015; Fischl et al, 2017). One such class of non-coding transcripts contains the nascent transcripts transcribed from the antisense strand of the gene, which are often rapidly degraded by exonucleases (He et al, 2008; Neil et al, 2009; van Dijk et al, 2011). Although antisense transcription within genes is a consistent feature of eukaryotic genomes (Mellor et al, 2016), it is not known whether it is simply a by-product of gene transcription, whether there are consequences of antisense transcription and, if so, whether these are conserved across species. Much effort has been expended to determine the function(s) associated with antisense transcription. For a small number of yeast genes, sense and antisense transcription appear to suppress one another and/or be reciprocally regulated (Hongay et al, 2006; Camblong et al, 2007; Houseley et al, 2008; Castelnovo et al, 2013). However, there is no obvious global relationship between sense and antisense transcription, as levels at the same gene are not correlated genome-wide, either positively or negatively (Murray et al, 2015) and a recent study found that, at the protein level, gene expression is unaffected by lowering levels of antisense transcription in the majority of the 162 genes studied (Huber et al, 2016).

As antisense transcription often proceeds into the sense promoter of its associated gene (Xu et al, 2011; Mayer et al, 2015) and does not appear to be contemporaneous with sense transcription (Castelnovo et al, 2013; Nguyen et al, 2014), we previously hypothesized that antisense-transcribing RNAPII might indirectly influence sense transcription by modulating the chromatin environment in the vicinity of the sense promoter. Thus, one round of
antisense transcription would be sufficient to leave an epigenetic signature and influence sense transcription. We identified in yeast a chromatin signature at the sense promoter and in the early coding region unique to genes with high levels of antisense transcription: high levels of nucleosome occupancy leading to a reduced nucleosome-depleted region (NDR), high histone H3 lysine acetylation and histone turnover, but low levels of histone H3 lysine 36 tri-methylation (H3K36me3), H3K9me3 and H2BK123 mono-ubiquitination, amongst others (Murray et al., 2015). Some of these features have been found associated with antisense transcription in mammals (Lavender et al., 2016). Conservation of chromatin features associated with antisense transcription between yeast and mammals will enable us to apply anything we learn in yeast about the mechanistic consequences of antisense transcription more broadly. Here we address the question of how antisense transcription influences sense transcription using a stochastic model of transcription and quantitative data from a single-molecule approach, RNA fluorescence in situ hybridization (RNA-FISH). This allows for the best understanding of the effects of antisense transcription on the dynamics of sense transcript production and processing at the individual cell level. We model RNA-FISH data obtained from engineered constructs expressing high or low levels of antisense transcription, but the same level of sense transcripts, thus mimicking the commonly reported situation where antisense transcription has little effect on steady-state transcript levels. We show that antisense transcription decreases rates of transcript production and processing while increasing transcript stability and, importantly, that these changes in transcription dynamics are directly influenced by the antisense-dependent chromatin signature. As we reveal a remarkably conserved chromatin architecture around the sense promoter and early transcribed region of yeast and human genes with antisense transcription, despite large differences in gene size, we suggest that the effect of antisense transcription is likely to be conserved between yeast and human genes.

Results

A conserved arrangement of sense and antisense transcription start sites in yeast and human genes

To address whether and how antisense transcription is conserved across species, it was necessary to map genic transcription start sites (TSSs) as either sense sites (sTSS), or antisense sites (aTSS), depending on their orientation relative to their proximal gene, and the extent of sense and antisense transcription downstream of these TSSs (Fig 1A). As many antisense transcripts are unstable, we used data from nascent transcript mapping techniques such as NET-seq (Churchman & Weissman, 2011; Nojima et al., 2015), PRO-seq (Booth et al., 2016) or GRO-seq (Core et al., 2008) to assess genomewide levels of transcription in Saccharomyces cerevisiae and HeLa cells. To map TSSs, we used Cap Analysis of Gene Expression (CAGE) data for HeLa cells (FANTOM Consortium et al., 2014), pooling the polyadenylated and non-polyadenylated tag data from nuclear, cytoplasmic and whole cell fractions, and TIF-seq for yeast (Pelechano et al., 2013), supplemented with data from cryptic unstable transcripts (Neil et al., 2009) and stable unannotated transcripts (Xu et al., 2009). From over 20,000 protein-coding genes, we identified 9,320 with a sTSS in HeLa cells. Of these genes, we found 2,468 (27%) with an internal aTSS; 1,008 (40%) of these aTSSs were within 500 bp of the sTSS, with a median distance of 632 bp (Fig 1B). Thus, a large fraction of active genes in HeLa cells show evidence of a productive, antisense-oriented transcription start site close to their promoter. We defined 5,222 yeast genes with a sTSS, of which 1,529 (29%) had an aTSS. The median distance between the sTSSs and aTSSs of yeast genes was 884 bp, 252 bp larger than in humans (Fig 1C).

Strikingly, in humans, the aTSS aligned more closely to the 1st exon–intron boundary than the sTSS, and with a much higher frequency than expected if aTSSs are randomly re-distributed over this region (Fig 1D). In fact, in 2,162 (88%) genes with antisense transcripts, the aTSS is closer to the 1st exon–intron boundary than it is to the sTSS. Using the same approach, we also found that the aTSS aligned much more tightly with the 1st exon–intron boundary than it did with the 2nd exon–intron boundary (Fig 1E) or to the 3' end (Fig 1F). In yeast, antisense transcription tends to initiate from the vicinity of the 3' region of genes (Fig 1G; Xu et al., 2011) rather than from introns (Fig 1H); 1,202 (79%) genes with an antisense transcript had an aTSS closer to their 3' end than to their sTSS (Fig 1G and I). Despite their distinct sites of origin in humans and yeast (1st intron–exon boundary and 3' end, respectively; Fig 1D, G, and I) and gene size (Fig 1J), the aTSS is at a similar median distance to the sTSS in humans and yeast (632 bp compared to 884 bp), suggesting a conserved arrangement.

Higher nucleosome occupancy at promoters of genes with high antisense transcription in yeast and humans

To examine how antisense transcription influences the chromatin and sense transcription in the vicinity of the promoter, we assessed three regions: 300 nucleotides upstream of the sTSS (the sense promoter), 300 nucleotides downstream of the aTSS (the antisense promoter) and the region between the two TSSs, which was broken into an equal number of bins. We compared the upper and lower quintiles of genes with an aTSS, giving us two groups of 494 genes in humans, and 306 and 307 genes in budding yeast. Firstly, we compared levels and distributions of sense and antisense transcription in the three regions using NET-seq, GRO-seq or PRO-seq, which are similar when comparing HeLa cells with yeast and the different techniques (Figs 2A, and EV1A and B). From this point, in Figs 2B–E and 3, data are related to profiles for NET-seq, while the PRO-seq and GRO-seq profiles are in Figs EV1 (relates to Fig 2) and EV2 (relates to Fig 3). Next, we examined a genome-wide map of nucleosome occupancy (MNase-seq). Both species had a NDR at the sTSS and a marked increase in nucleosome occupancy in the vicinity of the aTSS in genes with the highest levels of antisense transcription (Figs 2B, and EV1C and D). This suggests that antisense transcription may modulate promoter chromatin in both species without necessarily altering levels of sense transcription in the vicinity of the sense promoter (Figs 2C and D, and EV1E, F, and G). Indeed, nucleosome occupancy and sense transcription may well be disconnected (Nocetti and Whitehouse, 2016), despite the apparent association between sense transcription and nucleosome depletion at the promoter (Fig 2B). Nucleosomes also interact with one another in local space, one component of the chromatin conformation in the nucleus (Hsieh et al., 2015). To see whether the antisense-associated increase in nucleosome occupancy is related to the
We used Micro-C data in yeast, which identifies chromosomal contacts at the resolution of nucleosomes (Hsieh et al., 2015). For the 5,222 yeast genes that had an associated sense transcript as described above, we determined the level of gene compaction, as defined by Hsieh et al. (2015), normalized such that it is independent of gene length. Strikingly, we found an inverse association between intragenic contacts and antisense transcription. Genes with an antisense transcript showed a significantly reduced level of gene compaction, regardless of the level of sense transcription ($P = 1.2 \times 10^{-65}$, Wilcoxon rank sum test, Fig 2E), suggesting that antisense transcription favours a looser higher order structure. Varying sense transcription results in no significant change in the level of compaction ($P = 0.29$, 0.42, Wilcoxon rank sum test, Fig 2E).

**Figure 1. Antisense transcripts initiate at a similar distance from the sense TSS in yeast and humans, though from a distinct functional site.**

A. A schematic demonstrating how antisense TSSs (asTSSs) were defined in this study. Blue arrows represent possible sites of antisense transcript initiation, within the region inspected for the presence of asTSSs, as defined by CAGE.

B. The distribution of distances between stSS and asTSS in HeLa cells, for those 2,468 genes which had both an upstream stSS and an internal asTSS defined by CAGE. Shown is the median distance between the stSS and asTSS.

C. The distribution of distances between the stSS and asTSS in Saccharomyces cerevisiae (budding yeast), for those 1,529 genes that had both an overlapping sense and antisense transcript, defined by TIF-seq.

D. The position of the asTSS relative to the stSS and the end of the 1st exon in HeLa cells, to demonstrate which of the two points the asTSS aligns to preferentially. This position was defined as the distance between the stSS and the asTSS, divided by the distance between the stSS and the end of the 1st exon. The genes are the same as those shown in (B). The dotted line represents the average distribution from a thousand simulations, in which asTSSs for each gene were randomly reassigned to a base pair within the region shown.

E. The position of the asTSS relative to the end of the 1st exon and the end of the 2nd exon, for those HeLa genes in (D) that also had a second exon.

F. The distribution of distances between the 3′ end of genes and asTSS in HeLa cells, for the same genes in (B).

G. The position of the asTSS relative to the stSS and the 3′ end of the open reading frame of the 1,529 S. cerevisiae genes that have both an overlapping sense and antisense transcript. The dotted line was generated as in (D).

H. The position of the asTSS relative to the end of the 1st exon and the 3′ end of the open reading frame of those S. cerevisiae genes in (G) that also have an intron.

I. The distribution of distances between the 3′ end of S. cerevisiae genes and asTSS in, for the same genes in (C).

J. The distribution of lengths for human 1st exons, human genes, and yeast genes.
**Antisense transcription is associated with a similar unique chromatin signature in yeast and humans**

We next turned our attention towards histone modifications (Fig 3, NET-seq; Fig EV2, PRO-seq and GRO-seq; Appendix Fig S1). Levels of H3K36me3 and H3K79me3 were higher in the region bounded by the sTSS and atTSS for those genes with high sense transcription, in both species (Fig 3A; Ng et al, 2003; Pokhrolok et al, 2005). Strikingly, however, these two modifications are much lower in those genes with high levels of antisense transcription, in both humans and yeast (Fig 3A). This is despite the fact that the level of sense transcription is the same in the high/low antisense classes (see Figs 2C and D, and EV1E, F, and G). That these modifications should have reverse associations with sense and antisense in both yeast and humans is intriguing, and suggests there may be some fundamental difference to the two modes of transcription that is shared across species. By contrast, levels of H3K4me3 tended to be more evenly spread between the sTSS and the atTSS in genes with high antisense transcription compared to high sense transcription (Fig 3B). Levels of H3K4 lysine monomethylation (H3K4me1) tended to be lower with high sense or antisense transcription showing that not all modifications have reciprocal patterns with sense or antisense transcription. Finally, levels of H3 acetylation are increased in the presence of antisense transcription (Fig 3C), particularly in the region downstream of the sense TSSs.

Taken all together, one can see that despite the vast differences in size between yeast and human genes, they share a very similar arrangement in terms of where their antisense transcripts initiate relative to their coding-transcript start site, and in how antisense transcription associates with numerous shared chromatin features. We conclude that antisense transcription in the vicinity of the sense promoter is associated with increased histone lysine acetylation and nucleosome occupancy, and decreased histone H3K36me3, H3K79me3 and chromatin compaction, and that this unique architecture is conserved between yeast and humans.

Is there a consequence of this widespread and conserved antisense transcription initiating downstream from the sense promoter for the genes in yeast and humans that have it? How might it be changing gene behaviour? To address this, we developed a mathematical model that describes the dynamics of transcription and allows us to discriminate between transcriptional events at the sense promoter, the nucleus and the cytoplasm. When compared with experimental data, the model allows us to determine which parameters of sense transcription production and processing are affected by antisense transcription.

**A stochastic model for transcription**

Our stochastic model of transcription captures the production, processing and destruction of a transcript (Fig 4A) and builds on existing models of transcription (Raj et al, 2006; Zenklusen et al, 2008; Choubey et al, 2015). Within the model, a gene promoter is allowed to switch between an active and inactive state stochastically, with an activation rate $\alpha$ and an inactivation rate $\beta$. In the active state, transcription initiation occurs with rate $\gamma$. As a result, the “mean production rate”, that is the average rate of transcript initiation, is given by $\gamma/(\alpha + \beta)$.

Nuclear transcript processing, meanwhile, is modelled as a sum of reactions representing the advancement of RNAPII across the DNA as a series of stochastic jumps. The time to fully process a nuclear transcript is distributed as a sum of $N$ exponentials, corresponding to gene length, with parameter $k$ corresponding to elongation rate, $\Gamma(N,k)$. Our experimental protocol does not allow for the separation of nascent and nuclear transcripts; therefore, in our modelling framework, we do not differentiate between the two types of transcripts. As a result, the parameter $k$ is a conflation of both elongation rate and nuclear export rate. We refer to this parameter as the “nuclear processing rate”, representing the time for a transcript to go from initiation to export. Finally, transcripts are assumed to degrade in the cytoplasm with a constant half-life, decaying exponentially with degradation rate $\delta$.

We used this model to study the transcription dynamics of the inducible yeast gene GAL1 (Fig 4B). The first strain (high AS) contains an engineered form of the GAL1 gene that expresses a stable antisense transcript as a result of insertion of the ADH1 transcription terminator (GAL1:ADH1t; Murray et al, 2012, 2015). In the second strain (low AS), a 6-bp AT rich sequence within the inserted terminator region is scrambled (while retaining the overall base composition), resulting in a significant reduction in levels of the antisense transcript (Fig 4C), which is a consequence of reduced levels of antisense transcription (Murray et al, 2015), but no change in levels of sense transcripts (Fig 4D).

Two types of data were used to parameterize the dynamics of transcription. Firstly, we obtained the rate of degradation of cytoplasmic sense transcripts (Fig 4E), relying on the galactose-inducible and glucose-repressible nature of GAL1. We grew cells in galactose-containing media, then recorded the decreasing concentration of sense mRNA via Northern blot at multiple timepoints after cells were moved to glucose-containing media. By fitting an exponential curve to the data sets, we obtained the degradation rate $\delta$, which gives the half-life via the formula: $t_{1/2} = \ln 2/\delta$.

Secondly, we obtained the distribution of individual sense transcripts in the nucleus and cytoplasm of cells using RNA-FISH (Fig 4F). We probed cells grown in galactose-containing media for 2 h for the GAL1 sense transcript and counted the number of fluorescent foci within the nucleus and cytoplasm. Individual dots were assumed to represent at least one transcript, with the number of transcripts at a given dot determined by dividing the intensity of the dot by the median intensity of all foci. Several hundreds of cells were considered for a given experiment, and the distributions of nuclear and cytoplasmic transcript counts were obtained (Fig 4G). The nuclear distribution gives an indication of mean initiation rate relative to nuclear processing rate, or the fraction $\gamma/(\alpha + \beta) k$. The cytoplasmic distribution, correspondingly, tells us the mean initiation rate relative to degradation rate, or $\gamma/(\alpha + \beta) \delta$. Using the measured degradation rate $\delta$, we find the parameters that best fit the RNA-FISH data via the Kolmogorov-Smirnov test, sampling 1,000,000 parameter sets via Latin Hypercube (McKay et al, 1979) and sampling the 1,000 parameter sets with the best Kolmogorov-Smirnov statistic. The parameters obtained by simulation are then used to determine the mean initiation rate $\gamma/(\alpha + \beta)$ and nuclear processing rate $k$. The 1,000 best parameter sets obtained from the cytoplasmic data give a probability distribution for the expected true value of the parameter. The nuclear data show the likely corresponding ratio of mean initiation rate to nuclear
Figure 2. Antisense transcription is associated with changes in chromatin structure in both yeast and humans, but not with changes to the level of sense transcription.

A. The average levels of sense and antisense transcription determined for both HeLa and Saccharomyces cerevisiae using NET-seq. HeLa using GRO-seq and S. cerevisiae using PRO-seq. For each trio of panels, the left panel shows average levels around the sTSS, the right panel shows the average levels around the asTSS, and the middle panel shows the average level within thirty equal sized bins within the region bound by the sTSS and asTSS. In all cases, levels of transcription on the sense strand are shown in red, while levels on the antisense strand are shown in blue. Genes considered are those which contained an asTSS, as defined in Fig 1.

B. The average levels of nucleosome occupancy determined for both HeLa and S. cerevisiae using MNase-seq. Panels are grouped in threes and show average levels as in (A). The top panels compare two sets of genes—those with high levels of sense transcription (dark red), and those with low levels (pale red). The bottom panels show those genes with high levels of antisense transcription (dark blue), and those with low levels (pale blue).

C, D. Scatter plots comparing the number of sense and antisense NET-seq reads within the 300 bp window shown, in both HeLa cells and S. cerevisiae, for those genes with both an sTSS and asTSS. Shown for both species is the Spearman correlation coefficient, \( r_s \).

E. Boxplots showing the distribution of gene compaction in different sets of S. cerevisiae genes. Gene compaction was determined by summing the number of intragenic contacts, measured by Micro-C, and dividing by gene length. On each boxplot, the central mark indicates the median, and the bottom and top edges of the box indicate the 25th and 75th percentiles respectively. The whiskers extend to the most extreme data points. The numbers at the bottom of each box plot show the number of genes in that group.

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Figure 3. Antisense transcription has similar associations with chromatin modifications in both yeast and humans. 

A The average levels of H3K36me3 and H3K79me3 relative to levels of sense or antisense transcription in HeLa and Saccharomyces cerevisiae genes. For each trio of panels, the left panel shows average levels around the STSS, the right panel shows the average levels around the aTSS, and the middle panel shows the average level within thirty equal sized bins within the region bound by the STSS and aTSS. Genes considered are selected from those which contained an aTSS, as defined in Fig 1. Shown in red are two sets of genes—those with high levels of sense transcription (dark red), and those with low levels (pale red). Shown in blue are those genes with high levels of antisense transcription (dark blue), and those with low levels (pale blue).

B Average levels of H3K4me3 and H3K4me1, laid out as in (A).

C Average levels of H3K9ac and H3K27ac, laid out as in (A).
Antisense transcription influences rates of sense transcription initiation, transcript processing and sense transcript stability

We generated experimental data using the two strains in which GAL1 was subject to different levels of antisense transcription (Fig 4B). By obtaining transcriptional parameters in these two strains, we were able to compare how antisense transcription influences sense transcription and transcripts. Strikingly, the stability of the engineered GAL1 sense transcripts is higher with greater antisense transcription (t_{1/2} = 13.53 vs. 20.26 min for low vs. high AS; Figs 4E and 5A). Modelling of the RNA-FISH data reveals roles for antisense transcription in controlling the rates of initiation and nuclear processing of GAL1 transcripts/transcripts; both parameters were lower in the construct expressing higher antisense transcription (Figs 5B, and EV3A and B). The mean production rate was 0.425 min^{-1} in the construct with low antisense transcription, but 0.256 min^{-1} in the construct with high antisense transcription. Similarly, the nuclear processing rate (combining elongation and export rates) was reduced from 2.33 to 1.54 min^{-1} in the presence of antisense transcription. Thus, at the engineered GAL1 gene, antisense transcription does not alter overall sense transcript levels (Fig 4D) but does alter the dynamics of sense transcript production, processing and turnover.

Next, we asked if the effect of antisense transcription on sense transcription/transcript dynamics extends to other genes. We show that the effect of antisense transcription on transcript stability is not strictly limited to the engineered GAL1 genes used here (Figs 5C and EV3C). Using four different sources of data, we compared the stability of transcripts produced from 1,529 yeast genes with, and 3,693 without an antisense transcript (Wang et al., 2002; Churchman & Weissman, 2011; Miller et al., 2011; Geisberg et al., 2014). Remarkably, we observed significant increases in stability for sense transcripts produced from genes with an antisense transcript. Finally, we produced and modelled RNA-FISH data for transcript distributions (Fig EV3D) for five genes with varying levels of antisense transcription, modelled that data and expressed the elongation/export rate as a function of mean production rate to account for inherent differences in sense expression levels (Fig 5D). We observed a decrease in transcript processing rate as levels of antisense transcription increases, corroborating our observations at GAL1. Taken together, these data suggest compensating changes in rates of sense transcript production and sense transcript degradation as a result of antisense transcription.

We asked how antisense transcription alters sense transcript dynamics. It is possible that changes in transcript dynamics resulting from antisense transcription are a consequence of altered patterns of histone modification. To this end, we sought to assess whether experimentally modulating histone modifications could recapitulate the effects of changing antisense transcription, focusing on histone H3 lysine acetylation, as genes with high levels of antisense transcription tend to be associated with increased histone acetylation compared to genes with lower levels (see Fig 3; Murray et al., 2015). Final levels of histone acetylation are influenced by rates of acetylation and deacetylation. The histone deacetylase complexes Rpd3L (containing Rpd3, Pho23 and Hst2), Rpd3S (containing Rpd3 and Rco1) and Set3C (containing Set3 and Hst2) decrease overall levels of histone acetylation, control transcript dynamics at a small number of genes, but do not affect global gene expression (Pijnappel et al., 2001; Kim et al., 2012, 2016; Weinberger et al., 2012; Woo et al., 2017). We asked first which of these HDAC complexes control histone acetylation dependent on levels of antisense transcription and second whether one of these HDAC complexes, by its effect on histone acetylation, might modulate the same parameters as antisense transcription.
Figure 4.
Figure 5. Transcription dynamics change with high antisense transcription.

A GALI sense transcript turnover rates from the engineered GALI with high (left) or low (right) antisense transcription. The red lines show the best fit for nine experiments (see Fig 4E for details).

B Histograms showing the mean production rate (left), mean elongation/export rate (middle) and transcript degradation rate (right; error bars for degradation rates are RMSE of linear regression fit to exponential model) for the engineered GALI genes expressing high or low antisense transcription.

C Transcript stability for sense transcripts from 1,529 genes with an asTSS (dark blue) or 3,693 genes without one (light blue). The frequency plot shows the majority of transcripts have higher stability when expressed from genes with an antisense transcript.

D Elongation/export rate expressed relative to mean production rate for five endogenous genes ranked by levels of corresponding levels of antisense transcription in a 300 bp downstream of the antisense TSS as determined by NET-seq.
**SET3 deletion differentially influences levels of H3K9ac at genes that differ by the presence or absence of antisense transcription**

Acetylation of K4, K9 and K14 on histone H3 is higher at genes with higher antisense transcription (Fig EV4A). Next, we asked whether the change in histone lysine acetylation, following gene deletion of specific HDAC components, is different when considering those genes with or without an antisense transcript. Strikingly, following deletion of SET3 and HOS2 (Set3C) and RCO1 (Rpd3S), those 3,693 genes without an antisense transcript show a significantly larger increase in the level of acetylation than those 1,529 genes with an antisense (\( P = 6 \times 10^{-13} \) for set3A, Figs 6A and EV4A). Furthermore, the changes were observed at different regions of the metagene, consistent with where Set3C and Rpd3S are proposed to function (Kim & Buratowski, 2009; Li et al., 2009). Next, we asked whether increased acetylation could be explained by increased antisense transcription in the mutant strains. This was the case for the rco1 mutant (Murray et al., 2015) and so it was excluded from this study, but not for the set3 mutant (Fig EV4B). Thus, Set3C modulates acetylation at genes with low antisense transcription suggesting some redundancy in the effect of deleting SET3 and in the effect of antisense transcription.

**SET3 differentially influences levels of H3K9ac at GAL1 with high or low antisense transcription**

We investigated the effect of SET3 deletion in the presence or absence of antisense transcription at the engineered GAL1. Importantly, Set3C does not affect the levels of the stable antisense transcript at engineered GAL1 (Fig 6B). That there is no change in antisense transcription is confirmed by levels of transcription-associated histone modifications H3K4me2 and H3K4me3 in the four strains, which are low without antisense transcription and higher with antisense transcription and, importantly, do not change when SET3 is deleted (Fig EV4C).

Next, we asked how Set3C influences levels of H3K9ac at engineered GAL1 with high or low antisense transcription (Fig 6C). High levels of acetylated H3K9 in chromatin correlate with antisense transcription (Murray et al., 2015). As expected, the construct with high antisense transcription has higher levels of H3K9ac than the strain with low levels of antisense transcription, despite both constructs producing similar levels of the GAL1 sense transcript when induced (Murray et al., 2015; Fig 6D). On deletion of SET3, we observe a higher level of H3K9ac in the transcribed region in strains with low antisense transcription (Fig 6C). This neatly reproduces what we have observed genome-wide—that SET3 deletion has a greater effect on acetylation levels in the absence of antisense transcription.

We conclude that the effect of SET3 deletion on levels of H3K9 acetylation at the engineered GAL1 gene is unlikely to result from changes to antisense transcription, but from a direct effect on the chromatin. We hypothesize that antisense transcription buffers chromatin against the modulating effects of Set3C during sense transcription. Thus, following deletion of SET3, we would expect the transcription dynamics in the strain with low levels of antisense transcription to resemble those in the strain with high antisense transcription, assuming transcription dynamics are influenced solely by the chromatin.

**Altering levels of histone acetylation recapitulates the effect of antisense transcription on sense transcription dynamics**

RNA-FISH data (Fig EV4D) and degradation rates (Fig 6E) were produced for SET3 deletion strains expressing the engineered GAL1 gene with either high or low levels of antisense transcription. The experimental data were then modelled to estimate the parameters of sense transcription dynamics (Figs 6F and EV4E). Consistent with our hypothesis, deletion of SET3 in the strain with low levels of antisense transcription decreased the mean production rate, decreased the nuclear processing (elongation/export) rate and increased the stability of the mature GAL1 sense transcripts, making the transcription dynamics of the strain with low levels of antisense transcription behave more like the strain with high antisense transcription. Global levels of transcripts are buffered by opposing rate changes for synthesis and degradation resulting in no overall change, as observed previously (Dori-Bachash et al., 2011). We suggest that antisense transcription reduces the sensitivity of genes to deacetylation by Set3C, and this influences transcription dynamics. Thus, at GAL1, antisense transcription buffers gene expression against the action of chromatin modifiers such as Set3C. We conclude that sense transcription dynamics are variable and can be modulated by histone modifiers, and therefore histone modifications, in the vicinity of the promoter and early part of the coding region.

In summary, we show that antisense transcription has a conserved spatial and chromatin architecture in both yeast and human genes, focused around the sense promoter and early transcribed region. Modelling with quantitative data reveals that antisense transcription at the engineered GAL1 locus alters all measurable aspects of sense transcription by decreasing the rates of
Figure 6.
initiation and processing of the nuclear transcripts, and the cytoplasmic degradation rate. The effect of antisense transcription on sense transcript dynamics is observed at other genes and can be mimicked at GAL1 by simply increasing levels of histone acetylation in the vicinity of the promoter.

Discussion

Antisense transcription is a widespread feature of both yeast and human genomes. In this work, we use mathematical modelling to provide insights into the consequences of antisense transcription on chromatin architecture and sense transcript dynamics, and show for the first time that antisense transcription alters rates of transcript production and transcript degradation.

There is a tight, apparently counterproductive, coordination between the processes of production and degradation, widely observed in yeast and mammals (Das et al., 2017). For example, mutants such as rpB4A, which display reduced rates of transcription, compensate by reducing the rate of transcript degradation (Schulz et al., 2014). The coordination of transcription and transcript degradation is known to be influenced by promoter sequences, the Rpb7 component of RNA polymerase II and many factors involved in mRNA degradation (Enssle et al., 1993; Dori-Bachash et al., 2011, 2012; Sun et al., 2013). Assessment of steady-state mRNA levels in mutants that influence these rates often leads to the conclusion that these factors do not have much effect on gene expression, apart from the associated stress response (O’Dubhir et al., 2014). This would be entirely consistent with what we observe for antisense transcription and the set3 mutant, which do not change steady-state transcript levels at GAL1, but do change the epigenetic marks on the associated chromatin, and in doing so, alter the transcription dynamics. The set3.3 strain with low GAL1 antisense transcription showed raised levels of H3K9ac at GAL1 similar to those observed with high antisense transcription and a concomitant lowering of sense transcript production, processing and degradation rates to resemble those in strains with high antisense transcription (Fig 7).

Our model makes no assumptions about the mode of transcription initiation ab initio and can accommodate genes with “bursty” kinetics, as it allows for promoters to exist in both active and inactive states, or a constitutive model of production, and so would be applicable to analyse data from mammalian cells. Indeed, much transcription initiation in yeast and mammals shows bursting kinetics (Suter et al., 2011; Lenstra et al., 2016). In addition, our model extends previous models, incorporating a stochastic elongation rate, and as such, each gene displays a distribution of times from initiation to termination. One interesting finding is that our model infers a decrease in the rates of both sense transcription initiation and elongation/export in the presence of antisense transcription, yet we observe no difference in the NET-seq, GRO-seq and PRO-seq average profiles between genes with high and low levels of antisense. How could antisense transcription alter sense transcription dynamics without altering these nascent transcription profiles? These techniques offer a static, steady-state view of the locations of engaged polymerase across a gene. A corresponding decrease in initiation rate and elongation rate would result in the same number of polymerases on a gene at steady state. Any differences in polymerase profiles clearly come as a result of a number of complex interacting factors including physical blocks to transcription, the activity of elongation factors and stochastic dynamics of polymerase (Jonkers & Lis, 2015). Here we have analysed the production and processing of functional mRNA. As such, we look at the effect of antisense on the passage of the transcript through to the cytoplasm and on the rates of productive initiation, meaning that we ignore any initiation events that, for example, result in early termination. By contrast, NET-seq and GRO-seq likely convey a more complex story, in which the number of engaged polymerases does not correspond directly to the resultant number of steady-state RNAs. This is evidenced by the less-strong-than-expected Spearman correlations between nascent transcription reads and RNA-seq reads either in the 300-bp window downstream from the promoter (PRO-seq 0.52; NET-seq 0.78) or across the whole gene (PRO-seq 0.61; NET-seq 0.79).

The effects of antisense transcription on the sense promoter and promoter-proximal chromatin architecture are conserved between yeast and humans, despite large differences in gene size. In addition, antisense transcription initiates at a similar distance downstream from the sense transcription initiation site in both systems, suggesting that the effect of antisense transcription is focused on the promoter and early transcribed region of genes. This is where most control can be exerted over transcription dynamics, as the promoter and early coding region chromatin can influence initiation and the elongation phases of transcription, respectively. Moreover, we observe conserved changes to the promoter and promoter-proximal chromatin structure as the functional consequence of antisense transcription. These changes include increased lysine acetylation and increased nucleosome occupancy, which together could influence the residence time of transcription factors bound to the promoter or the composition of RNA polymerase II leaving the promoter. At the promoter itself, the altered chromatin features may also be reinforced by divergent upstream non-coding transcription (Marquardt et al., 2014). In support of a promoter focused function, we have recently shown that RNA polymerase II shows variable enrichment with elongation factors and that this a function of promoter sequences and associated transcription factors (Fischl et al., 2017). Paf1 enrichment on RNA polymerase II, for example, through its effects on the chromatin structure, affects how the encoded transcripts are decorated with RNA binding proteins that control transcript export from the nucleus.

Could antisense transcription function in gene regulation? By reducing production and increasing stability, as observed with high antisense transcription, the same final transcript response level can be achieved as with low antisense transcription, but the time taken to reach these final levels differs, and this can be a regulatory feature, for example, providing benefit in some bet-hedging strategies (Snijder & Pelkmans, 2011) or if rapidly varying conditions are expected. Indeed, antisense transcription has proposed role in fine-tuning levels of sense transcripts under different environmental conditions (Xu et al., 2011) and its production can be regulated (Conley & Jordan, 2012; Murray et al., 2012, 2015; Nguyen et al., 2014). Transcription elongation is not a smooth process from start to finish, with RNA polymerase pausing heterogeneously across the gene (Jonkers & Liu, 2015), and this is likely to influence the nuclear processing rate. Changing the nuclear processing rate does not affect the cytoplasmic distribution or the rate at which a cytoplasmic steady state is reached. However, antisense transcription-dependent changes to the nuclear processing rate could be tuning how much
other pathways can affect a transcript. For example, decreasing the elongation/export rate could make a gene more susceptible to control by a factor that relies on stochastic events during transcription or while a transcript is in the nucleus.

Why does Set3 modulate transcription dynamics predominantly at low antisense genes? We see increased levels of acetylation at low antisense genes when the integrity of Set3C is lost. Set3C may antagonize the action of the SAGA lysine acetyltransferase, as genes with high antisense show significantly more enrichment for the Spt3 component of the SAGA lysine acetyltransferase (KAT) complex (Murray et al., 2015), supporting a higher inherent level of acetylation at high antisense genes in strains with Set3C. This may be a reinforcing mechanism whereby once the levels of acetylation on chromatin are high, more KAT will be recruited to maintain these levels. A dynamic interplay between Set3C and the KATs, modulated by differences in histone turnover and chromatin compaction, could explain the different characteristics of high and low antisense genes and the particular sensitivity of low antisense genes to loss of Set3C. Being able to modulate transcription and transcript dynamics by manipulating the activity of a chromatin modifying enzyme strongly supports antisense transcription modulating the chromatin structure in the vicinity of promoters and this in turn affecting transcription and transcript fate. Pervasive transcription is not limited to the antisense strand of genes as studied here, but is also abundant at enhancer elements, at the 3’ ends of genes and throughout gene-rich

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**Figure 7. Summary.**

A Schematic outlining the influence of antisense transcription or SET3 deletion on levels of histone lysine acetylation (ac), steady-state transcript levels (3) and on the rates of transcript production (1), processing and export (2) and transcript decay (4). Line width indicates levels (3) or rates (1, 2, 4) with increased rate represented by extra lines.

B Relationship of the dependent variables to independent variables in our study. High antisense transcription is associated with a highly acetylated chromatin environment, resulting in slower sense transcription kinetics, namely the initiation rate and elongation/export rate. We mimic this behaviour by deleting SET3, increasing acetylation levels and we observe a reduction in initiation and elongation/export rates.
regions of many different genomes including plants, yeast, fungi, flies and worms (Ni et al., 2010; Kwak et al., 2013; Andersson et al., 2014; Nojima et al., 2015; Booth et al., 2016; Krzyczmonik et al., 2017; leiswaart et al., 2017). In some cell types, non-coding transcripts could facilitate RNAI-mediated degradation of the sense transcripts which might make uncovering the associations we have found in budding yeast, which lacks an RNAI system, more challenging. However, the functional consequences of most non-coding transcription are likely to be a result of the act of transcription, and its effects on associated chromatin, rather than via the transcript itself. In yeast, many non-coding transcription events are performed by a distinct RNA polymerase II complex, depleted for Paf1 and Set2 (leading to reduced Ser2 phosphorylation on the Pol2 CTD and reduced H3K36 methylation) amongst other factors (Fischl et al., 2017), explaining in part the unique chromatin environment associated with these events. Whether enhancers also contain a chromatin environment, for example high H3K27ac, dictated by non-coding transcription and how this influences enhancer function are questions for the future, but underscore the potential regulatory nature of non-coding transcription, as distinct from the encoded transcripts. Indeed, it is becoming clear that levels of pervasive transcription can be regulated in genomes (Mellor et al., 2016).

**Materials and Methods**

Jane Mellor is responsible for all reagent and resource requests. Please contact Jane Mellor at jane.mellor@bioch.ox.ac.uk with requests and inquiries and see Appendix Table S4 for reagent details and Appendix Table S5 for details concerning software and algorithms.

**Yeast culture and genetic manipulation**

Strains were streaked from glycerol stocks onto 2% agar YPD (1% yeast extract (Difco), 1% bactopeptone, 2% glucose) plates and grown (1–2 days, 30°C). Cell pre-cultures were then grown overnight in 5 ml YPD at 30°C. This culture was used to inoculate an appropriate volume of YPD culture at OD₆₀₀ 0.2 which was grown at 30°C, shaking at 200 rpm to OD₆₀₀ 0.45–0.5. To induce the GAL1 gene, cell cultures were centrifuged (900 g, 3 min) and then re-suspended in YPG (1% yeast extract (Difco), 1% bactopeptone, 2% galactose) pre-warmed to 30°C. Re-suspended cells were incubated (30°C, 200 rpm) for the specified time(s) before harvesting by centrifugation (900 g, 4 min). For the experiments to obtain the GAL1 sense degradation rates, after 2 h in YPG, cells were transferred back to fresh YPD pre-warmed to 30°C and 15 ml of samples was harvested at 0, 5, 10, 20, 30 and 60 min. Other experiments were done at OD₆₀₀ 0.6–0.8.

Genetic manipulation of strains was performed using the homologous recombination method (Longtine et al., 1998). For gene deletion strains, PCR products were made containing the HISMX or KANMX selection cassettes flanked at both ends by 40 bp of sequence homologous to sequences either side of the region to be deleted. Construction of the GAL1:ADH1 (high AS) and TATA mutant (low AS) strains has been described previously (Murray et al., 2012, 2015). Cells to be transformed were grown to log phase, pelleted, re-suspended in 450 μl 100 mM LiAc/TE and incubated (>1 h, 4°C). 100 μl of cell suspension, 10 μl of PCR product, 10 μl calf thymus DNA (Sigma D8661) and 700 μl 40% polyethylene glycol in 100 mM LiAc/TE were incubated (30 min, 30°C) then heat-shocked (20 min, 42°C). Cells were pelleted (5 min, 4,600 g), re-suspended in H₂O and plated onto appropriate selection media. DNA was extracted from the resulting colonies, screened by PCR and confirmed by sequencing.

**Chromatin immunoprecipitation (ChIP)**

Yeast grown to OD₆₀₀ 0.5 in 50 ml of YPD was transferred to YPG for 2 h before they were fixed in 1% formaldehyde in 45 ml PBS for 30 min at 22°C followed by addition of 125 mM glycine for 5 min. Cell pellets were collected by centrifugation (900 g, 4 min) before washing twice with 10 ml cold PBS. Cells were re-suspended in 500 μl cold FA-150 buffer (10 mM HEPES pH 7.9, 150 mM NaCl, 0.1% SDS, 0.1% sodium deoxycholate, 1% Triton X-100) and broken using 1 ml glass beads on a MagnaLyser (Roche; 2 × 1 min runs, 2,500 g, 4°C). Sample volume was increased to 2 ml with FA-150 buffer before shearing of the fixed chromatin by sonication using a biorupter (Diagenode, 30 min, 1 min on, 20 s off, medium setting). Chromatin was cleared by centrifugation (9,400 g, 15 min, 4°C), and 50 μl was diluted to 200 μl with FA-150 buffer and incubated with 5 μl of the following antibodies as appropriate: H3, H3K4me2, H3K4me3, H3K9ac (for details see the Appendix Table S4) in 1.5 ml siliconized Eppendorf tubes for 15–20 h rotating at 4°C. Bound chromatin was immunoprecipitated for 90 min at 22°C with 50 μl protein A-Sepharose pre-blocked with bovine serum albumin and sonicated salmon sperm DNA. Beads and attached chromatin were pelleted by centrifugation (640 g, 1 min) and washed with TSE-150 buffer (20 mM Tris–Cl pH 8.0, 150 mM NaCl, 2 mM EDTA, 0.1% SDS, 1% Triton X-100) for 3 min, TSE-500 buffer (20 mM Tris–Cl pH 8.0, 500 mM NaCl, 2 mM EDTA, 0.1% SDS, 1% Triton X-100) for 3 min, LiCl buffer (0.25 M LiCl, 10 mM Tris–Cl pH 8.0, 1 mM EDTA, 1% deoxycholate, 1% NP-40) for 15 min and twice with TE. After washing, chromatin was eluted from the beads for 3 h at 65°C with elution buffer (0.1 M NaHCO₃, 1% SDS). Addition of 350 mM NaCl and incubation for 3 h at 65°C reversed the cross-links before treatment of samples with RNase A for 1 h at 37°C and proteinase K overnight at 65°C. DNA was purified using a PCR purification kit (Qiagen) and eluted in 400 μl 1 mM Tris–Cl pH 8.0. Input DNA was diluted accordingly. Real-time quantitative PCR (qPCR) was performed using a Corbett Rotorgene and Sybr green mix (Bioline). Data [(IP—no antibody control)/input] were expressed as a percentage of the input and normalized to levels of H3 where appropriate. The primers used are listed in Appendix Table S1. All ChIP experiments were performed ≥2 times with independent biological samples.

**RNA extraction**

Fifteen millilitres of log phase yeast culture at a density of OD₆₀₀ 0.6–0.8 grown in the appropriate medium was pelleted (900 g, 3 min), re-suspended in 400 μl TES (100 mM Tris–HCl (pH 7.5), 100 mM EDTA (pH 8.0), 0.5% SDS) and 400 μl phenol:chloroform (pH 4.7) and incubated (65°C, 20 min, 22 g). The mixture was incubated (–80°C, 30 min). After spinning (15,900 g, 20 min, 4°C), the upper layer was transferred to 10 mM NaOAc pH 5.5/ethanol and incubated (–80°C, >30 min). RNA precipitate was pelleted from the ethanol solution by centrifugation at 15,900 g for 30 min. RNA was then dissolved in 50 μl diethylpyrocarbonate (DEPC) treated water and used directly for reverse transcription or stored at −80°C until required.
fifty millilitres yeast culture was grown in YPD to RNA fluorescence as loading controls. All Northern blotting experiments were repeated 20 min each at 65°C SSC/0.1% SDS, 0.1 M 40% formamide, 2× SSC for 5 min at room temperature and once with 0.05× SSC/0.1% SDS and 0.05× SSC/0.1% SDS for 20 min each at 65°C. Membranes were typically exposed to X-ray film for 1 h–1 week. For quantification, images were acquired using a FLA 7000 phosphorimag (GE Healthcare). Levels of the 18S and 25S rRNA species measured by ethidium bromide staining were used as loading controls. All Northern blotting experiments were repeated ≥ 2 times with independent biological samples.

RNA fluorescence in situ hybridization (RNA-FISH)

Fifty millilitres yeast culture was grown in YPD to > 0.45 OD600 before transfer to YPG for 2 h. 50 ml of cells at OD600 0.6 was pelleted (900 g, 4 min) and fixed with 4% (v/v) paraformaldehyde in PBS (45 min, 80 rpm, 22°C). Fixed cells were washed twice with 10 ml FISH buffer A [1.2 M sorbitol, 0.1 M KHPO4 (pH 7.5)] and re-suspended in 1 ml FISH buffer B (FISH buffer A, 20 mM ribonucleoside vanadyl complex (VRC), 20 μM 2-mercaptoethanol). The mixture was incubated (15–40 min) at 30°C with 15 μl lyticase (25 U μl⁻¹, Sigma) until >70% of cells were spheroplasted, as observed by microscopy. Cells were pelleted (900 g, 3 min, 4°C) and washed with and then re-suspended in 1 ml FISH buffer B without 2-mercaptoethanol. ~150 μl of cells was left to settle (30 min, 4°C) on poly-l-lysine treated coverslips. These were gently washed with 2 ml FISH buffer A to remove unattached cells and incubated (~20°C, >3 h) in 2 ml 70% ethanol. Samples were rehydrated twice with 2 ml of 2× SSC for 5 min at room temperature and washed with 40% formamide in 2× SSC. For the hybridization, 0.5 ng of each probe, 10 μg of E. coli rRNA and 10 μg of salmon sperm DNA were mixed and lyophilized in a SpeedVac. 12 μl of 40% formamide, 2× SSC, NaHPO4 pH 7.5 was added, and the probes were denatured at 95°C for 3 min followed by the addition of 12 μl of 2× SSC, 2 mg ml⁻¹ BSA, 10 mM VRC. Hybridization was performed overnight at 37°C in a parafilm-sealed chamber, where the coverslips with the cells facing down were placed onto 22 μl of the hybridization mixture. The coverslips were then subjected to a series of washes: twice with 40% formamide/2× SSC (15 min, 37°C); once with 2× SSC, 0.1% Triton X-100 (15 min, 22°C); once with 1× SSC (15 min, 22°C); and once with 0.05× SSC (15 min, 22°C). The Stellaris GFP probes were incubated overnight at 30°C, then washed several times in a 10% formamide solution and stained with a PBS solution containing DAPI. The coverslips were dipped into H₂O. Once dry, coverslips were mounted onto a microscope slide using ProLong Diamond Antifade Mountant with DAPI (Life Technologies), allowed to polymerize for 24 h in the dark and then sealed with nail varnish. Cells were imaged using a DeltaVision CORE wide-field fluorescence deconvolution microscope using a 100×/1.40 objective lens. 21–31 0.2 μm z stacks were imaged with an exposure time of 0.01 and 1 s for DAPI and Cy3 channels, respectively. All RNA-FISH experiments were repeated ≥ 2 times with independent biological samples.

RNA-FISH probe design and synthesis

For GAL1, DNA probes of ~50 nt and ~50% GC content were designed with five modified bases (amino-allyl dT) spaced by about ~10 nt included for the incorporation of the fluorophore (see Appendix Table S3 for probe sequences). Modified DNA oligos were custom ordered from MWG Eurofins. For the labelling of the probes, a total of 5 μg was purified using the QIAquick Nucleotide Removal Kit (Qiagen) and eluted with 40 μl of H₂O. The probes were then lyophilized in a SpeedVac, re-suspended in 10 μl of 0.1 M sodium bicarbonate pH 9.0 and added to the dye-containing tube (CyDye™ GE Healthcare, Cy3 PA23001). The tube was vortexed vigorously followed by a quick spin. The reaction was incubated overnight at room temperature with low speed shaking. The probes were purified using the QIAquick Nucleotide Removal Kit (Qiagen) and eluted with 100 μl of elution buffer (supplied with the kit). The concentration and efficiency of the labelling was measured using a spectrophotometer. Probes were stored in the dark at ~20°C. The labelling efficiency was calculated as described (Zenklusen & Singer, 2010). The remaining genes were fused at their 3' ends to S65T pFA6a-GFP sequence (Longtine et al, 1998), and the common GFP sequence detected using 27 × 20 nt probes labelled with Cy3 fluorophore obtained from Stellaris. The GFP probes were washed in a PBS/DAPI solution and were mounted in ProLong Gold without DAPI.

Yeast strains

All S. cerevisiae strains used in this study are listed in the Appendix Table S6. All strains and genetic manipulations were verified by sequencing or PCR-based methods.

Mathematical modelling

RNA synthesis and degradation were modelled as described in the main text. Four of the five parameters were sampled via Latin Hypercube with 1,000,000 sampling points. The degradation rate was sampled as described below. 10,000 cells were simulated for 500 min to reach steady state and the number of nuclear and cytoplasmic RNA recorded. The Kolmogorov–Smirnov statistic was used as a goodness-of-fit metric to compare simulated results to raw data. The best 10,000 parameter sets as judged by fit to nuclear and cytoplasmic RNA were then taken forward. For each strain, the modal value of the histogram of the mean initiation rate (on * init/(on + off)) was taken from the fits to the cytoplasmic data. The values for the nuclear processing rate were then determined by sampling data points from the ratio of mean initiation rate to nuclear export rate giving the fits to the nuclear data. Again, 10,000
parameters were sampled from the values determining the nuclear distributions, following the mean initiation rate distribution given by the cytoplasmic data.

**Quantification and statistical analysis**

**Bioinformatic analysis**

**Identification of sense and antisense TSSs in yeast and humans** Cap analysis of gene expression (CAGE) data in HeLa cells was obtained from the ENCODE repository on the UCSC Genome Browser (Rosenbloom et al., 2010), and used to determine genome coordinates of sense and antisense transcript start sites (sTSSs and aTSSs, respectively). Data were pooled from nuclear and cytoplasmic fractions, both polyadenylated and non-polyadenylated, and from whole cell extract, for which only polyadenylated data were available. CAGE cluster coordinates, determined with an HMM algorithm applied to the CAGE tag data, were obtained from the same source. To determine TSS coordinates in HeLa, we took the same approach presented previously by Conley and Jordan (2012). Clusters were ignored if they contained less than two overlapping CAGE tags, as it has been previously reported that two or more overlapping tags represent validated TSSs (Carninci, 2006; Faulkner et al., 2007). The TSS coordinate of a given cluster was taken to be the base with the highest density of mapped CAGE 5’ ends. As an added step, TSSs were excluded from all subsequent analyses if they contained less than three NET-seq reads in a 200-bp window immediately downstream, within the same orientation. To determine the sTSS of a given protein-coding gene, we scanned within a region 500 bp upstream of the left-most annotated TSS, and 500 bp downstream of the right-most annotated TSS. In the case of multiple sTSSs, the one with the highest CAGE density was taken to be the predominant sTSS. aTSSs were determined by scanning between the sTSS and the annotated transcript end site; again, the aTSS with the highest CAGE density was considered the predominant aTSS in the case of multiple candidates.

Budding yeast sTSSs and aTSSs were determined using transcript isoform sequencing (TIF-seq) (Pelechano et al., 2013), using the list of major transcripts provided, supplemented with the list of cryptic transcripts from Neil et al. (2009). For each gene, the sTSS was derived from the sense transcript which had the highest number of supporting NET-seq reads in YPD, and which completely encompassed the open reading frame. The aTSS was taken as the antisense transcript with the highest number of supporting NET-seq reads, and which overlapped the open reading frame in the antisense orientation.

To assess whether aTSSs were better aligned to the sTSS or the end of the 1st exon in HeLa, we determine for each gene the distance between the sTSS to the aTSS, and expressed it as a fraction of the distance between the sTSS and the end of the 1st exon. We compared the resultant histogram to a randomly generated distribution, in which the aTSS for each gene was randomly reassigned to a base within the region shown in Fig 1D. This approach was repeated in HeLa using the end of the 2nd exon in place of the 1st, to assess whether aTSS showed preferential alignment to the 2nd exon over the sTSS. It was also repeated in yeast, using the 3’ end of the open reading frame in place of the end of the 1st exon (Fig 1G).

**Correlating sense and antisense transcription** NET-seq data were obtained in HeLa cells from Nojima et al. (2015), specifically their data obtained using an antibody against all forms of Pol II, phosphorylated and unphosphorylated. NET-seq data in yeast were obtained from Churchman and Weissman (2011). To compare sense transcription levels between genes with and without an aTSS, we calculated the average number of NET-seq reads per base pair within the 1st exon, and compared the distribution between the two gene groups using a Wilcoxon rank sum test. Correlations between the transcription levels of different sorts of transcript were calculated by determining the Spearman correlation coefficient between the numbers of NET-seq reads in the 300-bp windows shown in Fig 2C and D. The same approach was taken with the GRO-seq (for HeLa) and PRO-seq data (for yeast), obtained from Core et al. (2014) and Booth et al. (2016), respectively.

**Assessing ChIP levels around sense and antisense TSSs** Genome-wide levels of histone modifications and nucleosome occupancy were obtained from the following sources: For budding yeast, genome-wide levels of H3K36me3 and H3K79me3 were from Kirmizis et al. (2009) (GSE14453). Levels of H3K4me1 and H3K4me3 were from Kirmizis et al. (2007) (GSE8626). Levels of H3K9ac and H3K27ac were from Weiner et al. (2015) (GSE61888). Nucleosome occupancy levels were from Kaplan et al. (2009) (GSE13622). Genome-wide levels of gene compaction, determined using Micro-C, were from Hsieh et al. (2015). Levels of H3K9ac in deletion strains of various histone modifying enzymes were from Weinberger et al. (2012) (SRA051855.1). For HeLa cells, genome-wide levels of H3K36me3, H3K79me3, H3K4me1, H3K4me3, H3K9ac and H3K27ac were obtained from the ENCODE experiment matrix. Nucleosome occupancy levels were from Klir et al. (2015) (GSE65644). Levels of H3 histone modifications are not normalized to levels of H3. We assessed average levels only in genes with an aTSS, comparing the two quintiles with the highest and lowest levels of antisense transcription (determined by NET-seq) in a 300-bp window placed immediately downstream of the sTSS. We wished to simultaneously assess levels upstream of the sTSS, downstream of the aTSS and in the region between both TSSs. To account for the varying distances between sTSS and aTSS, we broke this region into a hundred bins, calculating the average ChIP level within each bin.

**Comparing gene compaction levels** Gene compaction levels in budding yeast were obtained from Hsieh et al. (2015). Different gene groups were compared as discussed in the Results.

**RNA-FISH analysis**

**Software** Image quantification was performed using custom Matlab (MATLAB Statistics and Image Toolbox Release 2015a, The MathWorks, Inc., Natick, MA, USA) scripts based in part on elements of FISH-quant (Mueller et al., 2013) and CellProfiler (Carpenter et al., 2006), and utilizing MIJI (https://imagej.net/Miji) and MJ (http://bigwww.epfl.ch/sage/soft/miji/) to import data from Fiji (Schindelin et al., 2012). The custom scripts allowed for greater automation of the quantification process than is possible with FISH-quant and the algorithms were tailored to our data.
Due to slight differences in experimental protocol, including model of microscope, between the data sets including the engineered forms of \( \text{GAL1} \) and the sets including GFP-tagged genes parameters and methods for analysis also differed between these groups. In the following description, the engineered constructs expressing high or low antisense at \( \text{GAL1} \) and with or without the set3A mutation will be referred to as the \( \text{GAL1} \) set and the GFP-tagged genes (\( \text{TIM17, ATP4, GCV3, URA4, HMS2} \)) will be referred to as the endogenous set.

**Deconvolution and background subtraction** Images were deconvolved with a conservative deconvolution method and 10 cycles (\( \text{GAL1} \) set) or 15 cycles (endogenous set) using DeltaVision Softworx software. DAPI and Cy3 channels for the images were processed separately. Images were background-corrected with the following procedure. The median of all pixel intensities for each channel and each image, \( p_{\text{med}} \), was found. This was chosen as it was observed to generally be close to the modal value of the distribution. A measure of the spread around this value was also found by constructing a metric similar to the standard deviation from all pixels with intensities less than or equal to the median intensity. The median plus this spread value was taken to be the background value and subtracted from all pixels, \( \text{background} = p_{\text{med}} + \sqrt{1/(N_i-1) \sum((p_i - p_{\text{med}})^2)} \), where \( i \) runs over pixels with intensity less than or equal to \( p_{\text{med}} \). \( N_i \) is the number of pixels with intensity less than or equal to \( p_{\text{med}} \) and \( p_i \) is the intensity of pixel \( i \). Thus, the new intensity of \( p_i \) was \( p_i - \text{background} \) where \( j \) runs over all pixels. Any pixels that had negative intensity following this were set to 0.

**Foci identification and thresholding** Candidate foci in the FISH (Cy3) channel were initially identified using Piotr’s Matlab toolbox (https://pdollar.github.io/toolbox) nonMaxSupr function with a 1 pixel radius for detection. Images from each biological repeat and strain were processed together. To distinguish foci from random clustering of fluorescently labelled molecules, foci intensities were compared between the strains under testing and a double knockout strain, \( \text{gal10-1} \), which has no sequences to which the FISH probes should hybridize. In each experiment, histograms of all foci intensities as returned by the nonMaxSupr algorithm were constructed (bin width 250 for the \( \text{GAL1} \) set and 50 for the endogenous set, normalized by probability). For each strain, a tentative intensity cut-off was taken to be the first bin in which there was 10 times more signal in the strain than the knockout (with manual adjustment for obvious outliers). Within each experiment (which could contain multiple strains), the final cut-off value was taken as the mean of the tentative cut-off values. All foci with intensities less than the cut-off value from a set were not considered in all further analysis.

The deconvolution, background subtraction and foci identification are performed by the supplied FindAndAnalyseFoci Matlab function. This function should be run on all strains and the knock-out from a single experiment before a threshold for valid foci is determined. This threshold can then be found using the DetermineCutoffs Matlab script.

**Automated nuclei detection** A separate script automatically identifies nuclei and cells and quantifies the foci that fall within the nuclear and cellular boundaries. The first step of the process is to identify the nuclei in three dimensions using the DAPI channel. The procedures followed here allowed for improved detection of individual nuclei that differed in brightness or were very close to neighbouring nuclei. The DAPI channel of each image was scaled to the minimum and maximum intensity pixels, that is \( p_i = (p_i - \text{min}(p)) / (\text{max}(p) - \text{min}(p)) \), where \( i \) runs over all pixels, \( \text{min}(p) \) and \( \text{max}(p) \) denote the minimum and maximum pixel intensities of the set, respectively. The scaled DAPI images then have a Gaussian filter applied using Matlab function imgaussfilt3 with a smoothing-kernel standard-deviation value of 2 for the \( \text{GAL1} \) set and 2.5 for the endogenous set. At this point, the analysis for the two sets diverged considerably and so they are described separately. For the \( \text{GAL1} \) set, for each processed image, Otsu’s method for multiple thresholds, Matlab function multithresh, was used to give six threshold levels and the image was segmented into seven levels around these using the Matlab function imquantize. The segmented images thus contained pixels with values from 1 to 7. Each segmented image was then restricted to a subsection of the available \( z \) stacks by setting any pixels in \( z \) planes below or above certain values to zero, to avoid any errors due to using overly blurred portions of the image. For images with 31 \( z \) stacks, images were typically restricted to include only pixels from \( z \) planes 12 to 22, inclusive, and for images with 21 \( z \) stacks, images were typically restricted to \( z \) planes from 2 to 20, inclusive. These values were manually adjusted in some cases to allow for off centred focusing, but the same planes were used for all images of a particular strain taken in a single experiment.

The segmented levels were cycled through from the 3rd to the 7th levels. For each level, a 3D logical image was formed from the pixels with value equal to the value of the level. The \( z \) stacks were then cycled through and all holes (areas with pixels with value 0 inside areas with value 1) were filled, Matlab function imfill with flag “holes”. Then, all 3D-contiguous regions (with 26 connectivity) of pixel value 1 that had more than 6,000 or fewer than 50 pixels were removed by setting all pixels in the region to zero, using Matlab xor and bwareaopen functions. Any remaining 3D-contiguous regions (26 connectivity) were then labelled using the Matlab function bwlabel. For levels lower than the final level, an identical procedure was performed on the level immediately above, without the final labelling step. Each labelled section was then cycled through, and if there was no overlap with any non-zero valued pixels in the segmented level above, it was deemed as a good candidate for a nucleus and saved (the centroid was determined with the Matlab function regionprops and flag “centroid”, and each coordinate was rounded to the nearest integer). Any overlap with the level above signified the existence of a superior candidate or superior candidates in this region, and this potential nucleus was not saved. This process was repeated until the final level in which no checking against a higher level was possible.

Once all good nuclear candidates had been identified in this way, any nuclei that were very close together were merged with the following procedure. The Euclidean distances, measured in pixel coordinates, between all centroid locations were calculated. A list of all non-equal pairs of centroids that had a distance of less than or equal to six between them was created. This was done by having two nested loops: the outer loop cycled over the centroids from \( t = 1 \) to \( N_t - 1 \) and the inner loop cycled over \( j = (i + 1) \) to \( N_t \), where \( N_t \) is the number of candidate centroids. Any \( i, j \) pairs from
Automated cell detection The 2D nuclei identified above were used as seed points to identify cell outlines. Cell masks were identified using the CellProfiler function IdentifySecPropagateSubFunction from the MEX compiled file supplied with the developer’s version of CellProfiler 1.0. Cells were identified with a combination of the DAPI signal, FISH foci signal and autofluorescence of the cells observed in the FISH channel. The IdentifySecPropagateSubFunction takes a number of inputs: a set of seed points for cells; a 2D image with varying intensity; a 2D logical image; and a regularization factor which determines how to weight between the 2D images and distance to the nearest seed points when determining cell boundaries. A regularization factor value of 0.0001 was used in all cases.

The 2D images with varying intensities were constructed by combining processed DAPI and FISH channels in the following way. For each channel of each image, the sum in the z direction was taken to get a flattened image and the maximum and minimum intensities observed in this image were found. Each pixel in the image was then normalized as \[ p_i = (p_i - \min(p))/\max(p) - \min(p) \]. For each image, the normalized flattened channels were averaged, and then, this average had a Gaussian filter applied (Matlab function imgaussfilt with smoothing-kernel standard-deviation value of 2).

The 2D logical images were constructed starting in a similar way by creating normalized and flattened images as above prior to the averaging step. Each channel for each image had a Gaussian filter applied; the DAPI signal used a smoothing-kernel standard-deviation value of 5 and the FISH channel used a value of 2. Each filtered channel for each image then had a threshold generated by taking the lowest value from a three-level Otsu’s method thresholding (Matlab function multithresh with three levels). Each of the filtered channels for each image was then converted into a logical image with pixels having an intensity less than the threshold being set to 0 and the rest being set to 1. The channels for each image were then combined using the logical OR operation.

The nuclei and cells were then further processed to remove anything touching a border of the image as cells touching the border are likely to have part of the cell out of the image and using them for data collection could bias the results. First any 3D-resolved nuclei that touched the border in 3D were removed (Matlab function iclearborder). Following this, any cell masks that were touching the border were removed. Then, any 3D nuclei considered as being too large (i.e. containing more than 3,000 pixels for the GAL1 set or 20,000 pixels for the endogenous set) were removed. The size-based removal was done after cell detection as large detected nuclei often corresponded to multiple nuclei close together and keeping these causes the corresponding cells to be detected as one large cell, which often aided in assigning the correct boundaries to nearby cells. Then, any cells that did not overlap with any nuclei were cleared and any nuclei that did not overlap with a cell were also cleared. Finally, any cells that had two or more nuclei were removed along with the corresponding nuclei. This case can happen rarely when 3D nuclei do not touch but overlap when collapsed onto 2D resulting in a merged nucleus. These were removed as it is likely that there would be some cytoplasmic overlap also in this case.

Foci classification and mRNA quantification Cells were first divided into nuclear and cytoplasmic components. The nuclear components were formed first by flattening the 3D-resolved nuclei masks (taking the maximum over the z stacks) and then extruding them to the allowed z stacks (the same z stack limits used when restricting in the nuclei detection phase). The 2D cell masks were
then extruded to the same allowed z stacks and had the nuclear area within them set to zero, which gave the cytoplasmic components. Foci whose centre pixel lay in the nuclear region where classified as nuclear transcripts and foci whose centre pixel lay in the cytoplasmic region were classified as cytoplasmic transcripts. Any foci lying outside these regions were discounted from all further analysis.

In order to quantify the foci in terms of RNA molecules, an intensity value for all accepted foci was calculated by taking the mean of the 27 pixels immediately surrounding and including the central pixel (found by rounding each coordinate of the output of the nonMaxSupr function to the nearest integer). For a strain and image set from a single experiment, the median of these intensity values was calculated and was taken to correspond to a single RNA molecule. There is no reason that a FISH focus should contain only a single RNA, especially in the nucleus, but we assumed that foci most commonly contained a single RNA. Each focus intensity was then converted into a corresponding number of mRNA molecules by dividing by the median intensity and rounding to the nearest integer. Note that this can result in foci being classified as containing zero RNA molecules and provides an additional filtering step similar to the initial cut-off based on knockout strains.

The detection of nuclei and cells, and the quantification of foci is performed by the supplied Matlab script DetectCellsAndQuantifyFoci. This script should be run after the FindAndAnaylseFoci function and DetermineCutoffs script as it uses the data generated in the first script and the cut-off generated after averaging the determined cut-offs over an experiment. The cut-off must be manually changed in the DetectCellsAndQuantifyFoci script before running. Scripts and images are available from https://doi.org/10.17632/dhnv/j4xs5d.1.

**Chromatin immunoprecipitation**

Real-time quantitative PCR (qPCR) was performed using a Corbett Rotorgene and SYBR green mix (Bioline). qPCR was performed in triplicate for each sample and quantified using a standard curve. Histone modification data ([IP—no antibody control]/input) were expressed as a percentage of the input and normalized to levels of histone H3 at each amplified region. Data are presented as averages of ≥2 biologically independent experiments, with error bars representing the standard error of the mean.

**Northern blotting**

Raw images for quantification were captured using a FLA 7000 phosphorimager (GE Healthcare). Mean intensity of band was quantified using Fiji/ImageJ. Normalized levels of RNA were obtained from Northern blots. Data were tested for normality using a Kolmogorov–Smirnov test; all p values were > 0.7 indicating no evidence to suggest the data were not normally distributed. As such, unpaired t-tests were used to compare levels of RNA between strains across multiple experiments. Degradation rates were obtained by fitting results across six timepoints to an exponential using MATLAB fit function. Root mean square error was taken as goodness-of-fit metric and correspondingly used as the standard deviation of the estimator of the exponential decay term. To sample degradation rates for the purposes of modelling, degradation rates were sampled from a Beta distribution with maximum and minimum values given by the 95% confidence intervals of the estimator with standard deviation equal to the root mean square error of the estimator.

**Data availability**

Images for RNA-FISH experiments, computer codes and all source data are available from Mendeley https://doi.org/10.17632/dhnv/j4xs5d.1. Computer code is provided as Computer Code EV1.

**Expanded View** for this article is available online.

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**Author contributions**

Project conception: J.M and AA; strain construction, GAL1 RNA-FISH, chromatin immunoprecipitation, Northern blots: FSH; other RNA-FISH: MW; GFP tagging of strains: PL; bioinformatics: SCM; modelling: TB, SR, ES and AA; image analysis: AA. All authors involved in interpretation of the data. J.M and SM wrote the paper with input from FSH, TB and AA.

**Conflict of interest**

J.M acts as an advisor to and holds stock in Oxford Biodynamics plc, Chronos Therapeutics Ltd., and Sibelius Natural Products Ltd.

**References**

Andersson R, Gebhard C, Miguel-Escalada I, Hoof i, Bornholdt J, Boyd M, Chen Y, Zhao X, Schmid C, Suzuki T, Nitin E, Arner E, Valen E, Li K, Schwarzfischer L, Glatz D, Raithel J, Lilje B, Rapin N, Bagger FO et al (2014) An atlas of active enhancers across human cell types and tissues. Nature 507: 455–461

Battaglia S, Lidschreiber M, Baejen C, Torkler P, Vos SM, Cramer P (2017) RNA-dependent chromatin association of transcription elongation factors and Pol II CTD kinases. Elife 6: e25637

Booth GT, Wang IX, Cheung VG, Lis JT (2016) Divergence of a conserved elongation factor and transcription regulation in budding and fission yeast. Genome Res 26: 799–811

Bose DA, Donahue G, Reinberg D, Shiekhattar R, Bonasio R, Berger SL (2017) RNA binding to CBP stimulates histone acetylation and transcription. Cell 168: 135–149 e122

Camblog J, Iglesias N, Fickentscher C, Dieppois G, Stutz F (2007) Antisense RNA stabilization induces transcriptional gene silencing via histone deacetylation in S. cerevisiae. Cell 131: 706–717

Carninci P (2006) Tagging mammalian transcription complexity. Trends Genet 22: 501–510

Carpenter AE, Jones TR, Lamprecht MR, Clarke C, Kang IH, Friman O, Guertin DA, Chang JH, Lindquist RA, Moffat J, Golland P, Sabatini DM (2006)
CellProfiler: image analysis software for identifying and quantifying cell phenotypes. Genome Biol 7: R100
Castelnovo M, Rahman S, Guffanti E, Infantino V, Stutz F, Zenzlunen D (2013) Bimodal expression of PHO84 is modulated by early termination of antisense transcription. Nat Struct Mol Biol 20: 851–858
Chouby S, Kondor J, Sanchez A (2015) Deciphering transcriptional dynamics in vivo by counting nascent RNA molecules. PLoS Comput Biol 11: e1004345
Churchman LS, Weissman JS (2011) Nascent transcript sequencing visualizes transcription at nucleotide resolution. Nature 469: 368–373
Conley AB, Jordan IK (2012) Epigenetic regulation of human cis-natural antisense transcripts. Nucleic Acids Res 40: 1438–1445
Core LJ, Waterfall JJ, Lis JT (2008) Nascent RNA sequencing reveals widespread pausing and divergent initiation at human promoters. Science 322: 1845–1848
Core LJ, Martins AL, Danko CG, Waters CT, Sipeal A, Lis JT (2014) Analysis of nascent RNA identifies a unified architecture of initiation regions at mammalian promoters and enhancers. Nat Genet 46: 1311–1320
Das S, Sarkar D, Das B (2017) The interplay between transcription and mRNA degradation in Saccharomyces cerevisiae. Microb Cell 4: 212–228
van Dijk EL, Chen CL, d’Aubenton-Carafa Y, Gouveneuve S, Kwapisz M, Roche V, Bertrand C, Silvain M, Legox-Ne P, Loellet S, Nicolas A, Charneau P, Morillon A (2011) XUTs are a class of Xrn1-sensitive antisense regulatory non-coding RNA in yeast. Nature 475: 114–117
Dori-Bachash M, Shema E, Tirosh I (2011) Coupled evolution of transcription and mRNA degradation. PLoS Biol 9: e1001106
Dori-Bachash M, Shalem O, Manor YS, Pilpel Y, Tirosh I (2012) Widespread promoter-mediated coordination of transcription and mRNA degradation. Genome Biol 13: R114
Enssle J, Kugler W, Hentze MW, Kulozik AE (1993) Determination of mRNA fate by different RNA polymerase II promoters. Proc Natl Acad Sci USA 90: 10091–10095
FANTOM Consortium, RIKEN PMI, CLST (DG1), Forrest AR, Kawai H, Rehli M, Baillie K, de Hooij MN, Haberle V, Lassmann T, Kulakovsky IV, Lizio M, Itto M, Andersson R, Mungall CJ, Meehan TF, Schmierer S, Berton N, Jorgensen M, Dimont E et al (2014) A promoter-level mammalian expression atlas. Nature 507: 462–470
Faulkner CJ, Kimura Y, Daub CB, Wani S, Plessy C, Irvine KM, Schroder K, Cleonan N, Steptoe AL, Lassmann T, Waki K, Hornig N, Arakawa T, Takahashi H, Kawai J, Forrest AR, Suzuki H, Hayashizaki Y, Hume DA, Orlando V et al (2009) The regulated retrotransposon transcriptome of mammalian cells. Nat Genet 41: 563–572
Fischl H, Howe FS, Ferguson A, Mellor J (2017) Pafl has distinct roles in transcription elongation and differential transcriptional fate. Mol Cell 65: 685–698 e688
Geisberg JV, Moqtaderi Z, Fan X, Oszolak F, Struhl K (2014) Global analysis of mRNA isoform half-lives reveals stabilizing and destabilizing elements in yeast. Cell 156: 812–824
He Y, Vogelstein B, Velculescu VE, Papadopoulos N, Kinzler KW (2008) The antisense transcriptomes of human cells. Science 322: 1855–1857
Hongay CR, Grisafi PL, Galitski T, Fink GR (2006) Antisense transcription controls cell fate in Saccharomyces cerevisiae. Cell 127: 735–745
Housley J, Rubbi L, Grunstein M, Tolleryv D, Vogelauer M (2008) A ncRNA modulates histone modification and mRNA induction in the yeast GAL gene cluster. Mol Cell 32: 685–695
Howe FS, Fischl H, Murray SC, Mellor J (2017) Is H3K4me3 instructive for transcription activation? BioEssays 39: 1–12
Hsieh TH, Weiner A, Lajoie B, Dekker J, Friedman N, Rando OJ (2015) Mapping nucleosome resolution chromosome folding in yeast by micro-C. Cell 162: 108–119
Huber F, Bunina D, Gupta I, Khmelinskii A, Meurer M, Theer P, Steinmetz LM, Knop M (2016) Protein abundance control by non-coding antisense transcription. Cell Rep 15: 2625–2636
Ietswaart R, Rosa S, Wu Z, Dean C, Howard M (2017) Cell-size-dependent transcription of FLC and its antisense long non-coding RNA COOLAIR explain cell-to-cell expression variation. Cell Sys 28: 622–635
Jonkers I, Lis JT (2015) Getting up to speed with transcription elongation by RNA polymerase II. Nat Rev Mol Cell Biol 16: 167–177
Kaplan N, Moore IK, Fondufe-Mittendorf Y, Gossett AJ, Tillo D, Field Y, LeProust EM, Hughes TR, Lieb JD, Widom J, Segal E (2009) The DNA-encoded nucleosome organization of an euakaryotic genome. Nature 458: 362–366
Kfir N, Lev-Maor G, Glaiach O, Alajem A, Datta A, Sze SK, Meshorer E, Ast G (2015) SF3B1 association with chromatin determines splicing outcomes. Cell Rep 11: 618–629
Kim T, Buratowski S (2009) Dimethylation of H3K4 by Set1 recruits the Set3 histone deacetylase complex to S′ transcribed regions. Cell 137: 259–272
Kim T, Xu Z, Clauer-Munster S, Steinmetz LM, Buratowski S (2012) Set3 HDAC mediates effects of overlapping noncoding transcription on gene induction kinetics. Cell 150: 1158–1169
Kim JH, Lee BB, Oh YM, Zhu C, Steinmetz LM, Lee Y, Kim WK, Lee SB, Buratowski S, Kim T (2016) Modulation of mRNA and IncRNA expression dynamics by the Set2-Rpd3S pathway. Nat Commun 7: 13534
Kirmizis A, Santos-Rosa H, Penkett CJ, Singer MA, Vermeulen M, Mann M, Bahl J, Green RD, Kouzarides T (2007) Arginine methylation at histone H3R2 controls deposition of H3K4 trimethylation. Nature 449: 928–932
Kirmizis A, Santos-Rosa H, Penkett CJ, Singer MA, Green RD, Kouzarides T (2009) Distinct transcriptional outputs associated with mono- and dimethylated histone H3 arginine 2. Nat Struct Mol Biol 16: 449–451
Krzyzczonik K, Wroblewska-Swinarska A, Swiezewski S (2017) Developmental transitions in Arabidopsis are regulated by antisense RNAs resulting from bidirectionally transcribed genes. RNA Biol 14: 838–842
Kwak H, Fuda NJ, Core LJ, Lis JT (2013) Precise maps of RNA polymerase reveal how promoters direct initiation and pausing. Science 339: 950–953
Lam MT, Li W, Rosenfeld MG, Class CK (2014) Enhancer RNAs and regulated transcriptional programs. Trends Biochem Sci 39: 170–182
Lavender CA, Cannady KR, Hoffman JA, Trotter KW, Gilchrist DA, Bennett BD, Burkholder AB, Burd CJ, Fargo DC, Archer TK (2016) Downstream antisense transcription predicts genomic features that define the specific chromatin environment at mammalian promoters. PLoS Genet 12: e1006224
Lenstra TL, Rodriguez J, Chen H, Larson DR (2016) Transcription dynamics in living cells. Annu Rev Biophys 45: 25–47
Li B, Jackson J, Simon MD, Fleharty B, Gogol M, Seidel C, Workman JL, Shilatifard A (2009) Histone H3 lysine 36 dimethylation (H3K36me2) is sufficient to recruit the Rpd3S histone deacetylase complex and to repress spurious transcription. J Biol Chem 284: 7970–7976
Longtime MS, McKenzie A, Demarini DJ, Shah NG, Wach A, Brachat A, Philippens P, Pringle JR (1998) Additional modules for versatile and economical PCRs-based gene deletion and modification in Saccharomyces cerevisiae. Yeast 14: 953–961
Marquardt S, Escalante-Chong R, Pho N, Wang J, Churchman LS, Springer M, Buratowski S (2014) A chromatin-based mechanism for limiting divergent noncoding transcription. Cell 157: 1712–1723
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Mayer A, di Iulio J, Maleri S, Eser U, Vierstra J, Reynolds A, Sandstrom R, Stamatosyanopoulos JA, Churchman LS (2015) Native elongating transcript sequencing reveals human transcriptional activity at nucleotide resolution. Cell 161: 541 – 554

McKay MD, Beckman RJ, Conover WJ (1979) A comparison of three methods for selecting values of input variables in the analysis of output from a computer code. Technometrics 21: 239 – 245

Mellor J, Woloszczuk R, Howe FS (2016) The interleaved genome. Trends Genet 32: 57 – 71

Miller C, Schwab B, Maier K, Schulz D, Dumcke S, Zacher B, Mayer A, Sydow J, Marcinowicz L, Doilan L, Martin DE, Tresch A, Cramer P (2011) Dynamic transcriptome analysis measures rates of mRNA synthesis and decay in yeast. Mol Syst Biol 7: 458

Mueller F, Senecal A, Tantale K, Marie-Nelly H, Ly N, Collin O, Basyuk E, Bertrand E, Darzaqz X, Zimmer C (2013) FISH-quant: automatic counting of transcripts in 3D FISH images. Nat Methods 10: 277 – 278

Murray SC, Serra Barros A, Brown DA, Dudek P, Aylings J, Mellor J (2012) A pre-initiation complex at the 3′-end of genes drives antisense transcription independent of divergent sense transcription. Nucleic Acids Res 40: 2432 – 2444

Murray SC, Haenni S, Howe FS, Fischl H, Chocian K, Nair A, Mellor J (2015) Sense and antisense transcription are associated with distinct chromatin architectures across genes. Nucleic Acids Res 43: 7823 – 7837

Murray SC, Mellor J (2016) Using both strands: the fundamental nature of antisense transcription. Bioarchitecture 6: 12 – 21

Neil H, Malabat C, d’Aubenton-Carafe Y, Xu Z, Steinmetz LM, Jacquier A (2009) Widespread bidirectional promoters are the major source of cryptic transcripts in yeast. Nature 457: 1038 – 1042

Ng HH, Ciccone DN, Marshead KB, Oettinger MA, Struhl K (2003) Lysine-79 of histone H3 is hypomethylated at silenced loci in yeast and mammalian cells: a potential mechanism for position-effect variegation. Proc Natl Acad Sci USA 100: 1820 – 1825

Nguyen T, Fischl H, Howe FS, Woloszczuk R, Serra Barros A, Xu Z, Brown D, Murray SC, Haenni S, Halstead JM, O’Connor L, Shipkovanova G, Steinmetz LM, Mellor J (2014) Transcription mediated insulation and interference direct gene cluster expression switches. Elife 3: e03635

Ni T, Tu K, Wang Z, Song S, Wu H, Xie B, Scott KC, Grewal SI, Gao Y, Zhu J (2010) The prevalence and regulation of antisense transcripts in Schizosaccharomyces pombe. Plos One 5: e15271

Nocetti N, Whitehouse J (2016) Nucleosomal repositioning underlies dynamic gene expression. Genes Dev 30: 660 – 672

Nojima T, Gomes T, Grosso AR, Kimura H, Dye MJ, Carmo-Fonseca M, Proudfoot NJ (2015) Mammalian NET-Seq reveals genome-wide nascent transcription coupled to RNA processing. Cell 161: 526 – 540

O’Duibhir E, Lijnzaad P, Benschop JJ, Lenstra TL, van Leenen D, Groot Koerkamp MJ, Margaritis T, Brok MO, Kemmeren P, Holstege FC (2014) Cell cycle population effects in perturbation studies. Mol Syst Biol 10: 732

Pelechano V, Wei W, Steinmetz LM (2013) Extensive transcriptional heterogeneity revealed by isof orm profiling. Nature 497: 127 – 131

Pijnappel WWMP, Schaft DA, Rogeuve A, Shevchenko A, Tekotte H, Wilim M, Rigaut G, Seraphin B, Aasland R, Stewart AF (2001) The S. cerevisiae SET3 complex includes two histone deacetylases, HOs2 and Hst1, and is a meiotic-specific repressor of the sporulation gene program. Genes Dev 15: 2991 – 3004

Pokholok DK, Harbison CT, Levine S, Cole M, Hennett NM, Lee TI, Bell GW, Walker K, Rolfe PA, Herboldsheimer E, Zeitlinger J, Lewitter F, Gifford DK, Young RA (2005) Genome-wide map of nucleosome acetylation and methylation in yeast. Cell 122: 517 – 527

Raj A, Peskin CS, Tranchina D, Vargas DY, Tyagi S (2006) Stochastic mRNA synthesis in mammalian cells. Plos Biol 4: e309

Rosenblum KR, Dresser TR, Pensive M, Barber GP, Meyer LR, Pohl A, Raney BJ, Wang T, Hinrichs AS, Zweig AS, Fujita PA, Learned K, Rhead B, Smith KE, Kuhn RM, Karolich D, Hausler D, Kent WJ (2010) ENCODE whole-genome data in the UCSC Genome Browser. Nucleic Acids Res 38: D620 – D625

Schindelin J, Arganda-Carreras I, Frise E, Kaying V, Longair M, Pietzsch T, Preibisch S, Rueden C, Saalfeld S, Schmid B, Tinevez JY, White DJ, Hartenstein V, Eliceiri K, Tomancak P, Cardona A (2012) Fiji: an open-source platform for biological-image analysis. Nat Methods 9: 676 – 682

Schulz D, Pirkl N, Lehmann E, Cramer P (2014) Rpnb subunit functions mainly in mRNA synthesis by RNA polymerase II. J Biol Chem 289: 17446 – 17452

Snijder B, Pelkmans L (2011) Origins of regulated cell-to-cell variability. Nat Rev Mol Cell Biol 12: 119 – 125

Sun M, Schwab B, Pirkl N, Maier KC, Schenk A, Failmezger H, Tresch A, Cramer P (2013) Global analysis of eukaryotic mRNA degradation reveals Xrn1-dependent buffering of transcript levels. Mol Cell 52: 52 – 62

Suter DM, Molina N, Gastfeld D, Schneider K, Schibler U, Naef F (2011) Mammalian genes are transcribed with widely different bursting kinetics. Science 332: 472 – 474

Tisseur M, Kwapisz M, Morillon A (2011) Pervasive transcription – lessons from yeast. Biochimie 93: 1889 – 1896

Wang Y, Liu CL, Storey JD, Tibshirani R, Brown PO (2002) Precision and functional specificity in mRNA decay. Proc Natl Acad Sci USA 99: 5860 – 5865

Weinberger L, Voichyek Y, Tirosh I, Hornung G, Amit I, Barkai N (2012) Expression noise and acetylation profiles distinguish HDAC functions. Mol Cell 47: 193 – 202

Weiner A, Hsieh TH, Appleboim A, Chen HV, Rahat A, Amit I, Rando OJ, Friedman N (2015) High-resolution chromatin dynamics during a yeast stress response. Mol Cell 58: 371 – 386

Werner MS, Sullivan MA, Shah RN, Nadadur RD, Grzybowsky AT, Galat V, Mozokvit IP, Ruthehnburg AJ (2017) Chromatin-enriched IncRNAs can act as cell-type specific activators of proximal gene transcription. Nat Struct Mol Biol 24: 596 – 603

Woo H, Dam Ha S, Lee SB, Buratowski S, Kim T (2017) Modulation of gene expression dynamics by co-transcriptional histone methylations. Exp Mol Med 49: e326

Xu Z, Wei W, Gagnier J, Perocchi F, Claude-Munster S, Cambong J, Gufiganti E, Stutz F, Huber W, Steinmetz LM (2009) Bidirectional promoters generate pervasive transcription in yeast. Nature 457: 1033 – 1037

Xu Z, Wei W, Gagnier J, Claude-Munster S, Smolik M, Huber W, Steinmetz LM (2011) Antisense expression increases gene expression variability and locus interdependency. Mol Syst Biol 7: 468

Zenklusen D, Larson DR, Singer RH (2008) Single-RNA counting reveals alternative modes of gene expression in yeast. Nat Struct Mol Biol 15: 1263 – 1271

Zenklusen D, Singer RH (2010) Analyzing mRNA expression using single mRNA resolution fluorescent in situ hybridization. Methods Enzymol 470: 641 – 659

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